Conservation research in the African rain forests

a technical handbook

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EDITED BY

Lee White & Ann Edwards

Illustrations by ***Kate Abernethy, Richard Parnell & Deborah Haines***

Bonobo
Pan paniscus

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Paristolochia flos-avis

This manual is dedicated to the people who have participated in the WCS Regional Ecological Training Program in Central Africa.

The forest's future is in your hands.

Male sitatunga
Tragelaphus spekei

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Preface

The rain forests of central and west Africa, with the multitude of plant and animal species found within them, are one of the world's greatest biological treasures, and represent one of the most valuable assets for many countries in equatorial Africa. Rain forests are valuable because they serve so many life-sustaining functions. They provide food such as fruit, nuts and meat to the people who live in and near them. They provide building materials and medicines for local uses, as well as timber for export. Intact rain forests stabilise soils, reducing erosion and hence providing clean water to drink, and play a key role in the regulation climate, both locally and globally. The beauty, diversity and rarity of rain forest species attract tourists and scientists from all over the world, as well as inspiring unique and lasting cultural traditions among the people of forested African countries.

The African rain forest still covers a vast area, stretching from Guinea in the west across to the coast of east Africa, but it faces a wide range of threats. The rain forests of east and west Africa have already been greatly reduced by human activities in the last century or so, and today little natural forest vegetation survives outside protected forest reserves, wildlife sanctuaries and National Parks. The central African forest block remains largely intact, but even the most remote areas are likely to be affected in the near future by the combined forces of deforestation and exploitation. As human populations increase steadily, more and more land is needed for agriculture; and as technology advances, exploitation for timber, meat and other forest products becomes more and more intensive and damaging.

Under this scenario, protected areas and their management staff have a crucial role to play if biological diversity is to be conserved. However, just as there are a wide variety of habitats and vegetation types within the forest, protected areas are designed to fulfil many different roles and face a wide variety of threats. In order to successfully manage protected areas in the African rain forest zone (or elsewhere), management staff must have a sound knowledge of the objectives which lead to their creation, of their current status, and of relevant trends (particularly threats) within and around protected areas. They also need a sound practical knowledge of how to manage financial, mechanical and human resources.

Many protected areas have been established throughout forested Africa, with reserve boundaries, hunting restrictions and certain management goals, among other things, clearly described in legal documents. However, these protected areas often do not function as they were intended, to protect the natural resources contained within them. Boundaries are not clearly marked on the ground. Local people remain uninformed about which activities are legal or illegal within the protected area. No one is certain which species may be living in the protected area, specifically endangered species, or whether populations of those species are increasing or decreasing in number. No one has determined how fast agricultural lands are turning primary forests into secondary bush, or whether hunting to fill the cooking pot is having a significant effect on animal populations. Finally, the goals of many protected areas, the guiding principles by which management decisions should be made, are unclear or out of date.

Often the lack of trained personnel, proper equipment, and sufficient funds are blamed for the ineffective way that protected areas are managed in central and west Africa. These obstacles, though significant, should not be allowed to prevent protected areas from being managed effectively. The key to making educated, realistic, effective management decisions is to base those decisions on information. Furthermore, the key to gaining outside support for management and conservation efforts is the effective presentation and dissemination of information. Throughout protected areas in central and west Africa not enough is known, and what information may exist is rarely readily available.

The purpose of this manual is to assist people working in African forests to collect information from many different sources and use it effectively for management and conservation. The manual is designed so it can be used alone, without the need to refer to other reference books. However, many references are listed throughout the manual and whenever available they should be consulted. Furthermore, the techniques outlined for data collection and data analysis generally do not require sophisticated technical equipment. It is hoped that with this manual people will feel more confident collecting and interpreting biological and socio-economic information about the forest and its inhabitants, whether that information is used to direct management actions or whether it is used to rally support for continued natural resource protection. It is not possible to describe methods for all types of research possible in tropical rain forest. If we had, each chapter in this manual would have become a book (or several books!) in its own right. We have therefore selected priorities for research and present methods commonly used by researchers and conservation managers working in the African forest zone. In this way we hope to:

1. Help protected area managers to establish goals and determine priorities for management and to design research programmes appropriate to these objectives.
2. Outline simple, current, commonly used methods for collecting information, specifically on population densities and behavioural ecology of larger animals, vegetation, physical features of the land, weather patterns, and the numbers, impacts, needs and expectations of people that use and/or live near protected areas.
3. Present guidelines for analysing field data realistically and with confidence.
4. Present guidelines for interpreting and storing information with the aim of making it useful and accessible to a variety of audiences.

Yellow billed hornbill
Tockus flavirostris

If you see a copy of this manual in a colleague's home, at your head office, or on a researcher's shelf, and feel it would be useful for you and your colleagues, it is available free of charge from the **Wildlife Conservation Society**. The only 'price' we put on it is that you send us your comments in order to help us revise and improve any future versions. We hope it will encourage protected area managers and researchers alike to get to know the African rain forest and to work together towards its conservation and sensible management.

Lee White
Ann Edwards

Chapter 1
Protected area management and the role of research

Annette Lanjouw, Ann Edwards & Lee White

Introduction

In order to manage the different ecosystems in a protected area efficiently and securely one needs to know and understand how they function and what impact humans may have upon them. During a regional evaluation of training needs in 14 African countries many protected area managers expressed the need for more training in methods of biological inventory and collection of ecological data (Pitkin, 1995). This manual is intended to help protected area managers (and conservation scientists) working in the African rain forest to evaluate and monitor animal and plant populations and to assess the impact of humans on the ecosystem. It is a tool which should help the reader to better understand the natural resources for which they are responsible. It will not only guide the reader through the processes of research design and data collection, but will also cover the preliminary analysis and interpretation of data and the key phase of presentation of results. Furthermore, it may well help the reader to obtain either financial or political support for sound land management, be it species protection, resource extraction, leisure, maintenance of ecosystem functioning or some combination of these.

Before considering the different types of information which are necessary for the efficient management of a protected area located in the African rain forest, it is useful to consider the different roles that protected area managers are expected to fulfil and the varied functions of protected areas.

The role of a protected area manager

If you are already a protected area manager, or are considering a career in conservation management or research, you will hopefully already have quite a good idea of how research fits into protected area management, and into the day to day activities of a protected area manager. However, before plunging into this methods manual it is a useful exercise to pause and take a moment to think about the following questions:

What are a protected area manager's responsibilities?

Why does a protected area manager need to have a detailed knowledge of the protected area where he/she work?

Why does a protected area manager need to understand and be able to influence research being conducted in his/her protected area?

What biological and socio-economic information does a manager need to manage the protected area in which he/she (will) works successfully?

To begin to answer these questions, let us look at an example of a *job description* for a protected area manager. The following example is based on the newly created Uganda Wildlife Authority:

According to the Uganda Wildlife Authority, the main responsibilities of a protected area manager are to:

- direct the management of the ecosystem;
- design and implement relevant applied research and ecological monitoring, and ensure that management activities respond to the findings of this research programme;

- ensure the safety of people within the protected area;
- ensure that wildlife are adequately protected within and in the vicinity of the protected area;
- ensure the development and implementation of a community conservation programme, to build local support for the protected area and to enable neighbouring communities to benefit in appropriate ways;
- ensure the development of a Tourism & Commercial programme, where relevant, to improve visitor satisfaction and maximise profitability of the protected area, consistent with UWA policies and programmes;
- ensure the proper management and maintenance of the protected area's infrastructure, vehicles, machinery and equipment;
- apply UWA policies to management of the protected area and share relevant experiences in order to contribute to the future development of UWA policies;
- supervise the planning and monitoring of protected area management;
- liaise with major stakeholders in the protected area and with potential collaborators, relevant donors and non-government organisations;
- develop a competent and well-motivated team of protected area staff;
- ensure proper financial management of the protected area.

Daunting isn't it?

Imagine you are arriving for the first time in a protected area to which you have been posted. What are the most fundamental questions about its management which you should ask and find the answers to during your first month at your post? Consider for example:

- Why was the area classed as a protected area? What is its legal status - which activities are illegal and which, if any, are permitted? Are the criteria for its establishment as a protected area still relevant today?

- What are the management objectives for the area? Are they clearly stated and justified? Is there a management plan for the area?

- What is known about the area and, equally important, what is not known? What information needs to be collected in order to establish a realistic action plan for the protection of the resources within the protected area?

This chapter will give a framework for systematically answering these questions. It will start by describing the different types of protected area, emphasising the fact that they do not all have the same objectives. By understanding that protected areas can serve diverse functions you will be better able to establish and justify you own management priorities - and this is the first stage when initiating a management strategy. You will then be lead through the process of developing a management plan for your protected area. This process is extremely useful, not only because it results in a document that coordinates and directs actions, but because it also obliges you to think about the landscape and its resources in great detail. In particular, do we know what actually exists in the protected area? What is the extent, the size and the status of these resources? Who uses them (humans or animals)? What is the impact or potential impact of man on these resources? By considering these unknowns during the process of development of a management plan a manager and his/her team of researchers, advisors and technicians will begin to appreciate the great value of research for management. Furthermore, this will provide a guideline for the development of important research questions and the conception and implementation of research projects.

The roles of protected areas

According to the International Union for the Conservation of Nature (IUCN, 1994) the definition of a protected area is: «An area of land and / or sea especially dedicated to the protection and maintenance of biological diversity and of natural and associated cultural resources, which is managed through legal or other effective means».This is a worthy, but very broad goal for management. In order to physically manage an area, more specific objectives are required.

Protected areas are established and managed to meet a range of multiple objectives. Each protected area will have a priority objective, for which it is being managed. It can also have a range of secondary objectives. Typical objectives include:

Protecting representative examples of rare, threatened, fragile or otherwise valuable habitats and ecosystems.

This may include:

- areas of intact rain forest surrounded by logging concessions or farmland, as is the case for many of Africa's rain forest protected areas;
- areas of savanna or large clearings in areas dominated by rain forest, such as the coastal savannas in Petit Loango and Iguela in Gabon, or the network of clearings in Odzala, Congo, both of which support concentrations of large mammals;
- areas with fragile montane floras and faunas, which also have high tourist potential, such as the forests of Mount Cameroon or the Rwenzori Mountains in East Africa;
- areas known to be the home of many endemic species of plants and animals, such as the Korup National Park in Cameroon - these areas are often called centres of biological diversity;
- areas with a diverse complex of habitat types, such as the Conkouati Reserve in Congo, which contains coastal lagoons, mangroves, freshwater swamps and dry land rain forests;
- wetlands of particular importance for migratory birds, such as the Saloum Delta National Park in Senegal / Gambia;
- landscape or geophysical features of aesthetic or scientific value.

Protecting rare, threatened or otherwise valuable species.

This may include:

- rare, endangered or endemic species of plants and animals which are restricted to one or a few protected areas, such as the okapi in the Ituri Forest, RDC, or the pygmy hippo in Tai National Park, Ivory Coast;
- 'flagship' species, such as elephants, gorillas, or the congo peacock, which attract a lot of public attention and can be used to generate support for other conservation activities;
- wild plant species related to domesticated food plants, such as the many species of wild coffee, *Coffea* spp. and the yams, *Dioscorea* spp.;
- wild relatives or forms of domestic animals, or species with recognised potential for domestication, such as red river hogs;
- species harvested for food, medicine or other uses, which are likely to become over-exploited, including rare timber trees, rattans, crocodiles, bush meat etc.;
- species vital for fulfiling functions on which other harvests depend, such as forest dwelling bees which pollinate crops such as coffee and cocoa;
- species providing known or potentially useful drugs;
- species which are useful research models for studies of human behaviour, physiology, etc., such as chimpanzees.

Maintaining ecosystem function.

This could be by:

- reducing the intensity of floods and droughts, which are common in areas where there has been significant deforestation;
- protecting the soil from erosion by maintaining vegetation cover;
- limiting the extremes of local climates - in areas where there has been widespread deforestation streams may stop flowing in the dry season and local weather patterns may be altered;

Maintaining biological productivity, thus ensuring the continuing availability of plant and animal resources.

For example:

- one of the major justifications for creating the Cross River National Park in Nigeria was to protect the important fishery grounds off the Cross River estuary, which would have been seriously disrupted if deforestation had lead to increased sediment loads in the river because of erosion;
- the only forests remaining in Ghana (and many other west African countries) are those that have been protected in forest reserves or national parks - had these areas not been delimited and managed Ghana would have no timber production forests left, nor any forests in which medicinal plants, rattans or bush meat could be harvested;

Providing areas where rural populations can continue their traditional lifestyles.

For example:

- in Lobéké, in southeastern Cameroon, local people complained that forestry companies and immigrant hunters paid little or no heed to traditional land tenure, resulting in over exploitation of resources such as bush meat - a community reserve in which local residents have the right to practice their traditional activities has been proposed.

Providing opportunities for research and education.

*F*or example:

- tropical rain forests remain poorly studied and therefore represent a natural laboratory offering countless opportunities for original research projects for students of botany, zoology, ecology, geography, anthropology, archaeology, etc.;
- applied research, addressing questions such as how best to manage tropical forests for sustained timber or bushmeat production, is often undertaken in reserves, where conditions can be more easily controlled;
- for school children the best way to learn natural science is to see and experience natural phenomena, such as by visiting a reserve to see the different animals and plants which occur there.

Providing opportunities for cultural or spiritual enhancement.

For example:

- most central African cultures have evolved in close association with the forest and there are many beliefs and traditions associated with the forest - reserves ensure cultural continuity by providing people who no longer live within the forest the opportunity to visit and experience its sights and sounds;
- there is a growing interest in rain forest tourism, which depends to a great extent on protected areas;

- the forests of central Africa contain rich archeological remains which are a testimony to Africa's unwritten history, such as the network of earthworks around Benin City in southwestern Nigeria, which surpass the Great Wall of China as the longest (unmechanised) human excavation ever undertaken, some of which are preserved in the Okomu Reserve.

Providing opportunities for complementary rural development and the rational use of marginal lands.

For example:

- the soils of most tropical rain forests tend to be relatively poor and unfavourable for agriculture, but development of tourism in a reserve can supply employment for people living in the area;
- by managing forest reserves sensibly for sustained timber production, long-term economic development can be achieved without destroying the resource base.

Lophira alata

IUCN management categories

IUCN (1994) developed a classification system for placing protected areas into one of six categories (see below). This classification was created to provide a globally applicable framework to allow comparisons to be made and lessons to be learned across continents. Under this system protected areas are divided into six broad categories which differ primarily in the access available to the general public (including the extent and type of resource extraction permitted) and in the amount of active manipulation of the biological systems by management. It is useful to think about these categories in order to decide which one your reserve fits and then to assess whether the guidelines for selection of that category actually apply to your protected area. The categories are as follows:

Category 1 *Strict Nature Reserves / Wilderness Areas*

Protected areas managed mainly for science or wilderness protection. These two types of protected area are treated as sub-categories:

Category 1a *Strict Nature Reserves*

Areas of land and / or sea possessing some outstanding or representative ecosystems, geological or physiological features and / or species, available primarily for scientific research and / or environmental monitoring.

Objectives of Management:

- to preserve habitats, ecosystems and species in as undisturbed a state as possible;
- to maintain genetic resources in a dynamic and evolutionary state;
- to maintain established ecological processes;
- to safeguard structural landscape features or geological formations;
- to secure examples of the natural environment for scientific studies, environmental monitoring and education, including baseline areas from which all avoidable access is excluded;
- to minimise disturbance by careful planning and execution of research and other approved activities;
- to limit public access.

Guidelines for selection of strict nature reserves:

- the area should be large enough to ensure the integrity of its ecosystems and to accomplish the management objectives for which it is protected;
- the area should be significantly free of direct human intervention and capable of remaining so;
- the conservation of the area's biodiversity should be achievable through protection and not require substantial active management or habitat manipulation;

Category 1b *Wilderness areas*

These are protected areas managed mainly for wilderness protection. They should include a large area of unmodified or slightly modified land, and / or sea, retaining their natural character and influence, without permanent or significant habitation and should be protected and managed so as to preserve their natural condition.

Objectives of management:

- to ensure that future generations will have the opportunity to experience, understand and enjoy areas that remain largely undisturbed by human action over a long period of time;
- to maintain the essential natural attributes and qualities of the environment over the long term;
- to allow controlled public access so as to optimise the physical and spiritual well-being of visitors whilst maintaining the wilderness qualities of the area for present and future generations;
- to enable indigenous human communities living at low density and in balance with the available resources to maintain their lifestyle.

Guidelines for selection of wilderness areas:

- the area should be of great natural beauty or interest and be little altered or threatened by humans;
- the area should contain significant ecological, geological, physiogeographic or other features of scientific, educational, scenic or historic value;
- the area should offer outstanding opportunities for solitude, enjoyed once the area has been reached, by simple, quiet, non-polluting and non-intrusive means of travel (i.e. non-motorised);
- the area should be of sufficient size to make practical such preservation and use.

Category II *National Parks*

Protected areas managed mainly for ecosystem protection and recreation. These are natural areas of land and / or sea, designated to:

a) protect the ecological integrity of one or more ecosystems for present and future generations;
b) exclude exploitation or occupation likely to degrade the area; and
c) provide a foundation for spiritual, scientific, education, recreational and visitor uses, all of which must be environmentally and culturally compatible.

Objectives of management:

- to protect natural and scenic areas of national and international significance for spiritual, scientific, educational, recreational or tourist purposes;

- to perpetuate, in as natural a state as possible, representative examples of physiogeographic regions, biotic communities, genetic resources and species, to provide ecological stability and diversity;
- to manage access for educational, cultural and recreational purposes at a level which will maintain the area in a natural or near natural state;
- to eliminate and thereafter prevent exploitation or occupation likely to result in degradation;
- to ensure that the ecological, geomorphologic, sacred or aesthetic attributes which warranted designation are maintained;
- to take into account the needs of indigenous people, including subsistence resource use, in so far as these will not adversely affect the other objectives of management.

Guidelines for selection of national parks:

- the area should contain a representative sample of major natural regions, features or scenery, where plant and animal species, habitats and geomorphologic sites are of special spiritual, scientific, educational, recreational and tourist significance;
- the area should be large enough to contain one or more entire ecosystems not materially altered by current human occupation or exploitation.

Category III *Natural Monuments*

Protected areas managed mainly for conservation of specific natural features. These are areas containing one, or more, specific natural or natural / cultural features which are of outstanding or unique value because of their inherent rarity, representative or aesthetic qualities, or cultural significance.

Objectives of Management:

- to protect or preserve in perpetuity specific outstanding natural features because of their natural significance, unique or representational quality, and / or spiritual connotations;
- to the extent consistent with the first objective, to provide opportunities for research, education, interpretation and public appreciation;
- to eliminate and thereafter prevent exploitation or occupation counter to the purpose of the designation;
- to deliver to any resident population such benefits as are consistent with the other objectives of management.

Guidelines for selection of natural monuments:

- the area should contain one or more features of outstanding significance. Appropriate natural features include spectacular waterfalls, caves, craters, fossil beds, sand dunes and marine habitats, along with unique or representative fauna and flora. Associated cultural features might include cave dwellings, archeological sites, or natural sites which have heritage significance to indigenous peoples;
- the area should be large enough to protect the integrity of the feature and its immediately related surroundings.

Category IV *Habitat / Species Management Areas*

Areas of land and / or sea where active management interventions are undertaken so as to ensure the maintenance of habitats and / or to meet the requirements of specific species.

Objectives of Management:

- to secure and maintain optimal habitats so as to protect key species or groups of species, biotic communities, or physical features of the environment, where these require specific human manipulation for optimum management;
- to facilitate scientific research and environmental monitoring as a means to monitor sustainable resource management;
- to allow limited public access to inform people about key habitats and to demonstrate the work of wildlife managers;
- to eliminate and thereafter prevent exploitation or occupation counter to the aims of the management area;
- to deliver such benefits to people living within the designated area as are consistent with the other objectives of management.

Guidelines for selection of habitat and species management areas:

- the area should play an important role in the protection of nature and the survival of species, incorporating, as appropriate, breeding areas, wetlands, coral reefs, estuaries, grasslands, forests or spawning areas, including marine feeding beds;
- the area should be one where the protection of the habitat is essential to the well-being of flora, or to resident or migratory fauna, of national or local importance;
- conservation of these habitats and species should depend upon active intervention by the management authority, if necessary through habitat manipulation (c.f. Category 1a);
- the size of the area should depend on the habitat requirements of the species to be protected and may range from relatively small to very extensive.

Category V *Protected Landscapes / Seascapes*

Protected areas managed mainly for landscape / seascape conservation and recreation. They consist of areas of land, sometimes with coast and sea as appropriate, where the interaction of people and nature over time has produced a landscape of distinct character with significant aesthetic, ecological and / or cultural value, and often with high biological diversity. Safeguarding the integrity of this traditional interaction is vital to the protection, maintenance and evolution of such an area.

Objectives of management:

- to maintain the harmonious interaction of nature and culture through the protection of landscapes and / or seascapes and the continuation of traditional land uses, building practices and social and cultural events;
- to support lifestyles and economic activities which are in harmony with nature and the preservation of the social and cultural fabric of the communities concerned;
- to maintain the diversity of landscapes and habitats, and of associated species and ecosystems;
- to eliminate where necessary, and thereafter prevent, land uses and activities which are inappropriate in scale and / or character;
- to provide opportunities for public enjoyment through appropriate forms of recreation and tourism;
- to encourage scientific and educational activities which will contribute to the long term well-being of resident populations and to the development of public support for the environmental protection of such areas;

- to bring benefits to, and to contribute to the welfare of, the local community through the provision of natural products (such as forest and fisheries products) and services (such as clean water or income derived from sustainable forms of tourism);

Guidelines for selection of protected landscapes / seascapes:

- the area should possess a landscape and / or coastal and island seascape of high scenic quality, with diverse associated habitats, flora and fauna, along with unique or traditional land-use patterns and social organisations, as demonstrated by human settlements and local customs, livelihoods and beliefs;
- the area should provide opportunities for public enjoyment through recreation and tourism without changing traditional lifestyles and economic activities.

Category VI *Managed Resource Protected Areas*

Protected areas managed mainly for the sustainable use of natural ecosystems. They are areas containing predominantly unmodified natural systems, managed to ensure long term protection and maintenance of biological diversity, while providing at the same time a sustainable flow of natural products and services to meet community needs.

Objectives of Management:

- to protect and maintain the biological diversity and other natural values of the area in the long term;
- to promote sound management practices for sustainable production purposes;
- to protect the natural resource base from being alienated for other land-use purposes that would be detrimental to the area's biological diversity;
- to contribute to regional and national development.

Guidelines for selection of managed resource protected areas:

- at least two-thirds of the area should be in a natural condition, although it may also contain limited areas of modified ecosystems, but large commercial plantations are not appropriate for inclusion;
- the area should be large enough to allow sustainable resource exploitation without detriment to its overall long-term natural values.

The majority of protected areas in the African forest zone are either national parks (Category 2), or forest reserves in which natural resources are managed for a sustained harvest (Category VI). However, many protected areas in the African rain forest zone date back to colonial periods, and the original objectives may have been forgotten or become outdated. Furthermore, management of reserves for timber production has resulted in severe degradation of many managed resource protected areas (forest reserves) and if they are to continue to fulfil their intended role new management guidelines are required (see Box 1a). Thinking about why your protected area was established, what its original goals were, and whether these goals are still appropriate, is a productive way to start to think about what sort of information you will need in order to manage your protected area in an informed manner. If a management plan exists for your protected area the people who wrote it have probably already been through this process. This is the first place you should look for background information to help you in your reflections. Unfortunately, relatively few protected areas in the African forest zone have up-to-date management plans. Studying an existing management plan or thinking about the elements you would need to produce one for a given area, are good ways to start thinking about research needs.

Box 1a MANAGEMENT OF FOREST RESERVES IN GHANA.

From the mid-late 1960's to the early 1980's the forest reserves of Ghana were extensively degraded due to serious misjudgements by managers, which lead to blatantly unsustainable use (Hawthorne & Abu-Juam, 1995; Hawthorne, 2000). Whilst part of the reason for this mismanagement can be attributed to economic decline in the country, much of the reason was because guidelines regarding protection and sustainability were unclear and unexacting. During this period large areas of managed forests were replaced by farming and increased intensity of logging degraded forests, making them much more susceptible to damage by fire. As a result fires have severely damaged about 30% of Ghana's semi-deciduous forests (Hawthorne, 1994).

Since 1983 there has been an intensive effort to remedy the situation:

- An inventory of 1/400 of all remaining productive forest areas, including all trees down to a diameter of 5 cm, was undertaken in order to assess forest status and timber stocks and regeneration;
- A network of permanent research plots were established to monitor forest dynamics and growth;
- A field guide to trees was produced to enable forest managers to reliably identify tree species;
- Studies of the environmental impact of logging, fire and use of non-timber products were undertaken; and
- A botanical survey of all remaining forests established plant conservation priorities throughout the forest zone of Ghana.

These various applied research projects have contributed to a series of management plans and policy documents which are now guiding forest management in Ghana. Individual species deserving special conservation attention because they are rare or threatened at a national or international level have been singled out for partial or complete protection; habitats which are particularly rich, rare or fragile have been mapped and will be carefully managed or totally protected; and strict guidelines for timber and other forms of exploitation have been developed.

The work is not yet complete. In particular, attention needs to be paid to the status of wildlife in Ghana's forests, since uncontrolled hunting pressure has had a severe impact. However, the far-sighted research programme undertaken to date is an excellent example of how forest managers are attempting to formulate sound management options based on good scientific information, and could act as a model for other African states.

Developing a Management Plan

What is a management plan and how is it useful?

A protected area management plan guides and controls the management of protected area resources, the uses of the area, and the development of facilities needed to support management and use. It determines the development activities and management actions to be implemented in an area, and details mechanisms to assess how the success or failure of these activities should be measured. Central to the plan is a statement of *goals and measurable objectives* to guide the management of the area. These goals and objectives form the framework for determining what actions to take, when they will be taken and the budget and personnel needed to implement them. The plan should describe any specialised training needs of staff such that a competent team is maintained as and when transfers are made.

Usually a management plan is for 5 years, or a limited period of time, after which the plan is evaluated and modified. Annual workplans are developed during the implementation phase using the longer term management plan as a guide. The management plan is always subject to modification as new information is obtained, particularly regarding feedback on the effectiveness of the actions taken in the annual workplans.

Steps in the process of developing a management plan:

To assess whether an existing management plan needs to be updated, or to begin to write one where none exists, you need to understand how management plans are generally formulated.

1) Form the planning team: include a variety of people with expertise in planning, ecology, sociology, economics and various other resource sciences. Also include the protected area authorities and the people who manage the park, as well as those who will be affected by the plan. The team should consult with scientists, experts on tourism, educators, concessionaires and people living in and around the protected area.

2) State the objectives of the area: analyse the original reasons for creating the protected area and, if necessary, update the objectives as a function of modern conditions.

3) Gather basic background information: this includes legislation, data on biophysical features, cultural resources and socio-economic data. (These and the next points will be expanded below in the section on information required for the management plan).

4) Field inventory: consult and update existing data, gather new information and develop the information base to make informed management decisions.

5) Assess constraints: limitations of an environmental, economic, political, administrative, or legal nature, should be recognised and analysed, with an emphasis on realism.

6) Review regional inter-relationships: the protected area must be integrated as an essential element in the regional land-use pattern. The planning team must attempt to review the potential effects of development outside the protected area borders as well as the effects of the protected area on the region.

7) Divide the area into appropriate management zones: these may include core protected areas and surrounding multiple use zones which act as a buffer against degradation due to pressure from outside the protected area.

8) Review the boundaries of the area: consider boundary modifications that are realistic in terms of the ecological ideal, sociological or cultural ideals and political or administrative considerations.

9) Design the management programmes: address the questions *how* and *who* in the plan and develop the four major programmes of protected area management:

- resource management and protection
- human use
- research and monitoring
- administration

10) Prepare integrated development options: assess the infrastructure and investment that will be needed to accomplish the various programmes.

11) Outline financial implications: cost the different planning proposals and identify potential sources of funding.

12) Prepare and distribute a draft plan for comments.

13) Analyse and evaluate the plan in the light of comments from all interested parties.

14) Design schedules and priorities for action.

15) Prepare, publish and make public the finalised plan.

16) Monitor and revise the plan (and management activities) on a regular basis as a function of practical experiences during the course of management activities.

Research and management: key activities which compete for resources

Thinking back to the list of responsibilities of a protected area manager, it is evident that even if research is considered to be a priority, management staff will have many other responsibilities. Therefore, research activities need to be carefully planned so as to generate the maximum amount of information necessary for management, with the minimum investment of financial or human resources. This will be easier if the manager has a clear vision of the research needs in the area. Furthermore, if managers encourage research by universities or other non-profit organisations the budget for the protected area will not have to cover all of the costs. Therefore, providing that research proposed by 'outsiders' is not in conflict with management objectives it should be encouraged and supported.

The following chapter will describe the types of biological and socio-economic questions research can be used to answer. Below we make some suggestions about how to coordinate and manage research, particularly with respect to visiting researchers.

Identification of the information needed for management of a protected area

The management plan should include a section on research and monitoring. Each protected area will have different management objectives and specific management issues that will require special attention. A research component in the management plan, as well as in the protected area zoning plan, will help determine which areas will be used for research and what the research objectives of the protected area will be. All protected areas should establish a series of long-term protocols to monitor changes in populations of key animals and plants, important vegetation types, human population and resource use, water quality, weather and visitor numbers. Most of these studies can be conducted by protected area staff as part of their day-to-day responsibilities.

Research projects proposed by outside researchers should be evaluated and approved by the management authority of the protected area before they begin.

The management authority should ensure that research projects are compatible with the objectives of the reserve. They may need to request modification of a project to make it more relevant to management needs or to complement other research projects in the area.

Activities of researchers should be monitored by management authorities

The protected area management authority must retain control over all activities in the protected area. Researchers should submit written progress reports and occasional verbal presentations to the protected area staff to keep the managers up-to-date with their activities.

There should be clear guidelines for research projects which involve collection of specimens

Collection of plant and animal materials in the protected area must require specific permission of the management authority. In addition, the manager may request duplicate specimens and identifications to be deposited in the protected area or national herbarium or museum. Material to be taken outside the country may also require a special permit.

Outside researchers should submit preliminary and final reports with particular emphasis on the relevance of their findings for management

At the end of the study, the researcher should submit a preliminary report on his or her studies before leaving the protected area and a final report once the study is complete. These reports should contain a section on the relevance of the findings to management of the protected area and the conservation in the area.

Furthermore, each researcher should be asked to send copies of all scientific and other publications that arise from the research to the management authority as they appear in press. Summaries written in popular style could be used for interpretative literature on the park. In addition, researchers should leave copies of qualitative and quantitative data for the files for management purposes. However, managers should realise that data are extremely valuable for researchers - you can insist on the production of reports but should be flexible about the provision of whole data sets.

Adequate priority should be given to social and economic research programmes

One of the largest problems facing protected area managers today involves the impact of local populations, tourists and other resource users on the protected area and the response of local populations to restrictions imposed by the protected area on their activities. Research on human aspects of protected area management should be given support. Managers need to be able to quantify the economic benefits that protected areas provide, through conservation of biodiversity, protection of watersheds, tourism, etc. as well as the impact of economic activities on the reserve.

Promotion of the protected area as a site for research activities

Research projects which seem to be only of academic interest with no immediate benefit for management should be encouraged providing they do not deplete limited resources or interfere with management practices. Results of little apparent interest may become valuable when comparisons are made over time or between sites. The presence of internationally recognised scientists can help to protect an area or species. Infrastructure such as research stations or special study areas will facilitate scientific research. An open dialogue should always be maintained between managers and researchers.

The protected area authority should consider the appointment of staff research scientists

If finances permit the appointment of a full-time researcher this person will ensure better coordination and integration of management and research activities. In addition to undertaking his or her own research, which should include long-term monitoring as well as other punctual projects, the researcher should play an active role in coordinating visiting researchers and ensuring that they respect the conditions described above. Their research will consist of subjects which are important for management but which may be of little academic value. Furthermore, the presence of a permanent researcher will help the management authorities to refine research priorities.

A final note of encouragement

The sad reality is that human and financial resources in many African protected areas are extremely limited, and most management is done in a ‘seat of the pants’ or ‘crisis management’ fashion. Management staff react using their best judgement to situations as and when they arise. Very few managers have the luxury of complete biological inventories, good vegetation maps, in depth knowledge of species’ ecological requirements or seasonal movements, or even of the human pressure on their protected area. In some cases they have no vehicle, let alone a computer, a GPS unit or a set of satellite images.

Protected area management and the role of research

However, by going through the process of setting priorities for research you will be better able to benefit from the presence of researchers who are working in your protected area and to lobby for increased financial, equipment or human resources. Furthermore, this manual will demonstrate that not all research is necessarily expensive, nor time consuming; simply by living and working in a protected area you will be in a position to collect unique and useful information during day to day activities. The methods and case studies in this manual will hopefully open your eyes to the many ways in which research can be of benefit for management and encourage and enable you to begin to gather such information in a methodical and rigorous way.

Okapia johnstoni

Chapter 2
Research priorities and design of research programmes

Lee White and Ann Edwards

Introduction

Information on biological processes and human activities can be an extremely valuable tool for a protected area manager. Up to date, rigorously obtained information gives the manager a window into the reality of the forces at work within the protected area. Not only does information help a manager determine appropriate plans of action for managing the resources within the protected area, but when used effectively, information also can bring more support to the protected area in the form of increased tourism, scientific research, staffing and funding.

Conservationists need to take into account a complex array of immediate and long term factors when planning their research programs. Immediate problems are generally given priority but one must not lose sight of the need to monitor long-term trends. In areas where poaching, logging, or agriculture threaten wildlife and their habitat, we need to know the extent of the problem and the effects on vegetation or animal populations. If we are to monitor the success of conservation efforts we need to know animal densities or the extent of human encroachment before those efforts began and to monitor any changes (hopefully increases in animal densities and decreases in human encroachment). If we are going to protect large mammals such as elephants we need to know their habitat requirements - do they need to migrate out of protected areas on a seasonal basis, or will protection result in increased elephant densities due to influx of populations into protected areas, resulting in vegetation destruction ? Where there is a lack of basic knowledge the emphasis is often put on inventory - but if inventory is undertaken in such a way that it can be repeated it becomes a (potential) means of monitoring.

This chapter offers an overview of the kinds of questions that can be asked using the field methods contained in later chapters and leads you through the process of planning a research project. Interesting questions are often surprisingly simple. How many are there? Where are they found? What changes are there over time, or in the presence of certain types of disturbance? As you read the rest of this manual you may feel overwhelmed by the details of methods and analysis. For that reason it is important that you have a clear vision in your mind of the information that you need and the methods you are going to use to get it. Ultimately, the strength of your findings will depend upon how well you thought out your research questions and if the methods you used to address them were appropriate and well executed. Only if research is well planned and carried out will it yield results upon which you feel confident you can base management actions and decisions.

There are two key stages in any research programme. First, preliminary observations need to be made, or inventory work undertaken, in order to detect interesting patterns or trends. Next, experimental studies need to be designed and implemented in order to explain these results. The first phase is essentially descriptive. It involves documenting patterns of distribution and abundance (or intensity) of plants and animals (or human activities) and perhaps the detection of trends, such as decreases in animal numbers or changes in patterns of human activity. The second phase seeks to explain these preliminary observations - why have animal densities decreased? why are patterns of human activity changing? In order to answer such questions hypotheses need to be formulated and tested. For example, you might predict that animal densities have decreased due to increased hunting pressure, or that humans are travelling further into your protected area to hunt because animal numbers have decreased significantly close to their villages.

Both of these questions have obvious implications for management, but in both cases alternative explanations might be possible. Perhaps animals have been affected by a disease which has resulted in high mortality, or humans are travelling deep into the reserve because gold has been found in the interior, or because a logging company which employed a large number of workers has gone out of business. In order to be able to make firm conclusions your research must by carefully designed to address a particular question.

Much of this manual is concerned with description of pattern. This is partly because the rain forests of Africa, particularly the extensive forests of central Africa, are poorly known. In many protected areas in the forest zone managers have little baseline information to guide their activities. In most cases patterns of distribution and abundance have not been (adequately) documented, and it is highly probable that even quite significant trends will go undetected. Furthermore, in many instances knowledge of the pattern is sufficient to guide management activities and an understanding of process is an unnecessary luxury. For example, if managers know that a rare species of plant or animal of particular interest is restricted to the southern part of their protected area, where it is threatened by illegal exploitation, they can focus their protection activities on this area. They probably do not need to know **why** the species only occurs in the south. On the other hand, a scientist interested in the same species would not be satisfied simply to learn that it was restricted to the south, but would then want to explain this pattern by testing hypotheses based on these preliminary observations.

This manual is aimed primarily at wildlife managers and researchers whose work is intended to assist directly with management. There is a bias towards the challenge posed by the vast forests of central Africa, where managers have to cope with extensive, uncharted, inaccessible protected areas about which little is known. Consider the challenge of gathering information in the Salonga National Park, in central DRC (previously Zaire), covering 36,000 km^2 of inaccessible rain forest about which little is known. Simply documenting pattern over such an area is a major challenge, but without some baseline information it is difficult to make management decisions. Management priorities in such areas are unlikely to involve a great deal of highly detailed research aimed at explaining ecological pattern and process. Rather, a broad understanding of distribution patterns and threats will be sought and used to direct conservation activities in as efficient a manner as possible. However, the standardised methods we recommend are likely to be of use to managers and scientists alike and whilst, given the breadth of the topic no single volume can be complete, we hope that managers in particular will find answers to many of their questions about research in this manual.

Phase 1: description

A great deal of purely descriptive information is extremely valuable for both conservation managers and research scientists alike. In the case of managers this information is most often used to direct daily activities, whilst research scientists will use it to formulate hypotheses, which they will test by undertaking further research. Obviously priorities will vary in different places, depending on factors such as management objectives, plant and animal communities present, or the nature of human activities in the area. The following list gives some of the types of descriptive information which may be of particular interest, but it is by no means an exhaustive list!

Biological Questions

Species Presence/Absence:

What animals and plants are found in the protected area? Is species diversity higher in certain areas or habitat types?

Priority species for management

It is never possible to study or monitor all species so priority species need to be selected. These species might be rare, threatened or endemic (restricted to the area), they may act as indicators for changes in the ecosystem as a whole, they may be umbrella species which, if protected will result in the protection of many others, or keystone species (see Box 14a), which play a key ecological role, or they may be sacred.

Species Distribution

Where are these plants or animals found and how do distribution patterns relate to environmental factors? What are the patterns of distribution of different vegetation types? Are there seasonal patterns of animal movement between habitat types or in and out of the protected area?

Species Abundance

How many plants or animals are there of interest in different parts of the protected area or in different habitat types?

Current Status of Species or Habitats

How well are they surviving? Are they in danger of extinction? How do the species inside the protected area compare in abundance and distribution to the same species elsewhere?

Long-term Trends of Species or Habitats

How are distributions and abundances changing over time? How will they change in the future?

Socio-economic Questions

Resident and Neighbouring Human Populations

How many people live in and around the protected area? Where do they live? Are populations increasing or decreasing? What is the ethnic composition and age structure of human populations? How do these populations earn a living?

Use of the Protected Area

What resources do people exploit in and around the protected area? Which parts of the protected area do people visit? Are resources harvested for domestic use or commerce? How are patterns of exploitation changing? Does exploitation have a negative impact on the resource?

Seasonal Activities

How do human activities such as farming or hunting vary at different times of year? Are there specific activities which are restricted to certain seasons?

Crop Raiding by Wild Animals

In some places wildlife has a negative impact on human cultivation or causes negative attitudes for other reasons. In order to deal with such problems you will need information on the severity of the impact. Farmers almost invariably overestimate the losses due to crop raiding or livestock killing, so these need to be quantified before management actions are taken.

Research priorities and design of research programmes

Obtaining this descriptive information is rarely simple and may actually require a major research effort. For example, if the management authorities in Salonga National Park suspected that there had been a major burst of poaching activity, they would have to undertake a sampling programme to estimate the total number of elephants in the park. They would have to plan their sampling program in such a way that it was representative of Salonga as a whole, so that their estimate would be accurate. They would need a good knowledge of how to sample, how to census elephants in rain forest vegetation, how to analyse their data and how to extrapolate their findings to the whole of Salonga.

The greater part of this manual is aimed at giving conservation managers and researchers the knowledge necessary to achieve such a task. The emphasis is on collection of good quality data to document plant and animal distributions and numbers and human activities in and around protected areas. Preliminary experimental design and data analysis are also covered. The Lopes program written by Peter Walsh is distributed with this manual to help with data analysis. A companion manual covering more aspects of data analysis is planned.

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Genetta tigrina

Phase 2: experimental design and hypothesis testing

In some instances managers will wish to follow up descriptive research with more detailed investigations of particular issues (research scientists will usually adopt this approach). For example, a general survey in Gabon revealed low chimpanzee densities in logged forest compared to unexploited areas (Tutin & Fernandez, 1984) but did not determine the reason for the decline. Chimpanzee numbers could decline for a number of reasons. It could be due to increased hunting pressure in areas with new logging roads; to decreased food availability resulting from damage caused by loggers to important fruiting trees; chimpanzees might have moved out of logged forest to avoid humans (in which case densities would increase in surrounding areas); or there might be a different reason for the decline (see Box 14g). This is an important issue for chimpanzee conservation, since the majority of the forests where chimpanzees are found will be logged in the near future and a manager might therefore decide to investigate this issue in further detail. Such a research project would normally be undertaken in a number of distinct steps which would typically be as follows:

Preliminary phase: observation that chimpanzee densities are lower in logged forests.

Step 1: Formulate one or more hypotheses, to explain this decline.
Our hypotheses in this instance might be:

- Hypothesis 1: Increased hunting pressure in logged forest results in chimpanzees being killed;
- Hypothesis 2: Damage to vegetation during logging results in decreased food availability;
- Hypothesis 3: Chimpanzees leave logged forest;

Step 2: Make predictions based on the hypotheses which can be used to assess their validity.
In this case these might be:

- Prediction from Hypothesis 1: Logging in areas such as reserves where there is no hunting will not result in a decline in chimpanzee numbers;
- Prediction from Hypothesis 2: Chimpanzee numbers will decline gradually after logging. Older and younger chimpanzees will die first, birth rates will fall and there may be a greater die-off at some stage after logging, during a period of ecological stress such as a particularly long dry season;
- Prediction from Hypothesis 3: Chimpanzee numbers will decline immediately after logging but densities in surrounding areas will increase.

Step 3: Collect relevant data.
We might choose to monitor chimpanzee populations in a reserve where hunting is effectively controlled. We could select an area scheduled for logging and monitor chimpanzee densities before, during and after logging in this area and in surrounding areas. We would have to select or develop an efficient census method and implement a sampling program designed to detect changes in density of the magnitude expected based on data from the preliminary observations of Tutin & Fernandez (1984).

Step 4: Make a preliminary summary of the data.
Compiled and analyse the data in order to assess whether or not there were changes in density in the logged areas and / or in surrounding unlogged areas.

Step 5: Analyse the data using statistical tests.
Use appropriate statistical tests to assess whether or not observed differences between sampling periods are statistically significant (such tests assess how much confidence can be assigned to the results).

Step 6: Accept or reject the hypotheses.
If there were no change in chimpanzee density as a result of logging in this study we would accept the first hypothesis. If densities showed a gradual decline and there was no detectable change in surrounding areas we would accept the second hypothesis. If there was an immediate decline in logged forest and an increase in surrounding areas we would accept the third hypothesis.

Of course, it is possible that none of our hypotheses is the correct one, or that a combination of factors result in the decline of chimpanzees. The better our knowledge of chimpanzee biology and of how logging changes the forest, the more likely we are to be able to develop sensible hypotheses and design sampling strategies to test them effectively.

Post hoc analyses

Given the lack of baseline information and the logistical problems of working in large forested protected areas this stepwise research design will be an unattainable luxury for most protected area managers except in unusual circumstances. However, the survey methods described in this manual have been developed by researchers with a good deal of experience of working in Africa's rain forests and they are predisposed to providing some of the information such an approach would produce. For example, when collecting data on density of animals it is recommended that you also collect information on human sign and vegetation types. This will enable you to evaluate whether or not there is a relationship between differences in animal density observed in different areas and these two factors, which, for example, have proved key determinants of elephant density in some parts of Africa (Barnes et al., 1991). This should enable researchers to maximise their efficiency when working in vast, unexplored study areas where no baseline observations are available to direct research.

Practical considerations and common pitfalls

Research priorities will obviously depend greatly on the specific aims of your conservation project and on the goals of the protected area in which you work. However, there are a number of practical considerations and pitfalls which are common to most research projects and should be carefully considered.

Practical considerations

The total time available

Protected area managers tend to have many different responsibilities which can come into conflict with and take precedence over research protocols. Furthermore, managers and researchers alike face many logistical problems associated with trying to work in remote, inaccessible areas. The time available for research must be carefully evaluated during the planning phases of any research project. In addition to time for field work, sufficient time must also be factored in for planning and logistics and for data analysis. It is also vital that you factor in time for unexpected events, such as malaria attacks, vehicle breakdown, bad weather, civil unrest, etc.

Time periods available

Your research needs to be geared to any other commitments you may have on a weekly, monthly or annual basis and to any seasonal effects on your ability to work in the field. For example, if seasonal flooding is likely to mean that certain parts of your study area will become inaccessible this needs to be worked into your research design. Time spent planning for such factors is always time well spent.

Study areas available

In areas where site access can be difficult be extremely careful when choosing study areas. A remote site which is only accessible along a road maintained by a logging company may become isolated in the middle of your study if the company moves on, changes its work plan, or goes out of business.

Personal motivation

Research in the African rain forests tends to be demanding both physically and mentally. If researchers are not interested and motivated to undertake a study they will probably not collect good information, resulting in much wasted time, effort and resources. If designing a research project yourself be sure to choose a subject which interests **you**. If planning a study which will involve research assistants think carefully about their level of education, training, interest and motivation. Too many studies have failed because research assistants were either poorly trained or motivated, resulting in poor quality data or even data sets that were made up whilst research assistants lounged under a tree in some quiet, secluded spot.

Common pitfalls

Underestimating the time required

Many research projects begin with unrealistic aims which are far too ambitious. If research design is based on overly optimistic predictions of time availability, accessibility, motivation, good health, etc. then it is unlikely that sufficient data will be collected to answer the questions being addressed. New study areas are easy to add but half-complete data sets are generally of little value. All researchers, particularly inexperienced ones, attempt to do too much!

Relying on equipment

Relying too heavily upon equipment which may stop working and cannot be repaired or replaced is usually a bad idea. Make use of technology but do not become a slave to it. When your GPS stops working be sure you can continue using a compass and topofil. Electronic equipment is particularly susceptible to the hot, humid conditions of the African rain forests.

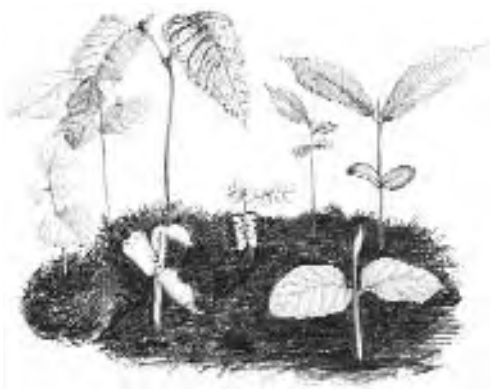
Involving too much identification

Surveys of a few, easily-identifiable species or species groups are much easier than surveys of whole communities. Carefully choose species or groups which are most likely to enable you to answer specific research questions.

Insufficient planning of how the data will be analysed

This is a common and major fault of many research projects. It is vital to carefully consider how data will be analysed during the planning phase of any research project, since this is likely to influence methods of data collection. Think carefully about what the study is intended to show and how you plan to use the data to do this.

Before you start to plan a research project you need to have a basic understanding of sampling theory and data analysis. The next chapter will lead you through these topics.



Seedlings germinating in a pile of elephant dung

CHAPTER 3
An introduction to sampling

Lee White & Ann Edwards

Introduction

It is easy to ask the questions: "How many hornbills are in the Dja Reserve?" "How often do male mandrills fight?" "How many wild mango trees are found in mature secondary forest?" But in most cases the exact answers can not be known. Instead the answers are estimated, as accurately as possible, using sampling methods. To sample something means to count the number of animals, plants, people, things or events that are observed while following a predetermined sampling methodology. If this methodology is well designed (see below) then the small number of animals, plants etc. counted will provide an accurate estimate of all the animals or plants that exist in the study area. The key to obtaining estimates you can trust is to start by developing a robust sampling design that may be unique for each new study. All the methods described in this manual are sampling methods.

Sampling area and sampling units

When sampling the density of plants or animals, the area to which your final density estimates apply is called the «sampling area» (when sampling a village of people, or the behaviour of a specific group of animals «population» is used instead of «sampling area»).

The sampling area may be, for example, an entire protected area, or just the area within a five kilometre radius of a single village. It may be restricted to a single habitat type, such as swamp forests, or steep hillsides, or it may include all habitat types. Similarly, the population to be sampled may include all villages in and around a protected area, or just one village or ethnic group of special interest. The researcher designing the study can define the sampling area or population in any way he or she chooses, but the sampling area must be clearly defined from the beginning and this definition must be borne in mind throughout the design and implementation of data collection and the analysis and interpretation of the results. The sample is the only entity for which you will be able to draw firm conclusions, since everything not included in the sample has not been studied (but see 'extrapolating your results below').

The smaller sections, in which plants or animals are actually counted, are called «sampling units». In this manual two types of sampling units are described, plots and transects (see below). Similarly, when selecting which households within a village to interview, or which animal to observe or weigh, each household or animal selected is a «sampling unit».

Plots and transects

Plots and transects are both sampling units. The main difference between plots and transects is that all objects detected within a plot are recorded, but with transects only objects detected from the centre line are counted.

A plot is an area of known size, within which every item of interest is counted. They can be any shape, but for convenience are generally either circular (measured a certain distance from a central point), square or rectangular. Because plots usually cover small areas, they are used to study things that do not move, such as trees. For example, botanical surveys often use plots of 1ha (100x100m) to study tree species diversity (see Chapter 8).

A transect is a trail, often many kilometres long, from which objects are seen and counted (see Chapter 11). The length of the transect is known, but the width of the transect is estimated, based on the distance to the observed objects. Transects are good for counting animals in forests because they allow a researcher to cover a lot of area quickly.

In the various methodologies which will follow in this manual advice is given as to whether you should choose to use transects or plots.

Representative sampling

The location of plots or transects, or the choice of particular households or plants should be «representative» of the larger sampling area or population. If the sampling units studied are representative it means they will contain the same kinds of things in the same proportions (or densities) as the sampling area. In other words, they accurately represent the sampling area. If the households interviewed are representative of the population different viewpoints will be expressed in the same proportions as would have been if every person in the village had been interviewed.

Of course, it may be difficult to be sure that the plots or transects are representative of the sampling area, or that a sample of households or individuals is representative of the whole population, since the area or population may themselves be poorly known. If this is the case, the most important aspect of sampling is to avoid introducing any bias because of the way you sample. For example, if you decide to do a survey of duiker numbers in a large, remote block of forest, but you only census close to roads, because this is the only access into your sampling area, your sample will not be representative of the area as a whole. Hunting pressure and habitat alteration by humans are likely to be centred along roads. Therefore the duiker population is likely to be lower in areas close to roads and your duiker population estimate will be biased, because it will underestimate the true density of duikers in the sampling area.

The simplest way to avoid bias is to locate the plots and transects at random (see below) within predetermined blocks or sections. When the locations of sampling units are chosen at random, and enough sampling units are sampled, then it is valid to assume that each habitat type or ethnic group, or each feature or attitude, has been sampled in the correct proportions and that the results are therefore representative of the entire sampling area or population.

Random versus systematic sampling

There are two ways to choose sample locations or units:

«Random sampling» is a method which is based on the principal that if each part of an area or each entity has an equal chance of being sampled it is unlikely that any systematic bias will affect the sample, and it should therefore be representative of the entire sampling area or unit. To do this you must establish a numbered system which represents all possible sample units (see below).

«Systematic sampling» is a method which is based on the principal that regular patterns rarely occur in nature. Therefore, systematic, or regularly-spaced sampling will often produce a fairly representative sample even though it is not truly random. It has the advantage that it is generally easier to locate plots which are evenly distributed than to locate random locations.

If, instead of randomly or systematically sampling plots, transects or other sampling units, they are grouped together for convenience, it is unlikely that data will be representative of the sample area or population. For example, if plots or transects for a vegetation study are intentionally located near trails or roads, the true variation of the forest will not be sampled. Existing trails and roads are not placed randomly. Major roads are purposely placed to avoid swamps and steep slopes, and to pass near villages. Therefore, when vegetation plots only sample areas close to existing roads they will be dominated by species which thrive in flat, dry areas which are heavily used by humans. They will not sample other habitats where species composition will probably be very different. As a consequence the data collected will not be representative of the sampling area as a whole.

Sampling bias

When results are not representative of the sampling unit they are intended to describe they are said to be biased. For example, if wildlife surveys are conducted along or close to roads the results are unlikely to be representative of your study area - they will be biased. Biased results will result in mis-representation of public opinion in questionnaire surveys, and in under- or over-estimates of animal or plant densities. This will probably lead to managers taking inappropriate management decisions, or in researchers coming to false conclusions.

Choosing locations for plots and transects

A simple procedure for picking random locations for transects and plots is as follows:

Transects

Transects are generally straight lines traced through the forest using a compass to maintain an exact bearing. In this manual transects are used primarily for animal censuses. Since animal densities often vary with vegetation type, it is a good idea to locate transects so they cross major topographic features, such as large rivers and mountain ridges, because this will give a more representative sample of all habitats. Alternatively you might choose a random direction, by picking a random number between 0 and 360. Then follow that bearing on a compass.

Having decided the bearing of your transects, draw a «baseline» which cuts through the area to be sampled, but perpendicular (90°) to your chosen bearing. This baseline will enable you to select random starting locations for your transects. Measure the length of the baseline on the map and then select a starting point along that line for each transect using random numbers (Box 3a).

Box 3a GENERATING RANDOM NUMBERS

To randomly select plots from a grid, or numbered households in a village, or a starting distance along a baseline we use random numbers. This is easy to do using a digital watch with a stop-watch on it. If your stop-watch measures to 1/100 of a second, run it for about 5s and then stop it without looking at the face. Take the last digit as the first number in your series and repeat this as often as is necessary. If the stop watch only works in 1/10 of a second, wait about 10s. If you wanted to select 10 numbers between 0-99 you should stop the watch 20 times and take each pair of figures as a random number in the desired range. For example, you might obtain the following series:

4, 8, 3, 7, 7, 6, 1, 5, 7, 0, 5, 9, 5, 9, 9, 3, 0, 4, 7, 3

Hence, the random numbers between 0-99 are: 48, 37, 76, 15, 70, 59, 59, 93, 04, 73

Note, numbers from 0-9 all start with a zero. If a number is repeated, is the case for 59, rather than sampling this plot or household twice you should discard duplicates and add another pair. If you only want numbers from 0-75 discard all pairs above the maximum and add additional pairs (in this example 76 and 93, as well as 59, would be discarded and more random pairs generated with the stop watch). If you wanted numbers up to 750 you would use combinations of three figures and discard any combinations greater than 750. etc.

If nobody in your team has a stop-watch you can write figures from 0-9 on pieces of paper, fold them, put them in a hat, and select them without looking in the hat having shaken them around to mix them. Replace each piece of paper after noting the number.

Plots

To locate a plot randomly within an area super-impose a hypothetical grid the size of the plot onto a map of the area to be sampled. Number these plots and select the number of plots you require using random numbers (Box 3a).

You can randomly select from any population in this way if all items can be numbered. For example, if you wanted to undertake a survey of how much fish was in the diet of people living in a town with 145 houses, you would first assign a number to each house and then select 25 (or whatever the appropriate sample size was - see below) houses at random for a questionnaire survey (see Chapter 15).

Alternatively, transects and plots can be located at set distances along a baseline, or every fifth (or tenth, or third etc.) plot can be selected from a grid. Such a «systematic» sampling design is unlikely to overlap the natural variability of the sampling area or population.

It is the responsibility of the researcher to state clearly in a final report what choices were made based on pre-existing information (for example, sampling the entire protected area using three blocks), and what choices were made at random (for example, choosing the starting points and directions at random within each block, and then locating the plots every 100 m along that line).

Dividing the sample area into blocks or sections

To assure that no section of a large sample area is left unsampled, the sample area or entity (such as a group of animals, or a town) can be divided into sections or blocks (Fig. 3.1), then the sample units (the plots, transects, households etc.) can be chosen randomly inside those blocks. For example, a large sampling area can be divided into north, middle and south sections, and each section can be sampled using 50 km of randomly placed 5-km transects. Achieving good «coverage» in this way will allow you to test whether density is homogeneous across the whole sample area. If it proves not to be then it is possible to «post-stratify» the sample (see below). Good coverage also improves your chances of obtaining a reasonable precision from a sample because variation between widely spaced samples is expected to be higher than between closely spaced samples.

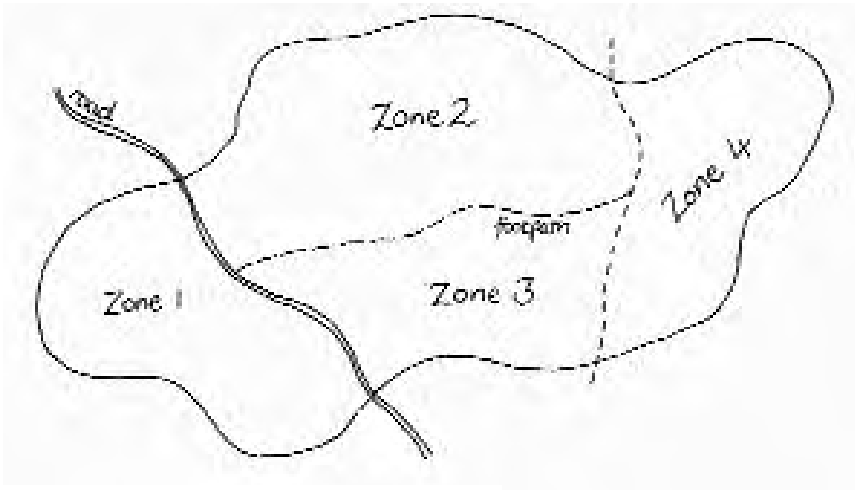


Figure 3.1. Dividing a large sample area into sections or blocks.

Stratification

To stratify something means to separate that thing into distinct categories (referred to as strata). Sampling should be stratified when you suspect that some environmental or human factor has a substantial influence on the value of the variable being sampled (see Box 3b). For example, in planning a study to estimate the abundance of elephants in your reserve you may suspect that elephant density is different in primary forest, secondary forest, and swamp forest. Stratifying the reserve into three strata based on these forest types and sampling separately in each stratum will yield two advantages. First, the sample for each stratum can be used to predict density (or abundance) specifically for that forest type across the entire reserve. The ability to make habitat specific density estimates may be important for guiding management or protection measures. Second, if density really does differ by forest type then samples within each forest type should cluster more tightly around their respective stratum means than they would around a pooled mean for all forest types. Thus, making separate density estimates for each stratum should produce a more precise estimate of elephant abundance for the entire reserve than making a single estimate for all forest types.

Any environmental factor can be used to define sampling stratum, including general spatial location, habitat type, or proximity to points of high human activity. Stratification can also be temporal. For instance, samples collected during the dry season might form one stratum and samples from the wet season constituting a second stratum.

Post-stratification

Sometimes during the course of a study you come to suspect that a certain environmental factor that you had not recognised or been aware of beforehand has an influence on density. In this case you can try stratifying samples according to this factor after the data collection is complete (this known as post-stratification). However, you should make separate density estimates for each stratum only if an appropriate statistical test shows that the samples from that stratum differ significantly from other strata. A test of significance is necessary because small samples taken from exactly the same underlying density can differ substantially. Only stratify if a statistical test indicates that the difference between strata is so large that it could not be due simply to sampling error. Incidentally, strata created before the onset of sampling should also be tested. If strata are not significantly different data from them should be pooled.

Box 3b STRATIFICATION: A CASE STUDY OF ELEPHANTS AND HUMAN DISTURBANCE

Between 1985-1988 Richard Barnes and his colleagues undertook a nationwide elephant survey in Gabon using dung counts (e.g., Barnes et al., 1995). They had planned to stratify the country by vegetation type using a large scale vegetation map. They undertook a pilot study and discovered that human density probably has a greater effect on elephant density than vegetation type, so they decided to stratify the country by human density instead. During their nationwide survey they discovered that elephant density was significantly higher in parts of the country with low human population, as they had predicted following their pilot study. They also discovered that there was a strong relationship between elephant density and the distance to the nearest road or village, and that there was a weaker relationship between elephant density and the proportion of secondary vegetation in an area (Figures B3.1 and B3.2).

From Figure B3.1 it is evident that elephant density increases steeply in the first 10 km from either roads or villages and less steeply in the second 10 km. Thereafter the fitted curve levels off. If you are planning an elephant census and there is a map of roads and villages for your study area, you could base your sampling methods on Barnes' findings and stratify your

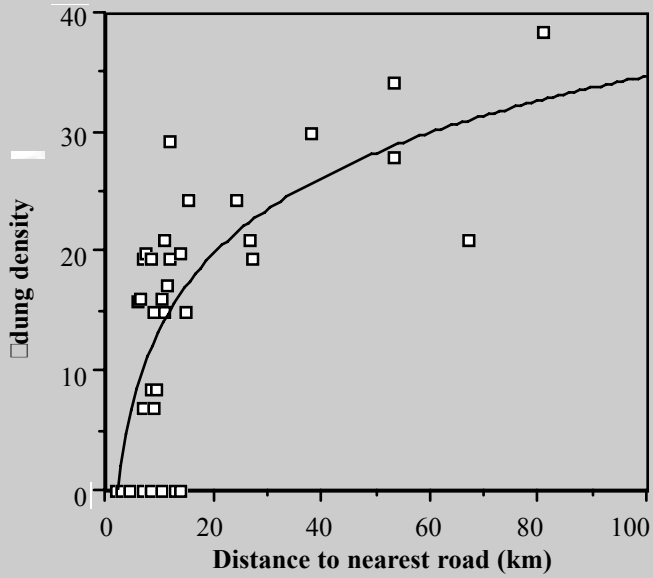


Figure B3.1: Relationship between elephant density and distance to nearest road. The relationship with distance to the nearest village is very similar.

transects according to roads and villages. You would do this by marking strata on your map as follows: 0-10km, 10-20km and further than 20km from the road. Then you would sample within each stratum in proportion to its expected abundance (stratum area multiplied by expected density).

Given the relationship between elephant density and secondary vegetation shown in figure B3.2, if you have a vegetation map with secondary forest marked on it, or if you can map secondary vegetation from aerial photographs or a satellite image of your site, you can further stratify within each 10 km band. If you then sample these various strata you will be able to extrapolate your results with a fair degree of confidence to your whole study area (see below).

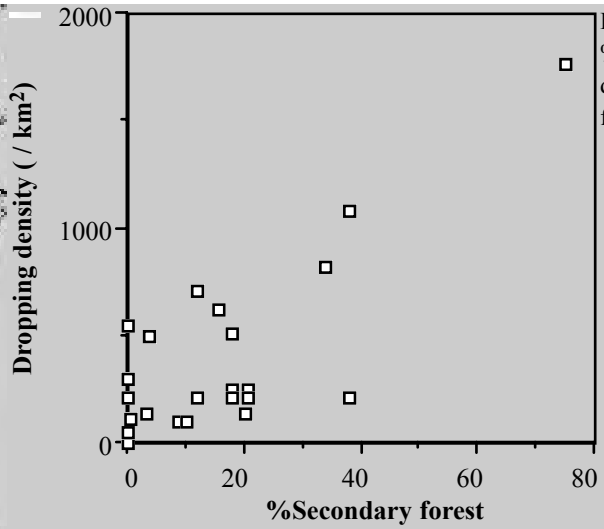


Figure B3.2: Relationship between %secondary forest and elephant density on transects at least 7km from the nearest road.

Figure B3.3 shows an example of a stratification of Gabon using 4 km contours located in concentric bands either side of roads and major rivers. A band 7.5km wide has been delimited either side of these access routes, where elephant density is assumed to be negligible. Areas of savanna and dense human habitation, where elephants were thought not to occur, are included in these low density bands. There is no vegetation map that includes all secondary vegetation on a national scale so this could not be added to the stratification.

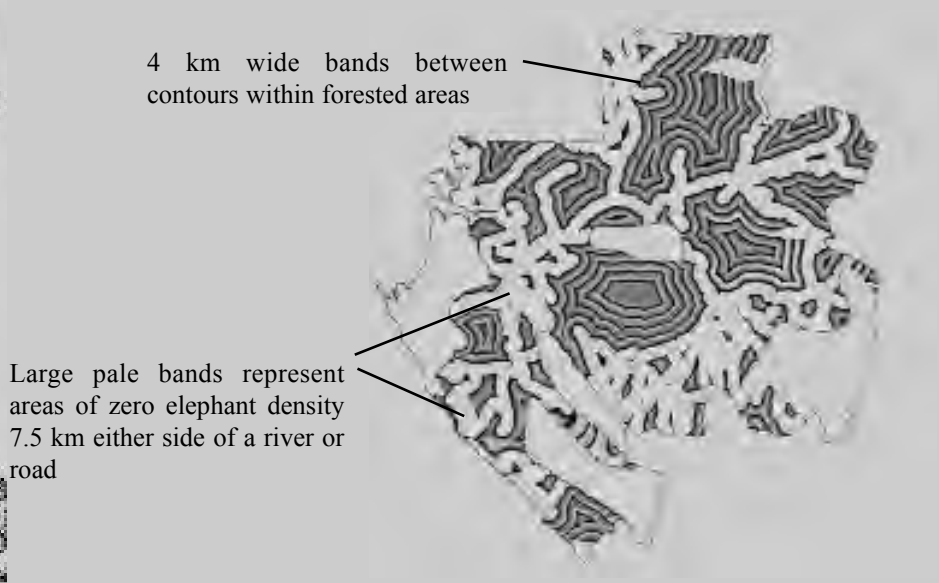


Figure B3.3: GIS generated map of Gabon showing computer-generated contours at 4 km intervals which represent distances from roads and major rivers in the known range of the elephant.

The importance of sample size

Sample size refers to how many transects or plots are sampled. For example, if you count trees in 50 plots then your sample size is 50.

Sample size is important because as the sample size increases, the precision of the estimate increases (Fig. 3.2), due to a decrease in the variance (see p. 42). In other words, the rule of sample size is that «more is better». However, this must be balanced against the amount of time, money and personnel available.

For example, if you pass through an area where the true density of an animal species is 10 animals/km^2, on the first pass through the area you may estimate 30 animals/km^2, or you may estimate 0 animals/km^2. If you make many passes through the area the mean of the passes should provide a better estimate than any one pass. In fact, as the number of passes increases the mean should converge on the true density.

The only way to decide if the number of samples is large enough is to use a statistical model of the sampling process to determine if the variance of the estimate is adequately small (see below). However, it is evident in Figure 3.2 that after about 20 censuses there is little change in the cumulative mean sighting frequency of white-throated monkey groups, a good indication that sample size was sufficient.

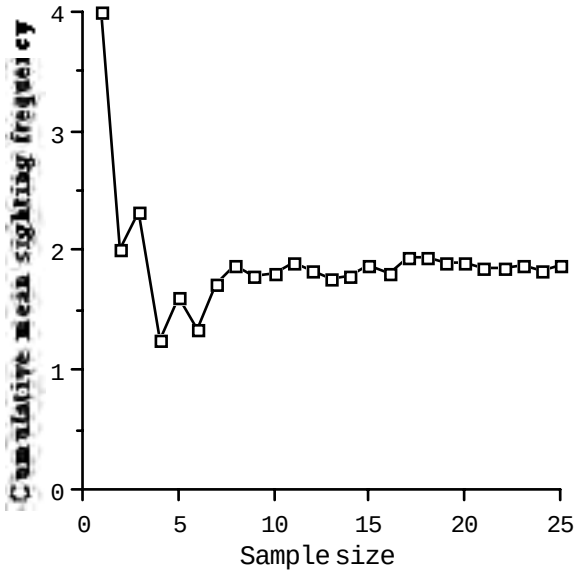


Figure 3.2. Plot of mean sighting frequency of groups of white throated monkeys, *Cercopithecus erythrogaster*, on a 5-km transect in Okomu Wildlife Sanctuary in Nigeria. Note that as the sample size increases, the mean estimate becomes more consistent.

Seedlings germinating in a pile of gorilla dung

CHAPTER 4
An introduction to data analysis and interpretation

Lee White & Ann Edwards

Data collection is not an end in itself. Data are not collected just so they can be filed away. After data are collected they need to be analysed and interpreted, and then made available to fellow managers or researchers in the form of reports or publications. Unless you complete all these stages you will have wasted your time.

Data analysis is the process of taking the numbers collected in the field and summarising them into a numerical or graphic form that is easy to interpret. This analysis or summary is called the 'results'. Results alone do not have biological meaning until they are put in a biological context. Data interpretation is the process of giving biological meaning to the results. This is done by evaluating the results in light of past research, and in the context of your own knowledge of the biological system you are studying.

The process of determining, «what do the data tell me?», and just as importantly, «what do the data not tell me?», makes data analysis and interpretation interesting and challenging.

Reliable results come only from reliable data

Results are only as good as the data they come from. Therefore, before doing any analysis, review the methods used to collect the data, and review the conditions under which the data were collected. Do the data represent quick guesses or the result of careful study? Were measurements taken accurately? Were observations made by well-trained field assistants? Were the methods used appropriate for the species being studied? What were the assumptions of the methods used (see below)? For each species studied, was the study undertaken at the appropriate times of the day, and the appropriate times of the year? Were enough samples taken in the right places throughout the sampling area in order to make them representative? Is the sample size large enough to provide narrow enough confidence limits (see below)?

There are no perfect methods nor perfect data. However, errors in data collection should be minimised whenever possible.

Assumptions

Whatever the methods of data collection and data analysis they all are based on certain assumptions. Assumptions are conditions that are considered to be true while the data are being collected or analysed.

For example, when plots are used to estimate the density of trees in a primary forest it is assumed that all the trees inside the plots will be counted, no trees outside the plot will be counted and no tree will be counted twice. These assumptions equate to meticulous data collection. When censusing moving objects, however, the assumption that no animal will be counted twice can not always be discounted even if you are careful during data collection. In both cases you will make the assumption (after appropriate stratification and random sampling) that the plots or transects sampled will be representative of the forest as a whole.

Different methods make different assumptions. When choosing between two methods, it is best to choose the method for which you can meet more of the assumptions. Theoretically you could use either line transect or plot sampling to estimate animal density in an area.

The plot method assumes you will see every animal in the plot. Plot sampling is not a good choice for estimating density in the forest because animals will often run out of the plot before they are detected. On the other hand the line transect method does not assume that you will see every animal within a given distance from the trail. Instead, the method assumes that you will see most of the animals near the transect, and fewer animals as the distance from the transect increases. When walking through the forest looking for animals this is a realistic assumption. Therefore, the assumptions of the method can be met and the results can be trusted.

Types of measurements

Three different types of measurements are described in this manual and it is useful to have an idea of what these are from the very beginning. Technically we say there are different levels of measurement.

Interval level measurements

These are what most people think of as «real» measurements, obtained by physically measuring, weighing or counting something, such that the magnitude of the difference between different measurements can be assessed. Interval level measurements can be either continuous or discrete. A variable is said to be continuous if it can assume an infinite number of values on some interval of the line of real numbers (numbers that can be expressed as fractions, such as a length or weight). Discrete variables can only assume an integer number of values so that between any two points on the real line there are always a finite (albeit large) number of discrete values (such as the number of trees in a plot or people in a village).

Ordinal level measurements

These are ranked measurements which make it is possible to say one measurement is bigger or more important than another. However, with ordinal level measurements it is not possible to assess the size of the difference. For example, the quantity of leaf fragments in elephant dung piles might be scored as zero, very few, few, common, and abundant. Similar scales might be used to measure things such as the open-ness of a forest under-storey, the age class of mandrills (where you can distinguish infants, juveniles, sub-adults and adults, but not assign actual ages in years and months), or the degree to which human populations living close to a reserve feel negatively about certain animals.

Categorical (or nominal) level measurements

These allow objects or opinions to be assigned to a specific category but these cannot be ranked relative to one another. For example, a gorilla nest might be classed as «zero», «herbaceous», «mixed» or «tree» (see Chapter 12), or a child may be a «boy» or a «girl», but these measures have no rank relative to one another.

Properties of ordinal measurements

Many of the methods described in this manual produce ordinal level measurements, so it is important to understand the properties of this type of data. Such information will almost always consist of a range of values. These can be visualised by plotting a frequency distribution (see Figures 4.1-4.4). A frequency distribution is a way of graphically presenting the distribution of the data collected.

If you take the time to study these frequency distributions, you will see that the patterns of distribution of the variables differs between examples. Can you suggest why this is? You need to understand in what ways these patterns differ if you are to make valid comparisons between your own samples.

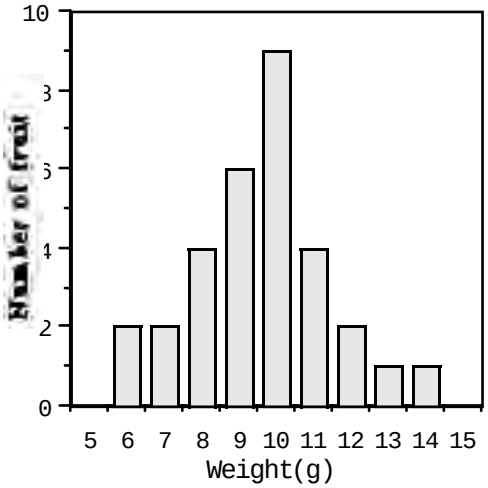


Figure 4.1: Weights of ripe fruits of the forest tree *Pseudospondias longifolia,* an important food for primates (R. Parnell, unpublished data).

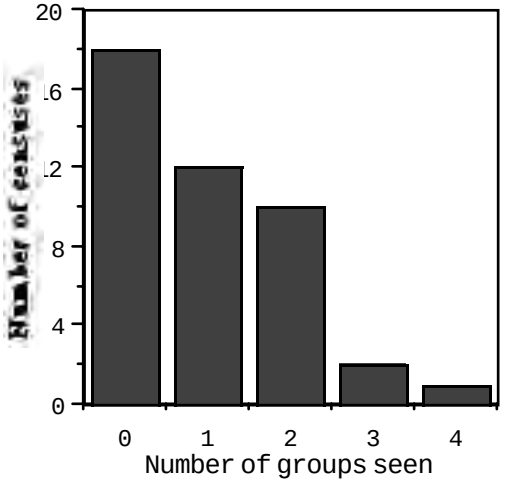


Figure 4.2: Number of groups of black colobus monkeys seen on 43 repetitions of a permanent 5-km transect (data from White, 1992)

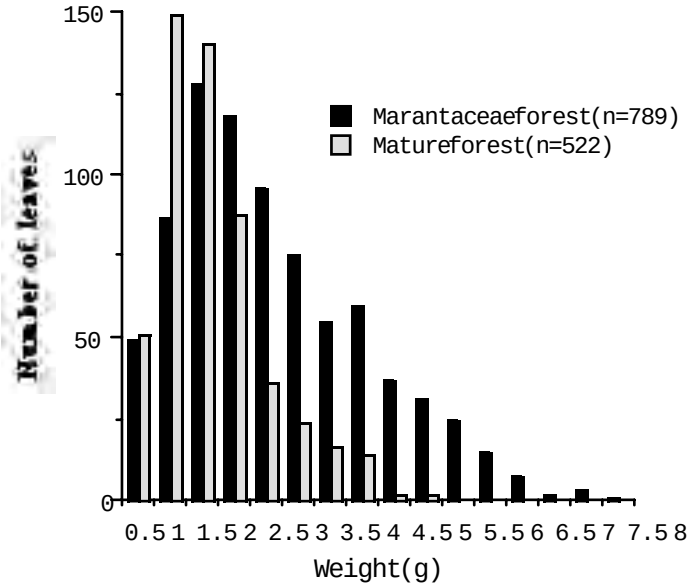


Figure 4.3: Weights of all leaves from 50 *Haumania liebrechtsiana* stems in two forest types in the Lopé Reserve, Gabon (data from White et al., 1995).

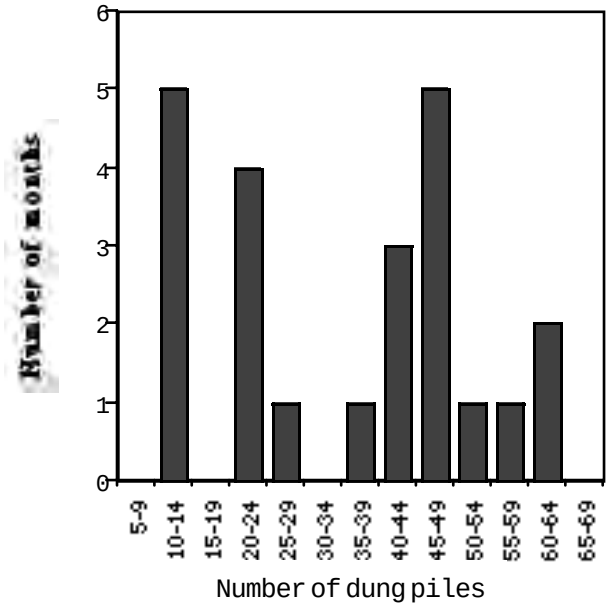


Figure 4.4: Number of new elephant dung piles each month on a 5-km line-transect monitored for 24 months in the Lopé Reserve, Gabon (from White, 1992).

Different frequency distributions

A frequency distribution is simply a representation of the distribution of the values of a particular data set. These data can be presented in a number of ways:

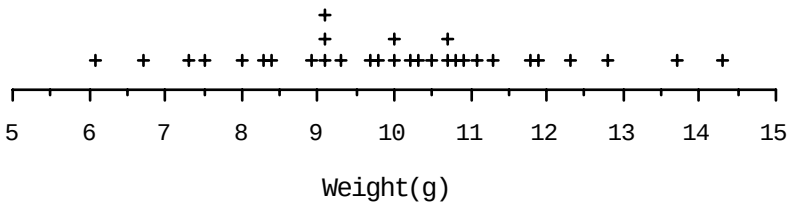
a) the raw data;
b) a lineplot of the raw data;
c) the data grouped into categories.

Figure 4.1 above is a graph of the data grouped into categories.

a)

Weight of fruits (g)	6.1	14.3	9.8	10.7	10.5	9.1	9.3
	11.3	10.8	8.3	7.5	10.0	13.7	10.9
	9.1	12.3	10.2	11.9	8.9	7.3	11.8
	12.8	8.0	6.7	10.0	11.1	10.7	9.7
	9.1	10.3	8.4				

b)



c)

Weight range (g)	6-6.9	7-7.9	8-8.9	9-9.9	10-10.9	11-11.9	12-12.9	13-13.9	14-14.9
no. of fruits	2	2	4	6	9	4	2	1	1

The frequency distribution describes the degree of spread (the range) of the values in a data set and the pattern of distribution of the data across this range. In Figure 4.1 the data are distributed more or less symmetrically on either side of a central cluster of values. In figures 4.2 & 4.3 the distribution is asymmetrical and in Figure 4.4 no pattern is evident. The nature of the frequency distribution can provide a great deal of information about a sample and changes in the frequency distribution over time can be used to demonstrate trends. For example, if the frequency distribution of the weights of elephant tusks confiscated from poachers were observed to gradually shift over several years towards lower weights, it would suggest that all large tusked (older) males had been killed and that poachers had started to kill younger males and females. Similarly, if the frequency distribution of the distances hunters travel from their village or camp shifted towards larger distances, it would suggest that areas close to the village have been over-hunted.

In many cases biological data sets are expected to conform to particular types of frequency distribution which have well understood properties. Some of these are described below.

The normal distribution

Figure 4.1 shows a frequency distribution of a continuous interval level variable that has been divided into discrete categories. If the occurrence of each event is independent of prior occurrences within the sampling unit (measurements are not influenced by one another) then the data are expected to fit a normal distribution. The normal distribution is always symmetrical around the mean (or average) value (see below) and can be described by a mathematical equation. The shape of a normal frequency distribution is determined by the mean and the standard deviation (a measure of the spread of the values - see below). Data such as the weight or body length of a sample of animals of the same species, age and sex, or of ripe fruit of any given species generally conform to the normal distribution, as do the distances of the houses in a village from the village square, or the distance people travel to hunt or collect forest produce, or the size of fields cleared for farming.

In some cases continuous variables differ from the normal distribution. When a frequency distribution graph is plotted it is not symmetrical but is distorted in some way. Such deviations from the normal distribution can provide interesting information for a manager or researcher. For example, tusk size in bull elephants in a given part of their range should be normally distributed. If poachers have killed most large tusked bulls the frequency distribution will be asymmetrical.

The Poisson distribution

Figure 4.2 shows a frequency distribution of a discrete interval level variable. When discrete interval level data are independent of one another and occur in a range which is theoretically limitless, they are expected to conform to the Poisson distribution (Figure 4.2 shows a typical Poisson distribution for a variable with a low mean, whilst Figure 4.3 is typical of data with a higher mean). Like the normal distribution the shape of the Poisson distribution can be described by a mathematical equation, but in this case the data are not distributed symmetrically about the mean. Rather, data are bunched on one side of the mean and there is a long ‘tail’ on the other side. The Poisson distribution has a very specific meaning. It applies to processes in which the probability of an event occurring is constant across time or space. Put another way, it is applied to processes in which there is a constant rate of occurrence per unit time, distance, or area. This is why the Poisson distribution pops up so much in random sampling situations: if events are randomly distributed they have a constant probability of occurring (across either time or space). For example, say you cut a ten kilometre transect and randomly distribute 40 elephant dung piles along the transect so that there is a mean rate of 4/km. Assume that you have a 50% chance of detecting any one dung pile so that on an average you will detect 20 dung piles on the 10 kilometre transect. What is the probability that on a particular trip you will detect only 12 dung piles? The Poisson distribution describes this probability.

The Poisson distribution can also be applied to events that occur with constant probability through time. For instance, if gorilla nests have a constant probability of becoming undetectable through time and a mean decay time of 65 days, then the probability of a nest becoming undetectable after 50 days can be calculated from the Poisson distribution for a mean of 65.

As the mean rate of occurrence gets larger the shape of for the Poisson distribution gets more and more similar to the Normal distribution. A major reason that the normal distribution gets used in statistical tests so much is that it is a good approximation to the Poisson, which is the probability distribution for randomly distributed events, but has some convenient mathematical properties that the Poisson distribution lacks.

To conform to a Poisson distribution a variable must have a mean which is relatively small compared to the maximum possible number of events in a sampling unit (i.e., the event must be rare) and the occurrence of each event must be independent of prior occurrences within the sampling unit. If the occurrence of one event enhances the probability of a second such event, you will obtain a clumped (sometimes called a contagious) distribution. If one event reduces the chances of a second such event in the sampling unit you will obtain a uniform (sometimes called a repulsed) distribution.

Data sets which tend to conform to a Poisson distribution include the number of primate groups or nest sites encountered on a transect, the number of dung piles decaying per unit time in a decay experiment, the frequency with which a rare animal such as a gorilla or leopard is killed by hunters, or the number of times per year that plantations close to a village are raided by elephants.

Other distributions

Figure 4.4 shows the number of elephant dung piles which appeared on a transect in Lopé during monthly counts undertaken over two years. No clear pattern emerges from the frequency distribution whereas one would have expected the data to conform to a Poisson distribution. It is possible that the sample size of 23 counts was too low and that this is the cause of the problem. However, inspection of the figure suggests this is unlikely. Observational data from Lopé show that the number of elephants seen in the forest correlates with the number of dung piles seen (White, 1994a) and that there is a distinct drop in elephant density in the long dry season, when elephants are said to move to swampy areas along large rivers elsewhere in Gabon. Hence, this distribution reflects the fact that elephant numbers are changing from month to month. If 23 censuses were undertaken in the dry season we might see a Poisson distribution corresponding to the first peak on the figure whilst 23 censuses in the wet season might give a similar distribution centred on the second peak. The end result however is a bimodal distribution, a distribution with two distinct peaks.

Bimodal distributions occur fairly frequently in biological data sets, particularly with temporal data in a seasonal environment. Another example would be the weights of bull elephants in museum collections. In this case there would be two distinct peaks corresponding to the distribution frequencies of forest and savanna elephant bulls, since forest elephants are significantly smaller than the savanna sub-species.

Another common distribution discussed in many textbooks on statistics for biologists is the binomial distribution, where there are two possible criteria which are mutually exclusive, as is commonly the case with genetic characters such as albinism. Some questionnaire data might conform to this distribution as will presence/absence data from plots.

Effect of sample size

If your sample size is small (see Chapter 3) it can be difficult to assess the frequency distribution of the data since the pattern will be diffuse (Figure 4.5). You could enhance the shape of the frequency distribution by lumping several weight categories together, for example, place all infants between 3.0 and 3.2kg in one category, all infants from 3.2 to 3.4kg in another, etc.

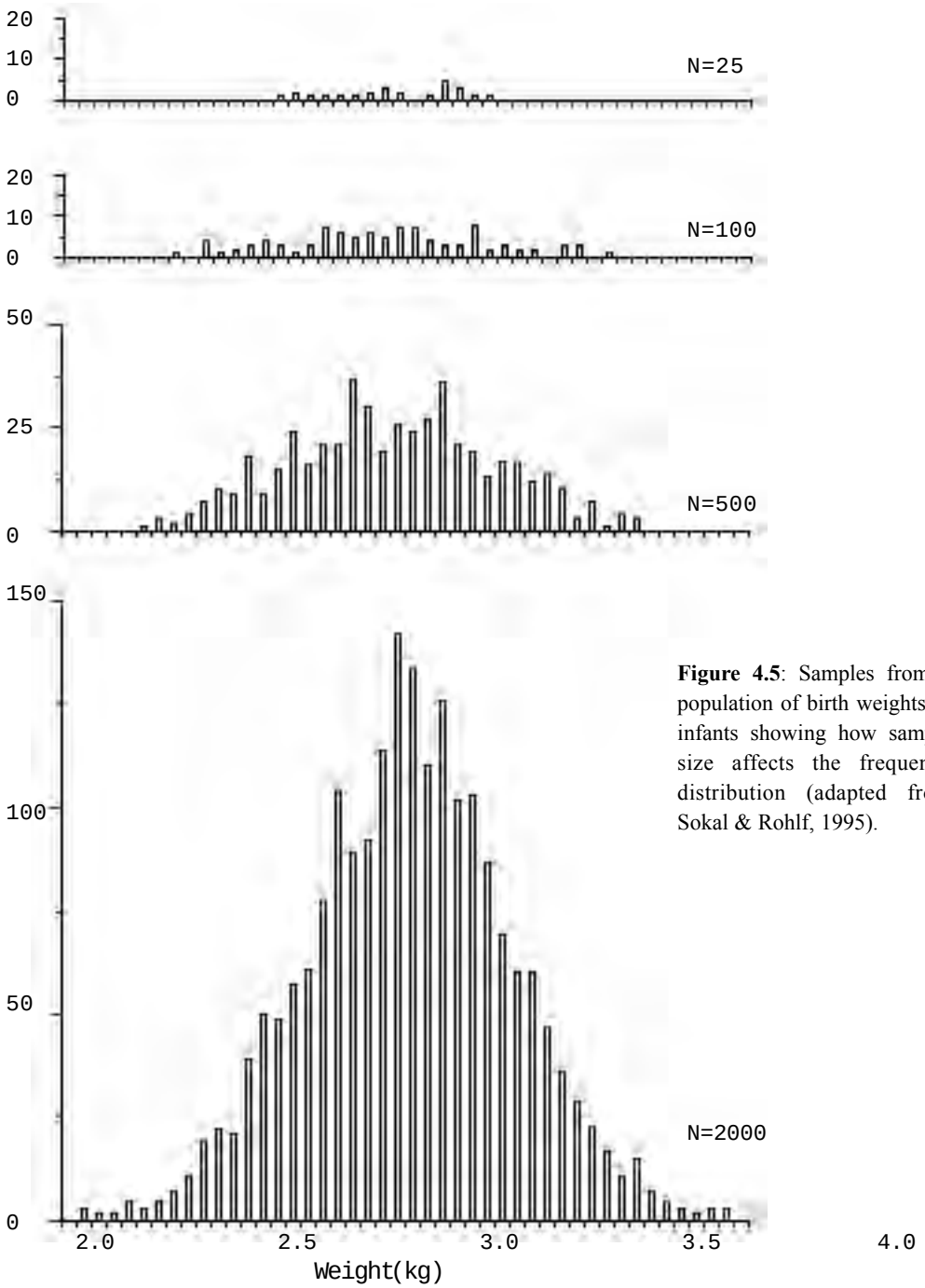


Figure 4.5: Samples from a population of birth weights of infants showing how sample size affects the frequency distribution (adapted from Sokal & Rohlf, 1995).

Basic statistics: mean, median, mode, range

Mean, median and mode are different ways of describing the "centre" of your data set. The range describes how variable the data set is. All data sets should be reported using indicators of both the "centre" and the variation.

Arithmetic mean

The «arithmetic mean» (commonly referred to simply as the 'mean' or 'average') is the most commonly used statistic. When a density or other result is documented in a report, that number is probably the mean, unless otherwise stated. The mean is calculated by adding all the data points in a sample and dividing by the sample size. When reporting the mean, "round off" the value so it has the same number of decimal places as your data.

For the example in Figure 4.2, there were 31 fruits:

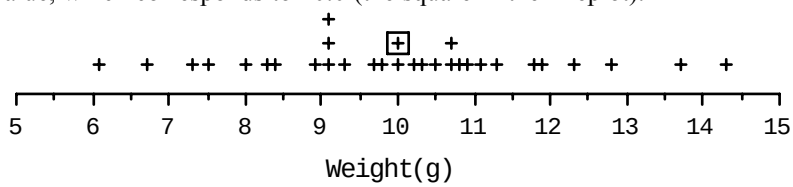
Weight of fruits (g)	6.1	14.3	9.8	10.7	10.5	9.1	9.3	11.3
	10.8	8.3	7.5	10.0	13.7	10.9	9.1	12.3
	10.2	11.9	8.9	7.3	11.8	12.8	8.0	6.7
	10.0	11.1	10.7	9.7	9.1	10.3	8.4	

The sum of the weights = 310.6g

The mean weight = 310.6 / 31 = 10.0g (actually 10.019)

Median

The median is a statistic which is particularly useful to describe frequencies which are not symmetrical about the mean. When the data are put in numerical order, the median is the value with an equal number of values on either side of it. Thus the median divides the frequency distribution into two halves. If we arrange the fruit weights above in order the median value is the 16th value, which corresponds to 10.0 (the square in the lineplot).



If data are only available in size classes the median value is the mid-point of the class in which the middle value occurs.

Mode

The mode is the most common or frequent value in a sample. On a frequency distribution this is the value corresponding to the peak, although sometimes two or more values are equally numerous and the distribution is referred to as bimodal or multimodal, respectively. For the fruit weights the modal value is 9.1g.

For normally distributed data with large sample sizes, the theoretical values of the mean, median and mode are the same, whilst for skewed distributions they can differ significantly. Figures 4.6 a & 4.6 b are symmetrical and the mean, median and mode are therefore the same. Figure 4.6c is asymmetrical and the mean, median and mode have different values. Say Figure 4.6c represents records of the number of times an elephant defecates per day. If you were asked one day to bet how often the elephant would defecate you would be best advised to choose the mode. If however you wanted to use these data to calculate elephant density (see Chapter 13) the most appropriate measure would be the mean.

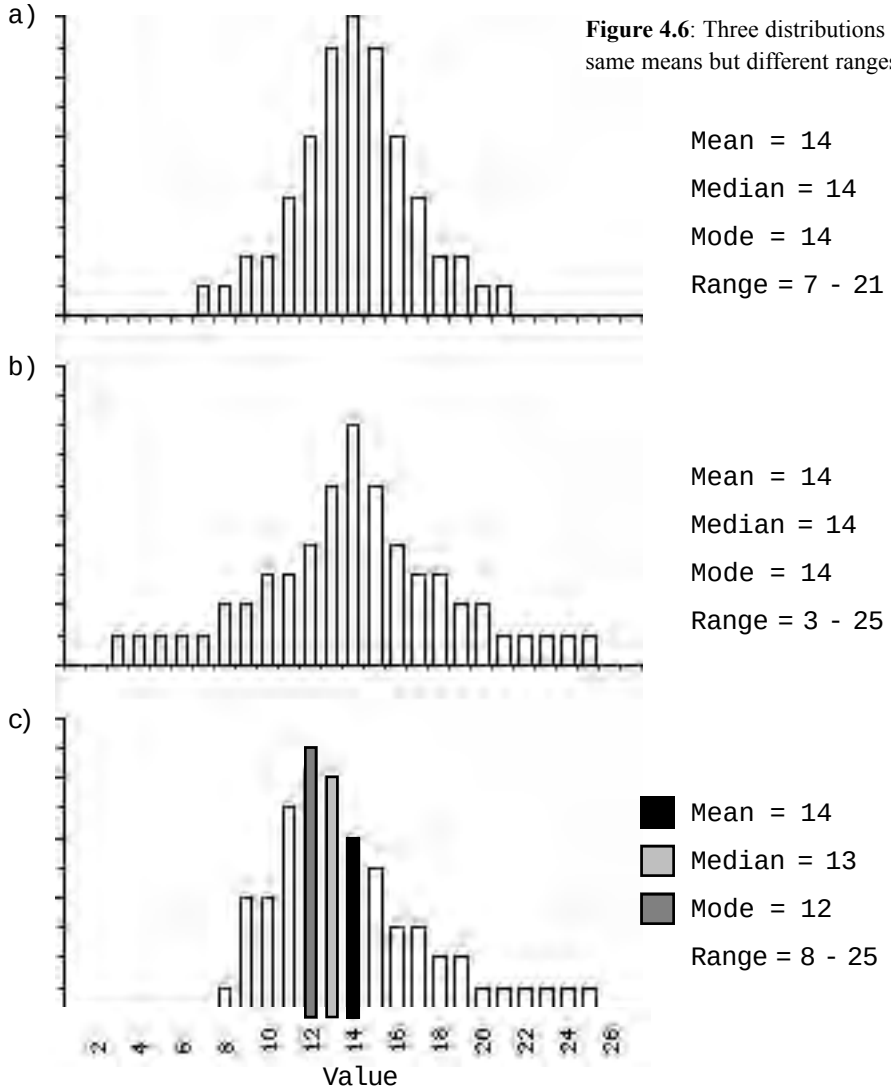


Figure 4.6: Three distributions with the same means but different ranges

Range

The range is the simplest way to present the variation in a dataset. The range is the maximum spread of all the data and is represented by the smallest and largest data points (Figure 4.6). Note that Figures 4.6a, 4.6b and 4.6c represent distributions with the same means but different ranges.

Accuracy and Precision

The accuracy is the closeness of a measured or computed value to the true value. The precision is the closeness of repeated measures to one another (the variance is a measure of precision). Samples can be accurate but not precise (have a mean close to the true mean but with high variance), precise but inaccurate (low variance but with a mean which is far from the true value) or both accurate and precise or inaccurate and imprecise. Your aim when sampling is to produce an estimate which is both accurate and precise.

Accuracy can be increased by ensuring the methods used are well suited to the species studied. For example, if you are counting the number of monkeys in each group you observe, but the females in the group tend to sneak away quietly while the males stay to noisily defend the group, you may consistently under-count the number of monkeys in each group. Therefore, your density estimate would have a negative bias (see below), resulting in low accuracy.

Accuracy also can be increased by reducing errors in the way information is collected through careful training of field assistants. For example, a poorly trained observer may correctly identify only 70% of the animals, whereas a better trained observer would get all identifications right (or note those instances where they were unsure).

Precision can be improved by increasing the sample size, and by consistently repeating the same technique (for example, using the same well-trained individuals each time).

High accuracy is needed for estimating total population size. A density estimate must be accurate before a manager can decide exactly how many individuals can be harvested sustainably from a population. High precision is needed when looking for trends since it allows the differences between two estimates to be detected. A manager must have precise data collected over many years before he or she can say with confidence that the density of a population is increasing, decreasing or staying the same.

Random Error and Systematic Error (Bias)

«Random error» is used to describe unexplained variability in the results of a study. Field studies experience many random errors. Random error will affect the precision (i.e., the variance) but not the accuracy of an estimate. This is because, if the error is truly random, there should be an equal number of incorrect values both above and below the true value.

«Systematic error» is used to describe a systematic or consistent increase or decrease in a result. This systematic influence is known as a «bias». If a result is consistently too high, then the result is said to have a positive bias. If a result is consistently too low, then the result is said to have a negative bias. A systematic error, or bias, will affect the accuracy of a result, but it will not necessarily affect its precision.

Statistical tests

Statistical tests are a means of analysing two or more sets of data and deciding if there are sufficient grounds to say that they are different. For example, you may census hornbills along line transects before and after logging. The mean encounter rates probably will differ in the two data sets. However, you need some means of knowing if the difference is due to random error, or if the density of hornbills changed during the course of logging. Statistics will provide the basis for making that decision.

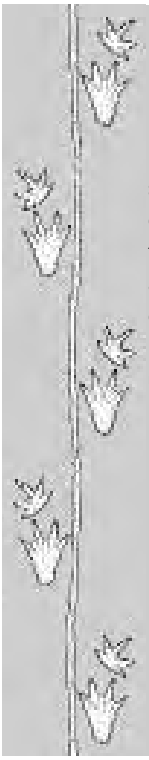
Chapter 16 will guide you through the use of some simple statistical tests. It is beyond the scope of this manual to lead you through all the statistical tests you might use. That would, quite literally, require an entire second volume. However, wherever possible we will describe how to analyse the data sets you will collect. The most complex analyses described in this manual are those for line-transect and "reconnaissance" (recce) density estimation (see Chapter 13). The computer program «Lopes», written by Peter Walsh, is provided with this manual to allow you to undertake these analyses. It works on computers running Windows '95 or Windows '98.

A fundamental concept in statistics is that of the frequency distribution of the errors in your data (see below). The first step in any statistical analysis is the plotting of a frequency distribution (e.g., Figure 4.6). If the errors in the data conform to a normal distribution there are a wide variety of tests, known as "parametric tests" that you can apply.

If the errors in your data deviate significantly from a normal distribution then they do not meet a primary assumption of all parametric tests and you can not reliably use them. In this case there are a different set of tests, known as 'non-parametric tests', which you can use. Sometimes your data will conform to a distribution which can easily be converted to a normal distribution by a mathematical transformation. For example, encounters of groups of primates along a transect, or decay of a sample of elephant dung (see above) will occur following a Poisson distribution. The data can be transformed to a normal distribution by taking the square root of the value of each observation, and then standard parametric statistical tests can be used to test for differences. If you are not sure whether your data fit a normal distribution it is safer to use non parametric statistical tests.

Statistics rarely give an absolute answer. Rather, they make a set of assumptions and tell you the probability that your two (or more) samples are taken from the same population given these assumptions. To report a statistically significant difference (you should avoid using the term 'significant' in your reports and papers unless referring to the results of a statistical test) the accepted standard is that there should be at least a 95% probability that the two data sets are different. If there is more than a 5% chance that they are from the same population any difference in the mean values cannot be considered statistically different.

The resolution of a statistical test varies as a function of the sample size and the variance of your samples. This is illustrated in Box 4a for primate census data. As a general rule unless you have a sample size of five or more plots, transect repetitions, weights etc. you will not be able to use statistical tests. For such small sample sizes you will only be able to detect very distinct differences, such as the difference between an area with no monkeys and an area with high densities. Minimum sample sizes for the various methodologies in this manual are suggested in the relevant chapters.



BOX 4a CALCULATING THE RESOLUTION OF CENSUSES.

When censusing dung, nests or animals your crude data unit is 'number of encounters per species per census'. For each species on any given transect this should conform to a Poisson distribution. If parametric tests are to be used first check that data do not differ significantly from that predicted by the Poisson probability. If they do not, use square-root transformed data for parametric tests. If testing for differences between transects first test to see if visibility differs between sites. If it does, adjust encounter frequencies for the transect with greater visibility down by a factor equal to the ratio of the two means of detection distance (see Chapter 13).

For some species encounter frequencies will be low and statistical resolution poor. Sampling resolution can be determined using a parametric formula (Anon, 1981; Janson & Terborgh, 1980; Skorupa, 1988):

$$D = 4\,(C.V.)\,/\,(N)^{0.5}$$

where,

D= minimum % difference in sample mean that indicates a statistical difference.
C. V. = coefficient of variation.
N = number of replicate samples.

Using this formula the difference in the mean encounter rate that indicates a statistical difference (ie., the resolution) can be calculated for each species in each site.

Hypothesis testing

There is a standardised, step by step sequence of reasoning that is always followed when doing a statistical test. Say, for example, we decided to census red colobus monkeys in a logging concession before and after logging, in order to see if logging had an effect on red colobus densities. The procedure we would follow is (see Box 16a for a more detailed explanation):

1) Set up a null hypothesis and an alternate hypothesis;

A null hypothesis is a hypothesis of «no difference». In this case it would be that «there is no difference in the sample populations before and after logging». This is usually denoted as Ho. Our alternative hypothesis, H_1 would be that there is a difference.

2) Look at the measurements obtained;

In order to distinguish between samples from two populations we need to be able to show that the mean (or median, or variance) of one is different to that of the other. If there is a large difference between the means and the data are tightly bunched on either side of the two means it is more likely that we will detect a difference than if the means are similar and the data are widely scattered. In this step you should display the data in the form of a frequency distribution. First look at the distribution of the two data sets. Then inspect the frequency distributions to see if they appear to be normally distributed (it is possible to statistically test whether data are normally distributed), to determine whether to use parametric or non-parametric tests.

3) Find out the probability of getting results at least as extreme as these if the null hypothesis were true;

Use a statistical test to assess the probability of obtaining two means (or medians or variances) as different as those obtained in your study.

4) If the probability is too small reject the null hypothesis and accept the alternative hypothesis.

In general, if the probability of getting a difference of the magnitude obtained in your study is 5% or less the difference is considered statistically significant and you would reject the null hypothesis. That is, there is a >95% probability that the difference is <u>not</u> due to chance or random events. Therefore, you would conclude that there was a statistical difference between red colobus densities before and after logging. Note that this does not mean for sure that there was a difference. A 5% significance level means that there is a 1 in 20 chance that the sample populations were actually the same and that the difference was due to random error in the sample. If the probability is a lot smaller, say 1% or 0.1% it would be a lot more convincing. There are several conventional methods for reporting statistical significance levels in reports and publications, as shown in Table 4.1

Table 4.1: Four conventions for denoting significance levels.

Less than 1 in 20	Less than 1 in 100	Less than 1 in 1000
5% level	1% level	0.1% level
$p<0.05$	$p<0.01$	$p<0.001$
$0.01<p<0.05$	$0.001<p<0.01$	$0.0001<p<0.001$
*	**	***

Classic statistical tests and likelihood tests

So called «likelihood» tests are being used more and more in preference to classical statistical tests. These tests involve resampling the data a great many times in order to compare alternative hypotheses. This is possible in great part because of the rapid improvement in computer technology, which makes it easy to do a large number of calculations quickly. Whilst the general hypothesis testing procedure is similar for these two types of tests there are some subtle differences:

- In classical tests the null hypothesis is that the samples are drawn from the same population. In likelihood tests the null hypothesis is that samples are drawn from populations with different means;
- Classical tests assesses how often we would observe such a high variance around pooled means if the samples really were from the same population. Likelihood statistics test how often we would expect to obtain the observed ratio of likelihoods if the null hypothesis were true.

When is it important to use statistical tests?

There are many instances when use of statistical tests will improve conservation management or research findings. For example, it may be considered important to monitor trends of bongo populations in a hunting concession. Your data may suggest that the population is declining, but in order to be sure and to convince sport hunters or politicians that management changes are necessary you need to know how sure you can be of your results. Alternatively you might want to assess the relative merits of two potential protected areas for the conservation of a rare monkey species. You undertake primate censuses in each area but can only make a choice between the two sites if you know how certain the data are suggesting one has a larger population than the other. As part of the same exercise you might have stratified your sampling by habitat type, but before making extrapolations to the whole of each area based on this information you need to test whether there are indeed different densities in the different habitat types you sampled. Statistical tests give you a rigorous, unbiased tool with which to demonstrate the quality of your information, to people who do not know your study site and have never applied the methods you have used.

More simple statistics: measures of variability and confidence limits.

Variance

Figure 4.6 demonstrates how two data sets with the same mean can have different ranges and frequency distributions. The «variance» of a sample is a measure which quantifies how widely or narrowly the individual variants are dispersed around the sample mean. In Figure 4.6 the top two graphs are of data distributed normally about the same mean, but which have different variances. If the values generally fall near the mean then the variance is low, as in Figure 4.6a. If the values generally fall both far and near from the mean then the variance is high. You can have more confidence in an estimate if the variance is low.

For the **normal distribution** the variance is equal to:

$$\text{Variance} = \frac{\text{Sum of (Each Value - the Mean Value)}^2}{\text{Sample Size - 1}}$$

It gives a measure of the «tightness» of the data about the sample mean. The difference between each value and the sample mean is squared because otherwise differences on either side of the mean would cancel one another out. The sum of the squared values is divided by the sample size minus 1 in order to correct for the fact that we are working with a sample and not the actual population. The method used to calculate the variance is different for different frequency distributions (see Box 4b).

Box 4b HOW TO CALCULATE COMMON STATISTICAL PARAMETERS

Normally distributed data:

The following statistics can be computed quickly using a hand-held calculator with statistical functions. Without a calculator the work is straightforward but time consuming. Work slowly and carefully. Keep all the sheets on which you do your calculations in case you need to check your work.

Ten 5km transects were walked looking for monkey groups. The following number of groups were seen on each transect.

Transect	Number of Groups	Transect	Number of Groups
1	8	6	7
2	0	7	3
3	3	8	5
4	1	9	3
5	6	10	4

Mean

The mean number of groups seen per transect is calculated by

1) adding all the groups seen on all the transects, and

2) dividing that number by the number of transects walked. (Note all the transects must be the same length.)

Mean = 8+0+3+1+6+7+3+5+3+4 / 10

= 40 / 10

= 4 groups/transect

Variance

Value = number of groups seen per transect

Sample Size = the number of transects = 10

Sum of (Each Value - the Mean Value)^2 = "Sum of Squares"

Variance = Sum of (Each Value - the Mean Value)^2 / Sample Size - 1

In order to calculate the variance, make a table of your data and calculate as follows:

Transect	Value	Value - Mean Value	(Value - Mean Value)^2
1	8	4	16
2	0	-4	16
3	3	-1	1
4	1	-3	9
5	6	2	4
6	7	3	9
7	3	-1	1
8	5	1	1
9	3	-1	1
10	4	0	0

Here, (Value - Mean Value)^2 = 58

Variance = Sum of Squares / Sample Size - 1

= 58 / 9

= 6.44

Standard Deviation

The standard deviation is the square root of the variance.

Standard Deviation = -Variance

= -6.44

= 2.53

Coefficient of Variation

The coefficient of variation presents the standard deviation as a percentage of the mean.

Coefficient of Variation = (Standard Deviation / mean) x 100

= 2.53 / 4.0 x 100

= 0.6325 x 100

= 63.25%

Standard Error

Standard Error = Standard Deviation / -Sample Size

= 2.53 / -10

= 0.80

Confidence Limits

Confidence limits combine the standard error with a probability factor. This probability factor allows you to say with a known degree of confidence (for example, 95%) that the true value should fall within a stated range of values (i.e., between and including the upper and lower confidence limits).

The confidence interval is the interval between the upper and lower confidence limits.

For sample sizes of 30 or greater, calculate the confidence interval using the probability factor Z. Z is the critical value of the standard normal distribution measured in units of standard deviations from the mean.

Probability	Value of Z
90%	1.645
95%	1.960
99%	2.576
99.9%	3.291

Confidence Interval = Standard Error x Z

For sample sizes of less than 30, calculate the confidence interval using a t-value taken from a statistical table (Table B4.1). Note that the t-value is determined by both the sample size and the specified confidence level.

Confidence Interval = Standard Error x t

If you desire 95% confidence limits on the above example then to find the correct t-value in Table B4.1, look down the column marked 95%, and across the row marked 9 degrees of freedom (df). (The degrees of freedom are usually determined by subtracting 1 from the sample size. The sample size was 10 in this example.)

This gives a t-value of 2.262.

Therefore,

Confidence Interval = Standard Error x t
= 0.80 x 2.262
= 1.81

Upper Confidence Limit = Mean + Confidence Interval
= 4.0 + 1.81
= 5.81

Lower Confidence Limit = Mean - Confidence Interval
= 4.0 - 1.81
= 2.19

These results mean that you can say that in 95% of cases the values 2.19 - 5.81 bracket the true mean.

Table B4.1 Student's "t"-values.
(df = degrees of freedom = sample size - 1)

df	90%	95%	99%	99.9%
1	6.314	12.70	63.65	636.620
2	2.920	4.303	9.925	31.598
3	2.353	3.182	5.841	12.941
4	2.132	2.776	4.604	8.610
5	2.015	2.571	4.032	6.859
6	1.943	2.447	3.707	5.959
7	1.895	2.365	3.499	5.405
8	1.860	2.306	3.355	5.041
9	1.833	2.262	3.250	4.781
10	1.812	2.228	3.169	4.587
11	1.796	2.201	3.106	4.437
12	1.782	2.179	3.055	4.318
13	1.771	2.160	3.012	4.221
14	1.761	2.145	2.977	4.140
15	1.753	2.131	2.947	4.073
16	1.746	2.120	2.921	4.015
17	1.740	2.110	2.898	3.965
18	1.734	2.101	2.878	3.922
19	1.729	2.093	2.861	3.883
20	1.725	2.086	2.845	3.850
21	1.721	2.080	2.831	3.819
22	1.717	2.074	2.819	3.792
23	1.714	2.069	2.807	3.767
24	1.711	2.064	2.797	3.745
25	1.708	2.060	2.787	3.725
26	1.706	2.056	2.779	3.707
27	1.703	2.052	2.771	3.690
28	1.701	2.048	2.763	3.674

Data sets conforming to a Poisson distribution.

If you predict that the frequency of monkey encounters will conform to a Poisson distribution then the variance for a Poisson distribution is equal to the arithmetic mean.

= 4 groups / census.

When the mean is large the Poisson distribution closely resembles a normal distribution and the methods described above for a normal distribution can be applied. If the mean is small but the product of sample size and the mean is >30, the standard error can be calculated as:

SE = -(sample mean /sample size)
= -(4/10) = 0.63

Hence, the encounter rate would be reported as 4 groups ± 0.63 per census.

If the product of the sample size and the mean is <30, tables are available to give confidence limits for samples with different means (Table B4.2)

Table B4.2: 95% confidence limits for the Poisson distribution.

Mean:	Lower limit	Upper limit	Mean:	Lower limit	Upper limit
0	0.0000	3.69	16	9.15	25.98
1	0.0253	5.57	17	9.90	27.22
2	0.242	7.22	18	10.67	28.45
3	0.619	8.77	19	11.44	29.67
4	1.09	10.24	20	12.22	30.89
5	1.62	11.67	21	13.00	32.10
6	2.20	13.06	22	13.79	33.31
7	2.81	14.42	23	14.58	34.51
8	3.45	15.76	24	15.38	35.71
9	4.12	17.08	25	16.18	36.90
10	4.80	18.39	26	16.98	38.10
11	5.49	19.68	27	17.79	39.28
12	6.20	20.96	28	18.61	40.47
13	6.92	22.23	29	19.42	41.65
14	7.65	23.49	30	20.24	42.83
15	8.40	24.74	35	24.38	48.68

Data sets which differ from both normal and Poisson expectations.

Data sets which do not conform to normal or Poisson distributions are fairly common for many of the sorts of studies described in this manual. This makes calculation of confidence limits difficult. In these instances you should use a non-parametric bootstrapping method as described in the main text (page 48).

For example, say you undertook 32 primate censuses and the mean number of encounters was 3.3 groups of red colobus per census. You would put your 32 data points into a computer. It would then pick 32 samples at random from your data and calculate a new mean (any point can be sampled several times - they are not skipped once sampled). It would then repeat this process another 999 times and the 25th and 926th smallest values respectively represent the lower and upper confidence limits.

To undertake such an analysis you need a computer since it would take far too long to do this by hand.

Standard deviation

The «standard deviation» has a very intuitive meaning; it is essentially the average deviation from the mean. For normally distributed data it equals the square root of the variance. When reporting a population's mean and variance it is standard practice to write it as the mean plus or minus the standard deviation, for example, 30±7.8. (Note that the standard deviation carries the same units as the mean, for example, elephants/km^2, or *Ceiba* trees/ha)

Coefficient of Variation

The «coefficient of variation» shows the standard deviation as a percentage of the mean. This single number indicates how widely or narrowly the values are clustered around the mean, regardless of whether the mean is large or small. The coefficient of variation is a way of describing numerically what Figures 4.6a and 4.6b illustrate graphically and allows you to compare variation for samples of different means.

Standard Error

If you took repeated samples of a given size from the same population how strongly would you expect the sample means to cluster around the population mean? Intuitively, you would expect the strength of clustering to depend on the sample size. Really large samples would do a very good job of estimating the population mean, so all sample means would be tightly clustered together. However, the mean for a small sample might, by chance, deviate from the population mean so that means for repeated samples would not be tightly clustered. The standard error is a mathematical way of expressing this intuitive idea. It is essentially the average deviation of sample means from the population mean. The standard error is calculated by using the within sample standard deviation as an estimate of the variability of the underlying population. This estimate of population variability is then weighted by dividing by the square root of the sample size (if you find the square root confusing just think of it in terms of dividing the variance by the sample size). Thus, for a given within sample standard deviation, the standard error decreases inversely with the square root of sample size.

Confidence Limits

Confidence limits are calculated values that fall above (upper confidence limit) and below (lower confidence limit) the mean. Confidence limits are a direct reflection of the variability of the data. Highly variable data lead to greater distance between the upper and lower confidence limits. This distance is called the confidence interval. The biologically true value of the thing being estimated (say, the density of elephants) has a known probability of falling within that confidence interval. That probability is called the confidence level and is routinely set at 95%. This means that if we repeated our observations 100 times (with the same samples sizes) we would expect our calculated confidence limits to bracket the true mean 95 times.

For example, if the mean density of elephants is estimated to be 2.1 / km^2, and the upper and lower confidence limits are 3.1 / km^2 and 1.1 elephants / km^2 respectively, we can conclude with 95% probability that the true density is between 1.1 - 3.1 elephants / km^2.

The confidence interval is important because it describes the precision of an estimate. It conveys how strongly the empirical data support a given estimate. If the data support an estimate strongly then confidence intervals are narrow. If data do not support an estimate strongly then confidence intervals are wide.

The are many ways to calculate confidence intervals. A simple approach is to assume that the means for repeated samples of the system are normally distributed with a standard deviation equal to the standard error for the estimate. The variable z is the distance from the mean (measured in standard deviations) of the standard normal distribution within which a critical percentage (e.g. 95%) lies. Now the standard normal distribution has a standard deviation of 1. Therefore, if we multiply z by the standard deviation of our normal distribution (the standard error of our data) we get the distance within which the critical percentage of our normal distribution lies. For example, the value of z for a 95% confidence level is 1.96. Thus, if our estimate is x with standard error y then the lower 95% confidence limit is x-1.96y, the upper 95% confidence limit is x+1.96y, and the confidence interval is (x-1.96 • X • x+1.96y), where capital X is the true value. A variety of other similar, but more sophisticated approaches use the standard error of your parameter estimate to establish confidence intervals.

Alternatively, resampling methods such as non-parametric bootstrapping can be used to place confidence limits on parameter estimates. In non-parametric bootstrapping you assume that the variability within your sample is a good approximation of the variability within the population from which the sample is drawn. You then simulate the process of drawing repeated samples from the true population by drawing repeated «psuedo-samples» from your original sample. Each new psuedo-sample is the same size as the original sample and is constructed by randomly choosing values from the original sample. Values are drawn with replacement: that is, a value that is drawn once is placed back into the «pool» and can be drawn repeatedly in a single sample. The parameter estimate (usually the mean) is made for each psuedo sample and the distribution of these estimates is used as an approximation if the distribution of estimates you would get by actually sampling repeatedly from the real population. 250 to 1000 psuedo-samples are usually considered sufficient to get a good estimate of confidence limits. If you perform 1000 psuedo-samples then the lower 95% confidence limit for your estimate is just the value of the 26th lowest psuedo-sample estimate. The upper confidence limit is the 975th highest psuedo-sample estimate. In general the lower and upper confidence limits are respectively n(1-a)/2+1 and n-n(1-a)/2, where a is the desired confidence level (represented as a proportion, e.g. 0.95) and n is the number of psuedo-samples. Confidence limits can be displayed in graphical form (Figure 4.7).

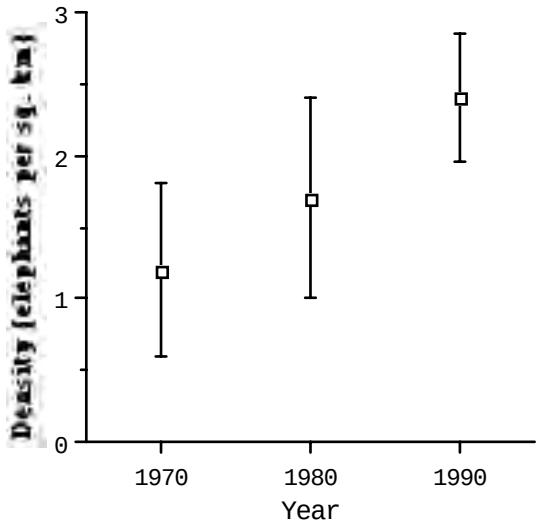


Figure 4.7. Mean density estimates, with upper and lower confidence limits, for three sampling periods.

Extrapolating from your results: what you can and can't say

What You Can and Can't Say about your Results

It is important that you make a clear distinction between what you can and can't say about the results from your study. What you can say is «given the methods used», the results «from this area», «collected at this time», «under these conditions» are as follows. You can not say anything more than that with certainty.

If you studied animals in a limited area, or in a single habitat type, then you cannot claim to have results for animals in larger, or more diverse, areas. For example, if you estimated the densities of elephants in primary forest in a 50 hectare section of a 200,000 hectare park, then you should not claim to know the density of elephants throughout the whole park, which contains swamp forests, secondary forests and patches of grassland.

The results of your study are accurate only for the time you undertook the study. For example, if you estimated primate densities only during the dry season, when fruits may be generally less abundant in the primary forest than in the secondary forest, you may find that primate densities were higher in secondary forest than in primary forest. Given those results you cannot state that primates are always found in secondary forests more than primary forests, because in the wet season this conclusion might not be true.

It is important to be very clear about when and where you undertook your study because sometimes your results may be affected by factors that might not become obvious until years later. For example, if you are estimating pig densities in the drier, western end of a park, you might not realise until years later that you counted them during an unusually dry year when they spent an unusually large amount of time in the wetter, eastern end of the park. After rainfall patterns have been monitored for many years, the discovery of low pig densities in the western end of the park during the year you studied them will be a very interesting finding. At that point your study will be most helpful if it can provide enough detailed information to make comparisons with later studies.

The important point to remember is to report the conditions of your study (time of day, time of year, locations, habitat types, average temperature and rainfall, etc.) as accurately as possible, and to report your results only in relation to those conditions.

The Process of Extrapolating from your Results

You can speculate and discuss the implications of your results (what the results suggest but do not show conclusively), and extrapolate the results of your study to larger areas (draw conclusions, based on the data, about areas for which you do not have data), as long as you clearly explain the steps you took on the way to making your conclusions.

This kind of data interpretation is not the same as results, and should not be presented in a «Results» section of a report (see Chapter 17). Results are based on field data and should be presented in the «Results» section without interpretation. A discussion of possible meanings of the results, and extrapolated estimates, are presented in the «Interpretation and Discussion» section, where the reader is free to agree or disagree with your ideas.

In the primate example above, you can suggest that a possible reason why primate densities were higher in secondary forest than in primary forest was because during the dry season more fruit may have been available in the secondary forest than in the primary forest. Given this possible relationship between fruit abundance and primate densities, you can then suggest that primate densities may be higher in primary forest during the wet season, when fruit is more abundant in the primary forest.

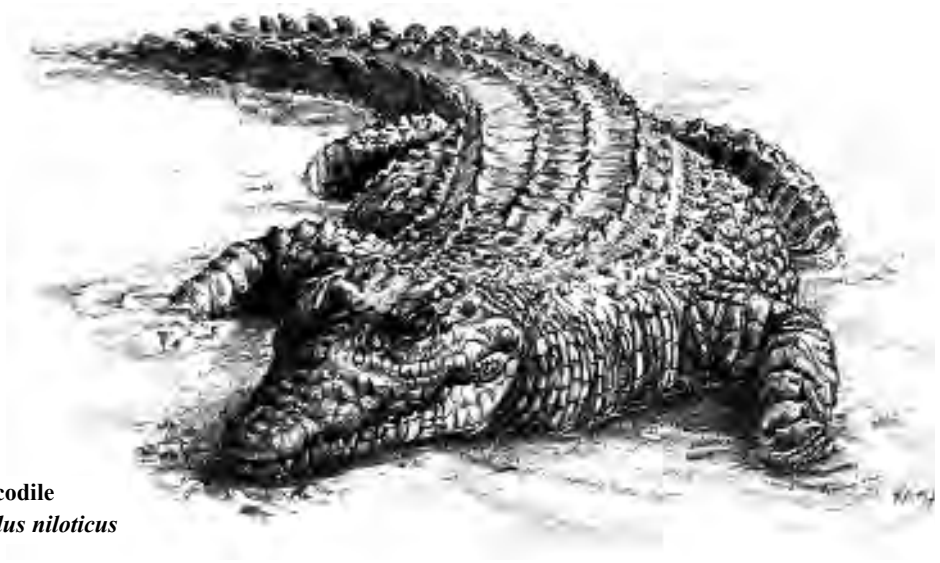
Note carefully that you have no data on fruit densities, and no data on primate densities in the wet season. However, the relationships you suggest are possible, and interesting to think about. And as long as you state clearly that you are only suggesting these relationships, and not documenting them as something you have observed, it is acceptable to put them in the «Interpretation and Discussion» section of a report.

In the elephant example above, you may be asked to estimate the density of elephants in the entire park. You can do this by extrapolating from your data, stating one assumption after another until you have enough assumed information to make a park-wide estimate. This is acceptable as long as you state your assumptions as «educated guesses», rather than as well-researched «facts».

For example, you know that the density of elephants in your study area was 0.4 elephants/km^2. Your first assumption can be that the density of elephants in all primary forest throughout the park is 0.4 elephants/km^2. Next, based on opportunistic observations noted in your field notes or log book, you assume that the density of elephants in secondary forest is half the density in primary forest (i.e., 0.2 elephants/km^2), the density of elephants in swamp forest is one fourth the density in primary forest (i.e., 0.1 elephants/km^2), and that there are no elephants in grasslands (i.e, 0 elephants/km^2). If the habitats in the park are found in the following proportions: primary - 75%, secondary - 10%, swamp - 10%, grassland - 5%, then you can assume the park-wide density of elephants is:

$$(0.4)(0.75) + (0.2)(0.10) + (0.1)(0.10) + (0)(0.5)$$
$$= 0.3 + 0.02 + 0.01 + 0.0$$
$$= 0.33 \text{ elephants/km}^2.$$

Producing estimates from incomplete data can be dangerous because the extrapolated «result» can be misused. Even though in your report you may clearly distinguish between what is known as a result of careful study, and what is speculation, your park-wide «estimate» of elephant densities may become valuable information. Such information may be used without caution by people who do not respect that the number is only an «estimate» and not the result of a careful, park-wide study.



Nile crocodile
Crocodylus niloticus

CHAPTER 5
Making observations and recording data.

Ann Edwards, Alan Rabinowitz & Lee White

Introduction

Having defined your research questions, making observations and recording data are the next steps in the research process. Observations of plants, animals, events, landscapes, disturbances etc. provide the data from which management questions are answered. However, it is a long road between making an observation and confidently answering a question of concern to management. If you are to negotiate that road successfully, you need to record relevant observations accurately and fully. If you are to achieve this you will have to think carefully about how to record observations and make the effort to do so rigorously. Making observations and recording information in a conscientious and rigorous way is something that must be learned and practiced, since poorly recorded observations can be useless or even misleading.

All managers and researchers working in reserves should make it a habit to systematically record observations of interesting or unusual events in a field notebook. Otherwise the details will be forgotten. It is not possible to record every observation, so you should define priorities. In some cases priority categories will already be defined by guidelines issued to wildlife managers by their head office, perhaps stipulating what information to include in monthly activity reports. If there are no clear guidelines, or if the existing guidelines need to be modified, you should think about and discuss the types of information that can be collected, which will help to answer specific management (or research) questions.

Once priorities have been determined, all researchers and managers should record the standardised information. However, collection of additional, unusual information should be encouraged. For example, a cape fur seal, *Arctocephalus pusillus*, was recorded off the coast of Gabon by conservationists in the Gamba complex of protected areas in 1996 (Thibault, 1999). This is the northernmost record of the species and it was not previously on the list of key species for the reserve. Staff with special interests, such as birds or butterflies, should be encouraged to keep written notes of their specialised observations.

Making observations and accurately recording information

Most information (including numbers) is interpreted in the mind of the observer before it is written down for others to see and interpret. The way events are seen, interpreted and recorded must be carefully controlled, so that whoever uses the information later will have no doubts about what it means. Bear in mind that the notebooks of scientists such as Charles Darwin have been catalogued and stored in libraries and can still be consulted today. You should aim to produce notes which you would be proud to have people consult in the future. With this in mind, it is of paramount importance that your notes should be clear and legible, and ordered in a logical fashion - not only for future generations, but for your own convenience. There is nothing worse than looking up a particular, half remembered event in an old notebook only to find that you cannot decipher your own notes! (see Figure 5.1)

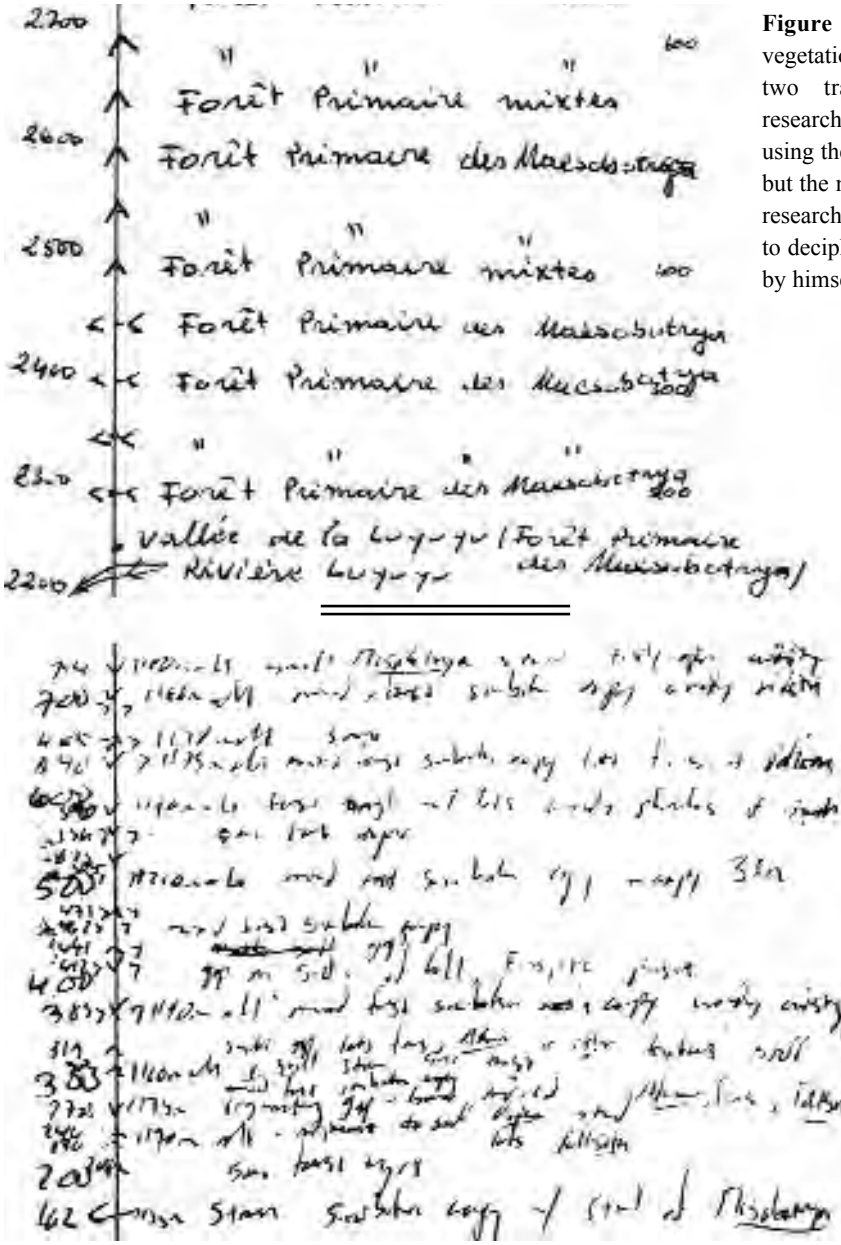


Figure 5.1: notes on vegetation changes along two transects by two researchers. They are both using the same data format but the notes of the second researcher are impossible to decipher (probably even by himself!).

The first step to taking good notes is to be a good observer. You must be clear about what it was you saw or heard, or just as importantly, what you didn't see or hear. Try to be as specific as possible while collecting the information (i.e., making the observation). When making an observation, separate in your mind what you have actually seen (or heard, smelled, etc.) from the possible implications of what you have seen. For example, if you see a monkey eating a bird, do not assume that the monkey killed the bird. The monkey may have found the bird when it was already dead. Record things exactly as you see them (i.e., the monkey was eating a bird). Do not assume you know what happened before (i.e., the monkey killed the bird), or what will happen next.

It is a good habit to record your thoughts, as long as they are clearly separated from what you actually see. For example, you can say a fruit was bigger than usual, or an animal was found in an unusual place. But this should only be recorded as a comment, clearly separated from the actual measurement or location. Later, by comparing actual measurements, you will be able to determine if the fruit was in fact bigger, or the animal was indeed in an unusual location. Recording what you see instead of what you think may be happening is particularly important when recording animal behaviour (see Chapter 14).

A second thing to remember when making observations in the forest is to focus on the important details first. Since animals usually flee when approached by people, try to focus on important identifying marks such as colouration, size, habitat and method of travelling. For birds, look at colouration, tail, wing and bill shapes, and markings around the eyes and throat. Review guide books to learn how to separate different species using simple distinguishing marks. Unusual behaviours should be given particular attention. Similarly, note important information first. When using data sheets fill in as many boxes as you can. Then add comments as necessary to allow the information in the boxes to be interpreted correctly.

Never attempt to guess the "right answer". It is better to say, "I don't know", and record nothing, than to record information incorrectly. Final reports may contain information about unidentified animals, or inconclusive observations, as long as the uncertainty is clearly stated. But no final report should contain untrue information. Incorrect data are inexcusable.

When recording information systematically, it is important to record what you didn't see as well as what you did see. Reporting that no animals were found in an area which was searched, or that no trees were in fruit, can be important. Such findings can suggest the question, why is this area without animals or fruit?

When recording measurements decide before you collect any data how accurately each measurement should be recorded. For example, if you are measuring the diameter of trees to the nearest centimetre, then the diameter of a tree measuring 25.3 cm should be recorded as 25 cm. Or, if you are recording measurements to the nearest tenth of a centimetre, a tree that measures exactly 29 centimetres should be recorded as 29.0 cm.

Field books, sketches and data sheets

Field Books

Field books are pocket-sized, bound or loose leaf notebooks, with blank or lined pages. If possible the paper and binding should be of good quality. Commercially available notebooks made from water resistant paper are particularly well suited to conditions in the field. Notes should be written using a sharp pencil. Pencil is permanent and not affected by water, whereas most inks fade and many are water soluble. Mistakes should be crossed out and not erased with a rubber. Erasing causes smudging and may result in the loss of valid information.

Field books are best for describing observations that are not easily described by lists of numbers or categories (for example, animal behaviour, descriptions of tracks, unusual events, daily activities, etc.) because they allow ample room for writing, sketches, maps etc.

Since different kinds of information can be entered into a single field book you should put extra effort into organising and standardising the contents. Every field book should have a title and the name of the observer on the front cover. Later, the dates covered in the book can be written on the front cover. Every page should be numbered. The first few pages should be left blank in order to leave room for an index of the contents of the book (for example, "Map of Trail System - p.8"). No page should be torn out. If something is incorrect, a large "X" should be made through that section or page.

Making observations and recording data.

Every field assistant should carry a field book, even if he or she is also filling out data sheets. Every guard on patrol should carry a field book. In it they should describe daily activities (where they have walked and when) as well as observations of interest (tracks, animal behaviour, trees fruiting, trees cut down illegally, snare lines, elephant carcasses etc.). The current date and the location should always accompany any recorded observation.

Sometimes it is advisable to carry more than one field book, each one covering a different subject. The first book should contain a daily record of movements and activities, as well as maps, sketches, and notes on unusual sightings or activities. Other field books can be dedicated to specific subjects under study, such as identifying tree species, or counting animals.

Sketches

Sketches add to the written information in field books. Sketches record information that can be difficult to write down quickly and accurately. Unidentified species of animals can be sketched and described for later identification from guidebooks. Take care to emphasise key distinguishing features, such as colour and shape. Accurate measurements should be written next to the sketch (see Fig. 5.2).

Sketch tracks together with measurements of their length and width. Also note the distance between individual tracks and the track arrangement. All of this information can help to determine which species made the tracks. It also may help determine the age, sex, and general condition of the animal (see Chapter 10).

Don't hesitate to make a sketches to add information to a report. For example, sketch the arrangement of bones from a leopard kill, before you disturb them, or sketch the layout of an illegal hunting camp, or a map of a stream system.

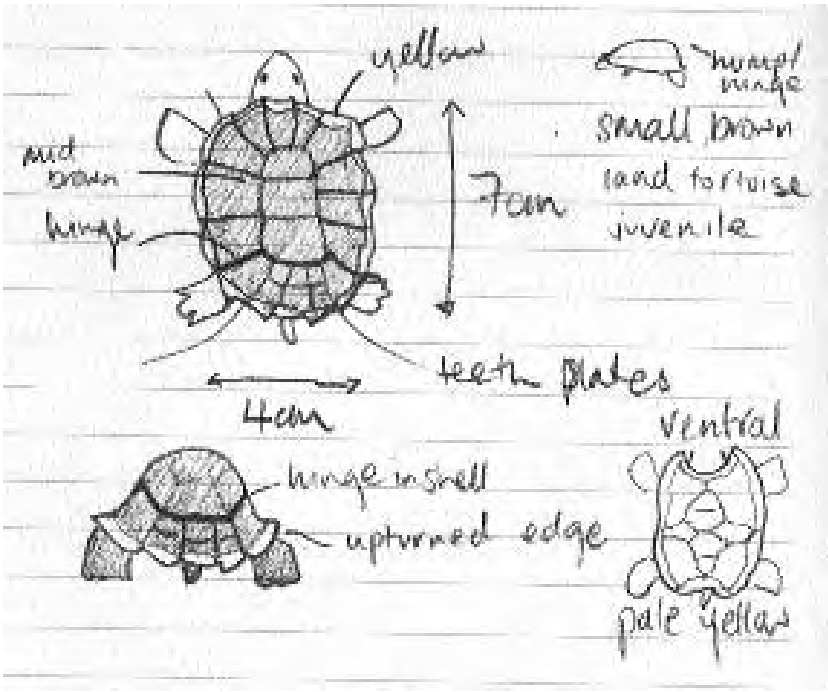


Figure 5.2: example of a sketch in a field book.

Data Sheets

Data sheets are forms that have been designed to help a researcher collect information in a standardised way. This allows information collected over time, or by different observers, to be comparable, a critical factor in studies employing many field assistants. Data sheets are particularly suited to studies where data collection is undertaken by assistants with a limited level of education.

Furthermore, data sheets allow a large amount of information to be summarised quickly. The information recorded usually is limited to just a few possibilities for each category (for example, "species" or "number seen" or "height"). Therefore, summarising and analysing the information is relatively simple. For example, averages, frequencies and trends are easy to determine, and unusual events or missing data are easy to detect. Since the structure of data sheets limits the kinds of information that can be recorded, every data sheet should have a section for comments. The comments section can be used to explain unusual data entries, or to record unexpected observations.

The top of every data sheet should have the following information: the type of data collected (e.g., primate census, weather records or observations from a vehicle), names of observer(s), date, and location of observations (for example, "transect B3"). Usually the following are also at the top of a data sheet: time when data collection started, time when data collection ended, weather and habitat type or types. Examples of data sheets (often called check sheets) are given in many of the chapters of this manual, whenever they are useful for data collection.

When filed in an office by subject and date, data sheets provide an efficient, organised means of storing and retrieving information (see Chapter 17).

Log Books

For rare or little known species, and unusual observations and events, each record is potentially valuable. If individuals note these down in their field book but later leave their job or are posted elsewhere that information will be lost to workers in the site where the observation was made. One way to avoid this is to ask people to make field notes which are kept in a central location. Over the years these will build up to represent a daunting quantity of information which will have to be sifted through in order to extract relevant observations. It is recommended that for rare species and observations, protection staff and researchers keep a central log book where all information is noted. This may be combined with cross references to more detailed descriptions in field notes where appropriate. For example, in Cross River National Park in Nigeria, all observations of gorillas, chimpanzees, drills, and red colobus monkeys might be noted in a primate book (including all sightings, group counts, observations of ventral infants, confirmed foods, poaching incidents etc.); all observations of red river hog, buffalo and elephants in an ungulate book; all observations of predators, particularly leopards, in a carnivore book; and all observations of the rare bare-headed rock fowl and their nests might go in a bird book. Other log books could be established for historical information gleaned from local people, about medicinal plants and other products people collect in the forest, for discoveries of pottery or charcoal in currently uninhabited parts of the forest, or for observations of rare animal behaviour.

Field notes

If all researchers and managers working in a protected area write up well organised notes each time they are in the field, it is possible to generate a permanent record of useful information (as well as giving a record of their daily activities). For example, make a list of key species for which spot sample data (see Chapter 14) should be taken for each sighting, or for which one should take 'recce' data (see Chapter 13) during foot patrols in the forest. If each person writes a set of summary field notes each time they go into the forest, describing all observations of a pre-determined list of animals and plants, this can be kept in a central location and serve as a source of information for all future workers at the site (see Box 5a).

Making observations and recording data.

These should be filed by date in a safe place. In addition, for certain key species all observations can be noted in a central log book, with cross references to field notes. In this way you can look observations up efficiently rather than having to search through all the assembled field notes.

Box 5a AN EXAMPLE OF FIELD NOTES TAKEN DURING EACH WALK IN THE FOREST AT THE *STATION D'ETUDES DES GORILLES ET CHIMPANZÉS* IN THE LOPÉ RESERVE, GABON.

1 Monday 18 July 1994

2 L. White & J. Dibakou

3 Mapping out east, Mandrills, Chimp Nests

4 SEGC-Airport Forest-Caillet's Est -Gellardo's Bridge-SEGC

5 Sunny

6 08:50 - 09:10 -we meet a large group of mandrills crossing in the FdA close to Pipta - counted 457 including, 47 ventral infants and 12 large males. Two swollen females, one with a very fine male escort at 1.5m. JD had seen 2 big males crossing as we arrived which I did not see, and a good few had crossed before us with some males bellowing already on the far side. Minimum group size was 500 and over 600 would not surprise me at all.

09:18 - Into Caillet's (Sacoglottis) Bosquet - mapping vegetation.

09:29 - group of *C. cephus* seen inside bosquet. Five individuals seen including a female with ventral infant. Estimate 10-12 in group. Feeding on *Cissus* fruits in a dense vine tower.

09:52 - Chimp nests and fresh jobbies - probably made by the same individuals over a period of three days. 9 nests close together: 5 m high in a *Diospyros zenkeri*; 10 m *Diospyros dendo*; 7 m *Dichapetalum barteri*; 5 m *D. zenkeri*; 6 m *D. zenkeri*; - all within 5 m of one another (see nest map) - could have been judged to be gorilla if older; 6 m *D. zenkeri* & 8 m *D. barteri* - a pair about 20m away; 7 m *D. zenkeri* & 10 m *D. dendo*.

10:13 - we surprise a group of buffalo resting in the forest. There are five or more - they grunt and flee out into the savanna to the SE. As they move off there is a *C. cephus* alarm close by.

10:20 - Chimp nests at end of gallery - one older, 2.5 m *D. zenkeri*; three fresh: 4 m *D. zenkeri*; 5 m *D. zenkeri*; 8 m *D. zenkeri*. Dung collected from the fresh nests for dietary analysis.

10:34 - A lone chimp nest about 3-5 days old, in a *D. barteri* at 6 m (see map for relative position of all these nest sites).

10:51 - *C. monticola* alarm calls and flees from a thicket of *Monanthotaxis*.

11:20 - Mixed group of *C. nictitans*, *C. cephus* and *C. albigena* at bottom of gallery leading into the Lopé river. Oil palm densities here must be c. 20-30 per ha. One of the monkeys had fed on new Iroko leaves rather like gorillas do.

13:08 - In Gellado's gallery on cut left from old BP road, *C. nictitans* and *C. cephus*.

13:41 - Vitex bosquet, *C. cephus* group seen and heard alarming.

14:55 - Exit to car

7 FT 335 mins.

(1=Date; 2=Observers; 3=Objective of the day and key words/summary of observations; 4=Route taken; 5=Weather; 6= Time and location of each observation; 7=Total field time).

Conservation of field note books

If field notes are written neatly, using permanent ink or pencil, clearly labelled and filed carefully once full, they constitute a valuable long-term record. In 1994 and 1995 Jefferson Hall coordinated a survey of eastern lowland gorillas in the Kahuzi Biega National Park. George Schaller had worked in the same area in 1959-60 and made his field notes from that time available, providing valuable insights into how the area had changed over 35 years (Figure 5.3).

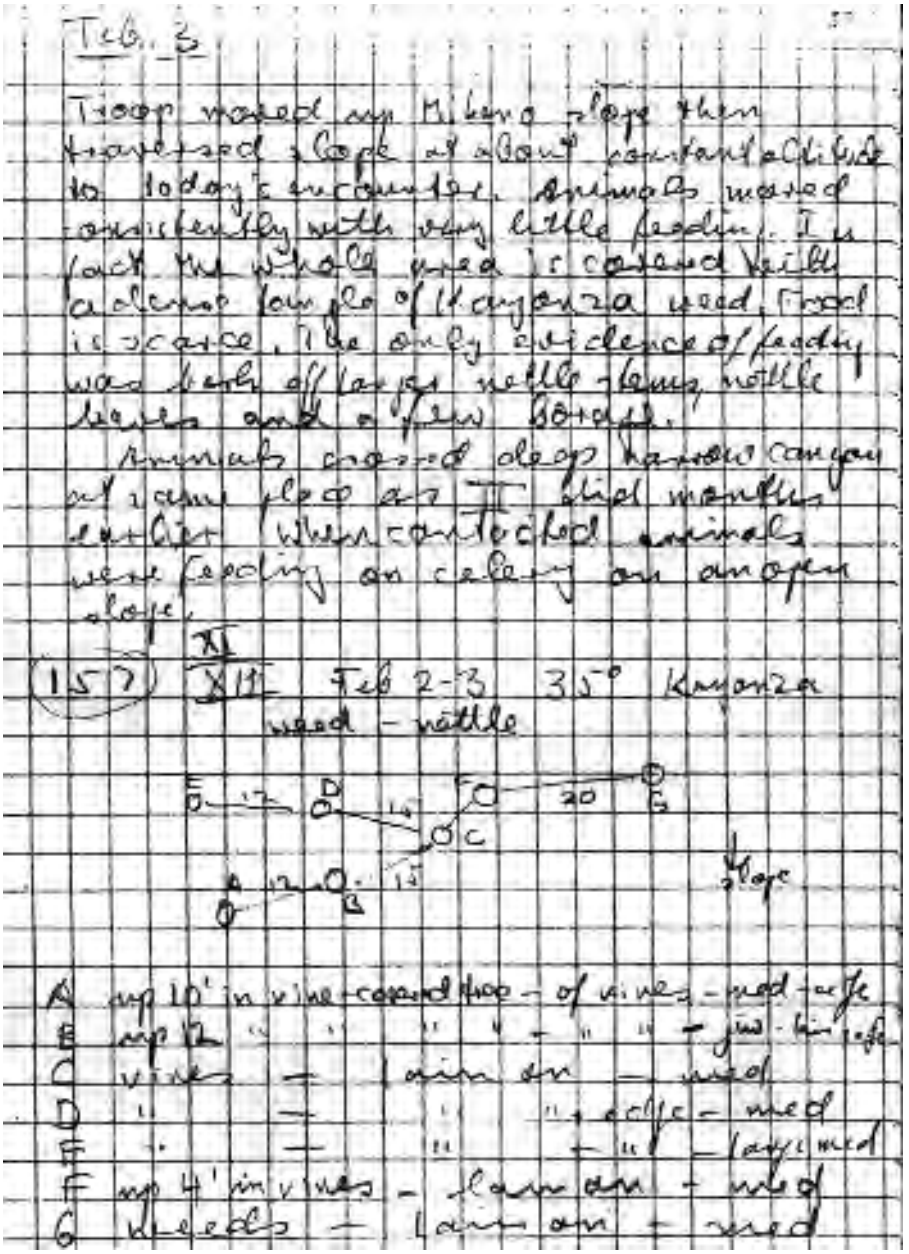


Figure 5.3: A page from George Schaller's field notes dated February 3, 1960. Will young researchers be able to consult your field notes in 35 years time?

Precision of observations

Always record observations precisely. For example, if measuring perpendicular distances to dung piles with a measuring tape all observations should be noted to the nearest centimetre. If using callipers to measure the length of seeds swallowed by hornbills all observations should be noted to the nearest millimetre. Decide what precision is reasonable prior to data collection and record all observations with the same accuracy (number of figures / decimal points).

When distances or ages are estimated this should be clearly stated in your notes.

Information collected during day to day activities

Managers of protected areas have many duties and few will have the luxury of devoting a large proportion of their time to research. However, many will be able to make useful observations about the wildlife in a protected area, or about pressure mounting around its limits. By following the guidelines in this chapter it should be possible to devise a system for recording opportunistic observations in order to generate information of use for monitoring conservation efforts, without greatly adding to the work load of protected area staff. Furthermore, by devising a system of reliable note taking you will be better able to monitor and evaluate the performance of field staff.

A good source of data which is often neglected is observations made whilst travelling by vehicle or boat. Many researchers and protected area managers spend a good deal of time in vehicles moving from point A to point B during their weekly routine. During the course of these journeys they will see animals moving within sight of the road, or pass road kills or hunted animals for sale. The frequency of these encounters may be correlated to animal abundances. Remember, when data are collected systematically, even from a vehicle, they can be used to detect trends or changes over time.

When managing a protection staff or research project it is important to keep track of project vehicles to ensure that they are not being mis-used. One way to do this is to keep a log book in the vehicle which must be filled in each time the vehicle is used. If project staff are routinely asked to do this they can, with little extra effort, add simple observations to the log. The quality of these observations is likely to vary according to a number of factors. For example:

- some personnel will make better observations than others;
- if there are more people in the car (more observers) more observations are likely to be made as those not driving will not be so fixed on the road ahead;
- observer efficiency will vary with speed;
- the number of observations will vary with time of day.

Therefore, data sheets should be designed such that these factors can be taken into account in any analysis. Bear in mind that if you are to analyse such data you need to be able to locate observations on a map and also that negative data (lack of observations) is as important as the observations themselves. A common practice within wildlife departments is for staff to record each animal they observe, but not necessarily to keep track of all of their observation opportunities. Therefore, during the dry season in the Lopé reserve, when staff spend a lot of time in the reserve because they have a savanna burning programme to conduct, there will be many observations of elephants, whereas in December, when most of their work is anti-poaching patrols along the main roads, with only occasional sorties into the well protected savannas, they will see many fewer elephants.

However, during the dry season most elephants leave the Lopé Reserve and move to swamps along the large rivers of central Gabon, whilst densities in December are high because the bush mango, *Irvingia gabonensis*, which is common close to the savanna edge, is in fruit. Hence, during 5 trips into the savannas in December they might see 12 groups of elephants whilst during 25 trips in the dry season 17 groups were seen. The encounter rate in December is 2.4 groups per trip and in the dry season it is 0.7 groups per trip, but if no record of the number of trips is taken the data would show that more elephants are encountered in the dry season!

Ask staff to note the following information when they decide at the beginning of the trip to record observations of animals from the vehicle:

- Date
- Name of driver;
- Names of passengers;
- Odometer reading (km) at start of trip; end of trip; and total distance travelled;
- Time of departure; time of arrival; total time of trip;
- Route taken;
- Time and distances of any breaks in the trip;

For each animal sighting ask observers to note:

- Time;
- Location - a description in words, and a map coordinate or GPS position and an odometer reading;
- Species;
- Number seen (record number of adult males, adult females, sub-adults / juveniles and infants if possible);
- Estimated total number;
- Habitat type where animals were observed (e.g., see Chapter 9).
- Any interesting observations, such as food, behaviour or species heard but not seen (see 'spot scans', Chapter 14);

This information can be summarised in a data sheet:

Date _____

Name of driver _____ Names of passengers _____

Odometer reading: start of trip _____ end of trip _____ Dist. travelled _____

Time of departure _____ Arrival time _____ Trip time _____

Route _____ Breaks _____

Time	Location	Species	No. / sex	Est. total no.	Habitat	Comments

Information can be recorded for dead animals in addition to sightings of live animals. For example, road kills should be noted and, if possible, collected, measured and preserved in your field museum (see Chapter 11). If many more tortoises are seen killed by vehicles at a certain time of year it may indicate a seasonal change in activity patterns or ranging. Note animals for sale in villages beside the road as this can be used to assess changes in hunting success with time and changes in the efficiency of hunting by season or over the years. For example, over a number of years you might be able to show that there are decreased numbers of animals for sale close to a major town, suggesting that populations have decreased due to over-hunting. In the same way the appearance of new settlements or other demographic changes should be noted.

CHAPTER 6
Maps, compasses, GPS units and the principals of navigation

Ann Edwards & Lee White

No conservationist or field researcher can do their job efficiently unless they understand the principals of navigation. This is particularly true for people working in the tropical rain forest, where visibility is restricted and large landmarks, such as mountain ridges, are generally obscured. Even on a well-known path, a forced detour around an angry elephant, or to go to the toilet, can leave you helplessly disoriented. However, if you do lose your way, you are carrying a compass and you have a good working knowledge of the area (such as knowing that a logging road running north-south is to your west, or that all rivers drain eastwards) you can never be lost. You simply have to follow a compass bearing towards a path, road or familiar river. If you ever do find yourself helplessly lost in a remote African rain forest, then follow any water course downstream and you should eventually find your way to areas used by people.

In 1985 Michel Fernandez was tracking a group of gorillas about 4 km south of the 'Station d'Etudes des Gorilles et Chimpanzés' in the Lopé Reserve. Unknown to him, a lone male silverback gorilla was also following the group. As the lone gorilla moved into a small clearing he probably thought he was approaching the resident silverback and his group. Instead he came face to face with Michel. In his confusion the gorilla charged forwards, took Michel's leg in his hands, and bit deeply into his calf. Adding insult to injury he also pulled at the red string emerging from Michel's shirt pocket, and snatched his compass. As the gorilla fled Michel began to search for his compass. He was in a part of the forest he had not visited before and knew he would get home much faster with the aid of the compass. It was not until his blood had filled his boot that he abandoned the search and made his way home.

Given the severity of his injuries, why did Michel linger, with blood pouring out of his wound, in an area where the gorilla which might attack again, in a seemingly vain search for a compass? Because Michel, a knowledgeable field worker, appreciated the true value of a compass. If you think Michel's behaviour strange, you probably need to read this chapter.

Compasses

What is a compass?

A compass is an instrument used to indicate direction. It is made of a magnetised needle that floats freely (often in a liquid). The needle always points North and is surrounded by a moveable dial on which the four "points of a compass" are written (**N**orth, **S**outh, **E**ast and **W**est). The dial is further divided into 360 equal sized parts, called degrees, the standard unit measure for directions and angles (Figure 6.1).

How do you use a compass?

To use a compass, hold it level so the needle can swing freely. When it stops moving, align the "N" on the compass dial with the red or dark tip of the needle. When North (N) is properly in place you can accurately read the other compass directions (South, East and West) from the dial. If you have a compass which uses a freely rotating card with compass directions written on it (instead of just a needle) you can read the compass directions straight from the card without further adjustments.

Figure 6.1: a compass

In addition to the four directions of North, South, East and West, compasses indicate direction using degrees. This allows for greater accuracy in defining a direction. There are 360° in a full circle. North always equals 0° (read zero degrees) or 360°, East equals 90°, South equals 180°, and West equals 270°. The circle can be further broken down into named sub-divisions: North East is exactly half way between North and East, at 45°; North North East and East North East are exactly between North, North East and East, at 22.5° and 67.5° respectively, etc. (Figure 6.2). Other directions are usually referred to by a number (called a bearing), for example, 49°.

To "take a compass bearing" means to align the compass with some distant object of interest and to read the "bearing" (for example, 86°) from the compass dial. Some compasses have sighting mechanisms or mirrors that allow you to look at the distant object and the bearing at the same time. This allows you to take a more accurate reading.

Because they depend on magnetism to work, compasses should not be used around large sources of iron or steel, or near electrical or magnetic currents. Therefore, compasses should not be used in or near a car, truck, or power lines. Even a nearby gun barrel carried by a park guard can affect a compass reading. If you must use a compass near any of these things move the compass around the object to see what effect the object has on the compass.

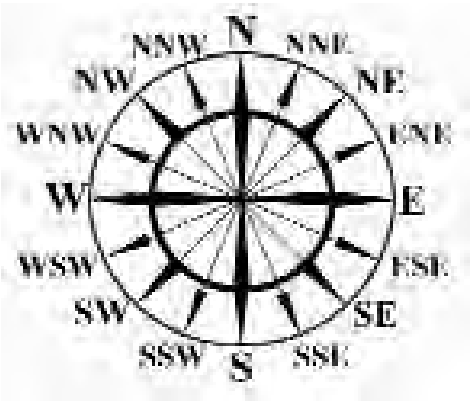


Figure 6.2: compass directions

Declination or magnetic deviation

A compass is the best instrument available for determining direction. However, compass needles are not pulled to the true (geographic) north pole, but to a heavily magnetised point 500 miles from it. The difference in compass angle between true north and magnetic north is called the "declination" or "magnetic deviation". The size of the declination changes between different places on the earth, and in any one place with time. Declination, which is always small in the tropics, is often pictured on the edge of carefully drawn maps along with a statement of the rate at which it will change (see Fig. 6.3).

When using a compass, there are times when it is important to account for declination, and other times when it is not. Small differences in compass angle have a greater effect over longer distances. Therefore, you should account for declination when walking long distances that must be accurately mapped or navigated (for example, a long transect or a park boundary). On the other hand, you can ignore declination when walking short distances, or when navigating using local landmarks. Declination also can be ignored when drawing small-scale maps (for example, of a small trail system). Around the equator if declination is ignored it typically results in errors of 50-100m over a kilometre (about 17m per degree of declination per kilometre travelled).

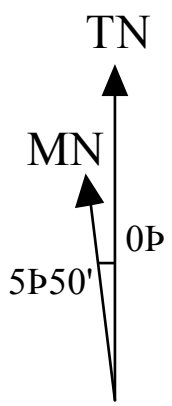


Figure 6.3. Symbol used on maps to show the magnetic declination of the area mapped. (From IGN Booué 2A)

Magnetic declinationı in relation to true northı on 1 January 1985.ı Declination decreases byı 6 minutes each year.

Correcting for declination

In the example in Figure 6.3 magnetic north is 5°50’ west of geographic north. We can correct for this declination in one of two ways:

1) The dial on most good compasses is fixed by a small screw which can be loosened, to adjust for declination. In this example adjust the dial such that when the red arrow points North the dial reads 360°-5°50’ = 354°10’.

2) If your compass cannot be adjusted for declination add 5°50’ to the bearing you wish to follow. For example, if you want to walk due east (90°) follow a bearing of 95°50’ on your compass. Alternatively, if taking a bearing, subtract 5°50’ from the observed compass direction.

Maps

Once you know the principals of using a compass you can make much better use of maps.

What is a Map?

A map is a representation of a landscape. It uses symbols to represent the features found on a piece of land. Different kinds of maps focus on different features. For example, a physical map focuses on features such as rivers, mountains, or vegetation. In contrast, a political map shows man-made features, such as the boundaries between countries, or the locations of cities, villages and roads.

Whenever available, maps should be consulted before the start of any management or research project since they can help to determine where the project should be situated. First, maps can indicate which parts of a proposed study area are inaccessible, due to the presence of large rivers, wide swamps or high mountains, or which areas are far from roads, trails or waterways. Second, general habitat types or physical features which may influence where a study should be conducted (for example riverine forest, high elevation forest, steep slopes prone to frequent disturbance from landslides, etc.) may be indicated on a map . There is always something new to learn from a map. Refer to them often, especially when new information is brought in from the field. Check the date of any map you use and decide if it is still relevant.

Aerial photographs and even satellite images are becoming increasingly available and can complement the information available from maps (see Box 9k).

What are Maps used for?

Maps are important tools for any land manager or field researcher. They provide a means of representing spatial information in an accurate and compact way. Maps are important tools for three reasons:

1) *Maps provide spatial information that cannot be conveyed in writing.* For example, a map can show how many rivers there are, their locations, if there are many in one area and few in another, which directions they flow, if they flow near mountains, forests or villages, etc. Many words would be needed to convey the same information and the description would still remain incomplete.
2) *Maps provide a simple, efficient way of recording and presenting spatial information collected in the field.* For example, the distributions of species or habitats, or the locations of trail systems, logged areas, or villages may be presented most effectively using maps.
3) *Maps allow someone to locate features in a new area.* An accurate map enables someone to walk into an area where he or she hasn't been before and find a feature drawn on the map, such as a proposed transect line, or a salt lick or clearing. When carefully interpreted, a map can allow someone to estimate how long it will take to get to a particular spot, and what the best route for walking there might be. Hence, you can use maps to navigate to and from areas of interest which cannot be accessed by road or river.

What features are important in a map?

All maps should clearly indicate orientation and scale. Furthermore, all symbols should be defined, and the map should be dated. Finally, the site of the map should be located within a larger known area.

Orientation

North, South, East, and West indicate direction throughout the world. On most maps, North is towards the top of the map, but this is not always the case. Therefore, any map should clearly indicate which way is North. A simple arrow pointing North with an "N" marked next to it is usually sufficient (Figure 6.4). Once North is defined, any other direction (East, West or South) is evident.

Figure 6.4. Example of a map orientation symbol.

Scale

A map must include a scale in order for it to be useful as a source of spatial information. Scale refers to what a given distance on the map represents on the ground. Maps drawn in the same scale can be compared easily, because any given feature (e.g., a lake, road, or the bend of a river) is the same size on both maps.

Scales are usually presented in two ways. First, the scale of a map can be presented numerically, as a ratio between the distance on the map and the distance on the land. For example, on a map with a scale of 1:50,000 (read 1 to 50,000) 1 centimetre on the map represents 50,000 cm (or 500m) on the ground.

Second, a scale can be presented graphically (see Figure 6.5). Graphic scales can be used to measure distances on the map itself. Place a piece of paper along the edge of the scale and mark on the paper each kilometre line (or other distance) with a pencil. Then move the marked paper to the area of the map you wish to measure. Alternatively, measure the scale with a ruler and calculate the distance that is equivalent to 1cm. Thereafter multiply the number of centimetres and millimetres measured with your ruler by the distance equivalent to 1cm. To measure the length of a meandering stream or road, lay a string on top of the curving line, then measure the length of the string using the scale or your ruler. Note that if you magnify or reduce the map using a photocopier the graphic scale remains true (if copied with the rest of the map) but the numerical ratio written on the map will no longer be a true representation of map scale.

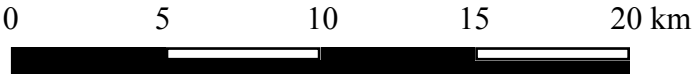


Figure 6.5. Example of a graphic map scale.

Locate the Site

Any map should have a title, for example "Korup National Park", or "Bandundu Province, RDC". Even with a title, sometimes it is difficult to determine the location of the area represented in the map. Therefore, for any map that will be read by people unfamiliar with the area, the site of the map should be located relative to the surrounding region. This can be done in two different ways, though both can be used for the same map.

First, a picture of the focal map can be drawn in relation to a larger map, and placed in the legend (see Fig. 6.6).

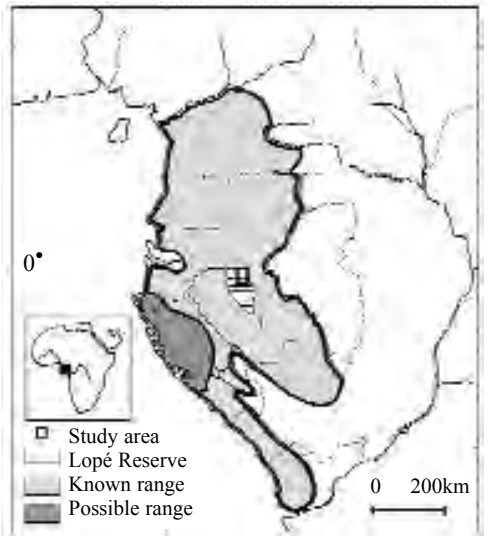


Figure 6.6. Locating the site of a map in relation to a larger known area (in this case the range of the mandrill, *Mandrillus sphinx,* in Africa).

Second, longitude and latitude lines can be included in the map (see Boxes 6a & 6b). Longitude and latitude lines are recognised worldwide. Therefore, anyone, anywhere reading a map can know the location of the region pictured. Longitude and latitude lines should be marked on any map that is widely distributed. Lines of longitude and latitude that form the borders of a map are generally marked in the corners. Other longitude and latitude lines can be marked along the edges. If you look at a topographic map of the area where you work you will see longitude and latitude clearly labelled every 5' for maps at a scale of 1:50,000 or every 10' for maps at 1:200,000.

Symbols

Map features are defined using symbols. Both man-made and natural features can be defined using symbols. For example, villages, roads, railroad tracks, wells, streams, mountain tops and waterfalls can each have their own symbol. Different features can be defined using different colours, or widths of lines, or different pictures, such as a triangle for a tree. Any symbol used in the map should be defined in the map's legend.

A legend is a box located near the edge of a map that has examples of each symbol, together with an explanation of what that symbol means. Sometimes a group of maps will have only one common legend, but these maps should always be found together, such as in an atlas.

Contour Lines

Contour lines are lines on a map that join all the places that have a common elevation. Contour lines define the presence of hills, valleys, streams and all the other vertical measurements of the land. Contour lines can indicate if an area is mountainous or flat, which way streams are flowing, or which way slopes are facing (see Box 6c). Contour lines (together with a scale) allow you to calculate the steepness of a slope or trail (see Box 6d).

The difference in elevation between one contour line and the next is called the "contour interval". Contour intervals remain the same throughout any one map. The contour interval can differ between maps and depends on the steepness of the area. In flat areas the contour interval may be only 10m, which means the elevational difference between one contour line and the next is 10m. In mountainous areas, the contour interval may be 50m or 100m.

Maps with contour lines are called "topographic maps" because they show the topography, or surface features, of the area. Topographic maps are widely used to navigate in natural areas where there are few roads or man-made features to act as landmarks.

Map making

Most maps available for wide distribution are accurately drawn in scale and direction. In addition to interpretation of images such as aerial photographs, many careful measurements are used to produce the maps available from government printing offices. If you choose to make a map with this level of accuracy you will have to invest a great deal of time and effort. If you do, you could produce a map that would be useful to many people for years to come. However, this level of accuracy is not always necessary. Sketch maps and overlays can be produced quickly and still be useful.

"True" maps

A "true" map is quantifiable because it is drawn accurately in scale and direction. Such a map should not be open to misinterpretation and can, for example, be used to calculate the size of an animal's home range, the density of villages in the park, the area covered by logged forest, or the length of trails available to tourists.

Sketch maps

A "sketch map" is a map showing the relative locations of objects without precise measurements of the distance or direction between them. To make a quick sketch of the houses in a small village you might take a compass bearing on the main street which passes through the centre of the village and then estimate the distance between houses at opposite extremes of this central axis. Once you have put these details down on paper you would then sketch in the relative positions of the remaining houses by eye.

Sketch maps are a useful way to record spatial information, but generally they are intended for limited use by the map maker. They are not for wide distribution because they are not drawn with precision, therefore, they are open to misinterpretation if read by someone else. Specifically, sketch maps are not quantifiable. In other words, it is not possible to take numerical measurements from a sketch map and use them to calculate distances, densities, areas, etc.

Overlays

An overlay is a way of using an existing map to produce a new map, or simply to add new information to an existing map. An overlay is produced by 1) placing a clean piece of paper over an existing map, 2) tracing the features you need as a base from the old map onto the clean paper (such as park boundaries, rivers and roads), and 3) adding new information to the new map (such as the distribution of primary forest, gorillas, or good agricultural soil, or the locations of houses). Overlays are useful because they can be made quickly and still contain much of useful information.

In recent years, sophisticated computer programs called Geographic Information Systems have been developed, which allow one to store and analyse data associated with a geographic positions (for example, sightings of a rare mongoose species, or locations of archeological sites) as different 'layers'. These programs allow one to store and analyse different types of data, such as sightings of animals, or sites of plant collections; road or river networks; vegetation or contour maps; and to produce versatile maps displaying various combinations of information.

Making a map

To produce an accurate map first you will need tools for measuring direction (a compass and a protractor) and distance (a meter tape for short distances, a hip chain for longer ones). You will also may need an altimeter or a clinometer for measuring elevation.

To map a trail or a boundary line, follow the methods outlined in Box 6e. When mapping a trail system, it may be useful to draw a sketch map, as well as record data on a data sheet, in order to describe the relative placement of trails, rivers, ridges and other features. When you map a trail system give each trail its own name or number and put that information on both the sketch map and the data sheet.

Where visibility is good, and you can see distant landmarks of known locations, use triangulation (see page 78) to locate features such as trailheads, buildings, bridges, etc. In places where there are many hills, care must be taken with distance measurements. Distances measured on maps represent horizontal (flat) distances on the ground. If you measure the distance between one point in a valley and another point on a hilltop, you are combining vertical and horizontal distance measurements. Therefore the actual distance between the valley bottom and the hilltop will be longer than the distance between these two points on an accurate map (see Figure 6.7). Where there are distinct elevational changes measure altitude using an altimeter and measure slope angle using a clinometer (see Box 6f). If you are mapping short distances, or an area with few hills, then you may be able to ignore this effect.

Box 6a LONGITUDE AND LATITUDE

Longitude and latitude lines provide a global, or worldwide, system for locating places on the earth. This system was developed to deal with the fact that the earth is a sphere, resulting in a curved (rather than flat) surface, which is difficult to divide into regularly shaped sections.

Longitude lines run North and South (see Figure B6.1). They number from 0Þ (read 'zero degrees') to 180Þ in two directions, East (E) and West (W). 0Þ runs through Greenwich, England, and passes near Accra, Ghana. Dakar, Senegal is at 17Þ W longitude (read 17 degrees west longitude), Kinshasa, RDC is at 15Þ E longitude and Nairobi, Kenya is at 37Þ E longitude.

Latitude lines run east and west (see Figure B6.1). They number from 0° to 90Þ in two directions, north (N) and south (S). 0Þ follows the equator and 90Þ is at the North and South Poles. Lagos, Nigeria is at 6° N latitude, Libreville, Gabon is near the equator at 0Þ latitude, and Brazzaville, Congo is at 4Þ S latitude.

To be more exact about a location, latitude and longitude lines each can be divided into smaller units, called minutes and seconds. One degree is divided into 60 minutes. One minute is divided into 60 seconds. Therefore 30 minutes equals one half of a degree, and 3600 seconds equals one whole degree.

Detailed locations can be written as follows: 37°22'30" E, 2°15'45" S (read 37 degrees, 22 minutes, 30 seconds east longitude, and 2 degrees, 15 minutes, 45 seconds south latitude).

Since maps are flat representations of a curved land surface they never can give a true representation of the shapes of different land masses, particularly when map scales are large. To allow for this distortion a number of different systems (called

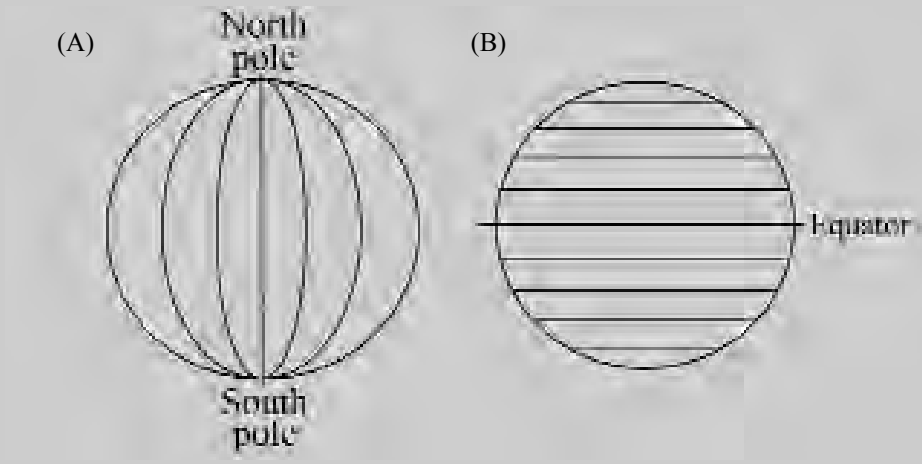


Figure B6.1. Longitude (A) and latitude (B) lines.

geodetic systems) are used in mapping to represent a flat projection of the land, since distortions differ in different parts of the world. All good maps state in the legend which system was used when plotting the map. This is generally of little concern, but when using a GPS unit to navigate with geographic coordinates (see Box 6g) it is important that the correct geodetic system is programmed into the setup menu. Failure to do this will result in inaccurate readings.

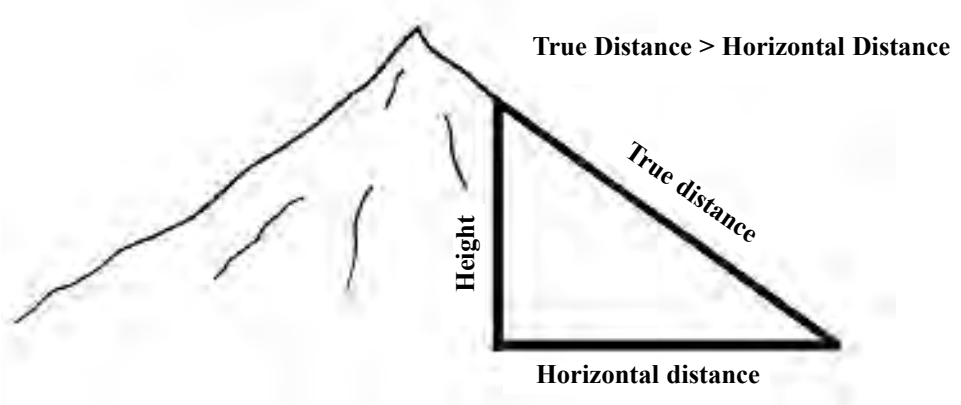
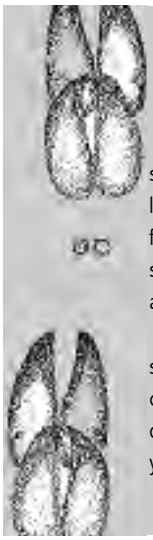


Figure 6.7. The ground distance along the slope of a hillside is longer than that same distance shown on a map, since maps can only show horizontal distances.



Box 6b UTM COORDINATES

Most modern maps are marked with two types of coordinates: geographic coordinates measured in degrees, minutes and seconds; and UTM (Universal Transverse Mercator) coordinates measured in meters. Geographic coordinates (latitude and longitude) are designed to locate positions on the spherical planet. So, as you can see from Figure B6.1, longitude lines are furthest apart near the equator and get closer together as one moves towards the poles. Therefore, one degree, minute or second of longitude does not correspond to any constant distance. This makes it difficult to plot geographic positions accurately onto a map.

The UTM system of coordinates measures distance in meters and adjusts for the changes inherent in the geographic system. This makes it more convenient to plot positions measured by a GPS unit. Using geographic coordinates you have to convert fractions of 60 into a decimal measure corresponding to the scale of whichever map you are using. The globe is divided into a number of UTM zones, which, like geodetic systems (see Box 6a), need to be programmed into your GPS unit if you are to obtain accurate positions. Again, the UTM zone features in the legend of all good maps.

Box 6c INTERPRETING LANDFORMS FROM CONTOUR LINES

Using Contour Lines to Indicate the Steepness of a Slope

When contour lines are close together, the slope is steeper than when the contour lines are further apart. This information can be used to determine the shape of a hillside as illustrated in Figure B. 6.2.

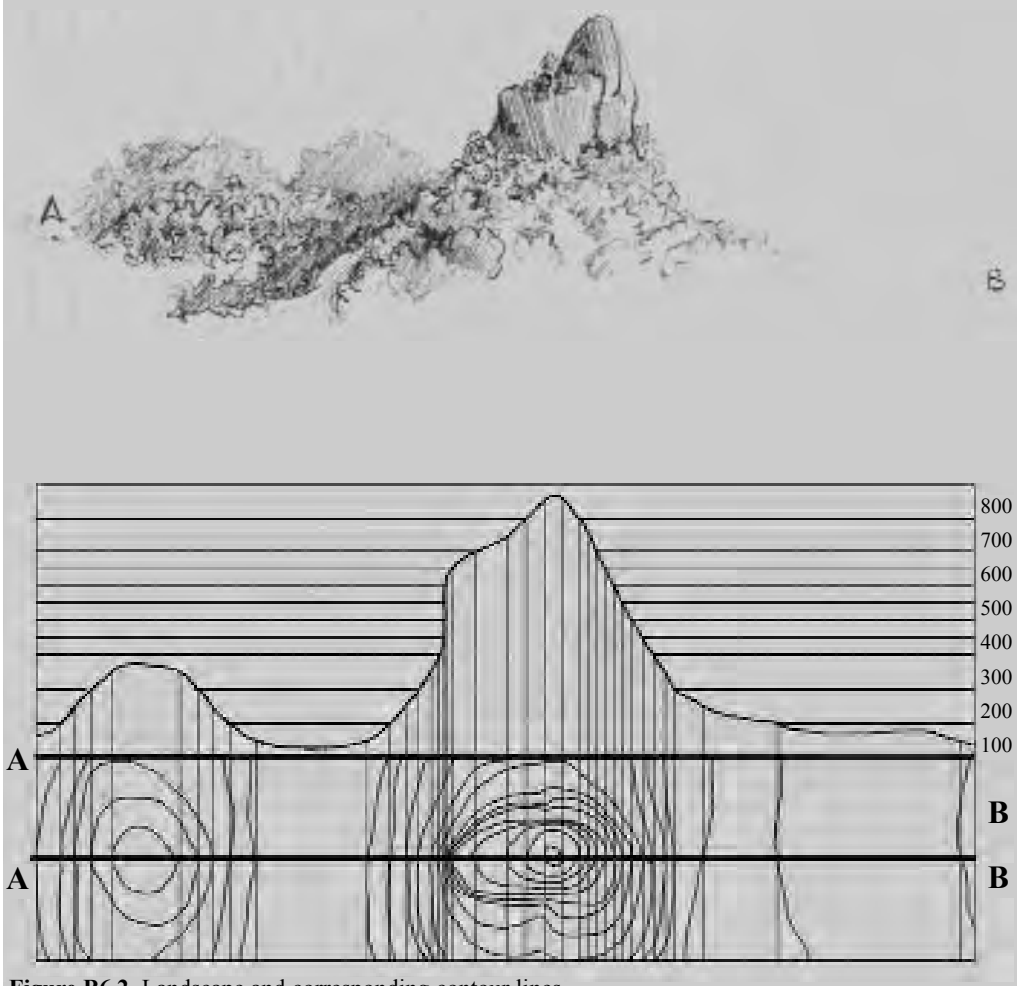


Figure B6.2. Landscape and corresponding contour lines

Reading the Shapes of the Landscape Using Contour Lines

Mountains, passes, ridges and valleys all can be distinguished by the contour lines of a map. Figure B6.3 shows a number of different landform features and the corresponding contour lines on a map. When interpreting contour lines from a map bear in mind that streams always flow downhill, and they always flow in valleys. Therefore, small stream beds, and steep-sided gorges, form small "V" shapes in the contour lines of maps, with the points of the "V's" pointing uphill. Steep-sided stream beds will form sharp "V's" . Shallow stream beds will form less distinct "V" shapes.

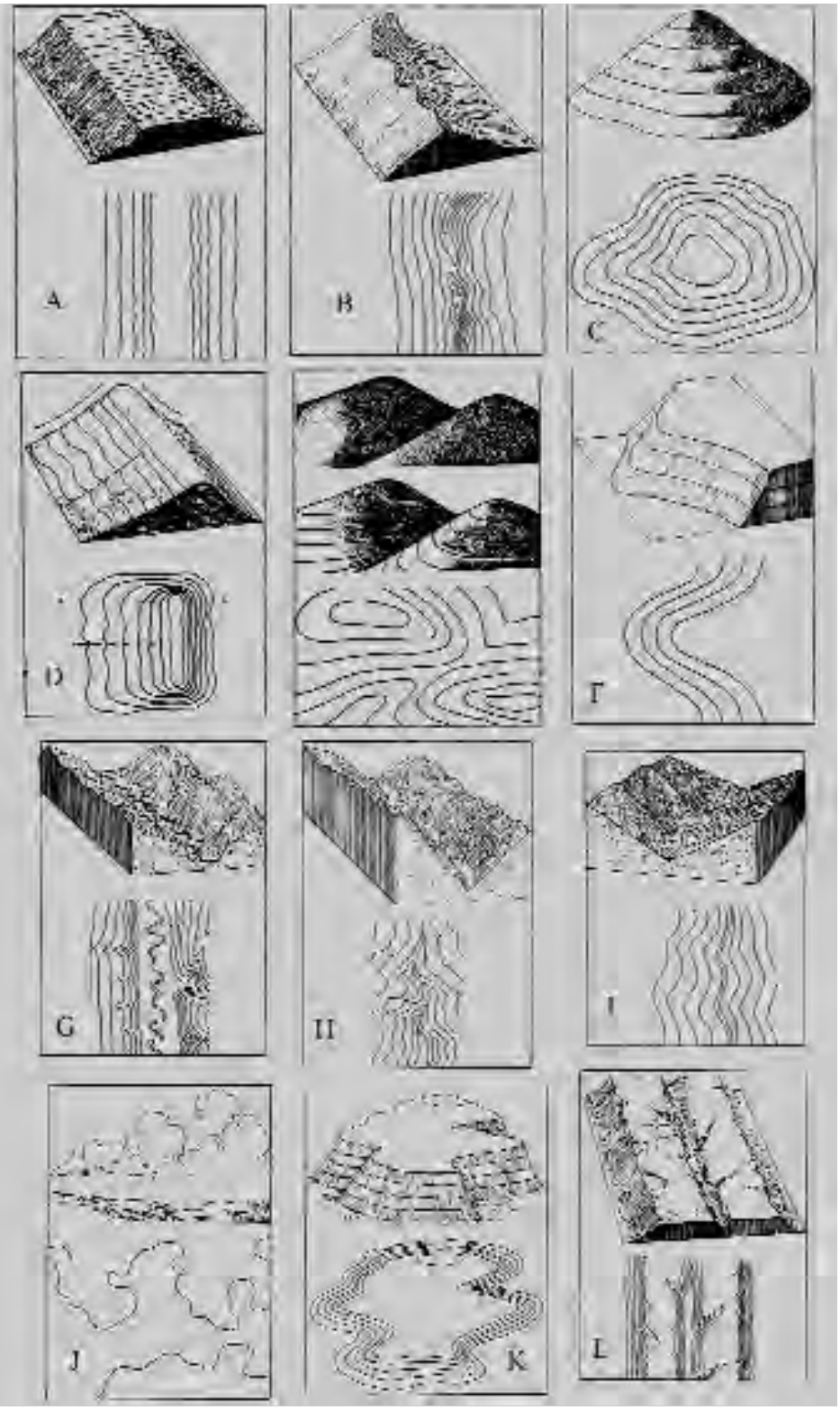


Figure B6.3. Various landscape forms and corresponding contour patterns..**A.** ridge; **B.** knife-edged ridge; **C.** hill; **D.** Escarpment; **E.** pass or col; **F.** spur; **G.** steep-sided valley; **H.** gorge; **I.** gentle valley; **J.** undulating plains; **K.** plateau; **L.** dissected plateau

BOX 6d CALCULATING THE STEEPNESS OF A SLOPE: GRADIENT AND PERCENT SLOPE

The steepness of a slope can be described in two ways: gradient and percent slope. A slope's gradient is the vertical distance (up and down) divided by the horizontal distance (flat along the ground), or

$$\text{gradient} = \frac{\text{vertical distance}}{\text{horizontal distance}}$$

The percent slope equals the gradient times 100, or

% slope = gradient x 100.

When a map is used, the vertical distance between two points is determined using contour lines and contour intervals. The horizontal distance is determined using the scale.

For example, the gradient and percent slope of a hillside with a vertical distance of 300m and a horizontal distance of 1km (Fig. B6.4) would be

$$\text{gradient} = \frac{300}{1000} = 0.3$$

and

percent slope = 0.3 x 100 = 30%.

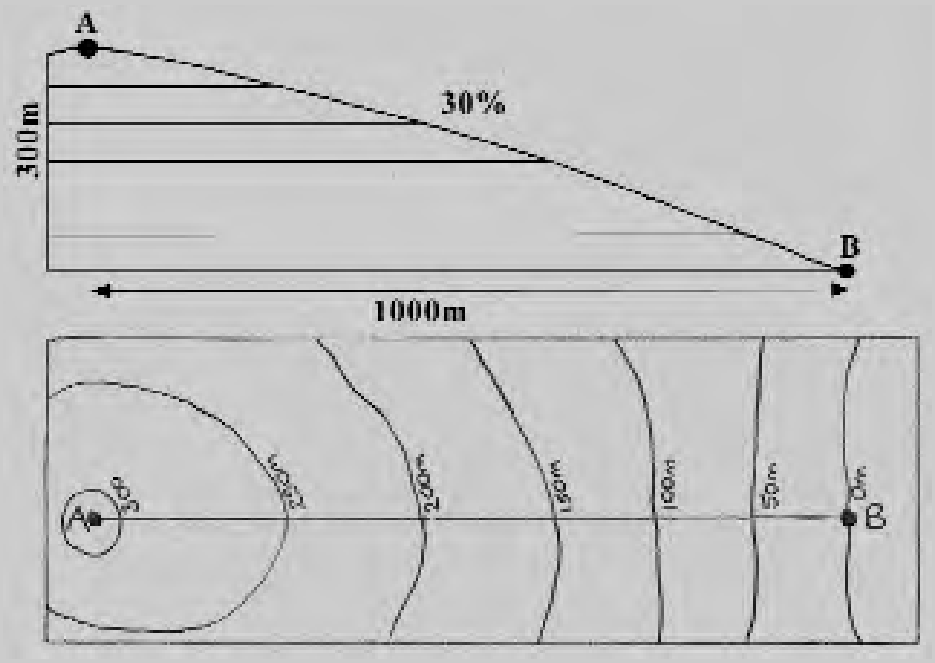


Figure B6.4. Calculation of a gradient from a contour pattern.

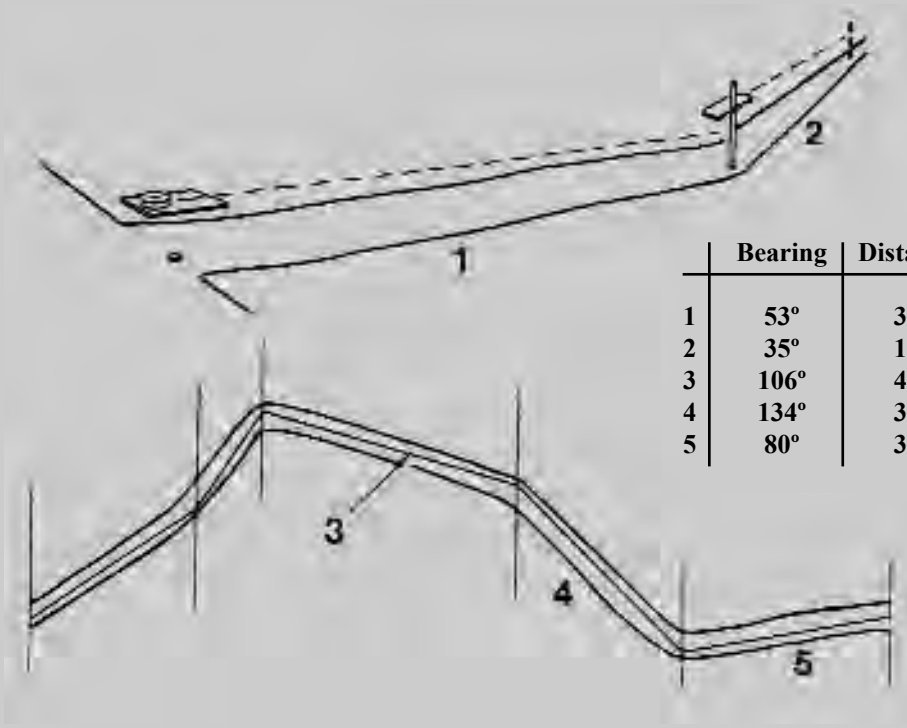
Box 6e MAPPING A TRAIL OR BOUNDARY LINE

Collecting Information in the Field

To map a trail or boundary line (such as that around the edge of an area of cultivation), first make a data sheet with four columns for the following information, 1) the compass bearing; 2) the gradient, measured using a clinometer (see Box 6f); 3) the distance to the next marker; 4) a column for notes, such as stream crossings, tree falls, trail intersections, etc. Include sketch maps as needed. You will need a compass, some way to measure distance and an assistant.

Stand at the start of the trail (or at any point on a boundary) and have an assistant walk as far as possible in a straight line along the path. Just before he or she turns any corners or goes out of sight, have your assistant place a 2m vertical stick in the ground to mark the spot. Take a compass bearing from where you are standing to the stick, and record that compass angle on your data sheet for the first measurement. Then measure and record the angle of slope by sighting through a clinometer at your assistant's head, if you are about the same height (or mark your eye level on the stick and sight to the mark). Then measure the distance between you and the stick, with a tape measure strung between you and your assistant and record that distance for the first measurement on your data sheet. Then advance to the spot where your assistant is standing (he or she should not move in the meantime). You should remain there as your assistant walks on. Take your second reading from where you are now standing.

Using information from your data sheet, draw the map of the trail (Fig. B6.5). You will need a protractor or compass to record the angles, and a ruler to record the distances (for example, 1cm on the map can equal 100m along the trail). Squared paper is helpful. Work first with a pencil and an eraser. Draw the final map in ink.



	Bearing	Distance
1	53º	35
2	35º	16
3	106º	44
4	134º	39
5	80º	36

Figure B6.5. Method for mapping a trail

Mapping the Information

To begin to make your map, first draw a long line lightly in pencil across the page to indicate the direction north. All of your other angle measurements will be based on this line. Place the centre of a protractor at the spot you want to begin. Mark that spot with a pencil. This is your first point. Turn the protractor so that 0° on the protractor is parallel (in line) with the line representing north on your map. Squared paper makes this much easier to do since all of the vertical lines can represent north. Read the angle or bearing for the first measurement on your data sheet. Place a small pencil mark on your map at that angle on the protractor. This is your second point. Remove the protractor. Carefully place a ruler between the two points now marked on your map, with zero placed next to the first point. Using the distance scale you have chosen, measure the distance along the ruler that represents the distance for measurement 1 marked on your data sheet. (For example, for a scale of 1cm to 10m, 25m along the trail equals 2.5cm on the map). Once you have drawn this line, you can erase or ignore the second point.

Next, place the centre of the protractor over the very end of the line you just drew. This becomes your new starting point (corresponding to the second marker in the field). Rotate the protractor until 0° is parallel with your direction arrow. Follow the directions above for the second measurement as you did for the first measurement.

Don't forget to include a scale, a legend (including a distinct arrow indicating north), a title and the date on the final map. It is also a good idea to include a description of how the data were collected.

Box 6f USING A CLINOMETER TO MEASURE VERTICAL ANGLES AND ESTIMATE HEIGHTS

Whilst a compass is used to measure angles in a horizontal plane vertical angles are measured using a clinometer. A clinometer is an instrument that appears at first glance somewhat like a compass. In fact, some compasses also contain an inbuilt clinometer that can be used when the compass is turned on its side. Clinometer needles or dials rotate in a vertical plane (unlike compasses which pivot horizontally). They measure the angle between the point at which they are held (A) and a second higher or lower point (B) which is viewed through the sight. If point B is higher than point A the angle (or the %) of slope at which the clinometer is held will be positive. If point B is lower than point A (you are looking downhill) it will be negative. When measuring the slope along a trail send an assistant ahead of you and tell them to stop just before they go out of sight. Then look through the sight of the clinometer and aim it at the part of your assistant's body, or at a mark on a pole held vertically in contact with the ground, that is level with your eye (if you aim for their feet you will introduce a bias of about 1.5m in your measurement since your eye is about 1.5m above the ground!). Repeat this process twice to be sure you get a consistent value and then record it (being sure to note whether it is in % or °). You might do this at every turn in a path (in which case you should also measure the distance over which the measurement is taken) or every 50m along a transect.

A clinometer can also be used to measure the height of an object, such as a tree. First find a spot where you have a good view of the base and crown of the tree in question, ideally at a distance about equal to its estimated height. Then aim the clinometer at the point where the main trunk enters the ground and record the angle (if there are large spreading buttresses try to aim for the centre of the trunk). Then repeat this process for the top of the canopy at a point directly above the trunk. Finally measure the distance from where you are standing to the base of the tree.

In order to calculate tree height from angles and distances you need a basic knowledge of trigonometry. Consider the right angled triangle in Figure B6.6.

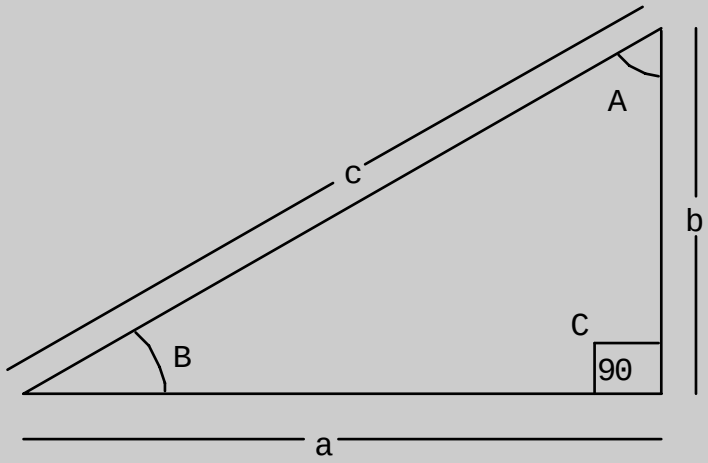


Figure B6.6. a right angled triangle

The various angles and distances in this figure are related to one another in the following manner.

For angles:

A° + B° = 90

90 - A° = B°

90 - B° = A°

For distances:

$a^2 + b^2 = c^2$ (i.e., the square of a plus the square of b equals the square of c)

a = c sine A, or c cosine B

b = c cosine A, or c sine B

tangent A = a / b, or a = b tan A or b = a / tan A

To calculate the height of the tree on level ground (Figure B6.7) first measure the height "a*", which corresponds to the height at which the clinometer is held (eye level). Then calculate the height "a". To do this measure "angle A" with your clinometer. Then measure "distance b" with a tape measure. Say, for example, b = 25m and angle A is 54°.

You know that a = b tan A

From a scientific calculator you can find tan 54° = 1.376

Hence, a = 25 x 1.376

= 34.4m

Hence, the tree height is 34.4 + 1.5 (your eye height) = 35.9m

Calculating the height of the tree on sloping ground is more complicated because to measure the height "a*" one would have to climb up the trunk of the tree (Figure B6.8). Furthermore, you will not be able to measure the distance "b". In this case you should record the two angles "A" & "A*", either side of the horizontal (for example, assume they are 35° and 25° respectively). Then measure the distance c*, from your position to the tree (say it is 37m).

In this case a* = 37 sine 25° = 37 x 0.42 = 15.6m.

Furthermore, b = 37 cosine 25° = 33.5m

Distance a = b tan A = 33.5 x 0.70 = 23.5m

Total tree height is 15.6 + 23.5 = 39.1m

Figure B6.7. Measuring tree height on flat ground

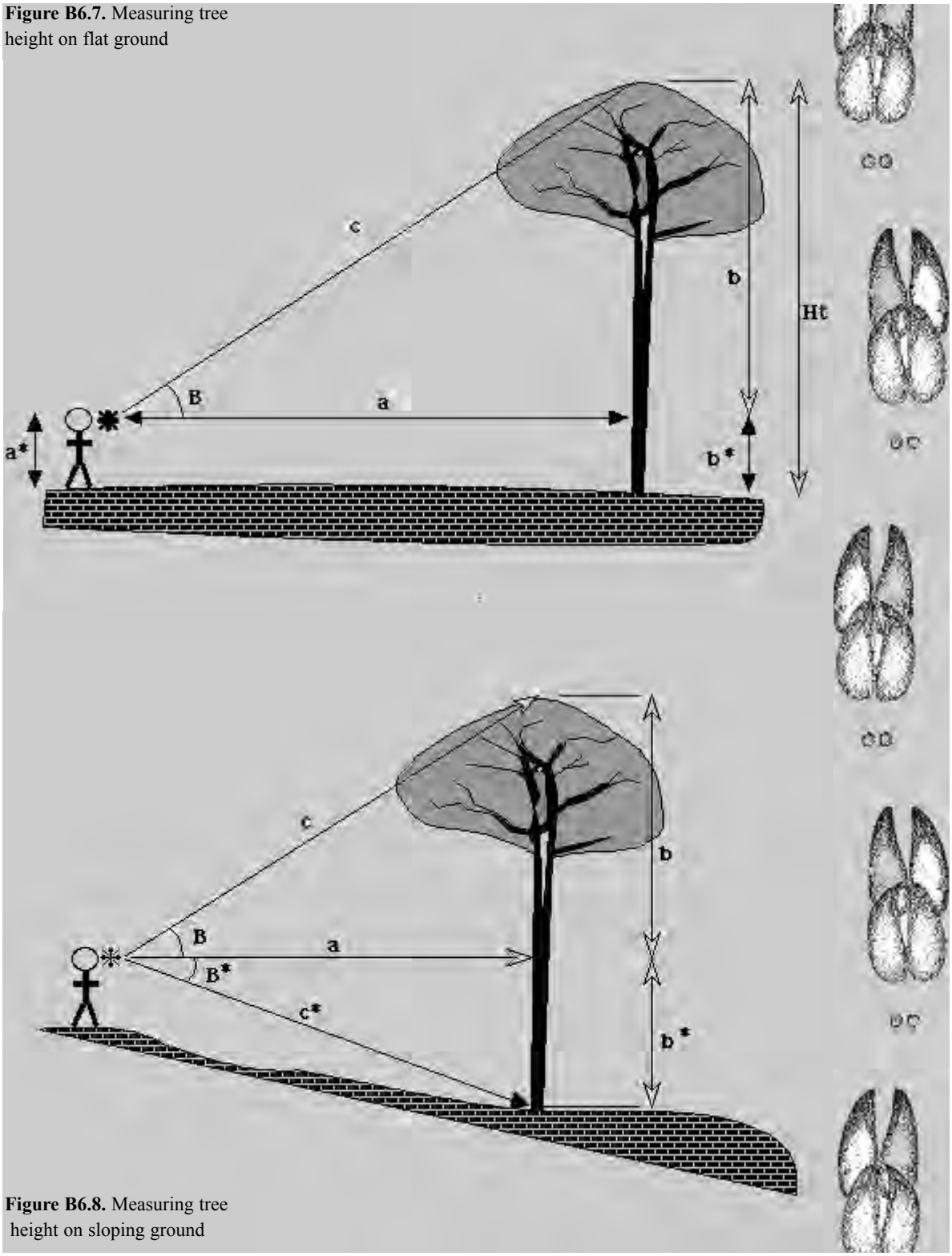


Figure B6.8. Measuring tree height on sloping ground

Finding your way using a map and compass

To find your way in an unknown, roadless area you will need a map and a compass. Before you start walking, use the map to look for the easiest or desired route to your destination. The shortest distance between two points is a straight line. But if large rivers, swamps or steep slopes fall along that line, it might be easier and quicker to plan a route around them. Waterways and ridgetops are often good places to walk, as major animal trails tend to follow these features. Try to minimise the number of contour lines that you cross so that you minimise the number of hills you must climb.

The best way to walk in an unknown area without getting lost is to walk from one clearly identifiable landmark to another, such as from a large bend in the river, to a hilltop, and then to a recognisable clearing; or alternatively from summit to summit along a ridge.

Mark your route on a map in pencil. Use straight lines where there are no paths to follow. Straight lines will allow you to use your compass to walk a compass bearing.

Calculating a bearing

You can use either a protractor or a compass to calculate the bearing you should follow to go from point A to point B on a map:

• First draw a faint pencil line joining points A and B;

• If you are using a protractor, place the centre point of the protractor over point A. Without moving the centre of the protractor from that point, rotate the protractor until 0° is parallel (in line) with the north arrow on the map. Then read the number written on the protractor that covers the pencil line you have drawn (Figure 6.8).

• To use a compass, line the north on the scale of your compass along map north. Then rotate the body of the compass (holding the scale still) until its north is in line with the line you have drawn between A and B, and read the bearing off the scale.

A **B** **C**

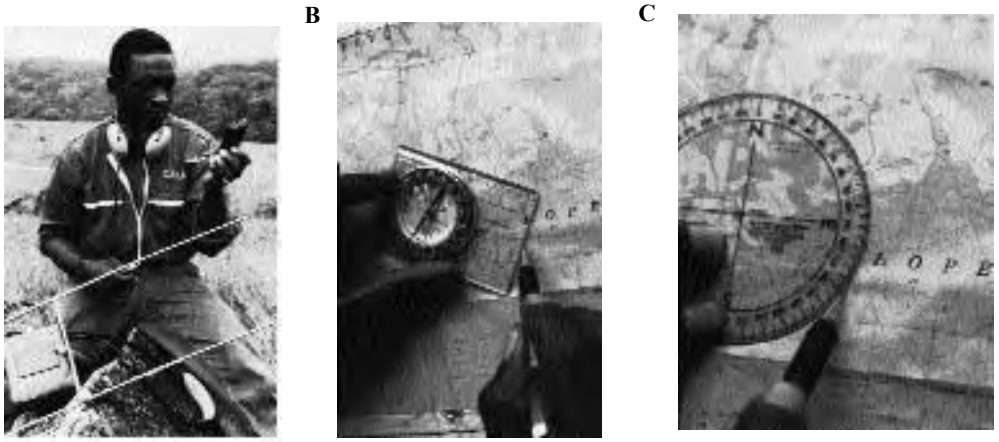


Figure 6.8. A. Taking a bearing with a compass; **B.** Putting a bearing on a map (compass dial aligned with north of map, bearing plotted is 120°); **C.** Putting a bearing on a map using a protractor (N is aligned with map North, bearing is 120°).

Orienting a map

To use a map in the field first you must orient the map to either a compass or to visible features in the landscape. You can orient the map to your compass by turning the map around until magnetic north on the map is lined up with north on your compass. You can orient the map to visible features in the landscape, such as mountaintops, by turning the map so that from where you are standing distant features line up with the map. From there the map can help you identify other distant features, and the compass can give you bearings towards them.

Walking a compass bearing

When you walk a compass bearing you walk a straight line in a single, pre-determined direction. First decide on the map where you are and where you want to go. Calculate the bearing between the two points as detailed above. Once you have calculated the bearing hold your compass in front of you and rotate the scale of the compass to the correct angle. Hold the compass still and rotate your body around until the needle of the compass corresponds to the north on the scale, and your chosen bearing is pointing directly away from you.

Look ahead and choose a clear landmark, such as a large tree or rock (use the sight of the compass if there is one to help choose the best landmark). Walk to this landmark. You can deviate around small objects; fallen logs, wasps' nests etc., as long as you get to the chosen landmark and are careful it is the right one. When you get there stop and repeat the same process, using a new landmark. For long distances, as you make your way towards your destination check that the features you pass correspond to those you expect from the map. If you will be walking long distances a compass with a strap worn as a watch is convenient as it keeps both your hands free.

It is difficult to maintain an exact bearing in the forest. If you need to do so, for example, to demarcate the limit of a protected area or logging concession, you will have to cut a transect (see Chapter 12). This will allow you to check the accuracy of your progress. Otherwise, you are likely to be thrown off course by the natural features of the landscape.

Navigating around obstacles

If you are demarcating a park boundary or cutting a line transect on a compass bearing and you come to a big obstacle, such as a cliff, use the following procedure to maintain your compass bearing (see Fig. 6.9).

Turn and walk exactly 90° or 270°from the direction you were travelling. When you have gone beyond the cliff, measure the distance you have deviated from your bearing or transect and proceed along your original compass bearing. When it is possible, turn 90° or 270° again, and walk the same distance back to your original line. Then turn one last time back on your original compass bearing to follow your original course.

Alternatively, if you can see across the obstacle, send one of your colleagues to the far side and have him or her stand at the point which corresponds to the continuation of your line.

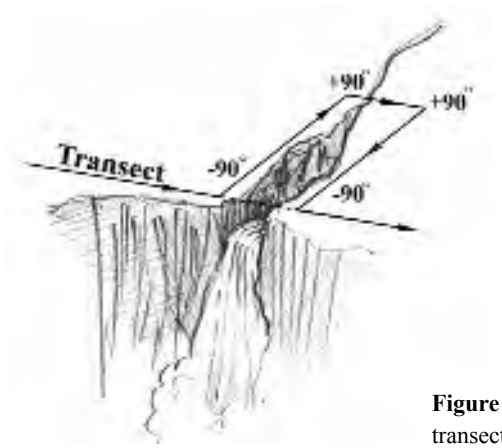


Figure 6.9: Deviating a line transect around an obstacle.

Deliberate offset

Given the high probability of navigational error the most efficient way to reach a predetermined destination is to use "deliberate offset". For example, if you are looking for the junction of two streams, you may attempt to walk a compass bearing straight to the stream junction. However, if you arrive at the stream and the junction is nowhere in sight, you may waste time travelling both upstream and downstream looking for the junction. If instead, you deliberately take a compass bearing to one side of the stream junction (deliberate offset), say to the south, then when you come to the stream you will know which way to turn to look for the junction. In this case you will turn north. This should save you time in the long run, but bear in mind that it is not infallible.

Determining an exact location through triangulation

Sometimes you may want to mark on a map the exact spot of a leopard kill, or an illegal hunting camp, or you may want to find your location if you are lost. If you do not possess a GPS unit (see Box 6g) this may be possible using triangulation. To find your location using triangulation, use a compass to measure the three directions to three identifiable landmarks, such as mountaintops. If only two landmarks are visible this method will work, but not as accurately as when three landmarks are used. If a number of landmarks are visible choose the "sharpest", such as a mountain with a well defined peak rather than a plateau. Estimating the distances to these landmarks is helpful but not necessary.

Plot on your map the back-bearing to each of the landmarks. A "back-bearing" is found by adding or subtracting 180° to a compass bearing (to keep the result between 0° and 360°). This gives you the angle you would get if you were looking in exactly the opposite direction, in other words, from the distant point back to the point where you are standing now. You should be standing at the intersection, or crossing, of the three back bearings (Fig.6.10).

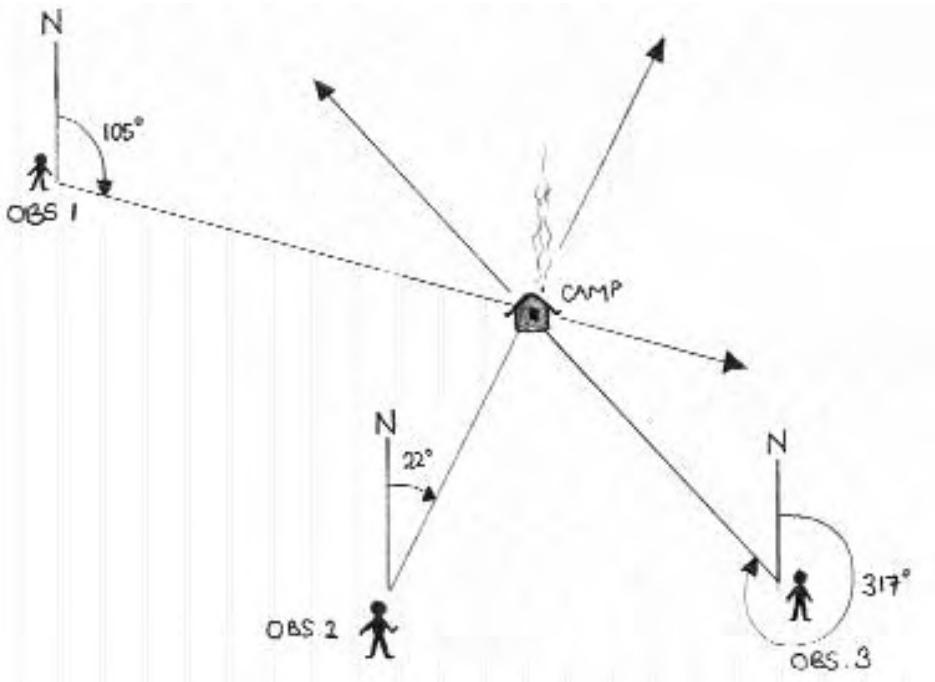


Figure 6.10. Using triangulation from three observers to plot the position of a poachers' camp.

Often these three lines do not cross at exactly the same place due to errors in measurements of bearings. Instead they delimit a triangle within which your location should lie. If the area is large try taking your measurements again to see where the error occurred, or add further landmarks.

The same method can be used to plot the source of a noise, such as a monkey call or a gunshot. If three observers in different positions hear the call or explosion, each can note the time (to be sure they are indeed hearing the same sound) and take a bearing. The location of the monkey or gunshot will be where the three bearings meet. Alternatively, if the noise continues for some time or is repeated intermittently a single observer can change his or her position and triangulate from repeated back-bearings.

Box 6g GPS UNITS

In the 1970's the U. S. Government began to build a satellite network to provide satellite-based navigation capabilities throughout the world. The network is called the Global Positioning System ("GPS"). Today there are 24 navigational satellites orbiting at about 17,500 km above the earth. Each satellite sends signals at the speed of light at regular intervals. At the moment each signal is sent the time according to an onboard atomic clock is recorded with a precision of one-billionth of a second. By registering the time the signal takes to reach a receiver on the ground a hand held GPS unit can calculate the distance to each satellite within receiving distance. Using this information the unit can calculate its position by triangulation. It needs to receive signals from three satellites to calculate a reliable position and if four signals are available the elevation of the GPS receiver can also be calculated (but beware that the error in the calculated altitude is often significant).

As satellites orbit the earth GPS units pass in and out of contact. They cannot receive signals from satellites orbiting on the other side of the earth, or those obscured by a physical feature such as a mountain range. Vegetation also interferes with the signals. Most GPS units currently on the market will only function well if you are in a large treefall gap or on a large road. One notable exception is the Garmin 12XL series that has an external antenna which permits constant satellite contact in anything but the densest forest vegetation. However, even this unit needs to be turned on in an open area and kept running so that it does not lose contact with the satellites. If you do not get a fix immediately try moving 10-20m to see if this enables you to pick up the signal. Alternatively, you can wait 10-20 minutes for the position of the satellites to shift. In low, dense vegetation try attaching your GPS to a pole to raise it above the foliage.

The position given by the GPS unit will either be in geographic or UTM coordinates (see Boxes 4a & 4b)and can be plotted on a map to show you where you are (see Box 6h). The position is not exact, but is generally accurate to about 100m. Accuracy can be increased greatly by having a second unit recording data in a known position, but this increases costs. Hand held GPS units are now relatively cheap (reliable units are available for about US$ 250-300) and are a great aid to navigation and data collection in large areas of unmapped rain forest. No researcher or protected area manager should be without one.

Note, however, that electronic devices are prone to break down when operated in humid forest environments. Never rely upon a GPS unit as your sole means of navigation. Always carry a compass and use it to track your approximate position. This way, if the GPS stops working or you run out of batteries you will still be able to to find your way using your map and compass.

Box 6h PLOTTING GEOGRAPHIC COORDINATES ONTO A MAP.

If you are lost in the forest and manage to get a position with your GPS unit, you will be able to rapidly assess your approximate position by looking at the longitude and latitude marked along the edges of your map (generally they are marked every 5' or 10' on 1:50,000 or 1:200,000 maps, respectively). If, however, you need to know your precise location you will have to plot the coordinates onto your map (for example, in order to locate a river confluence relative to your current position, where you have agreed to meet colleagues, or to check that you are indeed on the boundary of the reserve).

If your map has UTM coordinates and you are able to take a UTM reading, all you have to do is calculate how far you are from the marked north-south and east-west coordinates and measure this using the appropriate scale (for 1:50,000 maps, 2cm = 1km whilst for 1:200,000 maps, 0.5 cm = 1km). For example, if you are working on a 1:50,000 map in UTM Zone 32 and your GPS gives you a coordinate of 9977350S; 789600E, your position is 2350m south and 4600m east of the 9980000S, 785000E coordinate which is marked on the map. Since it is a 1:50,000 scale map, you measure 4.7cm south and 9.2cm east of the 9980000S, 785000E coordinate.

If your map or GPS unit only have geographic coordinates you will have to use these. For example, assume your GPS unit indicates you are at 00° 12' 04" S; 11° 36' 05" E and your 1:50,000 map has a mark at 5' intervals. You can plot your location on the map by measuring the distance from a known line, in this case 00°10'. Measure the distance between two 5' marks on the longitude scale. In this case the distance is 18.4 cm. Therefore, each minute corresponds to 18.4/5 = 3.68 cm and each second corresponds to 3.68/60 = 0.06cm. Therefore, your longitude is two minutes (2 x 3.68) plus four seconds (4 x 0.06) = 7.6cm below (south of) the mark at 00° 10'. Now measure the distance between two 5' marks on the latitude scale. In this case it is 18.45 cm. Hence each minute corresponds to 3.69cm and each second to 0.06cm. Therefore, your latitude is one minute (3.69) plus five seconds (5 x 0.06) = 3.99cm past (east of) the mark at 11°35'. Now measure 7.6 cm down from a point 3.99 cm along the line corresponding to 11°35' latitude and make a mark with a sharp pencil.

These two examples will put you in almost exactly the same position. Where are you? The first 10 people to reply correctly to this question will receive a free compass and tshirt.

Otter shrew
Potamogale velox

CHAPTER 7
Methods for recording the weather

Ann Edwards & Lee White.

Why is it important to record the weather?

Weather patterns strongly influence when plants produce flowers, fruits, or new leaves and help determine the quantities of fruits or flowers produced, and the number of seedlings that survive. These in turn affect the availability and distribution of food for animals, which influences when and how often animals reproduce or die and how often and where they move. On a short term scale, weather changes animals' local distribution, since they seek shelter from wind, rain or sunshine. Weather also can also influence how long animal signs last. All of these patterns, in turn, affect the way people use the forest. Therefore, weather data can enhance a researcher's ability to interpret data on species abundance, distribution, reproduction, mortality and behaviour.

The rate at which our climate is changing may be greater today than at any time in the last 10,000 years (Houghton et al., 1990). In particular, increases in the concentration of carbon dioxide and other so called 'greenhouse gases' are thought to be contributing to a gradual but steady increase in global temperature, which may have severe implications throughout the world (Box 7a). Unfortunately, due to lack of funds, many weather stations in Africa have ceased to collect regular data. If we are to monitor the severity of climate change, we will need reliable weather data from as many areas as possible. Given this, weather data collected in African forests take on global significance.

Collecting basic weather information is simple and mundane (see below), but it should be collected every day without fail in order to establish a baseline of information from which to assess future changes. During short-term plant or animal studies, the weather may be recorded only during the duration of the study. However, short-term weather data may not be sufficient to determine the effect of weather on the species in question. If weather patterns are monitored over many years, and a study is undertaken within that time, then it will be possible to see if data were collected during periods of "usual" or "unusual" weather.

Only once the weather has been monitored consistently for a long time can the "usual" patterns of temperature and rainfall be determined. Then from that point, the "unusual" weather patterns will become obvious. If plants or animals are studied during an unusually hot period, or during an unusually long rainy season, then the unusual weather can be taken into account when the results of the study are interpreted.

Weather is analysed by looking for patterns. Daily temperatures or rainfall mean very little by themselves. Usually, temperature and rainfall are reported as monthly, seasonal or yearly means or totals. When graphed, these means and totals show patterns, for example, the pattern may divide the year into the "cool, rainy season" and "the hot, dry season".

Weather patterns become more interesting once they are compared to patterns observed for plants and animals. For example, how do patterns of fruiting and flowering in trees relate to patterns of temperature and rainfall (see Box 7b)? How do patterns of reproduction and mortality in birds relate to patterns of temperature and rainfall, and eventually to patterns of seasonal fruit abundance? How do patterns of distribution and migration in elephants, pigs and buffalos relate to patterns of rainfall?

Box 7a INCREASED TURNOVER RATES IN TROPICAL RAIN FORESTS?

In a paper published in the journal *Science*, Oliver Phillips and the late Al Gentry (1994) surveyed tropical forests worldwide and found evidence of increased turnover rates (mortality and recruitment) in all tropical rain forest regions over the last decade. One possible explanation is that increased levels of atmospheric carbon dioxide have accelerated plant production. If turnover rates are indeed increasing, there will be wide ranging implications for conservation of biodiversity. Unfortunately when this study was published there were only scant data from Africa. By monitoring botanical plots over time you may be able to contribute to future analyses of this sort (see Chapter 9). However, your data will be of limited use unless you have reliable weather records.

Box 7b WHAT STIMULATES FLOWERING OF TROPICAL TREES?

Over the last 10 years Caroline Tutin and her colleagues working in Lopé, Gabon, have been monitoring 578 individuals of 63 species of trees and lianes whose fruits are eaten by gorillas or chimpanzees (Tutin & Fernandez, 1993; Tutin & White, 1998). Each month all individuals are visited and scored for flowers; ripe and unripe fruits; and young, mature and senescing leaves. Throughout this study weather data have also been taken. As the study has evolved it has been possible to identify some of the cues which trigger flowering in this seasonal tropical rain forest. Some of the patterns observed include:

- overall patterns of fruiting within the community are correlated to the number of hours of sunshine per month (more sunshine = more fruit). This correlation was determined using insolation (sunshine) data from weather stations elsewhere in Gabon because the team in Lopé did not have equipment for measuring sunshine.
- minimum temperatures in the cool dry season (Figure B7.1, Box 7c) stimulate flowering for a group of species, including several species of *Diospyros* (ebonies). If the temperature does not drop to 19°C (as measured on a max-min thermometer attached to a tree at 1.3m above the ground, in the forest) there is no flowering (and hence no fruit crop). In two years when there were unusually low temperatures in the short dry season, which is generally the warm season, (descending to 19°C) fruit crops appeared at an unusual time of year. In two years when the dry season temperature remained warm (never descending to 19°C) flowering did not occur and there were no fruit crops.
- some species seem to need a combination of climatic conditions to stimulate flowering. For example, the forest tree *Uapaca guineensis* appears to flower whenever there are 10 consecutive dry, sunny days (dry, cloudy days do not initiate flowering).
- about 70% of plant species in the Lopé sample are thought to respond to climatic cues, but the remainder seem to have endogenous rhythms. For example, each individual of the tree *Duboscia macrocarpa* produces flowers every 17 months regardless of weather conditions. Interestingly, there is no synchrony between individuals so fruit is available all year round.

In some years there are complete failures of certain fruit crops due to the absence of climatic cues for flowering. Considering that some responses are highly sensitive to climatic cues (e.g., temperature sensitive ebonies); and bearing in mind that many cues have not yet been identified, Tutin & White (1998) suggest that there may be unexpected consequences of rapid climate change. For example, if global warming results raises minimum dry season temperatures in Lopé, many forest plants may stop flowering and fruiting, resulting in fruit shortages for frugivorous forest animals (see Box 7a for another example).

Methods

The methods for collecting basic weather data (maximum and minimum temperatures, humidity and rainfall) are simple. However, they require some specialised equipment, and someone available to collect data every day throughout the year. The effort involved in what seems a simple job is often under-estimated. If rain gauges are forgotten until the heat of the afternoon, then part of the rainfall evaporates before it is measured. Data sheets are easily misplaced or lost. Research assistants or park personnel must be trained to read thermometers and measuring cylinder scales accurately. Max-min thermometers cause problems because scales go in opposite directions (see Fig. 7.1). So, if you want useful weather records make sure the person responsible for recording data is reliable (best to do it yourself!) and be sure there is a back-up person on hand for days when he or she is ill or on holiday.

Basic weather data should be collected at the same place every day for as long as possible. If data are always collected at the same place, then factors that can affect weather data (such as elevation, or the nearness of trees or buildings) will not change. This location is called a "weather monitoring station". Ideally, a weather monitoring station should be in some central, easily accessible place. If the site is accessible, then it is more likely that weather data will be collected consistently over many years.

Figure 7.1. Maximum-minimum thermometer. Note scales on the right and left are reversed.

If the resources are available, more than one monitoring station can be established. For example, one station can be established at park headquarters. A second station could be set up (in a clearing) at a research station situated inside the forest. Deciding whether to set up additional weather monitoring stations depends on 1) the ability to monitor the station daily, and 2) the degree of change in environmental conditions between stations. Potential reasons for differences between stations include: the distance between stations; the change in elevation between stations; change in the amount of standing forest (for example, a village with no standing forest versus a primary forest versus a palm plantation), distance from the ocean, etc. It is well worth checking a rainfall chart in your national atlas to see if there is likely to be a rainfall gradient in your region.

Weather data should be collected at the same time each day. The standard time to collect weather information is in the early to mid-morning, before the afternoon heat - 07:30 is usually convenient. Humidity data should be collected twice a day at 07:30 and 14:30 (slightly earlier or later if these times are inconvenient for some reason).

The most efficient way to collect weather information is on a data sheet. The data sheet should have columns for all categories of information to be collected: the date; the time; the maximum temperature; the minimum temperature; temperature of dry thermometer; temperature of wet thermometer; the daily rainfall (if possible note time rain falls during the course of the day and add this information to your checksheet); and the initials or signature of the person collecting the data (if this varies). The data sheet should have 31 rows, one for each day of the month, with room to summarise the data at the bottom. The month, year and location of the weather monitoring station should be clearly noted at the top of each data sheet.

Temperature

Two temperatures are recorded for each day. The daily maximum (usually occurring in the afternoon) and the daily minimum (usually occurring in the night). In order to collect this information a special "maximum-minimum thermometer" is needed. A maximum-minimum thermometer is actually two thermometers joined into one unit. One thermometer records the highest temperature experienced since it was last re-set. The other records the lowest temperature experienced since it was last re-set. Therefore, to collect the data, simply read the temperatures next to the marker on each thermometer, record the information, then re-set the markers. This will give you the highest and lowest temperatures for each 24 hour period.

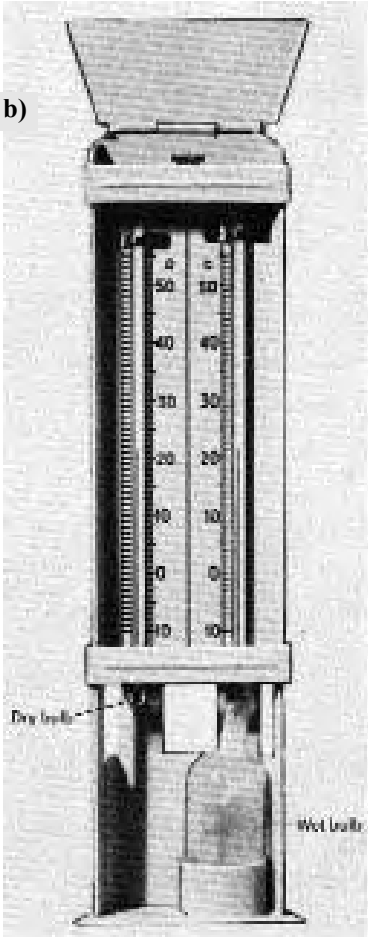
All thermometers should be placed in the shade. They should not be touched by direct sun or rain at any time of the day. You are interested in the air temperature, and not the temperature of the direct sun. It is best to build a small roof over thermometers placed in the middle of a clearing or on a tree trunk in the forest (see Figure 7.2).

Temperature data usually are summarised monthly, with separate summaries for the maximum and minimum temperatures. For example, all the maximum temperatures for April are added together then divided by 30 (April has 30 days) to get the average maximum temperature for April. The same is done for the minimum temperatures for April. Next these monthly averages are graphed in order to see yearly patterns (see Box 7c).

Humidity

Humidity is measured using wet and dry thermometers. The dry thermometer is simply a standard thermometer (rather than having a separate dry thermometer you can read the ambient temperature off your max-min thermometer). The wet thermometer has a tube of cloth (rather like a sock) over the bulb of mercury which sits in a container of water.

b)



a)

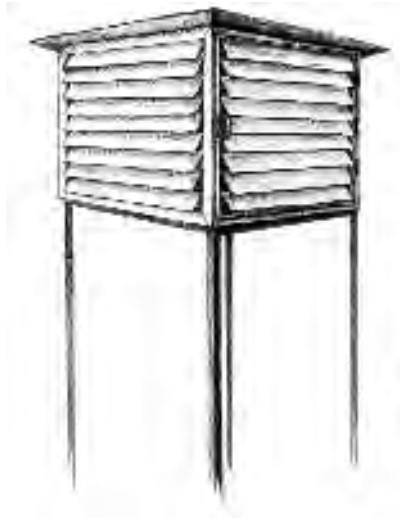


Figure 7.2. a) a simple structure designed to keep a thermometer protected from direct sun and rain; b) dry / wet thermometer.

You can make a simple wet thermometer using some absorbent cotton cloth and an empty camera film canister. Sew the material into the form of a tube which fits snugly over the bulb of a thermometer and extends down through a small hole pierced in the lid of the film canister. Keep the canister topped up with water. Water evaporates from the moist cloth, cooling the thermometer, resulting in a lower temperature (except when humidity is 100%). Humidity may be 100% in the early morning or during a rain storm (or you have let the water dry out!). Fan the bulbs gently and then take the readings once the temperatures have stabilised.

The difference in temperature between the wet and dry thermometers is used to calculate the humidity. The difference between the two thermometer readings is known as the wet bulb depression. For example, if you record dry and wet bulb temperatures of 29 °C and 23.5 °C respectively the wet bulb depression is:

29 - 23.5 = 5.5 °C.

Per cent humidity is obtained by looking down the column in Table 7.1 for a dry bulb temperature of 29°C (note that the dry bulb temperature is the same as the "ambient" temperature). Then look for the wet bulb depression of 5.5°C. The row and column meet at 61 which corresponds to the percent humidity (61%).

Table 7.1: % HUMIDITY, as determined by dry bulb temperature and depression of the wet bulb.

Dry bulb (°C)	**Depression of wet bulb (°C)**														
	0.5	**1.0**	**1.5**	**2.0**	**2.5**	**3.0**	**3.5**	**4.0**	**4.5**	**5.0**	**5.5**	**6.0**	**6.5**	**7.0**	**7.5**
10	93	87	81	74	68	62	56	50	44	38	33	27	21	16	10
11	94	87	81	75	69	63	58	52	46	41	35	30	24	19	14
12	94	88	82	76	70	65	59	54	48	43	37	32	27	22	17
13	94	88	83	77	71	66	60	55	50	45	40	35	30	25	20
14	94	89	83	78	72	67	62	57	52	47	42	37	32	27	23
15	94	89	84	78	73	68	63	58	53	48	42	39	34	30	25
16	95	89	84	79	74	69	64	59	55	50	43	41	37	32	28
17	95	90	85	80	75	70	65	61	56	52	47	43	39	34	30
18	95	90	85	80	76	71	66	62	57	53	49	45	40	36	32
19	95	90	86	81	76	72	67	63	59	54	50	46	42	38	34
20	95	91	86	81	77	73	68	64	60	56	52	48	44	40	36
21	95	91	86	82	78	73	69	65	61	57	53	49	45	42	38
22	95	91	87	82	78	74	70	66	62	58	54	50	47	43	40
23	96	91	87	83	79	75	71	67	63	59	55	52	48	45	41
24	96	91	87	83	79	75	71	68	64	60	57	53	49	46	43
25	96	92	88	84	80	76	72	68	65	61	58	54	51	47	44
26	96	92	88	84	80	76	73	69	66	62	59	55	52	49	45
27	96	92	88	84	81	77	73	70	66	63	59	56	53	50	47
28	96	92	88	85	81	77	74	70	67	64	60	57	54	51	48
29	96	92	89	85	81	78	74	71	68	64	61	58	55	52	49
30	96	93	89	85	82	78	75	72	68	65	62	59	56	53	50
32	96	93	89	86	82	79	76	73	70	67	64	61	58	55	52
34	96	93	89	86	83	80	77	74	71	68	65	62	59	56	54
36	96	93	90	87	84	81	78	75	72	69	66	63	61	58	55
38	96	94	90	87	84	81	78	75	73	70	67	64	62	59	57
40	96	94	91	88	85	82	79	76	74	71	69	66	63	61	58
42	96	94	91	88	85	82	80	77	75	72	70	67	65	62	60
44	96	94	91	88	86	83	81	78	75	72	70	67	65	63	61

Rainfall

Rainfall is measured in a specially designed and marked "rain gauge". Rain gauges should be mounted in a level place, a little above the ground (to avoid collecting splashing water). They should be located at least 30m from tall trees or buildings, since in a high wind these tall objects may affect the amount of rain reaching the rain gauge.

To read the rain gauge, hold the gauge level and read the number marked at the level of the bottom of the meniscus of the water (Fig. 7.3). (Most rain gauges record the amount of rain that has fallen in millimetres - some require you to pour the rain water collected into a calibrated measuring cylinder). Record the rainfall and then empty the rain gauge. Return it to its holder. Daily rainfall is measured as the amount of rain that falls in twenty-four hours. Therefore, it is important that the rain gauge be checked at the same time each day, even if it is raining at that time.

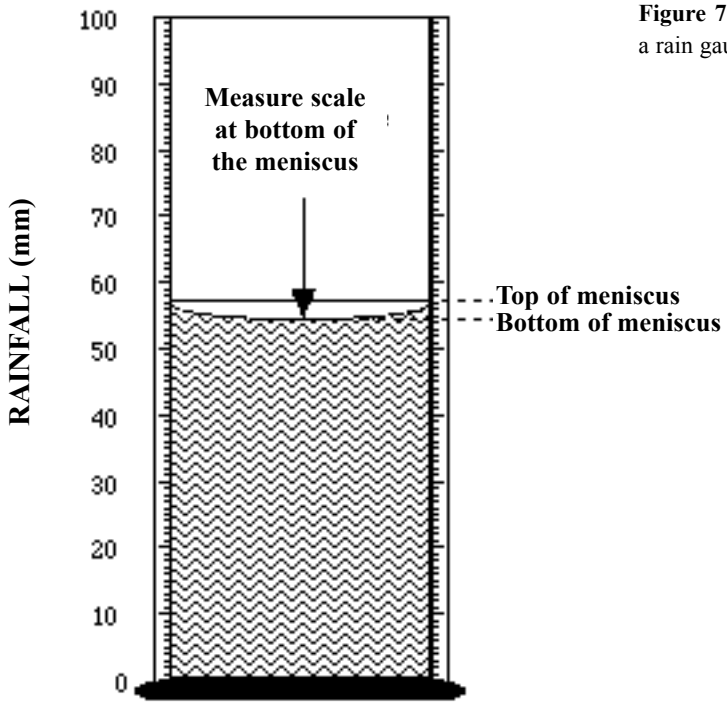


Figure 7.3. Reading a rain gauge

Rainfall usually is totalled (not averaged) monthly and yearly. For example, the amounts of rain that fell each day in April are added together to get the total amount of rain that fell in April. Rainfall also can be totalled seasonally (for example, over the four months of the rainy season, or the three months of the dry season).

When rainfall data are graphed together with temperature data, the periods when the forest is accumulating water, the periods when the forest is drying out, and the transition periods between these two states can be estimated (see Box 7c).

Water that accumulates on the ground over days and weeks can have a significant effect on certain natural systems, such as the height of rivers, the amount of time that small streams flow, the amount of soil that washes away, etc. Therefore, the total amount of rain that falls in one season, or in one year can be an important factor in the functioning of the local ecosystem, regardless of whether a lot of rain or a little rain fell on each individual day. For this reason, yearly averages are the most common way of summarising rainfall data. For example, one way to demonstrate that, indeed, it was an unusually wet year is to show that the yearly total rainfall was 500 mm above average.

Box 7c DESCRIBING SEASONAL WEATHER PATTERNS

The following method is useful for describing in which months an area is getting wetter, and in which months an area is getting drier (Walter & Leith, 1967).

1) First determine the average temperature for each month (add together all the maximum and minimum temperatures recorded in a month and divide by the number of temperatures recorded, for example, divide by 62 if maximum and minimum temperatures were recorded over 31 days).

2) Determine the total rainfall for each month (add together all the daily totals for one month - if you have several years of data determine the mean for each month).

3) Draw a graph as illustrated in Fig. B7.1. Put a scale for rainfall (measured in millimetres) on the left-hand axis. Put a scale for temperature (measured in degrees centigrade) on the right-hand axis. Note carefully that each 10 degrees in temperature should be the same height as each 20 mm in rainfall (so 20 degrees is the same height as 40 mm, 50 degrees is the same height as 100 mm, etc.). List the months along the horizontal axis. (for convenience, the scale for measuring monthly rainfall is sometimes altered above 100 mm, in order to accommodate months with high rainfall).

4) Plot your monthly average temperatures and monthly totals for rainfall on the graph. Draw a single line to connect all the rainfall data. Draw another line to connect all the temperature data.

5) When the rainfall line falls below the temperature line (light shading in Fig. B7.1), it indicates a drying period. When the total monthly rainfall is above 100mm (regardless of the temperature), it indicates a period of water accumulation (dark shading in Fig. B7.1).

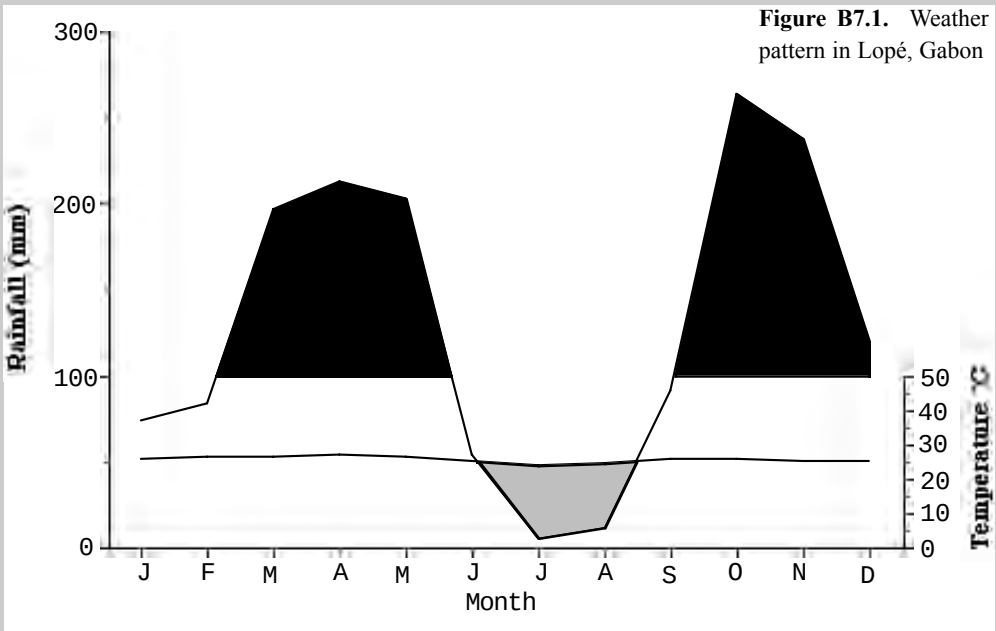


Figure B7.1. Weather pattern in Lopé, Gabon

CHAPTER 8
Collecting Botanical Specimens

Anthony Dold, Peter Phillipson, Rick Liesner, Peter Lowry & Lee White

Introduction

High quality plant specimens provide the foundation for any botanical inventory or ecological study. Unless you have a pressed, dried herbarium specimen (generally called a voucher) as proof that a species occurs at a particular locality and has been correctly identified, the validity of any inventory or plant food list for the animal you are studying holds little weight. There is a long tradition of botanical collecting in Africa, begun hundreds of years ago by early explorers. Plant collection is a science in itself, a building block for other studies, and a relaxing and stimulating pastime. In 1796 the great explorer and plant collector Mungo Park was stripped of most of his clothes and belongings by robbers whilst travelling in Nigeria. He wrote of his thoughts at the time:

> *"I sat for some time, looking around me with amazement and terror. Which ever way I turned nothing appeared but danger and difficulty. I saw myself in the midst of a vast wilderness, in the depth of the rainy season; naked and alone; surrounded by savage animals . . . . I was 500 miles from the nearest European settlement. All these circumstances crowded at once on my recollection; and I confess that my spirits began to fail me. I considered my fate as certain, and that I had no alternative, but to lie down and perish . . . At this moment . . . the extraordinary beauty of a small moss, in fructification, irresistibly caught my eye. . . for though the whole plant was not larger than the top of my fingers, I could not contemplate the delicate conformation of its roots, leaves and capsula without admiration. Can that Being (thought I), who planted, watered and brought to perfection, in this obscure part of the world, a thing which appears of so small importance, look with unconcern upon the situation and the sufferings of creatures formed after his own image? surely not!"*

He collected the moss, 'pulled himself together' (perhaps not in that order), and continued his exploration of Nigeria. The moss, a *Fissidens*, can still be seen today in the herbarium at Kew (Keay et al., 1964). The fact that specimens collected by the likes of Mungo Park and Charles Darwin continue to exist, and are still consulted by botanists, is a source of inspiration to any plant collector. Bear in mind when you collect and press a specimen that if you do your job well scientists centuries after your death will benefit from your work. But remember also that the quality of your collection and the notes that accompany your specimen need to stand the test of time and the critical eye of experienced plant taxonomists. Zoologists in particular have a reputation for collecting incomplete specimens (botanists require flowers and/or fruits, as well as leaves, to make sound identifications) or poorly preserved material, or worse still, no vouchers at all (e.g., see critique by Pannell & White, 1988).

Botanical collections are difficult to maintain, particularly in the humid tropics, where a multitude of insects and fungi represent a constant threat to your precious vouchers. It is therefore wise to collect several duplicates of each plant with a view to retaining one in your field reference collection, sending one to your national herbarium, and retaining at least one more which can be sent to the expert for that particular plant family in an international herbarium (this is also recommended in order to ensure that specimens are deposited at a wide range of herbaria, and are hence readily available for scientific study).

When he was a young peace corps volunteer, Mike Fay, now a field scientist working for the Wildlife Conservation Society in Congo, spent a year making a reference collection of botanical specimens, which was intended to be the start of a national herbarium in Central African Republic. At the end of his project he transported all of his specimens to Bangui, stored in the sheets of newspaper he had used to dry them (see below). He arrived late and left the specimens overnight in the building which was scheduled to become the herbarium. Next morning he returned to find that in the night thieves had broken in and stolen all of his newspaper to sell to market women as food wrappers - his hard won botanical collection was in disarray, piled up in one jumbled heap on the floor! (Mike did have duplicates so all was not lost).

The herbarium, with its orderly arranged specimens and complimentary library, is a vital resource in current botanical work for any field scientist. The preparation of pressed dried specimens is an ancient but simple and effective means of preserving a record of the earth's flora. This record provides an immense store of information that is a basic resource for botanical research, teaching and documentation and a building block for ecological research. An essential part of a botanist's training is to appreciate the value and uses of herbarium specimens, and be familiar with, and practiced in the basic methods and requirements of specimen preparation. Interested amateurs can also contribute in a significant way by preparing specimens, and specimen preparation is a good way to start learning the plants of a particular region. This practical guide to plant collecting and the use of the herbarium aims to provide basic information for the beginner, whether a botany student, a scientist who needs to work with plants, a professional conservationist or an interested amateur.

Why collect botanical specimens?

Collection of botanical voucher specimens is obviously a crucial aspect of plant taxonomy, but it is also important for many other disciplines, particularly as an aid to:

a) compilation of plant lists for a region, habitat or protected area;

b) identification of species which occur in botanical plots and transects;

c) identification of foods eaten or otherwise used by animals which are being studied (e.g., for construction of nests by gorillas or chimpanzees) , including identification of seeds and seedlings observed in dung;

d) identification of plants used by people, such as trees cut by loggers, leaves harvested as food or medicine, wood collected for fire wood or to make poles to support yams etc.; and

e) species identification of plants (or groups of species) which local people are able to recognise and name, such that you can benefit from the knowledge of local assistants.

As such, it is difficult for field ecologists and anthropologists to work effectively unless they know how to collect and document voucher plant specimens.

Collecting plant specimens

Herbarium specimens are permanent records of a species (or population) as it occurred at a given time and place. The value of every specimen is dependant on the care with which it is selected, collected, pressed, dried and labelled. A small number of really well preserved and well annotated specimens is far more valuable than a large number of poor specimens.

Each collection should:

1) be as representative as possible of the species in a given area, consisting of typical (or "ordinary") specimens and / or including specimens that show its range of variation,

2) have a collection label carrying certain essential data (the name of the collector, collection number, date, locality, habitat and botanical details likely to be lost on drying - see below), and,

3) should be well prepared and preserved.

Collecting equipment

Essential equipment

- compass and maps of the area (if available);
- secateurs;
- sharp knife for slicing bulbs and fruit, and sharpening pencils;
- a strong garden trowel and/or small pick for digging out bulbs etc.;
- clear/opaque large plastic bags (avoid black bags);
- hard-backed pocket-sized notebook and pencils (NB. use a sharp pencil to make notes as this gives a permanent record; never use ball-point pens);
- plant press, including press frame, straps, cardboard "corrugates", blotters and newsprint (see the description of a plant press below);
- hand-lens or magnifying glass.

Other useful items

- pole clippers/long-arm pruners if intending to collect trees;
- tree-climbing gear;
- binoculars (to search forest canopy for flowers or fruits);
- GPS unit;
- altimeter;
- tie-on tags that can be written on with a pencil;
- slotted cards for securing folded specimens;
- plastic screw-top bottles of liquid preservative;
- small envelopes for seeds;
- muslin or paper bags for fruits.
- photographic equipment;
- 70% alcohol for preserving pressed plants if drying is expected to be delayed;
- preservatives for spirit collections (see below).

What to collect

Sterile specimens, that is without fruits and or flowers, are of limited use. However, when undertaking botanical plots or observing animals feeding, the plants of interest will not necessarily have flowers or fruits and one is obliged to collect a voucher with only leaves. This may enable partial identification (or complete identification if you are fortunate) and certainly provides you with a valuable means of checking whether you encounter the same species again. But a sterile specimen is of little use for taxonomic study and such specimens are not generally maintained permanently in herbaria collections (although you may choose to conserve them in your own field herbarium - see below). In long-term studies sterile collections should be considered as a first step, and whenever possible you should monitor the plant and collect it (or another individual of the same species) again once it flowers and again when it fruits. If there is only a single flower or fruit, complement this with sterile material. On the other hand, when material is plentiful, collect extra flowers and fruits which can be used to supplement the main pieces and provide specimens for later dissection by specialists.

Look for variation in foliage, flowers and fruits on each plant and differences between plants and ensure that what you collect best represents this. Usually each collection will be made up of pieces of a single individual plant. In the case of dioecious plants (plants which have male and female flowers on separate individuals) make separate collections of male and female plants, cross-referencing them in your notes. For small plants several individuals will have to be collected, taking care to ensure that each collection consists of a single species (sometimes two or more very similar species grow near each other). If there is doubt, this must be noted, and, if possible separate collections made.

For herbaceous plants rootstocks and other underground parts are valuable and should be collected. For trees and shrubs bark and wood samples provide valuable additions. However, collection of wood involves inflicting wounds which can make the tree susceptible to disease and may even result in its death. So do not collect wood from plants intended for long-term monitoring.

Collect enough material to fill a herbarium sheet (which will be 46 x 26 cm - see below) and for each duplicate sheet you require. Don't make many little specimens. It is better to collect a single piece large enough to fill the sheet which can be supplemented by a sprig with additional flowers, fruits or different sized / shaped leaves.

To collect specimens from tall trees you will need pole clippers, climbing gear or other suitable equipment. If you do not have this, search the ground for fallen twigs, flowers or fruits (but use binoculars to check that they came from the right tree and note that the collection is material picked up off the ground).

When collecting a parasitic plant, search for and record the host. Better yet, make a collection of the host if possible.

Floating or submerged aquatic plants and delicate algae collapse in a mess when lifted out of water unless care is taken to "float out" the specimen onto a sheet of paper, which is submerged under the specimen and lifted gently, letting the water run off and leaving the plant lying in a natural position.

After collection, the specimens should be pressed before they deteriorate from wilting and before flowers wither, fall or close. Although in some cases plants need to be pressed immediately, most will last well enough for a few hours in a clear or light coloured plastic bag ("jute" or nylon rice / flour sacs are readily available in many places). It is useful to carry a small plant press into the field (a field press) so that the more delicate plants, or at least delicate flowers, can be pressed as soon as they have been collected. Alternatively they can be put in a folded sheet of tissue paper and pressed temporarily in a note book. The more durable specimens can be placed in large bags and pressed on return to your vehicle or base camp. When collecting in bags, first label the specimens and then fold them to the correct length for a herbarium sheet and place them firmly, but carefully into the bag. Do not just drop them in. This way separate collections will not become entangled and there will be less damage. To empty the bag turn it upside down and carefully dump all specimens on to the ground. Large, heavy plants are best put in separate bags. With experience a collector will soon know which plants will need pressing straight away. Obviously weather conditions influence how long a specimen will remain in good condition. Specimens in plastic bags will last longer if the bags are protected from too much movement, are not left lying in the sun, are relatively full of plant material and if the open end is tied with string to decrease evaporation. In some cases, after a day of collecting and pressing, wilted specimens may revive if left in water overnight and pressed the following morning.

Duplicate or replicate collections

Collectors often collect and press more material of a particular plant than will be needed by the herbarium at their home institution. The extra material is divided into sets which can then be distributed to other institutions as gifts or, if prior arrangements are made, in some cases as exchanges for other specimens, books or materials. The set of specimens that comprises each collection is called a set of duplicates. A collection that consists of a single specimen is referred to as a unicate. Collectors who have collected large replicate sets have thus left a very valuable legacy.

The main reasons why replicates should be collected, are as follows:

1. Collecting permits may require that the collector deposits a duplicate in a particular national or regional herbarium as a condition for the permission to collect specimens. Even if this is not the case it is good practice to give duplicate specimens to your national institution, where they will almost certainly be appreciated.

2. If numerous duplicates are placed in various herbaria a larger number of botanists will have access to your collections without the expense and delay of borrowing them or travelling to the institution where they are stored.

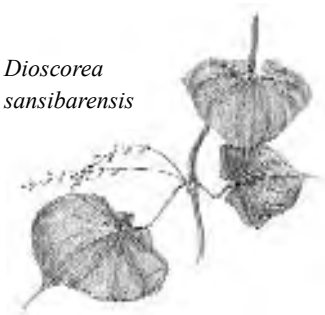
3.Duplicates sent to a specialist are likely to be identified quickly and accurately, and the value of each duplicate is enhanced if one of the specimens has been named by an expert who has access to and is working with authentically named collections. In turn, taxonomic research on the specialist's group benefits from your collection, which may represent a new population, or even a new species.

4. The more material of a particular collection that is available to the taxonomist, the better it can be interpreted.

5. You may be able to exchange specimens with international herbaria to obtain duplicate collections from other areas of interest to you, or in some special cases, in exchange for equipment or publications that are otherwise too expensive.

6. If a disaster such as a fire or a war destroys your specimens in one herbarium (a situation that unfortunately has occurred several times in the last few decades), your collecting efforts will not have been in vain if replicates have been distributed elsewhere.

The number of duplicates you decide to collect will depend on the number of herbaria with which you collaborate and the amount of time you have available and the facilities you have for drying and storing specimens. You will probably want to collect one duplicate for each institution, one for your reference collection and at least one more which can be sent to an appropriate expert. As a rule of thumb, aim to collect a minimum of 5 duplicates. Increase the number if you suspect that a species may be rare or new, or if you get the chance to collect a good specimen of something which is likely to be rare in herbaria, such as good material of a species with short-lived flowers or a large, recently fallen canopy tree with flowers or fruits.



Dioscorea sansibarensis

Field notes

The importance of good field notes for the plants collected cannot be overstressed. Although many collectors, past and present, have not set a good example, detailed accurate field notes add enormously to the value of the specimen for taxonomic research and may make the specimen easier to identify. Moreover, completely inadequate notes make an otherwise good specimen almost worthless! Field books for recording field notes are essential. They need to be sturdy to give a firm writing surface and, if possible, should be water resistant. Pocket-sized hard-backed notebooks are most convenient. Use pencils for note taking: they do not wash and are permanent. Propelling pencils are always sharp but ensure that they are equipped with a soft lead (carry spares). As with any data recording, take care to be legible - you will probably need to send a copy of your field book to the herbarium where the first set of your collections is stored, such that they can prepare herbarium labels, so the notes are not only for your own eyes. Keep an up to date copy of your book in a safe place, in case it is lost, stolen, eaten by termites, converted into a rat nest, or suffers some other disaster.

A description of the collection locality and botanical features that will not be preserved after the specimen has been pressed and dried should accompany each plant specimen. Record the collector(s) name(s) and the date of collection, and assign a unique number to each collection.

Take notes as methodically as possible in a consistent sequence and format. This will simplify the task of producing specimen labels. After noting locality details (one locality entry can cover several collections from a given site - make a clear note each time you change locality), note details for each specimen collected at the locality. Some collectors, particularly less-experienced ones, like to use blank forms which have a small space for each piece of information that should be recorded. These forms do not generally have enough space for good note taking and a "free-form" approach to note taking is preferable.

Notes should be taken in the field when you collect the specimen. Apply the following rule of thumb: *record any information that will not be apparent after the plant has been collected, pressed and dried.* For example, record flower or fruit colour, any odours or exudates, the height of trees, and other features such as buttress or stilt roots. However, you must be selective in making specimen notes; obviously time spent making notes must be balanced against time spent collecting and pressing specimens. At the same time, botanists who study and attempt to identify your material will need to know about some important features which you must record. Note-taking should therefore be concise but precise and limited to those features judged to be particularly important. Always leave a line or two between collection numbers so that the original identification, and later name changes and notes can be included. Detailed guidelines for the type of information to record are given in the following sections.

Collection numbers

Each collection must have a unique number which is recorded both in your field notebook and eventually on the label accompanying each duplicate specimen. The numbers should form a single running number sequence, starting at "1" and continuing unbroken. Together the collector's name and number provide a way of referring to a collection made of a particular species at a particular locality on a particular day. A combination of collector's name and number should only be used once. Thus even if you collect the same plant species at different times of the year from the same locality each collection will have a different number. The collections should be cross-referenced to each other on the specimen labels.

When two or more collectors are working together it is usual to use the number sequence of one of them, adding the other collector's names on the specimen label. Thus the specimen Mackanga & Dimoto 1189 would be number 1189 in Mackanga's collecting sequence, and was recorded by Mackanga in his field notebook. Dimoto may have helped gather or press the specimen. It is advisable to decide in advance how one is going to number specimens and divide the work before a collecting expedition.

In cases where it is discovered later that a single collection number consists of a mixture of species which will need to be separated (this often happens with Bryophytes and Algae), distinguish each with a suffixed letter e.g. Bourobou 1431a, Bourobou 1431b, etc. Be sure to update the field book accordingly.

The following list provides examples of the kind of information that should be provided for each collection. The locality and habitat information will be recorded once in your field book for each collection site, whilst the remaining information is specific to each species collected.

Locality

Record the locality as precisely as possible with a grid reference and an altitude reading obtained from a map or using a GPS (Global Positioning System) unit and an altimeter. Try to record grid co-ordinates to the nearest 10 seconds, or to the nearest 1/100 of a degree if using the decimal degree system, and altitude to the nearest 50m. Describe the locality in words including as many details as possible to enable someone else to relocate the exact spot. The actual site also needs to be described, for example: "Crest of hill behind forestry camp", one could go on to note: "200 meters west of source of small stream", however one should avoid using impermanent land marks in the description, such as temporary hunting camps. Draw a sketch map in your field book if this will clarify written information. Whatever the case may be, the more relevant details supplied the better - think about how you would describe to somebody hoping to visit your collection locality how to get there. This information should be noted methodically, and is usually printed in descending order of importance on final herbarium labels.

Habitat and ecology

- landscape topography (mountainous, rolling hills, coastal flats, etc.);
- slope (level, gentle, moderate, steep, etc.);
- position in relation to topography, (hill-top, mid-slope, valley bottom) and position within a vegetation type(e.g. forest margin, gallery forest, semi-montane forest);
- aspect (North, South, East, West, North-West, East-South-East, etc.);
- general vegetation type (degraded or montane forest, stream bank, etc. - see Chapter 9);
- soil/rock type;
- extent of water availability/degree of water-logging;
- salinity, alkalinity;
- associations with other species (list adjacent species and collection numbers of those you collect);
- successional status - does the site carry climax vegetation, a pioneer community or a successional stage?
- disturbance, human or other.

General comments for each specimen

- relative abundance, dominance, how many, how common, single or gregarious?
- variation in any features of the population;

Reproductive Ecology

- means of vegetative spread (stolons, rhizomes, etc.);
- observed pollinators (accompanying cross referenced insect collections are very valuable);
- seed and fruit dispersal (wind, water, animals, explosive);
- age structure of populations - is the species you collect represented only by mature plants, are seedlings present, etc. ?

Guibourtia tessmannii

Habit

- life form:
  - Herb: a non-woody plant rooted in the soil.
  - Vine: an herbaceous climber.
  - Shrub: a woody plant branching at ground level.
  - Treelet: a single-stemmed woody plant less than 2 m tall.
  - Tree: a single-stemmed woody plant greater than 2 m tall.
  - Woody vine (liane): a woody climber.
  - Epiphyte: plants growing on other plants, particularly large trees - may be ferns, mosses, lichens, non-woody (e.g., begonias) or woody (e.g., strangler figs).
  - Parasite: parasitic on another plant and lacking leaves.
  - Hemi-parasite: parasitic on another plant but possessing leaves.
  - Saprophyte: surviving on decaying organic matter.
- overall shape (especially trees and shrubs) and whether clump-forming;
- branching pattern.

Sap, latex or resin

- colour, stickiness, and amount of sap, latex or resin exuding from cuts.

Scent / odour of flowers, fruits or cut parts

Underground organs

- tap root, fibrous root, tubers on roots, extent of roots;
- rhizome - depth in soil, length and spacing of roots;
- bulb, corm or tuber - size, shape and structure (presence of outer covering - tunic and scales etc.);
- colour/texture/firmness/moisture content.

Stems and trunks

- number of trunks;
- total height, diameter at breast height (for trees that are unbranched to ca. 1.5m);
- height of stem before first branches;
- buttressed or not;
- bark colour, texture, thickness, lenticel colour;
- wood hardness, colour, grain;
- trunk-slash colour, texture, smell, exudate, etc.;
- shape in cross section - fluted, circular, etc.;
- internode length;
- persistent thorns or spines on trunk.

Leaves

- texture, smell, colours (both surfaces), glossiness, glaucescence;
- exudate or glands, stickiness;
- orientation in relation to stem or petiole e.g. "pendulous";
- note size and outline shape of large leaves when only a portion is taken, e.g. with palms;
- note any variation in leaves, for example basal leaves often differ from upper ones, young ones may differ in colour to mature ones, coppice shoots and seedlings often bear differently shaped juvenile leaves.

Inflorescence

- exudate or glands, stickiness;
- cauliflorous (flowers on trunk), ramiflorous (flowers on large branches), any other data on position, orientation or form that may be lost in prepared specimen;
- colour of axis.
- note size, shape and structure if only a portion can be collected.

Flowers

- Note if heterostylous or if plant is monoecious or dioecious, examine several specimens if possible;
- scent;
- exudate or glands, stickiness;
- calyx colour and texture;
- corolla colour*, shading and texture;
- filament, anther, ovary, style and stigma colours*;
- behaviour (e.g. open early and closed by noon).

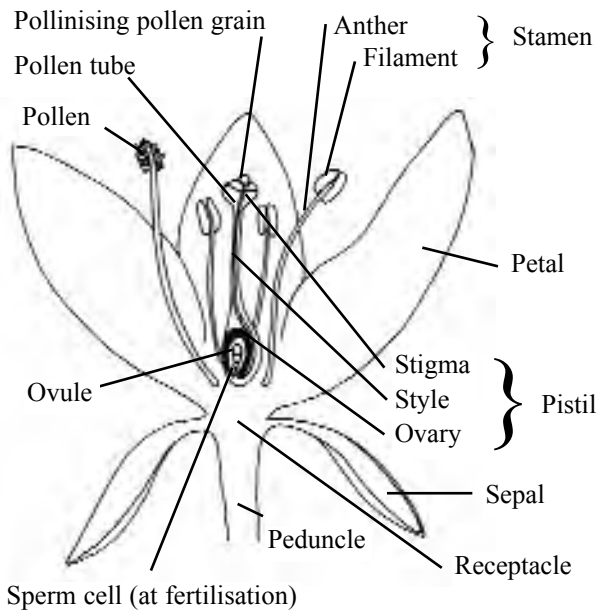
Fruit and seeds

- smell;
- colour*, texture;
- size, shape;
- seed coat colour* and texture;
- aril colour* and texture;
- dehiscence (explosive, disintegrating).

* colours can be described most accurately by referring to a colour chart such as the "Horticultural Colour Chart" (1938) issued by the British Colour Council and the Royal Horticultural Society (cite the specific colour chart used in your notes).

Descriptive Terminology

When describing characters of a plant for a herbarium label, regardless of whether it is a part of the plant collected or not, it is important to use the correct terminology. For example a statement such as "Flowers red" can be misleading when the only red parts of the flower are the petals. Features that are often lost or changed when a plant is pressed and dried need to be described correctly; the same applies to parts of a plant impossible to collect like the stem of a tree. Figure 8.1 illustrates the terminology used for a typical dicotyledon flower. Whenever you are uncertain of terminology add a sketch to your field notes.



Names

Try to assign at least a provisional name to each species in the field as an aide memoire. Even something descriptive such as "prickly legume" will help you sort out specimens if they get confused after pressing. Do not waste time trying to identify plants in the field; this should be done later back in the base camp or the herbarium, where you have access to reference material and literature.

Ethnobotany

Information about what people called plants and what they use them for can be recorded, but this must always be obtained from a reliable informant who has examined the actual plant you collect (show the informant the whole plant preferably still growing in situ). Avoid prompting the informant too much; people will often tell you the things they think you want to hear regardless of whether they are true. Having two or more informants can be useful, since they will be able to discuss the plants and reach a consensus between them in cases of uncertainty. Be tactful in dealing the informants, explaining why you want the information they are being asked to give freely. The following information can be gathered:

- vernacular names (be certain to note the language/dialect);
- human and animal foods;
- use for construction, in weaving baskets or roofs, as fuelwood, as a fish poison, for making canoes or household implements etc.;
- medicinal uses;
- folklore uses;
- religous uses.

Spirit collections

Although pressed dried herbarium specimens are an ideal way of preserving the majority of plants, some, e.g. orchids and succulents, make poor specimens when pressed. In such cases it is appropriate to preserve material in spirit. Flowers, whole inflorescences, soft fruits and fleshy rootstocks can also be usefully preserved in spirit (e.g. composite inflorescences become distorted and fan-like when pressed and the arrangement of the involucre is lost, or the exact aspect of the corolla segments in a Papilionaceous flower can become obscure). Since specimens retain their life-like appearance, spirit collections are valuable both for specialists, since the plant parts are preserved in their original 3-dimensional state, and for botanical illustrators. They also provide material for anatomical and micro-morphological analyses.

Preservatives

Whilst many herbaria use mixtures containing alcohol, formalin and acetic acid (see Bridson & Forman, 1992), 70% alcohol is generally available and works fine. Alcohol distilled locally for human consumption, which is widely available in many countries, is an acceptable temporary alternative.

Glass jars and vials with plastic stoppers are ideal (metal screw on type caps rust and corks don't seal perfectly). All spirit material should be cross referenced to a Herbarium sheet, and should be labelled in the same way, one label written in pencil inside the bottle and one on the outside, varnished for protection if possible. The collections should be checked for evaporation regularly. Strong plastic bags (e.g. whirl packs) can be used for collection in the field but specimens should be transferred to bottles as soon as possible.

Photographs and drawings

Illustrations are a valuable way of increasing the utility of plant specimens. Photographs taken of plants in their habitat can convey as much information as lengthy descriptions. Features of the habit of large plants such as tree architecture are difficult to describe in words, and photographs of whole plants or sketches of the specimen are very useful. Parts of plants that do not press well, such as succulent stems and leaves, fleshy fruits or complex flowers can also be depicted accurately in photographs or sketches. Colouration of flowers and fruits, and details of markings also are easily preserved in colour photographs (but note than some colours are modified on film, particularly shades of blue).

Photographs and drawings should always be cross-referenced to specimens, and either mounted on herbarium sheets or filed separately in herbaria that maintain separate collections of illustrative material. The label accompanying the voucher specimen should indicate that a photograph was taken. Transparency slides provide the best quality and most versatile reproduction.

Good botanical photography is not easy, and suitable cameras and accessories are needed. For the best photographs a flash is often used, especially for work in forests and for close-up work. This helps to overcome problems of poor depth-of-field in natural light. A piece of black fabric is invaluable to provide a neutral background for individual specimens. A macro (close-up) lens and a tripod may also be useful. Many books are available on the subject of nature photography for those wishing to develop these particular skills.

Field sketches are usually made in pencil. They are not intended as great works of art; technical drawings should depict particular features of the plant which will be difficult to observe on the final pressed specimen. It is important to use good quality drawing paper, since ordinary paper may not be of archival quality and will deteriorate with time. Do not redraw the sketch; preserve the original.

Pressing and drying

General Principles and Methods

Pressing should be done as soon as possible after the field notes have been completed (some people choose to make preliminary notes in the field, which they complete at the base camp). If you are working with a companion, one person can make notes while the other presses the specimens (for which note-taking is finished). Plant specimens are pressed by first arranging them in a folder of thin paper (newsprint is ideal), sometimes known as a "flimsy", taking into consideration the size of the final herbarium sheet (46 x 26 cm). When collecting in rainy weather shake off excess water from the specimen before pressing.

Carefully clean soil from rootstocks, using water if necessary, separating unwanted pieces and breaking dense clumps into smaller parts. Disentangle matted stems of climbing plants; their supports should be removed completely or trimmed to a minimum.

Specimens should not protrude from the folder and should be bent inward if necessary. Plants too long to fit the paper may be bent sharply into a "V", "N", or "W" figure, bruising or pinching the stem before bending will make it less likely to break and thicker stems can be partly cut. The folds can be secured to prevent them springing open with slotted cards. However, in the case of vines, long specimens should be simply curled around within the available space.

Spread flowers or inflorescences to show as many surfaces or views as possible. Section some flowers longitudinally and press flat to exhibit the inner parts and thereby reduce the need for difficult and damaging dissection of the prepared dried specimen.

Specimens may be trimmed to reduce bulk, to avoid having excessive overlap and to expose certain key characters, such as flowers, provided that sufficient material is left so that the pattern of branching, leaf arrangement, and other features are readily discernable. Be careful not to trim away all the leaf tips. Do not make a specimen composed of several small pieces; try to retain "organic connections" as much as possible. Densely clustered leaves may be reduced in number but always leave the bases of the petioles attached to the stem so that the original number can be determined. Be careful not to cut the petiole base and stem attachment of a compound leaf. Arrange specimens so that both upper and lower leaf surfaces are exposed, but try to retain a natural orientation of the plant parts as far as possible. No leaves should be left protruding out of the press - trim and fold as appropriate.

Excessively bulky and fleshy parts such as stems and fruits must be split and both parts included. Certain plants, such as those with large leaves (e.g. palms) and succulents, present particular problems for pressing. Those and other problem plants are discussed below.

Write the collection number on each folder conspicuously on the outside in the right hand corner of the long open edge, using an indelible marker or tie a numbered tag on every specimen (including each replicate). Write the collector's initials before the number to avoid confusion when the specimens are transferred to the herbarium, where other collectors' material is being stored. The specimens should remain in their numbered folders right through to the stage when they are mounted, to avoid them getting muddled with others and to prevent loss of small parts that may have fallen off (particularly seeds). Include only a single numbered collection in each folder, and use extra folders for each replicate specimen collected. Collect enough material to fill each replicate sheet, avoiding crowding or excessive overlapping.

The plant press

A plant press consists of two light wooden frames a little larger than a herbarium sheet, about 46 x 30 cm, in between which the specimens are sandwiched in their folders interspersed with "driers" or "blotters" (sheets of thick absorbent paper) and "corrugates" (sheets of corrugated cardboard or aluminium), which help to aerate the press and also help to keep the specimens flat. It is normal to have the corrugations in the sheet running across its width so that they run vertically when the press is laid on its side on a drier, to allow for convection of air through the press. The blotters and corrugates should be the same size, preferably the same as the press frames or a little smaller, and the folders should be no larger than the blotters and corrugates.

The usual system of making up a press is as follows: bottom half of press, corrugate, blotter, folder with specimen, blotter, corrugate, blotter, folder and so on. Complete the "sandwich" with a blotter, corrugate and the other half of the press frame. Strap up tightly using 2 straps, however do not use excessive force, the aim is to hold the specimens flat while they dry not to squeeze the moisture out of them!

When building a press always keep the numbered side of the newspaper facing upwards. The size of a press is limited by the length of the straps or the size of the drier, but avoid building presses too large if you are travelling long distances on bad roads, because they tend to work loose and specimens can be damaged, or the press can break open.The press is then placed on a drier. Excessive heat causes the specimens to become brittle but on the other hand insufficient heat will leave the specimens wet for too long and leaves tend to fall off, colour is lost and mould grows. The press should be checked regularly and wet blotters replaced as often as possible. The straps should be tightened if necessary (they almost always become loose as the plants dry); failure to do this will result in distorted or shrivelled specimens, which are less easily stored and are susceptible to damage. Some collectors like to leave specimens in the press overnight and rearrange them as necessary the following morning, before commencing the drying process. This results in better quality specimens but adds to your processing time.

Figure 8.2 A botanist with field equipment, cutting a specimen with botanical poles, trimming specimen with secateurs and arranging it in a field press, taking notes, closing press with gentle pressure, preparing specimens for drying (with help from a colleague), placing press in drier and finally removing pressed, dried specimens.

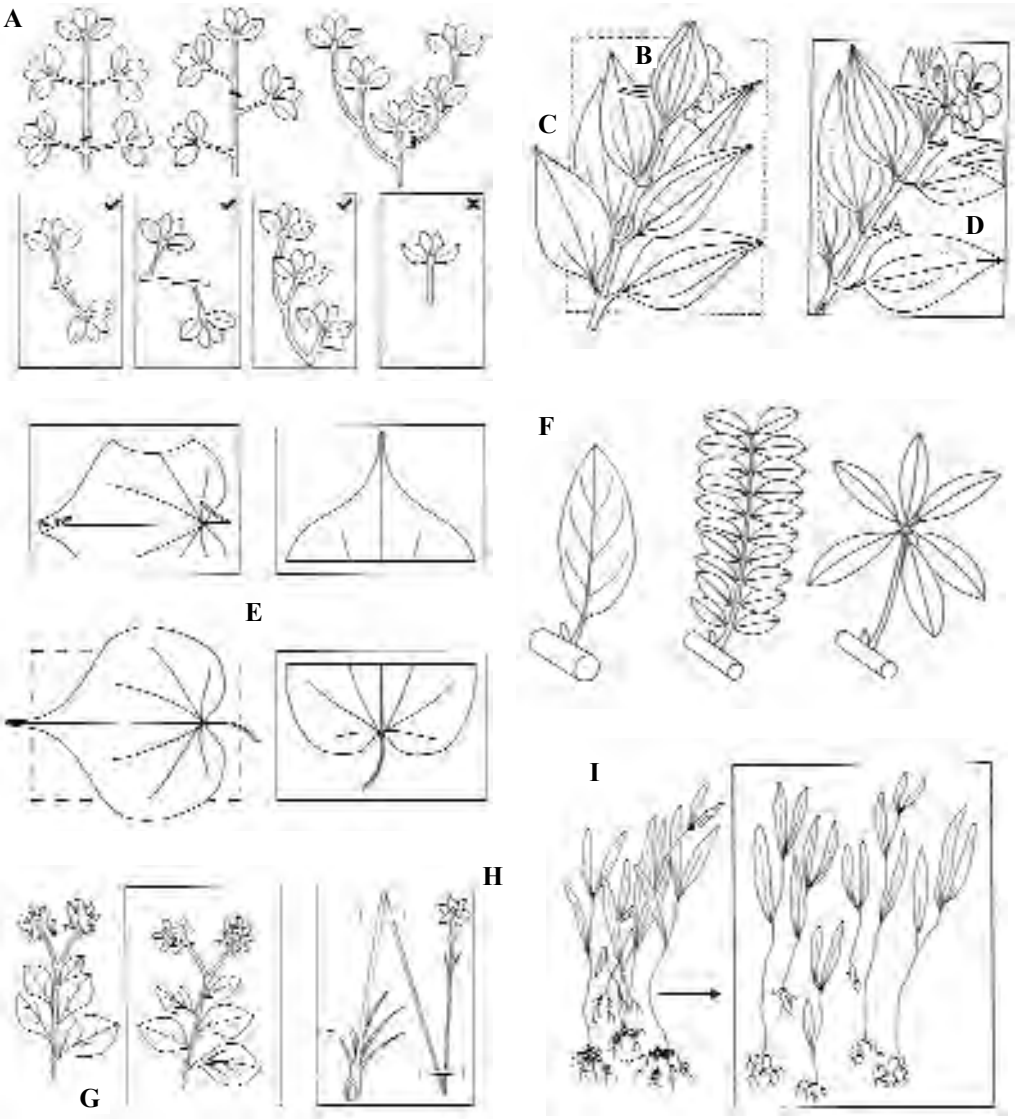


Figure 8.3. Some tips for making good plant specimens:
A. cut specimens such that the branching pattern is visible; **B.** remove leaves to display flowers or fruits; **C.** If leaves extend out of newspaper cut of fold them, but leave the leaf base and tip intact; **D.** if several leaves overlap remove one or more such that the base and tip of all pressed leaves are visible (cut leaves which are being removed such that it is evident where they were located); **E.** with large leaves that will not fit on a herbarium sheet cut and fold to fit, or make two sheets of a single leaf; **F.** show attachment of simple or compound leaves and always leave compound leaves entire (the folioles on one side of the leaf can be removed if there are too many); **G.** display both sides of flowers and leaves; **H.** make small incisions and fold stems cleanly rather than bending - pieces of card with cut slots can be used to hold the stem in place if necessary; **I.** for small plants fill the herbarium sheet with several individuals, include leaves and roots and avoid overlapping. (from Bridson & Forman, 1989)

The press is left on the drier for as long as it takes all the plants to dry thoroughly; some will not take as long as others and, together with spare press materials, these should be removed from the press in their folders when they are dry. Dried specimens are stored in their folders.

If it is impossible to start drying the plants immediately, specimens can be stored in their folders indefinitely by the use of alcohol. The specimens are put into folders as if they were to be pressed but are then tied in bundles with string. They should then be placed in a field press for 12-24 hours before making bundles for alcohol treatment; this will flatten the specimens. The bundles are placed in strong plastic bags and doused with enough 70% alcohol to completely moisten the folders, and the bags are carefully sealed. Each package must remain sealed until you are ready to put the specimens on a drier. The specimens are arranged in a press and drying can continue as normal. It is important to ensure that the sheets have been numbered with pencil or with ink that does not run or dissolve in alcohol. Note that specimens prepared in this way tend to discolour, and the use of alcohol presents a potential fire risk. It also removes or alters some chemical constituents, making the specimens less useful for chemical analysis.

When camping some distance from a road the additional weight of storing collections in alcohol is prohibitive and it is better to dry them before carrying them out of the field. Furthermore, few field workers in Africa have access to custom-built plant driers and a variety of innovative designs have been developed over the years. These all require a heat source, such as charcoal from a camp fire, or a gas or kerosine stove; and some means to isolate presses from the heat source to avoid excessive heating or fire, generally a shelf of sorts at a safe height (eg. Figure 8.4). Where electricity is available fan heaters are a good solution which produce excellent specimens; the press and heater are enclosed in a heavy tarp which channels warm air through the press, drying most plants in about 24 hours. If plants are dried in the field special precautions must be taken to ensure that they are not damaged when transported, as dry specimens are much more fragile than fresh or alcohol preserved collections.

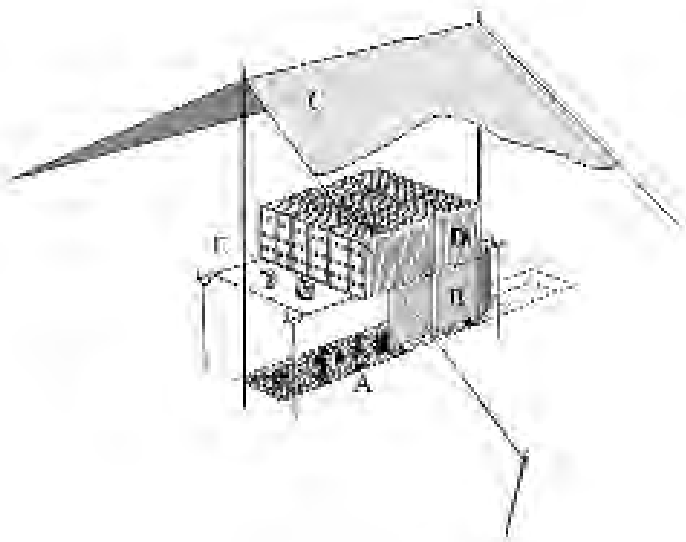


Figure 8.4. A drying system that can be set up in any field camp (Adapted from Bridson & Forman, 1989) A, trench filled with red hot embers from nearby fire; B, skirt of metal corrugates to conserve heat; C, tarp; D, full plant presses; E, large fruits drying.

Most herbaria will insist that new specimens brought in from the field are bundled and fumigated to ensure that no insect pests are introduced. Fresh, partly dried or alcohol treated specimens must be dried in an area which is isolated from the main herbarium. Material that is completely dried can be placed in a sealed plastic bag and left in a deep freeze for a minimum of 48 hours, or can be placed in a microwave oven (in both cases if the specimens are not totally dry they will be damaged!). When the plastic bag is taken out of the freezer it should be left for a few hours to return to room temperature before unsealing and removing its contents.

Pressing and drying problem plants

Although most plants will dry easily and need no special treatment, some others present problems and will require special care and effort. Plants with fleshy underground parts, with succulent stems or leaves, or with large or fleshy fruits, are particularly problematic. Not only are these plants bulky and difficult to flatten without squashing them and causing distortion, but they are also slow to dry and inevitably the drying process causes considerable further distortion to the fleshy organs as they lose moisture and shrivel. Added to this, succulent plants are well adapted to withstand drought conditions and frequently resist almost all of the collector's efforts to get them dry!

Many collectors shy away from collecting difficult plants, and as a result plants such as succulents are under-represented in herbaria. However the preparation of a good herbarium specimen out of difficult plants is a challenge which can be rewarded with considerable satisfaction if you compare your results with the miserable specimens that exist in most herbaria. Obviously plants that do not make good herbarium specimens benefit particularly from detailed accurate field notes, and the addition of photographs or sketches and the preparation of spirit collections. The following notes are intended as a guide to methods that can be used to deal with a range of problem plants.

Bulbs, corms and tubers can be sliced vertically and pressed separately if they are bulky. Only one rootstock per sheet is necessary.

Fleshy fruits can also be sliced, both horizontally and vertically, to show shape, internal structure, protuberances, etc. As cut surfaces tend to stick to specimen folders, pieces of waxed paper can be used to prevent this.

Seeds should be removed from a fruit and put in an envelope after drying so that they are not lost if they become detached. It is often much easier to count numbers of seeds in a fleshy fruit at the time of collection than after the fruit has been dried, so add this information to your notes.

Succulent plants can also be cut longitudinally or transversely, but note that more delicate species will flatten considerably on drying. They have to be killed in some way. Sufficient heat will do this, but may also cause blistering of the specimen, which may also become very brittle. Killing the plant before pressing by leaving it immersed in alcohol or in an acid such as household vinegar (the addition of a few drops of detergent per litre of acid helps in wetting the material) for a period depending on the degree of fleshiness. Alternatively specimens may be dipped in boiling water for a few minutes, excess water allowed to drain, and then the specimen is pressed.Some methods work better for certain plants, causing less discolouration and distortion; trial and error is the best approach. In practice, in the field, you may have to make do with whatever equipment and materials you have available. It is usually worth pressing some untreated flowers separately.

Very thick leaves and stems can be slit open and the soft tissues scraped out and discarded, after other relevant details have been noted.

NB. the method you use to prepare your specimens for drying should be recorded in your notes and noted on the label, such that botanists who wish to undertake chemical or anatomical studies will be able to evaluate the usefulness of your specimens.

Hard bulky specimens that cannot be sliced, such as thick woody stems, woody fruits and bark samples, can often be dried without being in a press at all. If pressing is required use rolls or pads of extra paper between the bulky pieces to ensure pressure is applied to the thinner parts in the press. Try to arrange the specimens in the press so that the bulky parts of adjacent sheets do not lie on top of each other. The use of aluminium corrugates, if available, for pressing these specimens is desirable since cardboard corrugates will be distorted and the corrugations will be crushed.

Spiny/thorny plants often have long spines (or thorns) protruding a various angles. Although some spines may need to be trimmed, try to retain as many as possible, bending them to the side so that they all lie in one plane. Make notes about their orientation.

Very large plants are impossible to press in their entirety. Portions of a suitable size must be taken off and pressed, but care must be taken not to fragment them unduly. Where stems, leaves or leaflets have to be trimmed, leave a few millimetres of the cut stump behind to show its position. When pressing very large plants, make several sheets rather than one mass of material on the same sheet. Particularly difficult in this respect are palms (Arecaceae) and certain other plants with enormous leaves and inflorescences. In these cases representative portions of each part must be collected, including the leaf-base and sheath, the mid-section and the apex, together with suitably trimmed portions of the inflorescence.

It may take several hours to prepare a good specimen of gigantic plant species, and hastily pressed unrepresentative fragments are virtually useless. Specimens of large plants will need to be mounted on a series of mounting boards in the herbarium, in which case each sheet should be marked with the same collection number and a unique part number e.g. *Diabata 428, sheet 1 of 3*. These specimens should be accompanied by detailed notes including overall measurements and/or sketches and photographs of the entire plant.

Delicate flowers (e.g. those of many Balsaminaceae, Convolvulaceae, Marantaceae, Zingiberaceae etc.) pose several problems:

- they need to be pressed as soon as possible;
- they sometimes shrivel almost instantly if their specimen folder is opened before they are dry;
- they often stick to the folder when they are dry.

Extra flowers should be collected if possible and placed between tissue or waxed paper, or mounted on gummed cards. Do not open the specimen folders until you are sure the flowers are dry (mark the folders in a particular way to distinguish them from other specimens). Preserve some flowers in spirit, prepare drawings, and / or take photographs if possible.

Structurally complex flowers such as those of Orchidaceae ideally should be preserved in spirit. Colour, fragrance and markings should be carefully noted. Some flowers may be dissected and mounted on gummed cards or split open and pressed separately.

Composite flower heads such as those of Asteraceae should be pressed so as to show both upper and lower surfaces, and an effort should be made to collect mature fruiting capitula as well as flowering ones. Some flower heads should be split open and pressed.

Blackening of specimens occurs in a wide range of plant families, but is common in certain Orchidaceae and Scrophulariaceae. In these plants chemical changes due to the presence of iridoid compounds cause the whole plant to become black in colour, and all natural colouration is lost. There is little that can be done, except ensuring rapid drying of the plant and preparing field notes, drawings and / or photos that indicate the original colours.

Leaf abscission (leaves fall off) occurs in many plants within hours of collection due to the plant's natural response to stress. Species of *Ficus* are particularly prone to this. With these plants rapid drying using heat is recommended to kill the specimens and prevent abscission. Alternatively, these plants can be killed by using the techniques recommended for succulent plants.

The Preparation of fungi

Only specimens of fungi that are in good condition should be collected. They should be placed in a flat bottomed box to minimise damage, and should be kept distinct in the field, preferably wrapped in newspaper, or in the case of smaller delicate fungi, in separate containers. Plastic bags should be avoided. It is important that the entire fructification is collected, including the base of the stipe and any volva if present (see Figure 8.5). Parasitic species must be collected with sufficient material of the host plant to enable this to be identified.

Gatherings should be made, if possible, to show all stages of development. Record the habitat for each collection, including associated plants and soil type. Make careful notes when material are fresh, indicating: colour, colour changes on bruising/drying, colour of broken flesh, presence/colour of any latex or exudate, consistency, odour, and size range of fruiting bodies, pubescence of any part of the fructification and for Agarics a note on presence or absence of striations at the margin of the pileus. Photographs of fresh material are desirable, including a vertical section to show the type of gill attachment.

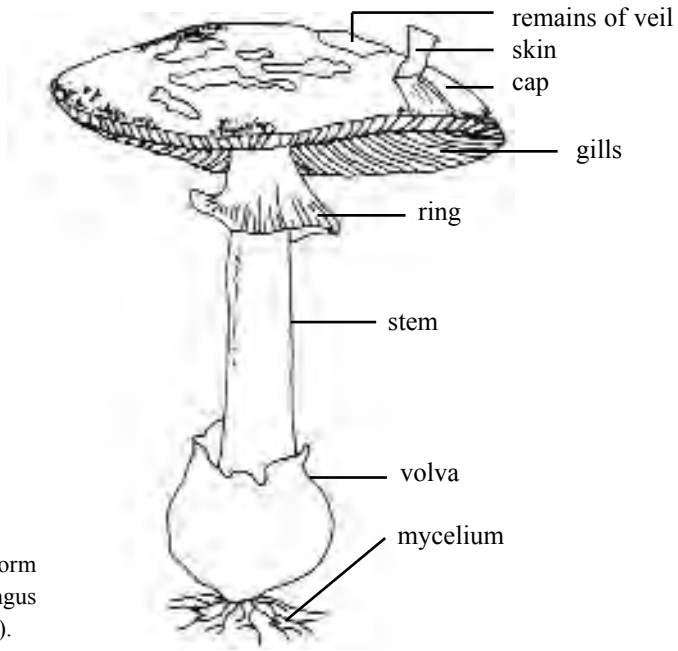


Figure 8.5. Form of a typical fungus (Basidiomycete).

Spore prints from at least one specimen in each collection should be taken from larger fungi. To prepare a spore print place the fruiting-body (with stipe removed in Agarics), hymenial surface downwards onto a sheet of paper or a glass slide and leave overnight. Place specimens under a glass jar or lid to prevent drying out. For Agarics with white gills use black or brown paper so that the white spores can be seen. Number spore prints and clearly cross-referenced to specimens.

It is important to dry fungi as rapidly as possible after collection as this helps to retain form and colour and prevents deterioration of the material. Cut larger Agarics in half to facilitate drying. Ideally a rapid constant flow of warm air (40 °C) over the specimens is recommended. If the temperature is too low insect larvae are likely to hatch, and if too high the fungi will cook. Do not press fungi in a plant press. Instead, spread the fungi on a wire-grid over a fan-heater or gas burner.

Store specimens in variously sized cardboard boxes as they are delicate and brittle. Avoid plastic or metal containers because dried fungi are particularly susceptible to insect attack Freeze them in a sealed plastic packet for 48 hours before incorporating into the collection.

The Preparation of lichen specimens

Prepare lichenised fungi (i.e., lichens) in the same way as fungi. Wrap them individually in tissue in the field to prevent damage, dry rapidly and eventually place them in paper envelopes with an appropriate label; they should never be pressed.

- Collect foliose (leaf-like) and crustose lichens with part of their substrate. Never peel them off since this would damage the thallus and rhizoids.
- Collect fruticose (branched) species with basal parts intact.
- Collect saxicolous (closely attached to rocks) species using a hammer and chisel or knife blade ie. they should not be removed from the substrate. Note the rock type.

The collection of delicate marine aquatic plants and algae

Collect seaweeds and delicate aquatic plants growing in marine habitats by wading for littoral species (at low water spring tide) and diving for sublittoral species (at high tide). It is as important to record habitat and location of seaweeds as for terrestrial flowering plants. Press and dry Algae in a plant press. Because of their wetness seaweeds are difficult to dry and susceptible to fungal damage, frequent changes of "blotters" are necessary and often the corrugates too. Seaweeds are best collected directly onto sheets of good quality paper to which they adhere, which are eventually glued to the final mounting board without the need for glue or paper strips. Wax paper may be useful to prevent the specimens from sticking to the specimen folders. Float small or delicate specimens onto sheets and arrange carefully with an artist's brush. For bulky specimens apply pressure sparingly at first and increased gradually over a period of a few days as the material dries.

The Preparation of bryophyte specimens

Place Bryophytes (mosses and liverworts) as soon as possible in paper packets, and never store for long in plastic bags, since this encourages the development of mould. Mats and cushions of bryophytes often consist of several species, sometimes with a dominant species and other more inconspicuous ones. Try to separate each species before they are dried or alternatively arrange to send all of the material to a bryophyte specialist, who can separate the individual species for you. Dry specimens as quickly as possible at a fairly low temperature as distortion of the cell structures (and therefore the specimen) may result at high temperatures. Do not press the specimens but leave them in their opened envelopes/packets to dry in a well ventilated place. File mosses in envelopes with a herbarium label on the front rather than on a mounting board.

The Preparation of pteridophyte specimens

Because unfertile Pteridophyte (ferns) material is very difficult to identify, only collect specimens with spores. Ideally the spores should be ripe (i.e., the sporangia should be releasing spores). It is also important to collect portions of fern rhizomes because their habit (e.g.,creeping or erect) and their covering of scales or hairs provide very important taxonomic characters. Also note the presence or absence of aerial roots. Information to be recorded about the fronds includes:

- length and width, and length of stipe (in large ferns of which the entire frond can not be collected);
- number of fronds arising together;
- whether they are produced synchronously or sequentially;
- how many make up a complete whorl.

And for tree ferns:

- the angle at which they are held to the trunk;
- if they are shed from the trunk or are held when dead;
- how far apart they are spaced on the trunk or rhizome;

Some small mat-forming ferns can grow as a mixture of species with rhizomes intertwined. These should be examined and separated carefully before drying. In some cases a frond may be too big to fit onto a herbarium sheet and one side of the pinnae may be trimmed almost to the rachis leaving just enough to indicate their position. Note that both surfaces of the frond must be visible after the specimen is mounted, so press the material accordingly.

Transport of dried specimens

When breaking down a press keep specimens that are not fully dried, along with their corrugates, together as a unit, and place them back on the drier (moist blotters can be replaced with dry ones if necessary). Do not scatter them among another press that is being built. When all of the sheets are dry keep replicates together - open the first folder and place successive folders into this one, with all the open edges together and the numbers facing up where they are easily viewed. When removing specimens from the press hold it flat, or if necessary hold the open edges higher as loose flowers and fruit will then fall to the safer folded edge. Put all specimens into numerical order, checking at the same time that all sheets have been correctly numbered. Then separate specimens which are to be shipped to different institutions or retained in the field. Note in your collection book which institutions' replicates have been taken out and also note this on the outside folder. There are standardised abbreviations for most herbaria (e.g., Libreville -LBV!, Missouri Botanical Garden -MO!, Herbarium Vadense, Wageningen -WAG!).

Arrange any specimens to be shipped into bundles about 20 cm thick. Wrap each bundle firmly in more newspaper to prevent the specimens from slipping, and then fasten with string or tape. To prepare a bundle for shipping, overlap separate halves of two double sheets of full-sized newspaper, place the specimens in the centre of the overlapped sheets, fold the edges firmly around the specimens and fasten securely. Note your name and the range of collection numbers enclosed on the bundle and pack several bundles tightly into a strong cardboard box, putting them within sealed plastic bags with Napthalene (if available) if they are likely to take a long time to reach their destination, are likely to be exposed to moisture or may be stored for a long period. Enclose any large fruits or other parts which were dried separately or preserved in spirit. If you will be shipping specimens to a herbarium rather than delivering them yourself be sure that you send any information required by that institution along with the shipment and advise them once the boxes have been dispatched. Be sure to consult a national herbarium or research institution in the country where you are collecting, as well as the herbaria to which specimens are to be sent, to check whether special export or import permits are required.

A field herbarium

Whilst it is strongly recommended that you deposit replicates of your plant collections with active, well curated herbaria, it is also extremely useful to retain a set in the field provided you have an acceptable way to store and care for them. This will enable you to identify species which have and have not previously been collected at your site and will serve as a valuable reference for visitors as well a new researchers or field staff. It is unlikely you will have facilities to maintain an herbarium that meets international standards, although if you do that is ideal. If you are thinking about embarking on such a venture the references given at the end of this chapter will provide helpful information and advice. Before starting you would be well advised to visit your national herbarium and spend some time with the curators there, learning the basic principals of mounting and caring for herbarium specimens. "The Herbarium Handbook" (Bridson & Forman, 1992) provides a good general introduction.

The most compact and convenient alternative to developing a proper herbarium in the field is to retain relatively small specimens which can be mounted on A4 paper and filed in a standard lever-arch file (alternatively photocopy or scan plant material and keep a ‘paper herbarium’ in the field). This system has proven particularly effective for many inexperienced botanists as they begin to study tropical floras, and also allows for infertile specimens, fallen individual leaves from tall canopy trees, small sprigs tossed down by monkeys, etc. to be stored conveniently whilst you are waiting for the chance to collect a fertile specimen worthy of sending to a herbarium (Figure 8.6). Such files are compact enough to be stored in an air tight trunk, with silica gel and moth balls to protect them from humidity and insects (note that the silica gel must be heated periodically to renew its dessicating properties).

If you have more available space and want to make a more high tech herbarium, a sealed, air conditioned room is ideal in the humid tropics, particularly one which can be fumigated if insect infestations ever become a problem. Alternatively, store specimens in air tight containers with silica gel and moth balls to keep them dry and insect free.

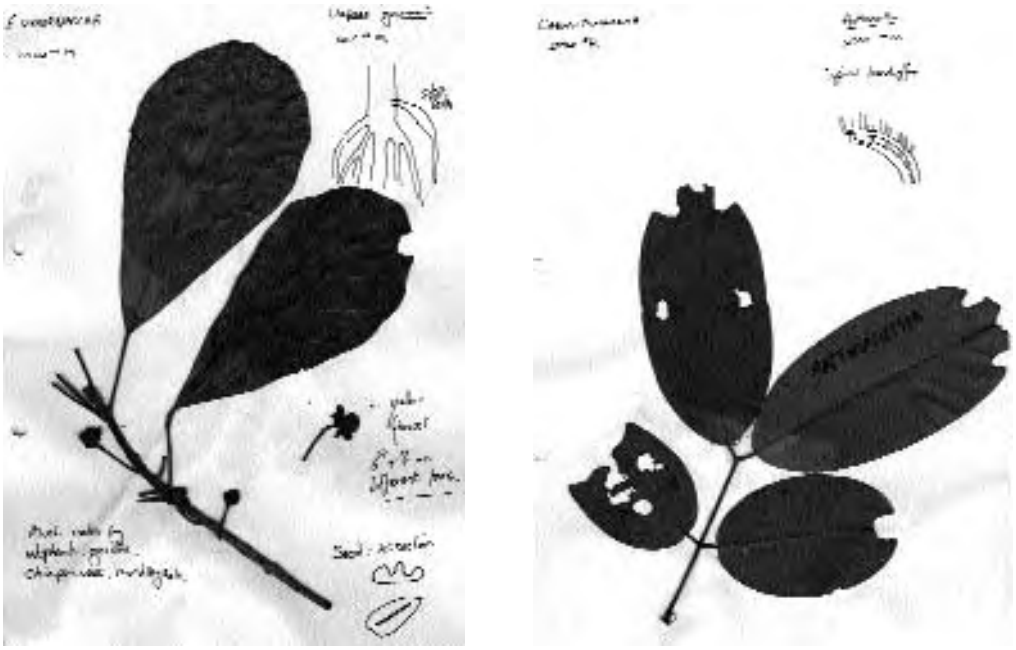


Figure 8.6. Two sheets from a field herbarium of dried plant specimens filed in a ring-binder, with notes to help identification, sketches of life-form etc.

Mounting and curating herbarium specimens

Dried specimens received from field biologists are mounted onto museum quality cardboard once they reach the herbarium where they will be stored permanently. Whilst we will not go into great detail here about mounting and curating herbarium specimens, it is useful to understand how your collections will be treated once they have been sent to a herbarium.

Labelling herbarium specimens

The label that is eventually attached to the herbarium specimen sheet is as important as the specimen it accompanies; without it, the pressed plant is no more than a piece of hay! The label provides essential information that is not available from the specimen itself. the label is transcribed from your field notes. It is customary to present this information in a logical uniform sequence, to facilitate rapid examination. Labels are printed on archival quality paper using permanent ink (or other printing medium). Note that ball-point pens are not suitable, since their ink fades with time. A typical label is shown in Figure 8.7. Labels are attached to the bottom right hand corner of the board on which the specimen is mounted (see Figure 8.8).

EX HERBARIO VADENSI (WAG)	**1.**
Plants collected by J.J.F.E.de Wilde & R.W. de Wilde-Bakhuizen	**2.**
no. 11874 d.d. 28.12.1996	**3 / 4**
Sterculiaceae	**5**
Scaphopetalum thonneri De Wild. & Th. Dur.	**6**
Det. M.E. Leal 12.06.1997	**7**
GABON, OGOOUÉ-LOLO, Lopé Reserve. Exploited high forest rich in Aucoumea. Along small creek. Half shade. Alt. 400m 0°46.0'S 11°46.5'E	**8**
Shrub, 2 m. high, growing in understorey of forest in shade on rocky soil. The leaf-pockets inhabited by small ants. Leaves thick papery, dull rather dark green above, beneath much paler green and with conspicuous venation. Corolla orange-yellow, purplish inside. Fruits 5-lobed in transverse section	**9**
5 replicates: -WAG!, -P!, -MO!, -LBV!, -SEGC	**10**

Figure 8.7. A typical specimen label.
1. Herbarium name, with international abbreviation in ( ).
2. Collector's name(s) **3.** Collection number **4.** Date of collection
5. Family. **6.** Name of plant as far as it has been determined (with authority if determined to species).
7. Name of person who identified the specimen and date of identification.
8. Locality data, in descending order (opposite to an address), including latitude and longitude, altitude and habitat/ecological notes.
9. Other data about the plant that cannot be obtained from the dried specimen,
10. Number of replicates collected and herbaria in which replicates have been deposited.

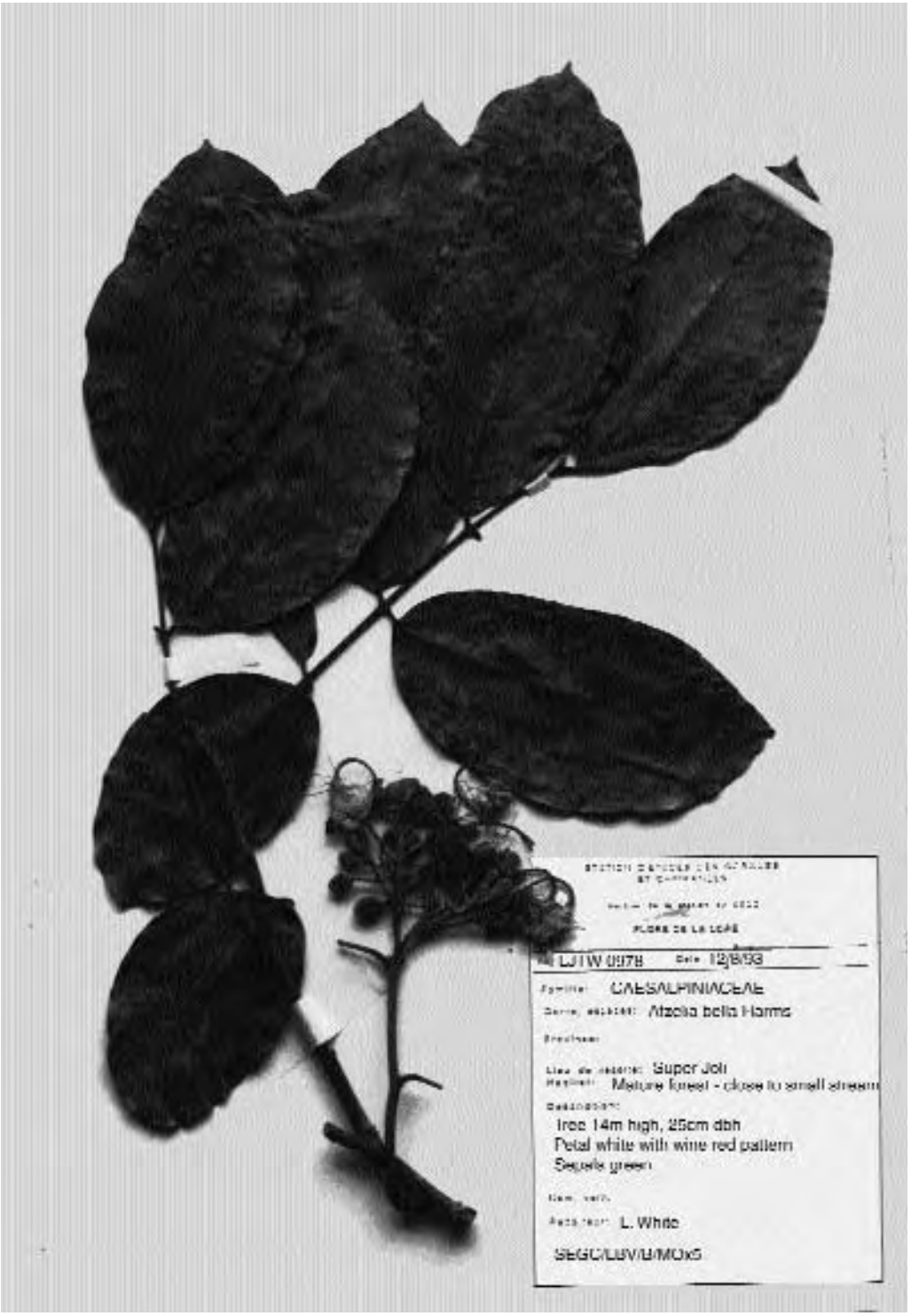


Figure 8.8. A mounted herbarium specimen and its label.

Mounting herbarium specimens

Herbarium specimens are mounted in order to:

1. Display the specimen and data to allow maximum observation.
2. Preserve the specimen by securely attaching it to a firm surface, but at the same time allowing for removal of small portions for more detailed study.

Once the collector gives specimens in their folders to an herbarium the labels are prepared and then mounted. Most herbaria require the collector him or her self to prepare the labels. Most herbaria use standard materials for mounting their specimens, but it is not always possible to obtain these in the field and you may be obliged to make the best of what is available locally for your field herbarium:

- Mounting Boards (46 x 26 cm): high quality white cartridge paper, preferably archival quality, acid-free and of 100% rag (fibre) should be used, but cost may be prohibitive.
- Small pieces of high quality white paper are used to make folded envelopes (specimen packets) used to store fragments of the specimen. Conventional envelopes are not recommended as delicate material can be damaged when removed for study.
- Gummed tape is used to attach the specimen to the mounting board, or the specimen itself is glued to the paper: do not use self-adhesive cellulose tape (Scotch tape or Sellotape), surgical-tape, or ordinary paper. Pre-gummed tape in rolls is cut into strips in the Herbarium.
- Specimens may also be attached by stitching with thread: strong linen thread is recommended, cotton thread is not a good alternative.

Once mounted, specimens are filed in the herbarium cupboards (sometimes called cases) according to a particular system. In most herbaria, each specimen is placed together with others of the same species in a paper folder called a species cover. The folder(s) for each species are then placed in alphabetic order, by genus; the genera are organised either alphabetically or in taxonomic order. Some larger herbaria provide separate species covers for examples of each species from different geographic areas, and sometimes the specimens are arranged in the species covers in chronological order of collection date, alphabetically by collector, or geographically. The aim is to store the specimens safely according to a system that facilitates future information retrieval.

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Treculia africana

CHAPTER 9
Vegetation inventory and description

Lee White & Ann Edwards

Why study vegetation distribution and abundance?

Conservation research and activities in African rain forests tend to focus on animals, particularly large mammals and birds. However, one cannot escape the fact that by their very nature it is the plants that define the forest environment. Tropical rain forests are home to a great diversity of plant species representing many forms and life-styles. This diversity in turn creates a wide variety of habitats and foods for animals. Plants increase the temporal diversity of the rain forest, since animal reproduction and migration usually are linked to the timing and location of fruit, flower and leaf production. As evidence of this, plant diversity tends to correlate well with overall species diversity. But given the wealth of taxonomic work which has been undertaken on plants in many areas (even if our knowledge is by no means complete), they tend to be more easily studied than other species-rich groups such as insects.

Patterns of plant abundance and diversity also are important to people. For example, loggers cut and remove large trees; and local communities often make use of and / or trade a vast range of foods, medicines and building materials from the forest. If such activities are authorised within a protected area, or even if they occur illegally, the impact of these extractions should be monitored to assess whether management goals are being met.

Botanical inventory

A botanical inventory is a study that aims to record a) the diversity of plant species found in an area; b) distributions of favoured habitats of each species; c) estimates of abundance of each species in the area of interest. An inventory is the necessary first step in any detailed vegetation study. It is also a valuable exercise in itself. For example:

- A botanical inventory provides biological information on the diversity and uniqueness of the area. It forms a basis for vegetation classification and mapping studies, both of which have obvious management applications;

- A botanical inventory provides the foundations for ecological or socio-economic studies, since you must first identify the plants in order to understand an animal's diet, or the impact of logging on a habitat (Box 9a), or patterns of food availability for frugivores, or use of medicinal plants by humans,;

- Undertaking a botanical inventory is a good training exercise, enabling you to become familiar with the botany of the area you work in. A botanical inventory will involve training personnel to recognise and collect plants. With the knowledge gained they will be better able to undertake other forms of botanical monitoring and vegetation mapping described below. Fortunately a botanical inventory can be undertaken on any schedule and during any part of the year, and be adapted to the time and personnel available;

- Finally, a well-conducted botanical survey in tropical moist Africa quickly takes on global importance since the botany of these regions is generally poorly documented.

Vegetation inventory and description

However, compilation of a plant list is a large undertaking, as there are probably at least 2000 species in most protected areas in the African rain forest zone. This is best done in collaboration with a national or international herbarium, which can take on much of the burden of collecting, preserving and identifying plant specimens. Before starting an inventory, try to contact institutions or individuals who have collected in your region, in order to establish how many specimens already exist and where they were collected. This will give some indication of what is already known about the area in question and where there are gaps in existing knowledge. Having compiled this information you will be better able to assess the need for further collecting and to decide what the appropriate level of effort should be (see Box 9b). Once you have completed this exercise you should try to ensure that records are kept of all future collections made, either by yourself and your team, or by visiting botanists and researchers. If you decide to establish a field herbarium it is a good idea to ask all researchers working in the area to provide duplicates of their collections.

Box 9a FOREST REGENERATION AFTER LOGGING IN UGANDA.

John Kasenene (1987, 2000) studied regeneration in gaps caused by logging in the Kibale Forest Reserve, Uganda in order to assess the rate of forest regeneration. Logging had occurred about 20 years before his study. He first mapped the locations and sizes of all gaps in one lightly logged, two heavily logged and one unlogged area. He then randomly selected 40 treefall gaps to study in each area. He studied seed rain (the quantity of seeds falling to the forest floor) and germination in two $4m^2$ plots in each of 20 randomly selected gaps and used 5m wide transects crossing the gaps to enumerate all saplings and poles (• 1.5cm - 10cm dbh). Once these samples had been established he monitored all plots and transects on a monthly basis for 20 months. He found that:

- the size of treefall gaps were statistically different between unlogged, lightly logged and heavily logged areas, with the largest gaps occurring in heavily logged forest;
- tree seedlings were poorly represented in soil seed banks;
- tree seedlings tended to be smothered by lush secondary vegetation in treefall gaps;
- large gaps created by logging were regularly frequented by elephants, which broke and otherwise damaged a high proportion of seedlings and saplings.
- abundance and diversity of rodents was significantly higher in logged forest (rodents are major seed and seedling predators in Uganda and have strong negative effects on regeneration, particularly of commercial timber species).

The end result is that, even 20 years after logging, there has been little regeneration of logged forests in Kibale. Hence any plans for cyclic logging on a pre-determined rotation will have to be modified (or abandoned). Kasenene's results are very important for forest managers in Uganda, since they show that regeneration might not happen automatically after logging. Managers should be monitoring regeneration (or lack of it) and studying the ecological processes which contribute to it, since they may have to undertake intensive management in order to promote regeneration in some forests. Given Uganda's reduced forest cover this should be a priority for their forest department.

Kasenene's study illustrates how vegetation (and mammal) surveys can contribute to our ability to manage tropical forests. It is a useful warning to all natural resource managers that they need to understand the ecological processes which sustain their ecosystem. Furthermore, it clearly illustrates the need for more applied research in the field of tropical ecology and forestry.

Box 9b BOTANICAL EXPLORATION IN THE LOPE RESERVE

The Lopé Reserve covers 5,320 km^2 in central Gabon. White et al. (in press) have tried to trace all botanical collections made there and to analyse the sampling intensity in different parts of the Reserve. To date about 3,700 collections have been made and just over 1500 species have been identified. Figure B9.1 shows how collecting has been spread across the Reserve. Clearly most collectors have concentrated their efforts in the northern parts of the Reserve. In the future collectors should try to visit the poorly collected southern parts of the Reserve, in order to produce a plant list that gives a more representative picture of the botanic diversity of the area. The analysis by White et al. has enabled researchers and managers to establish priorities for future botanical inventory.

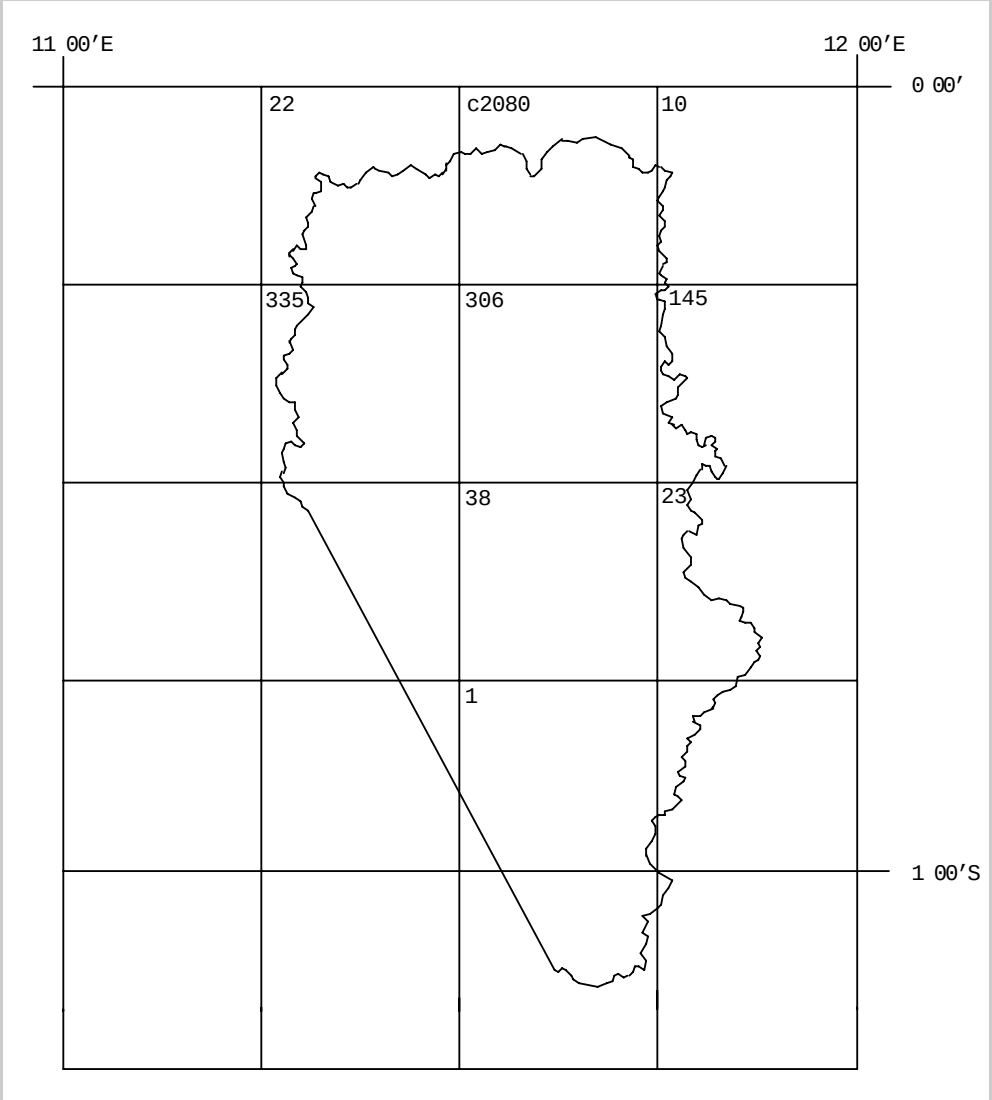


Figure B9.1. Number of botanical specimens collected in each quarter degree square in the Lopé Reserve, Gabon.

Chapter 8 gives comprehensive guidelines to plant collectors as well as suggestions of how to establish a reference collection in the field. Collection of plant specimens should be an on-going exercise. Definition and description of habitat types should be the next priority. This chapter details methodologies that can be used to classify and describe vegetation.

The first step inevitably is a qualitative or subjective classification. This may be followed by a quantitative study, if such is a priority at your site.

Qualitative description of vegetation

The differences between qualitative and quantitative habitat descriptions

Habitats can be described both qualitatively and quantitatively. A qualitative habitat description uses subjective descriptions instead of numbers. A qualitative description describes a habitat's most obvious features. For example, you can talk about the "high forest", or the "low closed scrub".

A quantitative habitat description uses numbers to describe the features of a habitat. For example, after collecting and analysing data on tree heights, you can say that one forest has an mean canopy height of 25m and another forest has an mean canopy height of 38m. A quantitative description is better for making detailed comparisons between habitats.

Qualitative habitat descriptions

Qualitative habitat descriptions have many uses:

- to place a plant or animal or event in a known, easily understood setting. For example, you could say that you saw congo clawless otter tracks in a swamp forest, or that the red river hog nest was in a light gap in Marantaceae forest;
- to produce a preliminary stratification of an area by vegetation type, prior to systematic sampling of animal density or plant communities. (see 'stratification' in Chapter 3);
- to look for trends in the way animals use different habitat types, for example from animal surveys which do not include detailed vegetation analysis (for examples, see Chapters 12 & 13);
- to give people a common set of terms to use when discussing the different parts of the forest. This allows for more efficient communication between colleagues. For example, you can say that the patrol will begin in the secondary forest near the road, pass through the logged forest and end near the clearing;
- to allow protected area staff to describe in general terms, to any person, the types of habitats that can be found in the area. This is important if protected area staff are going to communicate easily with visitors and other interested people. For example, you can say that most of the forest found in the park is tall and relatively open, with a sparse under-storey. However, the wetter areas are characterised by many palm trees and a denser under-storey.

Descriptive factors used in qualitative habitat descriptions

Basically, any striking differences found between contrasting forest or habitat types can be used to describe them. Some of the most widely used descriptive factors include:

- *Dominant or most common tree species*: Some forest types can be adequately described by listing the 1-3 most common tree species found in that forest, for example, the "*Gilbertiodendron dewevrei* forests" of central Africa. When there are no species that are found much more commonly than any other, the forest can be called a "mixed forest".
- *Indicator species*: Certain tree species, called "indicator species", can indicate growing conditions or past disturbance to the forest (see below).

- *Average canopy height*: A relatively tall forest may grow next to a relatively short forest. Although the cause of the difference in canopy heights may be differences in soil or water conditions, the descriptions of the forest types can remain "high forest" and "low closed scrub", because the difference in canopy height provides the most obvious difference between them.
- *Open or closed canopy*: Some forests exhibit a "closed canopy", meaning that there are almost no spaces between the leaves of neighbouring canopy trees. An "open canopy" refers to a forest where many spaces (gaps) in the canopy allow large patches of sunlight to reach the under-storey or the ground.
- *Average tree size*: Some forests, such as swamp forests or young secondary forests, are made up of many small trees (i.e., small trunk diameter). By contrast, other forests have greater proportions of large trees.
- *Under-storey density*: Some forests possess a thick, dense layer of ground vegetation, shrubs and small trees, while others are relatively free of under-storey vegetation.
- *Topography, or the presence of water*: Sometimes very different forest types grow on steep, rocky slopes than will grow on flat ground with deep soil. Similarly, different forests will grow in swampy, wet areas than on high, well-drained ground.
- *Disturbance*: Secondary forest is a term used to describe a forest that was disturbed and then regenerated naturally (see Box 9c). Other terms for describing disturbed forests include old farms, heavily logged forest, selectively logged forest, burned forest, etc.

Keeping qualitative habitat descriptions simple and consistent

Qualitative habitat descriptions should be simple. Each habitat type does not need to be described by every one of the factors outlined above. Instead, one or two characters that most obviously separate one habitat type from another should be used. For example, the Ituri forest of eastern Zaire can be described using four general habitat types: mixed forest, *Gilbertiodendron dewevrei* forest, swamp forest, and secondary forest.

Small patches that differ from the surrounding habitat do not always have to be described as a separate habitat type. Instead, the small area can be described as a note in the habitat description of the larger area. For example, you can describe a rock outcrop that is found within a large patch of tall, mixed forest. Or you can make a special note about a small burned area found within a large patch of old secondary forest.

Qualitative habitat descriptions are most useful if they are consistent. Everyone working in the forest should agree on how to distinguish between habitat types. As a group, forest guards, managers and researchers should take a walk through the different forest types and discuss their similarities and differences. Concentrate on those areas that may be hard to classify, such as boundaries between forest types.

Common habitat types in the African rain forest

The following habitat descriptions are widely used by researchers working in African forests:

- *Mixed Forest with open under-storey* - what many would call 'primary' forest, with many large trees, a high, unbroken canopy and sparse vegetation,cover on the ground, consisting mostly of shrubs.
- *Mixed Forest with closed under-storey* - a forest type which may be similar to old secondary vegetation, with many large trees but dense vegetation cover on the ground (note if under-storey is dominated by lianas, Marantaceae, or other vegetation);

Box 9c PRIMARY FORESTS AND SECONDARY FORESTS: THE STORY OF FOREST SUCCESSION

A "secondary forest" is created when there is wide scale disturbance to pre-existing vegetation, and then the forest is left to regenerate. Such disturbances, which result in the felling, destruction or removal of the existing trees, include fire, floods, land-slides, wide-spread blow downs from sudden, high winds, and the most common form of disturbance today, the alteration of land by people (cultivation, logging etc.).

As time passes since the initial clearing of the land, both the species composition and the structure of the forest will change gradually and (to a certain extent) predictably. This gradual, predictable change is known as "forest succession". Initially, the disturbed land will be covered by "weedy" (shade intolerant) plants and shrubs that thrive in open light conditions and grow and reproduce quickly. Within a year, a few, thin trees will appear above the dense ground vegetation. With time, a taller, more closed-canopy forest, characterised by many small and medium sized trees, will be observed, and the ground vegetation will become less dense. This forest can be described as a young secondary forest.

With the passage of time, a forest will appear that structurally may resemble a mature rain forest (see below), but will remain different in terms of species composition. This forest can be described as an old secondary forest. Once the shade-intolerant, canopy trees age and die, or once they become shaded by taller, mature shade-tolerant trees, the forest can be described as a mature forest.

When the shade-tolerant species become the most common species in the canopy (and mid-story), and there is no further large-scale disturbance, the structure and species composition of the forest as a whole may change very little over hundreds of years. This is because, the trees in the under-storey that will most likely survive to become reproductively mature trees are shade-tolerant species, the same species that currently dominate the forest canopy. An undisturbed forest that experiences little change in species composition or structure over time, is often called a "primary forest" although this is misleading since in Africa they are generally very old secondary forests (see Box 9f).

Tree-fall gaps will affect the species composition of a primary forest by allowing shade-intolerant species to reach the canopy. However, tree-fall gaps represent only small, isolated forms of disturbance, and are included as part of the natural structure and composition of a primary forest.

- *Monodominant Forest* - forest with the structure of mixed forest but where one species of tree is noticeably dominant (notable dominant species include *Gilbertiodendron, Garcinia, Berlinia, Uapaca, Lophira, Aucoumea, Julbernardia*)
- *Marantaceae Forest* - forest with a relatively closed upper canopy, sparse middle-storey and dense herb layer on the ground dominated by plants in the Marantaceae and Zingiberaceae - note the dominant herb species.
- *Liana Forest* - forest in which the middle-storey is dominated by lianes, often found close to major rivers.
- *Gallery Forest* - running along a river or stream, found both within larger blocks of forest, or isolated in savanna vegetation.
- *Coastal Scrub* - areas along the coast dominated by dense scrub with occasional, isolated trees showing signs of constant buffeting by the wind.
- *Mangroves* - forest growing in tidal swamps.
- *Montane or Semi-Montane Forest* - forest with relatively small trees festooned with mosses and lichens found at higher altitudes.

- *Inselbergs* - large granite outcrops surrounded by forest.
- *Forest on and close to rock outcrops* - which tends to have many small and medium sized trees growing in a dense, even canopied stand, with sparse under-storey consisting mostly of shrubs.
- *Swamps* - forest which is flooded all year round.
- *Seasonally Inundated Forest* - forest which is flooded during the wet season, but dries completely at other times of the year.
- *Marshes* - permanently wet or flooded areas dominated by monocotyledons such as *Raffia* or *Marantochloa*.
- *Clearings (elephant bais & swamp bais)* - open areas in the forest dominated by sedges.
- *Salines* - areas where elephants and other animals come to eat mineral-rich soil, creating openings in the forest.
- *Low Closed Scrub* - tends to grow around bais, dense thickets often with *Ancistrophyllum* palms present and occasionally open areas dominated by *Anthonotha macrophylla*.
- *Major light gaps* - where one or more large trees have fallen, resulting in a thicket of lush vegetation.
- *Savannas* - areas dominated by grasses or sometimes by ferns.
- *Mature Secondary Forest* - areas with large trees but showing evidence of past disturbance by humans, without food crops but sometimes with oil palm or mango trees still present.
- *Young Secondary Forest / Active or Recent Cultivation* - areas recently or currently cultivated by humans, where some food crops persist.
- *Logged* - areas exploited by foresters. Note the time elapsed since last logged if known, or estimate based on the size of secondary tree species growing along old roads.
- *Seismic* - areas affected by oil exploration or exploitation.

Indicator Species

Because certain tree species will grow to maturity only under specific conditions (e.g., waterlogged soil, light gaps, rock outcrops, river galleries, in villages) the presence of a mature tree of those species can "indicate" the actual or past condition of the forest. Such species are called "indicator species", because the presence of a tree of that species indicates, by its presence alone, that specific environmental conditions exist or that in the past a particular event such as a treefall took place.

Since the life span of some indicator species can be estimated, and since trees generally are affected most strongly by environmental conditions during their initial growing period (i.e., seedlings and saplings), the presence of certain species in the forest can be used to estimate how long ago the forest was cleared, flooded, burned, etc.

For example, *Musanga cercropoides* and *Fagara macrophylla* are both shade-intolerant species (see Boxes 9d & 9e), and thus indicate that light once reached the forest floor, and therefore that the canopy was disturbed. *M. cercropoides* will die before it is 25-30 years old, whereas *F. macrophylla* can live to be 100 years old. Therefore, the presence of *F. macrophylla* and the absence of *M. cercropoides* may indicate that some form of disturbance occurred between 25 and 100 years ago. This 75 year range might be narrowed further by looking at the number and condition of the *F. macrophylla* trees, and the estimating ages of other species growing nearby.

Different species can be used to indicate various past events or environmental conditions. For example, the presence of domestic oil palms (*Elaeis guineensis*) in a tall, closed canopy forest is a good sign that people once used that land for farming (see Box 9.6).

Table 9.1 lists a few indicator species, their estimated life spans, and what conditions they can be used to indicate.

Table 9.1. Selected indicator species and the type of initial growing condition they indicate

Species	Estimated Life Span	Condition Indicated
Musanga cercropoides	<25 yrs.	Disturbance
Macaranga spp.	15-20 yrs.	Disturbance
Anthocleista spp.	c. 20 yrs.	Disturbance
Trema guineensis	c. 20 yrs.	Disturbance
Fagara macrophylla	100 yrs.	Disturbance / Light gap
Ricinodendron heudelotii	100 yrs.	Disturbance / Light gap
Alstonia boonei	100 yrs.	Disturbance / Light gap
Hallea (*Mitragyna*) spp.		Wet or flooded ground
Nauclea pobeguinii & *vanderguchtii*		Wet or flooded ground
Macaranga schweinfurthii		Wet or flooded ground
Anthocleista vogelii		Wet or flooded ground
Raffia spp.		Wet or flooded ground
Guibourtia demeusii		Seasonally flooded areas
Uapaca spp.		Seasonally flooded areas
Domestic palms, bananas, mangoes etc.		Agriculture / old village

Box 9d SHADE-TOLERANT AND SHADE-INTOLERANT SPECIES

The ground in a tropical rain forest is covered with a multitude of seeds and seedlings waiting to grow. There is usually enough water and space on the ground for many of them to survive. However, the one element that is in short supply, and therefore limits their growth, is sunlight. Most tropical rain forests have a "closed" canopy, which means that the branches of canopy trees are close together. Because there are so few "gaps" in the canopy, not much sunlight reaches the forest floor where the seeds and seedlings need it. Tropical rain forest trees must compete for sunlight until they, themselves, reach the canopy. This competition has led different species of trees to adopt different "strategies" for growing early in life.

Some species, called "shade-tolerant" species, grow slowly with little available light, surviving in spite of the constant shade. Mature trees of these species tend to produce a small number of large seeds. The seeds are large in order to support a seedling long enough to produce leaves large enough to catch what little light is available. Shade-tolerant species tend to grow into trees that dominate the canopies of mature forests. Such canopy trees can live for hundreds of years. Therefore, even if only a few of the seedlings from each mature tree grow to become canopy trees, the species will reproduce successfully.

Other species, called "shade-intolerant" species, wait for direct sunlight to reach the forest floor. Then they grow very quickly. These species tend to produce many, many small seeds. They produce large quantities of seeds so that when by chance a tree fall occurs, some of the the seeds already will be in the right place at the right time and can start growing the moment the direct sun hits the forest floor. Shade-intolerant species grow tall quickly and reproduce at a young age. But each tree usually will not live more than a few decades.

At first, a tree fall gap will be dominated by shade-intolerant species, since they grow quickly in full sunlight. But as the shade-intolerant species grow and produce a more closed canopy, the shade-tolerant species will continue to grow beneath them, whereas shade intolerant species no longer regenerate and become rarer. As the shade-intolerant species age and die, the shade-tolerant species will once again come to dominate the undisturbed canopy.

Note that the strategies of shade-tolerant and shade-intolerant species represent extremes, and are used here to emphasise the compromises that are required of a tree growing with limited resources. Many tree species have growing strategies that fall somewhere in between the two extremes.

Milicia excelsa

Box 9e TREEFALL GAPS

Frequency of Tree Fall Gaps

A tree fall gap is formed when a large tree falls over in the forest, often bringing many other trees down with it. Tree fall gaps are important because they allow sunlight to reach the under-storey and the forest floor. Many tree species require this direct sunlight before they can grow (see Box 9d). Therefore, tree fall gaps can greatly influence the species composition of a forest.

The distribution, and sometimes the abundance of animals are affected by the distribution and abundance of tree fall gaps in a forest. Certain animals such as okapi, elephants and many bird species are attracted to tree fall gaps. Tree fall gaps may be attractive to these animals because the species of plants and young trees that frequently grow there can provide tender young leaves, or large quantities of sweet fruits, for the animals to eat.

The distribution of tree fall gaps is not always random. In other words, trees may tend to fall more often in some areas compared to others. For example, trees tend to fall more often on steep, rocky slopes with shallow soils, on ridge tops, or in swampy areas. This results in a clumped or patchy distribution of tree fall gaps. Trees also fall more often during certain times of the year. For example, trees fall more often when it is very windy, or when the soil is saturated with water.

In order to assess the frequency of creation of treefall gaps, record the presence of all gaps seen from transects. Since a fallen tree can remain visible for decades, in order to standardise your searching, include only those gaps made by a fallen canopy tree where the small branches of the fallen tree are still intact. (This indicates that the tree fell recently.) By recording only "large" and "new" tree fall gaps, comparable information can be collected from year to year. Determine the frequency by calculating the number of new tree fall gaps seen for every kilometre of transect.

For each fallen tree record the following information:

- the location of tree falls (distance along the transect);
- the length of the fallen tree, in order to get an indication of the size of the gap (you can also estimate the surface area of the gap);
- the species of the tree (if possible);
- an explanation as to why the tree fell (if possible): for example, the tree was growing on a rocky ridgetop; the tree was uprooted in shallow, swampy soil; the tree trunk was rotten in the middle and snapped in two, etc.

Treefall gaps offer a good opportunity to study forest succession and regeneration. Many interesting things can be learned if the structure and composition of plant species living in a tree fall gap are studied over time (see Box 9a). As such, your data may represent an important baseline for future studies of vegetation composition in gaps of different ages and sizes.

Box 9f HOW HUMANS HAVE CHANGED THE AFRICA RAIN FOREST OVER THE LAST 2,000 YEARS

Within the African rain forest, the oil palm, *Elaeis guineensis*, is found most commonly growing in and around inhabited or recently abandoned villages. Therefore, when oil palms are found in the forest it suggests the area was once inhabited by people, although some natural regeneration occurs in river and savanna gallery forests. In 1988 Mike Fay (Fay, 1997) found numerous palm nuts (he estimated hundreds of thousands) in the bed of a small stream in southwestern CAR. The forest along the stream was mature *Gilbertiodendron dewevrei* forest and no oil palm trees were observed in the area, although they do occur around villages in the region.

This was an intriguing puzzle. In order to try to find out more, Fay sent two samples of palm nuts to a laboratory in the USA to be carbon dated (a sophisticated analysis that estimates the age of once-living organic matter). The nuts were found to be 1590±50 and 1510 ±60 years old, showing that the trees which had dropped the nuts had grown in the 5th century AD. In the meantime Fay and his colleagues found more and more deposits of palm nuts in the adjacent forests along the borders between Central African Republic, Congo and Cameroon. Palm nuts were found in over 80% of streams during hundreds of kilometres of reconnaissance walks through what were thought to be undisturbed (primary) rain forests. A sample of 80 of these were analysed and gave a range of carbon dates between 2340 - 990 years BP.

Fay's conclusion is that the 'primary' forests of northern Congo and adjacent areas of Cameroon and CAR were, in fact, densely populated plantations up to about 1,000 years ago, when, for some reason, human populations crashed. Hence, in this case the oil palm was an indicator of dwellings that had disappeared many centuries before. Similar deposits have been found in many parts of west and central Africa and indicate that the forests of the region have had a turbulent past. Always bear this in mind when trying to explain distribution patterns within the forest and keep your eyes open for palm nuts, charcoal and pottery in road cuttings and stream beds.

Conservation is a long-term ideal. We are inevitably concerned with long-term monitoring in addition to documenting any short-term change. Many African rain forests, once considered pristine like Nouabalé Ndoki, have proved to have undergone marked change in the recent past. Jones (1955, 1956) discussed the history of forests in southwest Nigeria renowned for their large mahoganies and came to the conclusion that they had been farmland about 300 years before. In fact, new research has shown that these forests replaced a mosaic of farm and secondary vegetation when human populations declined about 700 years ago (White & Oates, 1999). In Ituri, RDC, Terese and John Hart recently discovered evidence of extensive fires, dated to about 2000 years ago, sweeping through an area now occupied by monodominant *Gilbertiodendron* forest. *Gilbertiodendron* forests were previously presumed to be an ancient formation, but the Harts were surprised to find that the charcoal was from other species, suggesting that the present vegetation is relatively young (Hart et al., 1996). In Lopé, Gabon, large areas of forest were savanna about 1,000 years ago. Forests only established in the wake of a crash in human populations, who had maintained savannas with fire for thousands of years (Oslisly et al., 1996). Furthermore, recent archeological finds in central Gabon indicate that forests have been much disturbed by humans over the last 2,000 years (Oslilsly & White, 1996).

Together, these examples illustrate the dynamism of what can appear a static system to short lived human beings. In the more distant past climatic change has caused wide scale movements in the borders between forest and savanna vegetation in Africa (e. g., Maley 1996) and this is reflected both by animal distributions (see Kingdon, 1990) and densities (White, 1994b).

Quantitative classification

A quantitative description of a habitat is a numerical description of that habitat's features. Generally, quantitative habitat descriptions are more detailed than qualitative habitat descriptions. For example, you can say quantitatively that 23% of the trees in the primary mixed forest are in the family Caesalpiniaceae, where the mean tree density is 579 trees/ha, and the mean trunk diameter (including only trees greater than 10 cm in diameter) is 32 cm.

Quantitative studies are extremely useful for many reasons, and can provide the kinds of detailed information that are needed to make effective, realistic management decisions. For example they can be used to (the list is almost endless but a few examples are given here):

- determine which species are common, and which ones are rare in any given area;
- quantify species composition in different habitat types, and hence refine and improve subjective vegetation classifications;
- quantify the density of fruit trees that provide important foods for animals.
- help determine if a timber species or other plant product is being exploited sustainably;
- assess how long it takes old farmland to return to mature forest;
- map the distribution of important forest types;
- provide a record from which all future changes can be assessed;
- provide the means to make detailed comparisons with other forests;
- investigate links between vegetation and animal numbers or movements;

Information that can be obtained from a quantitative study

Forest structure

The physical structure is one of the most obvious ways to describe a forest, and is important because it can help determine the types of species present. In particular, the quantity of sunlight that reaches the forest floor can strongly influence the species composition of the trees and other plants (see Box 9d), which in turn, can influence which animal species are found there.

In addition, certain animal species may be restricted to forests with specific physical characteristics. For example, vegetation that is dense enough to hide in, or trees that are strong enough to climb in.

Average canopy height

The true height of a forest's canopy is difficult to measure. Standing beneath a tree and guessing its height is not a reliable method, even though it is perhaps the most commonly used. Your estimates can be improved by using a clinometer to measure the height of a sample of trees whose heights you have estimated (see Box 6f). If tree / canopy height is important for your study it is best to measure all heights. Another way to estimate the average canopy height is to use a tape measure to measure the lengths of numerous fallen canopy trees, and then take the mean, or average, of those measurements.

Canopy cover

Canopy cover gives a measure of the amount of light penetrating through to different layers of the forest. As a rule, undisturbed forests have high canopy cover and there is only sparse ground vegetation in their gloomy under-storey. Secondary or disturbed forests tend to have incomplete canopy cover and dense ground vegetation. Many methods have been used to estimate canopy cover. Some researchers have used photographs taken vertically, with either a standard or fish-eye lens, and estimate the proportion of their pictures are covered by sky. Others have used light meters (or have used cameras to record the exposure required at a certain film and shutter speed as a measure of light availability). A simple method using a clinometer is given below (page 136-137).

Density of the herbaceous under-storey

Various studies across the rain forest zone of Africa have demonstrated the abundance of Terrestrial Herbaceous Vegetation (THV): ground rooted species in the subclass Monocotyledonae, in certain types of forest (e.g., Fay, 1997, whose definition is adopted here; Rogers & Williamson, 1987; White et al., 1995; Wrangham, 1986). In particular, some forests, often referred to as 'Marantaceae forests', have dense herb layers dominated by several species of THV.

THV is an important food for many large mammals, including elephants, gorillas, chimpanzees, bonobos and mandrills. Indeed, it seems that densities of at least some of these species (particularly elephants and gorillas) are generally highest in forests with abundant THV. In addition, abundant THV is often an indicator of past disturbance (such as agriculture or forest fires) or other forms of vegetation change (such as forest expansion into savanna areas).

Hence, it is informative to assess THV densities during vegetation inventories. This can be done in small plots ($1m^2$ or $4m^2$ are most commonly used) located randomly or systematically (see Chapter 3) along a transect or as nested sub-plots located within a larger plot. The quickest way to locate the boundaries of a small plot is to use a frame, or four sticks. The inside dimensions of the frame should equal the size of the plot (for example, 1m by 1m). Frames can be constructed of wood, metal pipe, plastic pipe or other sturdy material. Plot frames are easier to use if they are lightweight, and if they can be broken into smaller pieces for carrying. Circular plots can be used as an alternative to squares: place a stake in the centre of the plot and use a tape measure or a length of string cut to the appropriate length to determine which stems are within the plot.

Record the number of stems and species for each species of Marantaceae, Zingiberaceae, Commelinaceae, Arecaceae (Palmae), Araceae, Dioscoriaceae (wild yams), Cyperaceae and Poaceae (Graminae) (for the latter two it may be more convenient to note estimated % cover). Bear in mind that sampling the herbaceous under-storey of a Marantaceae forest will be time consuming. You will have done a good days work if you manage to count and identify all herbaceous stems in $200m^2$.

Figure 9.1: A classic Marantaceae forest.

Measuring the relative importance of different tree species: abundance and size.

Tree abundance is usually described in terms of density or basal area. Density gives a measure of the number of trees whilst basal area takes their relative sizes into account.

Measuring tree abundance: density

Tree density is one of the most common ways of quantitatively describing a forest because:
1) tree density is easily and accurately measured;
2) densities of various species can be used to classify different forest types;
3) knowing how many trees there are of each important species (importance can be measured in commercial, cultural or biological terms) can be very helpful for management.

The density of all trees above a minimum diameter (often • 10cm dbh) in the sampling area is determined by counting all the trees in a known-sized area (i.e. the total area of one or a series of plots) and dividing the number of trees by the area. For example, if 1000 trees were counted in 2 hectares, then the density of trees is 1000 trees divided by 2 hectares, which equals 500 trees/ha, or 50,000 trees/km^2.

The density of all the trees combined tells how densely the trees are packed in the forest. The total density of trees can indicate growing conditions. For example, recent disturbance from wind, fire or cutting may reduce the total density of trees. Also, the quality of the soil or the availability of water can affect the total density of trees.

Tree density also can be determined for each species individually. (The density of a single species is found by dividing the total number of trees counted of that species by the total area sampled.)

Estimates of how many trees there are for each species can suggest how many trees may be available (or not available) to harvest sustainably. Furthermore, abundance information for individual species allows detailed comparisons to be made between different areas (for example, between different habitat types, between areas experiencing different levels of disturbance, or between forests in different places). Abundance information for individual species also can be used to assess changes in species composition over time.

Note that density reveals nothing about the size of the individual trees (except that they are all above the predetermined minimum).

Measuring tree size: dbh and Basal Area

Collecting information on tree size, in addition to tree abundance, helps to determine the relative "importance" of two or more species. For example, one species may be a large, canopy-emergent species, while another may be only a medium-sized, mid-story species. Even though there may be the same number of trees of each species in the forest (i.e., the same density), the larger species will probably provide more fruit or wood, more climbing space for lianes, monkeys or ants, more nesting sites for birds and more crevices where epiphytic plants can grow.

For any given species the diameter of the trunk is usually related to both the height of the tree and the width of its canopy. Of these three measurements, trunk diameter is used most often to indicate a tree's size because it can be measured the most easily and accurately. (If specific ecological questions require information about tree height or canopy width then they certainly can be measured independently of trunk diameter).

Trunk diameter can be used to calculate size-class distributions (see Box 9g) and basal areas (see below).

Box 9g SIZE CLASS DISTRIBUTIONS

A size class distribution is a simple, graphic way of presenting the range of tree diameters found in an area. By displaying tree diameters in an orderly manner, rates of tree reproduction, growth and mortality can be better understood.

Making a Size Class Distribution Graph

To show graphically, or in picture form, the distribution of tree sizes observed in the forest, place the information in size classes. Size classes can be any size, but 10 cm size classes are common (for example, 10-19 cm dbh, 20-29 cm dbh, 30-39 cm dbh, etc.). Then, determine the number of trees (or calculate a density of trees per hectare) in each size class and make a chart (Table B9.1).

Table B9.1 Example of a chart used to make a size class distribution graph.

Tree Diameter (cm)	Number of Trees
10-19	457
20-29	191
30-39	119
40-49	82
50-59	35
60-69	26
70-79	25
80-89	17
90-99	5
110-119	5
120-129	6
130-139	2
>140	4

Finally, graph the size class distribution (see Fig. B9.2). First, mark the size classes along the horizontal axis. Second, mark the vertical axis, making sure you have enough room to include the density of trees found in the fullest class. Third, fill in the graph using information from the chart.

Interpreting Size Class Distribution Curves

Typically, size class distributions resemble a "reversed J-shaped" curve, where there are many small sized (or young) trees. This high number drops off rapidly at first, and then more slowly, as tree size increases. The curve's shape indicates that although many young seedlings and saplings are able to grow in the forest, most die off quickly leaving only a few medium sized trees, and even fewer large and very large trees.

Subtle changes in the shape of the "J" curve can be revealing. However, unless there are very distinct differences between two samples (such as one would find if one compared young secondary forest to undisturbed closed canopy rain forest) plotting size class distributions for all plants may obscure some of the information that can be obtained from this type of analysis. Fig B9.3 shows the size class distribution of an important timber tree, *Aucoumea klaineana* (Okoumé) growing in four forest types in Lopé, Gabon.

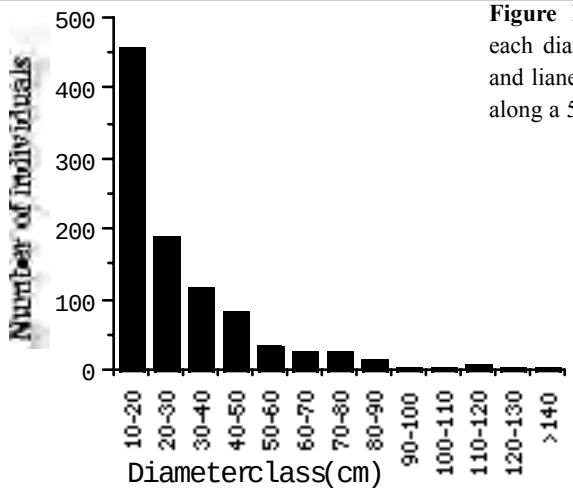


Figure B9.2. Number of individuals in each diameter class for a sample of trees and lianes • 10 cm dbh in a 2.5 ha sample along a 5 km transect (Tr. 4 in Fig. 9.8).

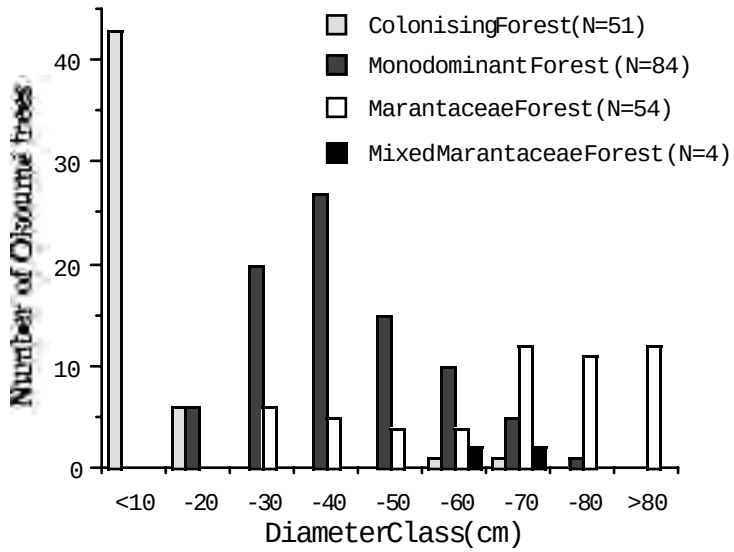


Figure B9.3. Size class distribution curves for *Aucoumea klaineana* in four forest types in the Lopé Reserve, Gabon. (from White, 1995 - five rectangular 20 x 40m plots were located randomly in each vegetation type and all plants • 5 cm dbh were counted).

This figure demonstrates that in 'colonising forest' there has been a recent establishment of Okoumé, with many small individuals present but very few large trees. In 'monodominant forest' Okoumé is common, particularly as medium sized (aged) trees, but it seems that there is no regeneration since no trees occur in the smallest diameter class. In 'Marantaceae forest' most trees are large (old) and smaller diameter classes are poorly represented and in 'mixed Marantaceae forest' only a few large (old) trees were found. These forest types are in fact stages in a natural succession from savanna to forest which occurs when savanna fires cease (see Box 9d). In this case size class distributions are a quick way of revealing things about a forest that otherwise might remain hidden. Think of them as pictures of the past.

Diameter at breast height (dbh)

The "diameter at breast height" or "dbh" is the standard way of measuring and reporting the size of a tree. To measure the dbh of a tree, place a measuring tape around the circumference of the tree trunk (see Fig. 9.2) at 1.3 m off the ground. If buttress or stilt roots are encountered at that height then the tape must be raised to the first place where the trunk can be circled without including any roots (Fig. 9.3). If a tree is going to be measured again in the future, in order to monitor its growth, the exact height at which the diameter was measured should be permanently marked (see below).

Because the distance around the tree (its circumference) and not the distance straight through the centre of the tree (its diameter) is measured, one of two things can be done in order to calculate the diameter. First, a specially marked "dbh tape" can be used to measure the tree's diameter. A dbh tape is marked in such a way that when it is placed around the tree trunk, the number read off of the tape is actually the diameter, and not the circumference. These tapes are available from companies that supply forestry equipment.

Second, the circumference of the tree trunk can be measured using a standard tape marked off in centimetres, and later the circumferences can be converted to diameters. To calculate a tree's diameter divide each measured circumference by pi (3.1416).

$C = 2šr = šd$; $r = C/2š$; $d = C/š$; $A = šr^2 = š(d/2)^2$

Translation Error

Figure 9.2 The circumference, diameter, radius and area of a circle.

Basal Area

Basal area refers to the area covered by a cross-section, or slice, through a tree trunk (see Fig. 9.2). Basal area is used instead of diameter when the sizes of the all the trees are added together (adding together tree diameters will not result in a meaningful number). Total basal area gives an indication of the total quantity of the measured trees (which later, if desired, can be converted to weight or volume). Basal area is used to determine the relative "importance" of a given species. In other words a few large trees may have a larger total basal area than many small trees. Similarly, those few large trees may provide more wood, climbing space or fruit and seeds than the many smaller trees combined.

The basal area of a single tree is calculated as follows. First, divide the trunk diameter by 2 to get the radius (for simplicity the trunk is assumed to be circular in cross-section, see Figure 9.2). Second, square the radius (the radius multiplied by the radius). Third, multiply that number by pi (= š = 3.1416).

For example, what is the basal area of a tree with a diameter of 40 cm? First, divide 40 cm by 2 to get a radius of 20 cm. Second, multiply the radius by the radius, or 20 cm x 20 cm equals 400 cm^2. Third, multiply that number by pi, or 400 cm^2 x 3.1416 equals 1256.6 cm^2 (=0.12566 m2; 1m2 = 10,000 cm^2).

Duboscia macrocarpa - a tree with a fluted trunk which poses a problem for dbh measures!

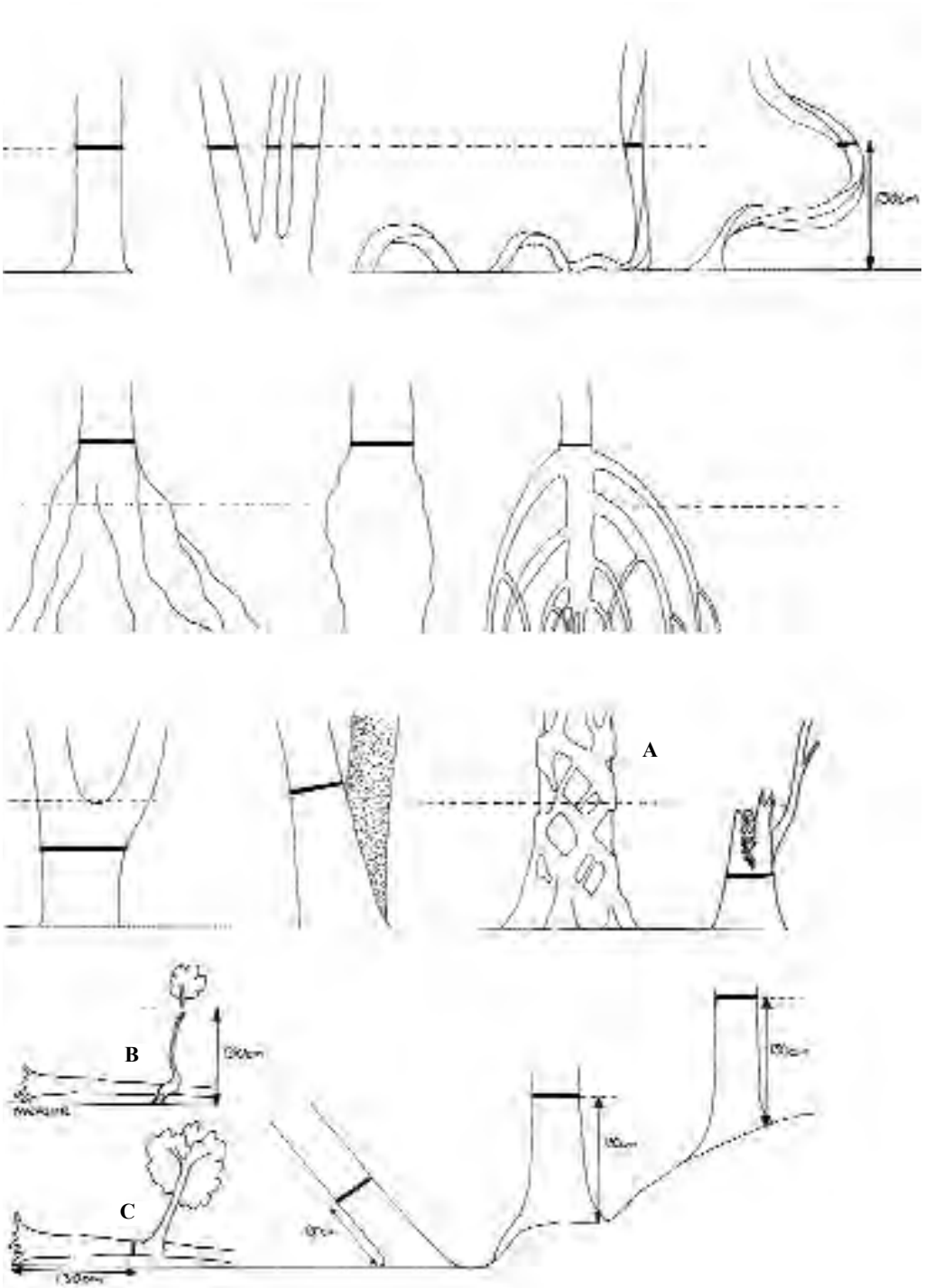


Figure 9.3 Measuring the dbh of 'normal' and problem plants
A. strangler fig - estimate diameter of the trunk where it leaves the host; **B.** dead fallen tree with rooted sucker - measure sucker; **C.** live fallen tree with unrooted branch - measure tree.

Equipment needs

The following equipment is needed for a quantitative vegetation inventory:

- Standard collecting equipment for botanical specimens (see Chapter 8);
- Camping equipment if you will not be working out of a base camp;
- 50 meter tapes;
- Claw hammers;
- Aluminium or galvanised nails[1];
- Numbered tree tags[1];
- Datasheets in a clipboard;
- Pencils and erasers;
- Sighting compasses;
- Clinometers;
- Hip chain with extra cotton line;
- 10m dbh tapes (see Box 9h to make a dbh tape if you cannot purchase one);
- Rolls flagging tape;
- Permanent marker pens;
- One litre tins of red paint;
- Wooden stakes of 1.5 meter length are also required if you intend to permanently mark your plots or transects. Note: the end of the stake should be painted red.

[1] the number of nails and tree tags should reflect the number of stems expected per plot. As a general rule always take more than you think will be needed as it is better to bring back unused equipment than be unable to finish marking the stems within the plot.

Initiating a quantitative vegetation study

A preliminary qualitative survey (perhaps as part of an animal census) can help define questions of interest and suggest appropriate locations to undertake more detailed quantitative botanical studies. The use of 5-km transects has proved a useful first stage in characterisation of vegetation in many sites, including Lopé (Gabon), Ituri and Kahuzi Biega (RDC), Lobéké and Korup (Cameroon) and it has been adopted as a standard method by the ECOFAC program, in Odzala (Congo), Monte Alèn (Equatorial Guinee), Dja (Cameroon) and N'gotto (Central African Republic). We therefore recommend this as the first approach in sites where little is known about the vegetation, in order to produce results that are comparable across sites and countries.

The rationale behind the use of a long, relatively narrow transect is that it will pass through many habitat types and provide a good overall measure of vegetation composition and variety. Furthermore, the length chosen here is ideal for censusing several groups of large mammal (see Chapter 11), so a number of types of information can be collected in the same area, increasing the value of all the data sets.

To locate transects, first study a map of the area (if no map is available make a sketch map yourself - see Chapter 6). In order to sample representative proportions of ridge, valley side and watercourses / marshes, providing a sample representative of the study area, the transect(s) should cut across the major drainage features of the area (Norton-Griffiths, 1978). That is, it should run approximately 90° (perpendicular) to the larger streams and rivers and mountain ridges. Start from a randomly selected point within your study area (see Chapter 3) and cut a transect 5 km long on a compass bearing crossing the drainage (see Page 220 for how to cut a transect). Cut a minimum of vegetation and be sure not to damage trees or other vegetation likely to be counted in your sample (or to be recruited in later years when samples are repeated). Mark the transect every 25m with coloured ribbon or stakes (at this stage the most convenient measuring instrument is a hip chain). Record GPS coordinates for the start and end points.

Data collection will then advance along the transect from one end to the other, or from a centrally located camp. When recording data measure all distances with a tape measure (this is more accurate than a hip chain). The locations of notable physical features (e.g., streams, marshes, rocky outcrops, old logging roads, ridges, treefalls, old secondary vegetation), or changes in soil type, should be recorded, and average gradient should be measured for each 50m section of each transect using a clinometer (see Chapter 6).

Measure diameters of all trees and lianes >10 cm dbh in a 5 metre strip, 2.5 m on either side of the transect. This gives a sample of 5m x 5km = 2,500m^2 = 2.5 ha per transect). Measures should be taken at 1.3m above the ground (diameter at breast height, dbh), or immediately above stilt or buttress roots, using a diameter tape. The centre of a tree has to be within 2.5m of the transect line for the tree to be included in the sample (Fig. 9.4). Record measures to the nearest millimetre. Cut a pole 1.3m long to measure to the height at which the diameter is to be measured. Figure 9.3 shows you how to deal with irregularly shaped or broken individuals. Take data for dead trees but record that they are no longer alive. Each tree should be labelled with a numbered tag. Use Aluminium or galvanised nails if possible (these are less likely to rust and allow rot to enter the trunk) and attach the tag exactly 30 cm above (or below, if there are not likely to be too many elephants rubbing against your trees) the point at which the dbh measure was taken. In this way future repeat measures can be taken in the same place. Where this is not done accuracy of future measurements will be much reduced. In Ituri, RDC tags are tied on to trees in a large botanical plot to avoid any increased mortality due to tagging and the point of measurement is painted on the bark.

Trees marked with **X** are outside the quadrat and are not measured

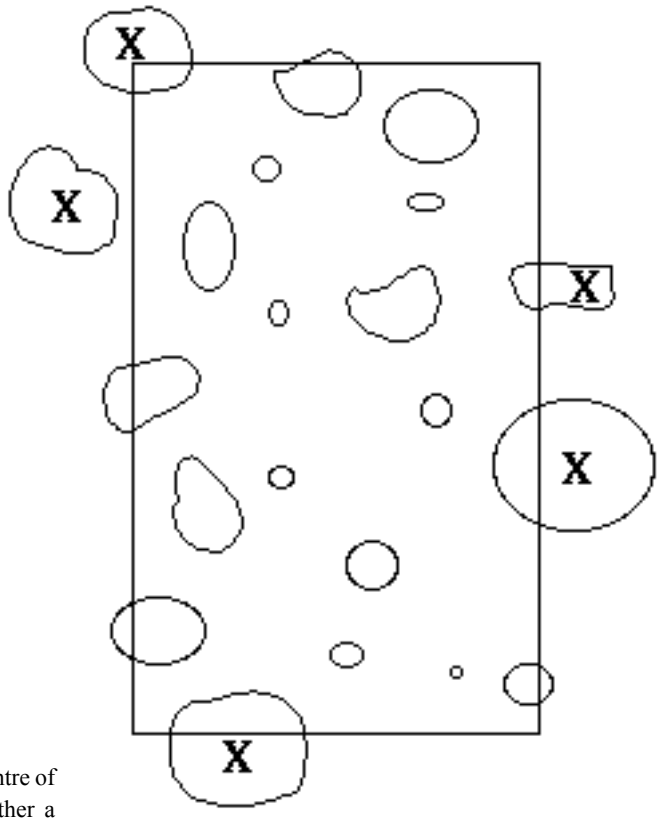


Figure 9.4 The location of the centre of a tree is used to determine whether a tree or liane is inside or outside a plot.

Vegetation inventory and description

Trees can be identified using a combination of sterile characters: general form (e.g., buttresses, stilt roots); bark texture; slash colour, smell and exudates (but only slash when necessary and keep the size of incisions to a minimum, since this can let in infections which occasionally lead to the death of trees!); leaf type and shape (see Box 9h). Whenever there is doubt about identification in the field, leaves should be collected and compared with herbarium specimens, and where possible collections of flowers or fruit with leaves made. A voucher specimen of each new species encountered should be taken.

Locate each individual using the distance along the transect and the distance to the left or right of the transect as coordinates. Estimate the height of each individual and the height to the first branch. If time allows these measures should be taken using a clinometer or equivalent. Repeat the process for all trees >60 cm dbh (optional - especially useful if studying logging ; threshold can be varied to suit the goals of your study) in a 50 metre strip, 25m either side of the transect. Note which individuals have suffered bark removal by elephants or other browsers and squirrels.

A long the length of the transect estimate canopy cover at four heights (0-2m, 2-10m, 10-20m, >20m). To do this one can use an adaptation of a classic method used by botanists, the point quadrat method (Greig-Smith, 1983 - Figure 9.5): Take measures displaced 2m off the transect at 5m intervals. Prepare a checksheet with five columns, one for distance along the transect and four for the height categories (Figure 9.6). For measures between 0-2m, use a 2m long pole to approximate a point; hold it vertically and if it touches vegetation at any point record "+", if not record "-". For measures above 2m, use a sighting clinometer to look up vertically. Use the cross wire in the sight of the clinometer as a "point". Score canopy as present "+" or absent "-" in each of the three height classes (estimated by eye) depending on whether or not the point in the sight projects upon vegetation. Thus 1000 points will be recorded at each height along each 5000m transect. Overall cover is scored as present if the point fell upon vegetation in any of the height categories.

Box 9h SPECIES IDENTIFICATIONS

The most important thing to remember when identifying a tree or plant species is to be certain of the identification. It is better to list a tree as "unknown" than to include an identification you are uncertain of, thereby leaving all of your identifications open to suspicion. Misidentifications will help no-one.

When recording a species' identification it is best to record the "scientific name". Scientific names have been standardised throughout the world. As a result, scientific names are the most reliable way of passing on information to someone not familiar with a particular forest. The use of scientific names allows for accurate comparisons between forests.

If a tree or plant cannot be identified to species, then identifying it to genus, or family, or "morpho-type" (species with similar features, such as similar fruits) also will be useful. If the scientific name is not known then local names can be used. If local names are used it is critical that all individuals of that species (and no other species) are called the same thing. The same local names may be used for several similar species, or species with similar uses, so beware.

If a local name is not known then "unknown 1" can be used for all plants that appear to belong to the same unknown species. After that, "unknown 2" can be used for plants of another unknown species, and so on. If plants of a single species are consistently identified as being the same, using either a local name, or "unknown 1", "unknown 2", "unknown 3", etc., then they can be counted and measured now, and the scientific name can be applied later.

If, even after much research, a species remains unidentified, then it should be listed in a final report as "unknown". Most studies that attempt to identify tree or plant species will list:

1) the percentage of individuals that were unknown; and

2) the number of species that were not identified (unknowns).

For example, if 300 out of 400 trees were identified, then 25% of the trees were "unknowns". These unknowns may have included 36 different species.

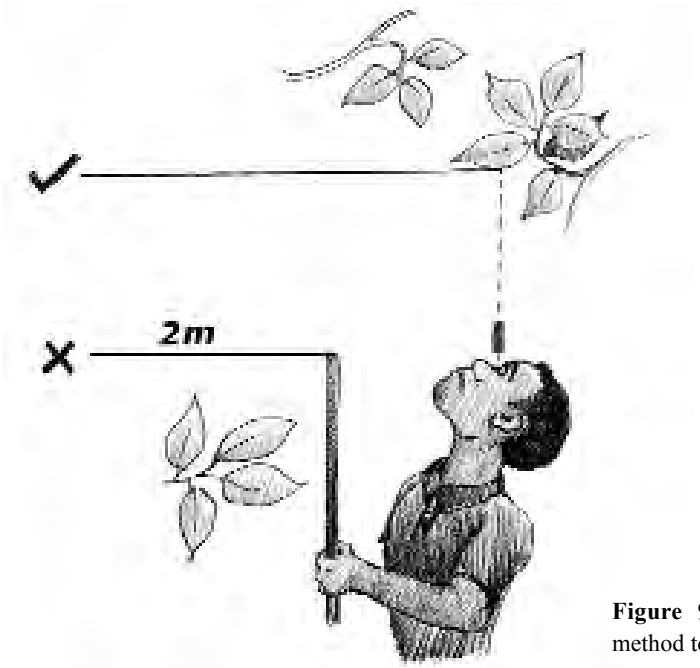


Figure 9.5 Using a point quadrat method to estimate canopy cover

Distance	0-2	2-10	10-20	>20
75	+	-	-	+
80	+	+	-	+
85	-	+	-	+
90	-	-	+	+
95	-	+	+	+
100	-	+	+	-
105	+	-	-	-
110	-	+	+	-
115	+	-	-	+
120	+	-	+	+
125	-	-	+	+

Figure 9.6: Part of a checksheet for measures of canopy cover.

In addition, count stem densities in $1m^2$ plots, located every 5m along the transect, of each species of monocotyledon encountered (particularly the families Arecaceae (palms), Commelinaceae, Marantaceae and Zingiberaceae). In order to avoid any effects of cutting the transect, displace plots two meters off the transect. To mark the plot make a frame or quadrat out of wood or metal, or put four stakes into the ground to mark the four corners. For Poaceae (Graminae) and Cyperaceae estimate % cover. Note also the dominant herb or shrub species in terms of canopy (leaf) cover.

Given that these long thin plots cut through several vegetation types, but do not necessarily sample all habitats in great detail, they represent the first stage in a detailed description of vegetation. As researchers become more familiar with looking at vegetation they will begin to pick up subtle variation which they did not previously notice. Aerial photographs should be inspected to check whether all vegetation types recognised on the ground can be detected from the air. Rare habitats that are poorly represented on transects can be sampled later using plots.

Plots or transects?

The transects described above are in fact long, narrow plots designed to sample a maximum variety of habitats. They are ideal for your first botanical survey in an unknown forest area (in forest/savanna mosaics you may have to use shorter transects or plots if you want to restrict the sample to forest vegetation). However, once you have classified a number of vegetation types which can be recognised in the field, or if you are working in an area for which vegetation maps already exist, it will probably be more appropriate to sample in plots, randomly located within specific vegetation types. Plot size will depend on the exact aims of your study (see Box 9i).

How big a sample should be collected?

When planning a vegetation sampling study one question which you are bound to ask is "how large a data set do I need to collect before I have a representative sample of the vegetation in my site?". Given the diversity of tropical rain forests you will never obtain a sample containing all plants and many rare species will be represented by only one or a few individuals. Furthermore, as you increase the size of your sample and your study area you will encounter new habitats and enter the distribution ranges of new species. Therefore, all you can hope to obtain is the species composition and abundance from a representative sample in your study area, or from the various habitat types within it.

One quick and simple means of assessing the completeness of your sample is to plot a species-area curve (see Figure 9.7). This involves dividing your sample into a number of equal sub-units and plotting a cumulative number of species encountered against the area sampled. If using many small plots each plot can be used as a discrete unit. If working on a long transect analyse it by 50m or 100m sections. Count the total number of species in the first plot or section. For the next sampling unit count the number of species found which were not in the first sample. For the third count the number of species not previously encountered in the previous two, and so on. When you have done this for all the plots or transect lengths plot a graph of total area sampled (x axis) against the cumulative number of species encountered (y axis).

Figure 9.7 Three theoretical species-area curves.

Figure 9.7 shows three theoretical species-area curves. Lines 1 & 2 pass through homogeneous areas (one habitat type). Line 3 passes through a heterogeneous area (more than one habitat type). Sample 1 was in a species rich area so the species-area curve is still rising after the completion of 30 plots. This suggests that the sample was not large enough. The forest where Sample 2 was taken was less species-rich and the species -area curve levels off well before 30 plots, showing that the sample was representative of this area. Sample 3 is not represented by a smooth line. In this case the species-area curve begins to level off after 10 plots (i.e., ten 100m sections of a transect) but then the researchers moved into a new habitat and encountered many new species (the transect crossed a river and entered seasonally flooded forest). A second new habitat was encountered after 25 plots and the curve was still rising after 30 plots. The fact that the line is still rising at the end of the transect suggests that 3km of transect (thirty 100m sections) was not long enough to provide a representative sample.

In a study in Gabon species area curves were plotted for five 5-km line transects by dividing each one into 100m sections. Figure 9.8 shows that there were distinct differences in species diversity on the five transects (Tr. 3 being the most diverse and Tr. 1 the least). As sample size (in this case length of transect sampled) increases, the number of new species (those not encountered previously on the transect) per unit area gradually decreases. Occasional increases (steeper sections in the curve) correspond to areas where the transect passes through a new habitat type. As the sample becomes more representative of the study site the slope of the curve becomes flatter. In theory one could continue to sample until the slope becomes zero (i.e., all species present have occurred at least once in your sample), but in practice this is almost never possible. Instead, as a 'rule of thumb' inspect the curve and see if it has begun to even out. In Figure 9.8, the sample in Site 1 (Tr. 1) is probably representative of the area as a whole as the slope of the curve has levelled off (although note the increase at 4 km as the transect crosses a large stream and enters a slightly different forest type). Similarly, in Site 5 (Tr. 5) the curve is levelling off and the sample is probably almost sufficiently large. However, in the other three sites new species continue to be encountered on a regular basis and these samples should be increased. Note particularly Site 2 (Tr. 2) where the curve seemed to be levelling off just after 3000m but then increased sharply (here the transect crossed a large river and entered a forest type which was quite different to that in the first 3 km of the sample!).

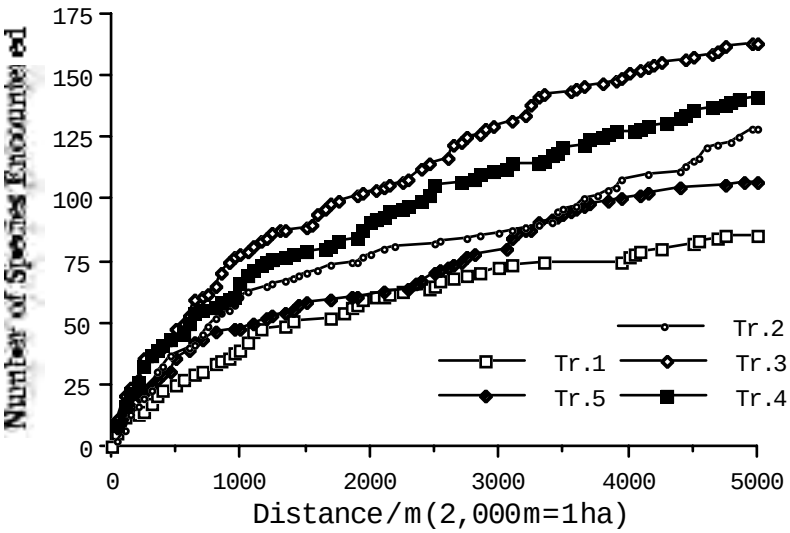


Figure 9.8 Species area curves for five 5-km line transects in the Lopé Reserve, Gabon on which all trees • 10 cm dbh in a 5m strip were counted, giving a sample of 2.5 ha (taken from White, 1992).

Repeating transects to collect mortality, recruitment and growth rate data

By repeatedly sampling the same vegetation plots over time (i.e., once every year or every five years) you will be able to collect data on:

- mortality rates of trees and lianes;
- recruitment rates of trees and lianes;
- growth rates of trees and lianes.

Mortality and recruitment data will give you an idea about the dynamics or turnover of the forest in your site. You will also be able to contribute data to pan-tropical studies which are being undertaken to assess the possible impacts of climate change (see Boxes 7a & 9j). Growth rate data for tropical tree species are rare. Without these it is difficult to design long-term timber harvesting programs. If you don't know how fast the trees grow how can you set rotation cycles? An extreme example is the case of the renowned African ebony, *Diospyros crassiflora*. Few data exist on its growth rate, but during 10 years, two individuals monitored in Lopé had mean growth rates of 1mm per year. Other species in the same genus grew even more slowly (e.g., *Diospyros zenkeri* which grew at a mean of 0.5 mm per year). Hence, one can extrapolate that an ebony tree of 1m diameter could be about 1,000 years old - how long a rotation cycle will be necessary to manage such a species?

By carefully recording the location of your vegetation samples and by tagging trees you make longterm monitoring possible, thereby increasing the value of your efforts. Transects or plots should be repeated at five yearly intervals, or more frequently if possible. The dbh of all trees should be remeasured and the reason for any mortalities noted if apparent. New recruits which were too small to be included in the sample on previous repeats should be identified, measured and tagged.

The next stage: large scale patterns and mapping

Integrating remote sensing and botanical studies

Once you have a good working knowledge of vegetation types, it is interesting and useful to produce a vegetation map for your study area. Vegetation maps are useful for many reasons, for example:

- to ensure botanical collections or other species diversity sampling have been undertaken in all habitats in the protected area;
- to delimit rare, fragile or particularly rich habitats which should be protected from logging or other human impacts;
- to identify key habitats for focal animal species, in order to select priority areas for conservation action;
- to help to explain seasonal movements of animals around, in and out of a protected area;
- to plan effective animal census programs by stratifying sampling effort (see Chapter 3);

A vegetation map can be produced from two types of information: 1) aerial or satellite imagery; and 2) observations undertaken on the ground (ground truthing). Most maps involve both stages. If aerial photographs or satellite images exist for your site then the next stage is to study them carefully to see what patterns can be detected. Some patterns in the images will be easy to classify, particularly areas where you are familiar with the vegetation. Patterns in areas you have never visited may correspond to those in areas you do know. But where new or unexplained patterns occur you will have to undertake qualitative or quantitative studies on the ground to interpret the imagery. Aerial photographs are generally of more use than satellite images. An alternative is radar imagery, which is very good for showing topography and of some limited use for vegetation mapping, but which may not provide the detail you are looking for (see Box 9k for more details).

Box 9i DETERMINING THE RIGHT PLOT SIZE

Plots can be any size or shape, but some sizes and shapes are more appropriate than others for studying trees in forests. Square or rectangular plots are used most often, but the length and width of the plots varies between studies. The shape and size of plots is often determined by balancing three factors:

1) the ability to determine plot boundaries with minimum effort;
2) reducing "edge effects";
3) expected scale of pattern in the vegetation.

Locating Plot Boundaries with Minimum Effort

When plot boundaries are located efficiently, the process of counting trees goes more quickly. Locating plot boundaries is simplified if a trail (transect) is used as a starting point for measuring, and the width of the plot is small. Therefore, long, narrow plots, with a trail running down the centre of them, are well designed for studies that must proceed quickly (for example, when counting trees over a large area).

A plot's boundaries should be located accurately, using a meter-tape or a hand-made measuring device. Such hand-made devices can include a string with knots tied at pre-determined distances, or for very narrow plots (for example, □ 5m wide), a stick of the appropriate length (e.g., 2.5m long).

Reducing "Edge Effects"

An "edge effect" refers to anything that affects trees on the edge of the plot that does not affect trees in the centre of the plot. Since it is possible to mistakenly include or exclude a tree standing on the plot boundary (see Fig. 9.4), an increase in the percentage of "edge trees" will probably result in an increase in sampling error.

When plots are used, edge effects should be reduced whenever possible since they are a potential source of error in the data. Edge effects are reduced in short, broad plots, because the proportion of trees on the edge is less than in long, narrow plots. Edge effects are also reduced in large plots and increased in small plots. The optimal plot shape is circular, since this minimises the edge:surface area ratio. Such plots can easily be measured using a tape measure or string of the appropriate length and a central stake. However, mapping individual trees or dividing the plot into sub-plots is more practical in square or rectangular plots. If you plan to study seedlings and saplings, the trail should not run through the centre of the plot since it may damage or remove some of the seedlings of saplings.

Expected scale of pattern in the vegetation

In relatively homogeneous vegetation plot size and shape are unlikely to alter your results. However, if working in an area with a complex vegetation mosaic with a number of distinct vegetation types, such as in a river delta, or an area where narrow forest galleries snake along water courses in a landscape dominated by savanna, plot size should be designed to sample homogeneous vegetation units.

Examples of Different Plot Sizes:

5m x up to 5000m: The shape and size of this plot is good if you want to survey a large section of unknown forest quickly and to obtain a representative sample of several vegetation types. You may decide only to collect limited information (e.g., only large trees of commercial value, or counting and measuring all trees but only identifying common species, etc.). The shape of this plot makes it easy to locate the plot boundaries from a trail, thereby allowing you to cover a large area in a short amount of time. However, edge effects in this shaped plot will be high and density estimates therefore may be inaccurate.

50m x 10m, or 50m x 20m: The shape and sizes of these plots are good if you want to gather detailed baseline information (i.e. tree density, tree size, species composition). These plot sizes represent a compromise between 1) easily locating the plot boundaries and 2) reducing edge effects. They are also likely to enable you to sample homogeneous units.

First measure 50m along the length of a trail. Then measure either 5m (for the 50m x 10m plot) or 10m (for the 50m x 20m plot) to both sides of the trail to locate the plot boundaries.

20m x 20m or 50m x 50m: The sizes of these square plots are good if you want to initiate a detailed study of forest dynamics, using permanent plots. Square plots are designed to reduce edge effects in order to maximise the precision of the results. Precise results are important if you want to assess changes over time.

A standard method has been proposed by Missouri Botanical Garden and is being applied in various sites in South America, Africa and South East Asia. A one hectare plot measuring 500m x 20m is established. Only stems of 10cm dbh or greater are marked and mapped. The choice of a relatively long rather than circular or square plot means that several vegetation types will probably be sampled, increasing the observed species diversity. Since it is a method which is being used in various sites, the value of data collected using these methods for comparison with other data sets will be increased. So if this shape is feasible at your site this may be a good methodology to adopt.

Box 9j APPEAL FOR INFORMATION ON PERMANENT PLOTS

As part of the Tree Growth Potential Information System (TROPIS) project the Centre for International Forestry Research (CIFOR) is compiling a catalogue of permanent plots in the tropics. They also offer a search facility to help put researchers and potential collaborators in touch with one another. The TROPIS index contains a summary of plot objectives, location, species, history and people involved. Anybody who requests a search of the index will be supplied with a list of plots matching their criteria, as well as the names and addresses of people to contact about the data. They hope that the service will stimulate collaboration between many researchers and encourage anybody currently establishing or monitoring permanent plots to contact them.

Contact: Jerry Vanclay, CIFOR, PO Box 6596 JKPWB, Jakarta 10065, Indonesia.
Fax + 62 251 326433; Email J.Vanclay@cgnet.com.

Pachypodanthium staudtii

Box 9k IMAGERY: MERITS AND COST.

David Wilkie

Once you have undertaken botanical inventories and defined vegetation types you may want to try to map these for your area. This will probably require the use of some form of imagery, in combination with more extensive ground surveys. African nations vary greatly in how comprehensive is their archive of aerial photography. For example, Cameroon has a relatively complete (nation wide) archive of historical aerial photography whereas Congo has only very limited photography. Contemporary aerial photographic coverage (within the last 10 years) does not exist on a national or regional basis, and would be prohibitively expensive to obtain and analyse. Therefore, unless you are lucky enough to be working in an area which is covered by aerial photographs, you may have to commission images, or convince somebody to donate them to you.

Selecting the most appropriate tools

The following is a very brief outline of the advantages and disadvantages of each source of remote sensing information (taken from Wilkie, in press).

For a more detailed description you should consult: Wilkie, D. S. & Finn, J. Y. (1996). *Remote sensing imagery for natural resources monitoring: a guide for first-time users.* Colombia University Press, New York.

Commercial aerial photography

Large format aerial photography (i.e. at least 70mm filmstock) is used in most countries throughout the world to generate topographic and land-use maps. Thus, archival panchromatic, colour, and IR (infra-red) aerial photography often can be purchased from national or state cartographic agencies. If photographs are not available over your area, commercial companies can be contracted to obtain photographs at the most appropriate time and scale. Aerial photography is most appropriate for applications that:

- have a small area or local perspective, < 1000km^2;
- constitute once off or baseline mapping; and
- are interested in detecting and identifying small features, <10m in diameter.

Advantages of aerial photography are: superior spatial resolution; little geographic rectification required; simple operation; and low cost of analysis equipment. Disadvantages of aerial photography are: limited spectral range photographic film; digital analysis only by scanning or digitising photographic prints or transparencies; difficulty in interpreting large volumes of data (large areas); data acquisition and analysis costs high per km2 for areas greater than 1000km^2.

Non-commercial aerial photography

Aerial 35mm photography is most often obtained on an ad hoc basis by the researcher. Using your own 35mm camera oblique photographs are shot through the open windows of a rented small plane flying over the target area. 35mm aerial photography is most appropriate for applications that:

- involve one or a few small areas, < 5km^2;
- require frequent, repeated coverage; and
- are interested in detecting and identifying very small features, <5m in diameter.

Advantages of 35mm aerial photography are: very inexpensive for micro-scale surveys (sampling) and monitoring, aircraft rental $100-$200 per hour; superior spatial resolution, <1:500 scale possible; simple operation; and low cost of analysis equipment. Disadvantages of aerial photography are: geographic rectification required for area estimation of features; digital analysis only by scanning or digitising photographic prints or transparencies; colour infrared film may require users to process and print their own negatives; limited spectral range of photographic film; and difficulty (in time and labour) in interpreting large volumes of data (large areas).

However, the advent of digital cameras that merge traditional 35mm photography with CCD digital data collection, adds ease of digital processing to the advantages of aerial 35mm photography. Though more expensive than photographic imaging with somewhat lower pixel resolution, digital cameras provide almost immediate access to images that can be transmitted electronically across telephone lines and computer networks, can be digitally processed and enhanced, and can be integrated easily into documents using desktop publishing software. The resolution of digital cameras is likely to match that of 35mm photography in the near future.

Aerial videography

Aerial videography can be obtained much like 35mm photography by pointing a videocamera or camcorder through the window of a rented airplane. More often, however, the videocamera is mounted outside the window pointing directly down, and is controlled remotely by the researcher within the plane (Sidle & Ziewitz 1990). Aerial videography is most appropriate for applications that:

- involve a few relatively small areas or samples along a transect or linear feature, < 50km^2;
- require frequent, repeated coverage;
- are interested in detecting and identifying relative large features, >5m in diameter.

As with aerial photography the spatial resolution of aerial videography can be increased by increasing the focal length of the camera lens or by reducing the altitude of the aircraft. Thus, though it is possible to use aerial videography to count poached elephant carcasses or even elephant dung in clearings, this high spatial resolution comes at a cost of a narrow field or view (i.e., narrow survey strip width). Advantages of aerial videography are: least expensive system for small to mid-scale surveys (sampling) and monitoring; ability to view imagery during image acquisition; visual and digital analysis possible; simple operation; visible to near-IR spectral range of video cameras; and low cost of acquisition and analysis equipment. Disadvantages of aerial videography are: low pixel resolution; requires relatively high light levels for image acquisition; and geographic rectification required for area estimation of features. However, technology and software for this technique are advancing fast and it is likely to be much used over the coming years.

Satellite imagery

Satellite imagery can be purchased from commercial companies (e.g., EOSAT, and SPOT Image) or from government agencies (e.g., United States Geological Survey, Eurimage). Images can be obtained in digital form (i.e., the image is stored on tape or disk and must be transferred to, and viewed on a computer), or as black-and-white or colour prints and transparencies. Though satellite imagery, at present, provides the most comprehensive regional scale land cover and land use information globally, these data are, however, not available prior to 1972 when the first Landsat satellite was launched.

Satellite imagery is most appropriate for applications that:

- have a large area, regional or global perspective > 20,000km^2;
- require several spatially separate areas to be surveyed, monitored or compared;
- require frequent, repetitive coverage;
- require or can take advantage of multispectral data; and
- are interested in features larger than the spatial resolution of the imagery.

Several remote sensing systems are suitable for assessing Africa's forest resources at different temporal and spatial resolutions. The AVHRR (Advanced Very High Resolution Radiometer) sensor provides daily coverage at 4km resolution, and 1km resolution at only $90 per scene. The high temporal resolution offers the greatest opportunity for obtaining cloud free imagery quickly and allows for very generalised vegetation mapping at a regional scale, but coarse spatial and spectral resolution (AVHRR was designed as a metereological sensor) preclude its use for detailed vegetation mapping.

A series of high spatial resolution LANDSAT and SPOT satellites have been in orbit since 1973 and 1986 respectively complete coverage for Africa still does not exist - because of cloud cover problems, the absence of a concerted effort to obtain data, and the dramatically increased cost of obtaining imagery since Landsat was privatised in 1984 (prices rose from $200 per scene to $3500). The future may not be so bleak however, at least regarding the cost of obtaining Landsat imagery, as recent legislation changes in the USA will result in much cheaper images being available. Now that Landsat 7 is launched imagery should be available to the public at the cost of duplication and handling (approximately $200). The SPOT system is fully operational with 4 satellites in orbit. However the price of SPOT imagery is $3000 for each image covering 60x60km.

In 1998, the MODIS scanning radiometer was placed in orbit as a component of the first of the Earth Observation System satellites. This satellite will provide high spectral (36 channels between 0.4-15m), and temporal (2 day cycle) resolution imagery at moderate spatial resolutions (250-1,000m), providing an opportunity for regional cloud-free vegetation mapping.

Advantages of satellite imagery are: wide spectral range (UV-IR); digital and visual analysis; digital and photographic (analog) output; wide dynamic range (brightness values) of detectors; quantitative biophysical measurements from radiometric information obtained from calibrated sensors; consistent sun angle, perspective and sensor response; easier to compare different scales and wavelengths; and semi-automated processing that makes use of full dynamic range of the data. Disadvantages of satellite imagery are: low spatial resolution relative to airborne and ground based sensors; and higher startup equipment costs.

Side Looking Airborne Radar (SLAR) and Synthetic Aperture Radar (SAR)

Radar imagery can be obtained either by plane or satellite. The preceding remote sensing systems all depend on the sun to illuminate landscape features. In contrast, radar systems provide their own source of illumination by transmitting an EMR signal in the microwave region. These microwaves are reflected back from landscape features and are detected by the sensor. The timing and intensity of the return signal is used to generate the final radar image. Launch of the Canadian satellite RadarSat in late 1995 provided imagery for the first time over many perennially cloudy areas. Radar imagery is most appropriate for applications that:

- involve areas of perennial cloud cover; and
- are interested in detecting and identifying relative large features, >50m in diameter.

Advantages of radar are: able to obtain data regardless of weather conditions Disadvantages of radar are: high cost of airborne equipment; satellite systems primarily experimental; moderate spatial resolution; image analysis and interpretation methods for natural resource management not well developed; and provides information on terrain/vegetation texture and water content only.

Choosing the most appropriate imagery

Choice of an appropriate source of remote sensing information depends very much on the size of the area to be surveyed and the level of detail required. All imagery available for Africa exhibit a trade-off between spatial detail (the smallest object that can be identified in the imagery), and the spatial coverage of each image. Figure B9.4 shows this trade-off graphically. A single NOAA-AVHRR image covers almost the whole of the Congo basin, but its spatial resolution obscures the fine detail within the landscape. In contrast aerial videography covers only an extremely narrow swath of the landscape but does so with great detail. Table B9.2 describes in very general terms the type of landscape features that can be identified at different map scales, and Table B9.3 summarises the trade-off amongst map scale, spatial resolution, and spatial coverage for the most commonly available sources of land cover and land use data for Central Africa.

More and more conservation projects in the African rain forest region are turning to 'high tech' tools such as satellite images to generate information and decide policy. However, managers and researchers in the field who do not have access to these technologies should not feel disheartened. Digital analysis of remote sensing information is able to detect patches of unique colour within a landscape, but without field based information the human interpreter is unable to identify which colour classes are associated with particular landcover types such as villages, farms, plantations, regrowth forest, and swamp forest. GPS assisted low altitude aerial videography (Marsh et al.1994; Sidle & Ziewitz 1990) will help greatly with image classification, by providing rapid and accurate field survey information over relatively large areas. However, this is no replacement for a robust knowledge of conditions on the ground.

The key to enhancing accuracy of, and confidence in, remote sensing image analysis of vegetation composition in the Congo Basin is to develop strategies to combine the work of field based conservation biologists with the image analysis expertise of climate change modellers. Therefore, any data you are able to collect on the ground, if recorded carefully and accurately, is as important as these other types of information.

Table B9.2: General guide to what features can be identified at various scales.

Scale	Features
1:500	Plant species identification, size of individual trees, uses of buildings, function of industries.
1:5,000	Volume of timber, wetland boundaries, outline minor tributaries, identify transport networks and property boundaries.
1:50,000	Outline areas of evergreen and deciduous trees, outline areas of forest associations, determine direction of flow of water, outline shorelines, local major transportation routes, measure areas of agricultural lands.
1:500,000	Regional vegetation and land-use classification.
1:5,000,000	Major river systems, continental vegetation zones, continental cloud cover

Table B9.3: Remote sensing systems - trade offs between spatial resolution and spatial coverage.

Sensor system	Platform	Map Scale	Spatial Resolution	Temporal Resolution	Spatial Coverage	Image Format
NOAA AVHRR	Satellite	>1:1,000,000	1.1 - 4km	12 hours	2,700km	Digital
Landsat MSS	Satellite	>1:500,000	79m	16 days	185km	Digital
Landsat TM	Satellite	>1:150,000	30m	16 days	185km	Digital
SPOT HRV	Satellite	>1:100,000	20m	5-26 days	60km	Digital
SPOT Panchromatic	Satellite	>1:50,000	10-100m	3-24 days	45-500km	Digital
Radarsat SAR	Satellite	>1:1,000,000	1.1 - 4km	12 hours	2,700km	Digital
Photography	Aircraft	>1:500	>0.10m	on demand	<20km	Hardcopy
Videography	Aircraft	>1:500	>0.25m	on demand	<10km	Digital
Digital photography	Aircraft	>1:500	>0.25m	on demand	<20km	Digital

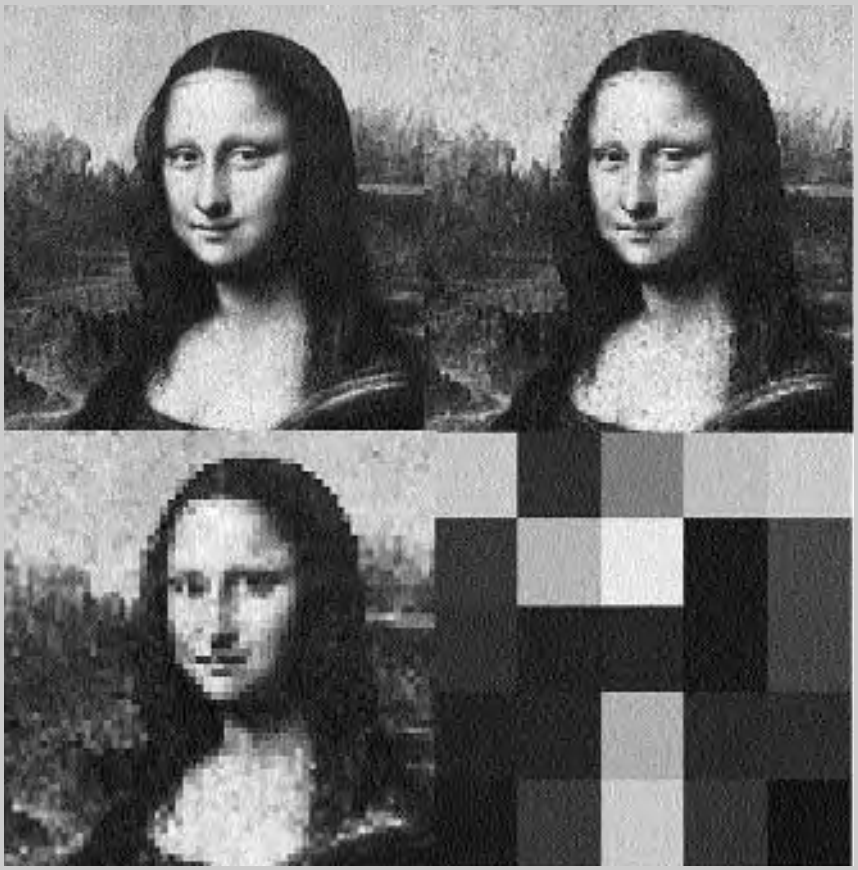


Figure B9.4 Schematic representation of the effect of different scales of resolution.

Phenology patterns

Phenology is the study of the temporal (time-related) patterns of flowering, fruiting and leaf production in trees and plants. The purpose of a phenological study is to better understand both the timing and the potential reproductive output (i.e. the quantity of flowers and fruit) of a species of tree or plant. This may be important from a botanical point of view (for example, one species may exhibit very different fruiting patterns in two different habitats). But it also can be important if you are interested in a species of tree that provides a significant food source to an animal, such as a hornbill, duiker or bat. Many of the large mammals and birds found in African rain forests are frugivores (fruit eaters). Therefore, seasonal patterns of fruit production are likely to have important implications for activity patterns such as ranging or reproduction, as well as limiting animal abundance and / or distribution.

Fruit fall phenology

Detailed studies of plant phenology are time consuming. However, "fruit fall" measures of seasonal fruiting patterns can be obtained without too much effort. The fruit fall method has the limitation that once fruit falls from the canopy it can be removed from the transect by terrestrial frugivores. Still, fruit fall data have been shown to give comparable results to in situ (on the tree) measures of fruit abundance in one African forest (Tutin & White, 1998).

Fruit fall phenology can be undertaken along 5-km botanical transects, or can be done simultaneously with a dung transect. Walk the transect on a fortnightly or monthly basis and count and identify all fresh ripe and unripe fruits fallen on a 1m strip. Remove all fruits which could conceivably remain intact until the next census. For each species collect a sample of ripe fruits and calculate the mean weight of a fruit (alternatively collect and weigh all fallen fruit). Count fresh fruit remains that have been fed upon by mammals or birds as well as entire fruits. Classify each fruit into a category (succulent, arillate, dehiscent, fleshy pods, wind dispersed, others). Figure 9.9 shows fruit fall data covering an annual cycle in the Lopé Reserve, Gabon.

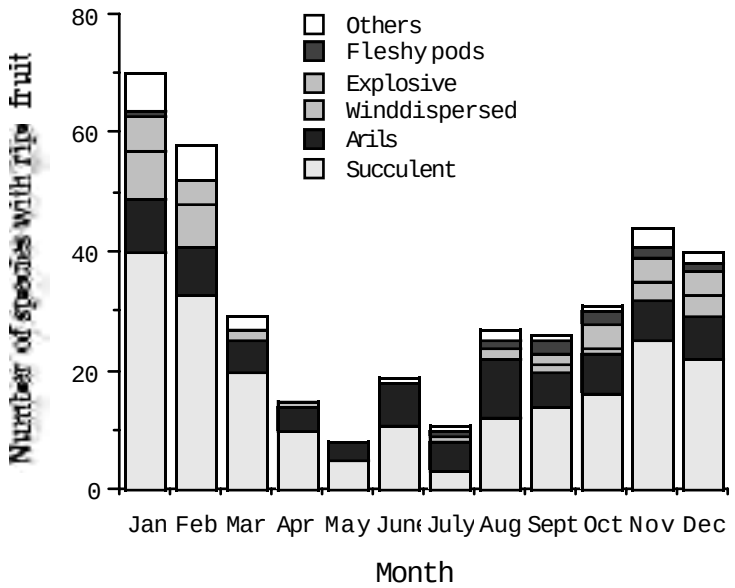


Figure 9.9 Summary data for five 5-km line transects monitored for fruit fall in the Lopé Reserve, Gabon. Fruits have been classified according to their physiology and dispersal mechanisms (from White 1994c).

Fruit fall phenologies undertaken simultaneously with censuses for animal densities can reveal relationships between fruit abundance and animal movements. For example, in Lopé, Gabon, this method was used to identify a limited forest type (covering c. 200 km^2) into which almost all the elephants in an area of 3000 km^2 migrate to feed on *Sacoglottis* fruit during the months of October and November (White, 1994a - Box 12d). Special efforts are now underway to protect this forest type from logging.

On the tree phenology

The fruit fall method measures fruits that have fallen to the ground. Other methods estimate quantities of fruit (and other plant parts) actually on trees and lianes. Such studies require more time than fruit-fall counts, but provide more information. If seasonal movements of animals are particularly marked and their understanding is important for management, such studies may be justified. However, bear in mind that fruiting, flowering and leafing are in constant flux and that you will have to take data at frequent intervals (generally 2 or 4 weeks) for a period of at least a year, to obtain meaningful data. This represents a major investment of time. However, if you think you will be able to undertake such a study the information generated is likely to be extremely informative. Bear in mind that there are often marked differences between years and that your results will be much more valuable if you are able to continue the study for a period of at least 2-3 years, and better still, 5-10 years.

What to record for each study plant:

Record the dbh of each plant at the beginning of the study. A list of characteristics is checked regularly for each individual studied and may include any or all of the following:

- new leaves, mature leaves, senescing leaves;
- flower buds, flowers in full bloom;
- unripe fruit in the tree, ripe fruit in the tree, unripe fruit on the ground, ripe fruit on the ground.

Locating study trees:

To begin a phenological study of trees first decide which species will be studied. Commonly studied tree species include those whose fruits are frequently eaten by particular animals of interest, commercially important species or those species that are found in high densities in the forest. Then find a minimum of ten reproductively-mature trees of each species of interest (larger samples are likely to give more representative results but are obviously more time consuming). If fruiting is of particular interest be sure to select female trees for dioecious species. Try to select trees in a "representative" (or "normal") setting (for example, make sure not all the trees are on the edge of large treefall gaps, or overhanging rivers, etc.) and that each canopy is clearly visible from at least one vantage point. It is best to locate these trees within easy viewing distance of a trail or transect. Each tree should be permanently marked. Making a map, or otherwise documenting the location of each tree, will help new observers find the study trees.

Estimating Quantities of Leaves, Fruits and Flowers:

Estimating the quantity of leaves, flowers or fruits found on each tree is difficult. A simple way to record estimated quantities is to use a three-point scale (see Table 9.3). A three-point scale is recommended if a number of observers are to be involved in data collection, particularly if observers will be people with relatively little research experience, since the simpler the system the more likely you are to obtain consistent results.

Table 9.3. Example of a three-point scale used for recording phenological information (Terese and John Hart, personal communication).

Characteristic	0	1	2
Mature leaves	none to very few	up to 75% full compliment	> 75% full compliment
New leaves	none	few or limited to 1-2 branches	numerous over most of canopy
Flowers	none	few or limited to 1-2 branches	abundant over most of canopy
Fruit in the tree	none	few or limited to 1-2 branches	abundant over most of canopy
Fruit on the ground	none	few	many and under most of crown

The advantage of a simple system such as this one is that data quality can be maintained even if many different observers are involved in data collection. The disadvantage is that the resolution is poor, and subtle changes may be overlooked. If the study of phenology patterns is deemed important and well trained staff are available to collect the data, a more detailed methodology can be used. In Lopé, Caroline Tutin and her colleagues have been using the following system during a 10 year study of phenology patterns:

Each plant is checked monthly. Its canopy is observed through binoculars from a known point on a regularly used trail (actually a mapped elephant path). The canopy is visually divided into four quarters. A score of "4" represents a situation where an individual has the maximum amount of fruit, flowers, or leaves, possible over its entire canopy. If it has a maximum crop over only half the canopy, or a crop equivalent to half the maximum possible over all its canopy, its score is "2". Hence scores of "1", "2", "3" represent a crop which is a quarter, half and three-quarters the maximum possible crop. To allow for the fact that plants sometimes only have a few fruits or senescing leaves, scores increase in a series as follows: 0, -, 0.5, 1.0, 1.5, 2.0, 2.5, 3.0, 3.5, 4.0. The score - is awarded when something is present, but represents less than 1/8 of the maximum possible crop. Each additional 0.5 represents an additional 1/8 of the maximum possible crop. The presence of insect damage, damage caused by animals during feeding, branch loss etc. are also noted. The scores for new, mature and senescing leaves (often detected due to a colour change) collectively add up to 4 (the whole canopy) unless some proportion of the canopy is bare. Hence, an *Irvingia grandifolia* tree given a score of NL (new leaves) 1.5; ML (mature leaves) 0.5; SL (senescing leaves) 1, would correspond to a canopy which only possessed 3/4 the normal leaf score. The equivalent of 1/4 of the canopy was covered by red senescing leaves, the score for pale new leaves was between 1/4 and 1/2 of the total canopy, whilst just a few normal mature leaves were observed. Figure 9.10 shows part of a filled out phenology data sheet.

This system may be overly detailed for your needs, but researchers with a long experience of taking phenology data find the added information useful.

Figure 9.10: Part of a phenology sheet from Caroline Tutin's study.

[1]Spp. code#	[2]Tag code#	Species	NL	ML	SL	FL	FRI	FRR	Comments
51	1	*Dacryodes buettneri*		4			1.5		
80	5	*Xylopia quintasii*		3.5		1.5	1		
59	4	*Duboscia macrocarpa*	3	0.5	0.5	4			
133	1	*Treculia africana*	1	3					insect damage
98	2	*Enantia chlorantha*	0.5	3.5					
71	1	*Irvingia grandifolia*		3.5					
98	1	*Enantia chlorantha*		3.5					
59	3	*Duboscia macrocarpa*		1	2.5				
43	4	*Diospyros polystemon*		3	0.5			1	leaves yellowing

[1] = species code number in computer data base; [2] =number of that individual; NL=new leaves; ML=mature leaves; SL=senescing leaves; FL=flowers; FRI=immature fruit; FRR=ripe fruit.

Pandanus candelabrum

CHAPTER 10
Information from animal tracks and trail

Richard J. Parnell

Everything that moves leaves some trace of its passing. This fact binds the whole of the animal kingdom and, while most of these traces are imperceptible or last no more than an instant, others, when observed and skillfully interpreted, can prove invaluable aids to those involved in wildlife management, conservation and research. This observation and interpretation is the 'art' of tracking.

Animals are generally hard to see, particularly in forest habitats. They use their highly tuned senses, stealth and sometimes camouflage to avoid predation or disturbance, or to maximise their chances of success in hunting. Many are also nocturnal (active at night) or crepuscular (active at dawn and dusk). In fact, most forest animals are hidden from us, and even some relatively large animals go unseen for most, if not all of their lives. However, by studying the tracks, trails and signs left by these animals, we can often learn a great deal about their lives and increase our chances of actually seeing them.

It is probably fair to say that the majority of people working with wildlife do not spend enough time studying tracks. Field work has often to be carried out within quite strict time limits, with workers frequently having insufficient time to familiarise themselves fully with the tracks and trail of their study animals before going on to try and answer detailed and complex questions about the animals' ecology or status. To get around this problem it is commonplace to seek the help of local hunters or guides whose knowledge of the fauna of their home areas is often considerable, having been passed down through many generations. Bear in mind that the knowledge of local people has not developed out of academic interest, but from the very real and immediate demands of survival. Of course, local guides are not always available and where available, it should be noted that in many developing nations, the gradual shift of bias from a village-based subsistence economy to a more urban, wage-based one has created a gap between the generations; the tracking skills of the grandchildren are often a pale shadow of those of their grandparents. Where guides are available, a knowledge of tracking will enable you to better judge the accuracy of the interpretations offered. If no skilled and proven guides are available, a basic knowledge of tracks and trail will help you obtain a better understanding of the animals you encounter and thus a more profound perspective when considering detailed questions.

This chapter aims to illustrate: a) how tracking can assist the field researcher or wildlife manager; b) where and how to find tracks; and c) how to record tracks and trail. At the end of the chapter, you will find a selection of animal track drawings made in the Lopé Reserve, Gabon.

Terminology

Throughout the text, the word 'track' refers to a single foot or hand print. A 'trail' normally refers to evidence of an animal's passing that can to some extent be followed, such as a series of tracks spread over some distance (also called a Track Pattern), perhaps including feeding remains, resting sites and feces. Some animals advertise their presence with territorial markings that may include sprays of urine, scenting with glandular secretions, feces, or the scoring of tree trunks and stumps. For the sake of simplicity, these markings, along with other signs such as shed hair and skin will all be included under the heading 'trail'. The term 'substrate' is used frequently throughout the text and refers to the surface on which a track is imprinted.

How tracks and trail can be used to identify and study animals

Species Identification

The first thing a track can tell us is the identity of its maker. Even a poorly defined track should indicate whether the animal was an ungulate (tracks with 'cleaves' or hooves) or of a family such as the canidae (dogs) or felidae (cats) which show tracks with soft pads. In order to be more specific in your identifications the tracks of each species have to be learnt ; there are no short cuts for this. Confusion can arise between closely related species, such as many of the medium-sized duikers, where few differences are discernible in the tracks. Even with perfectly made tracks, variation in foot shape and size between members of the same species could mask the true identity of an individual. Reference guides to animal tracks do exist, but for Africa, they can be disappointing. There are exceptions, although these usually confine themselves to areas of the continent where safari tourism is commonplace (see the booklist and reviews at the end of this chapter). Certainly for the equatorial rain forest there is very little help at hand. The best way of learning to identify tracks is to study them yourself. Whenever you see an animal, try to get a look at its tracks. If necessary, sit and wait in an area you know to be frequented by the animal. Positively identify the animal, then as soon as it has moved away, go in search of its tracks. The question of how best to record the tracks is dealt with later in the text, but for now, suffice it to say that you will have to record tracks from numerous individuals of each species to ensure that you know what a 'typical', entire track looks like.

With a positive identification one can confirm that a species is present in a particular area. The geographical distribution of some species has been shown to be much wider than previously thought, due to the discovery of their tracks in new areas (e.g., the spotted-necked otter and the serval in South Africa, Liebenberg, 1990; and spotted hyena in the rain forest of northern Congo, R.J. Parnell, pers. obs.). Of course, observation is also required to confirm the track interpretation if there is any ambiguity and especially if the animal has not previously been sighted in the area. Ground type, weathering and the movements of the animals themselves can all deform a track, making it appear larger, smaller, or even a completely different shape, so do not draw hasty conclusions from one set of prints alone.

In addition to revealing species identity, careful study and measurement of tracks can sometimes help establish, or confirm, the identity of individuals of certain species within a population. Several researchers have had success identifying individual leopards and other large cats from their tracks. In this way they have been able to map territories and establish minimum population densities with very few observations of the animals themselves.

Age Class and Sex

A common problem for protected area managers in African forests is the need to evaluate the impact of poaching on animal populations, or to assess whether hunting pressure is adversely affecting the population structure of target species. One way to do this is to monitor the age structure of the population. Highly stressed populations may stop reproducing, resulting in the absence of infant and juvenile tracks; or hunters may selectively target large males of certain species, such as elephants, resulting in an absence of large tracks. By monitoring such trends over time, or by comparing proportions of tracks of different sizes in areas where hunting pressure differs, you may be able to identify and hence react to management problems.

By studying tracks the age of an animal can be estimated with some accuracy up to adulthood, but this can only be done when the dimensions and form of an adult track are known, and when the degree of sexual dimorphism (one sex being larger than the other) is understood. For instance, an adult male, or 'silverback' gorilla is considerably larger than an adult female gorilla and this difference in size is accurately portrayed in their tracks.

Problems can arise though in the case of sub-adult males, as for some time as they develop towards full maturity, their tracks can look identical to those of adult females. Many mammals show sexual dimorphism to some degree, so age-classing tracks is seldom straightforward. Among infant and juvenile animal tracks, things are usually easier, but once again, it will take some time to amass a reference collection of tracks with confirmed ages. A pitfall to be aware of is when the adult tracks of one species closely resemble the juvenile tracks of a larger species. In mammals, be particularly wary of this if you find a juvenile track but cannot find any sign of the mother's tracks close by. Examples might include the similarity between the tracks of a young bushbuck, (*Tragelaphus scriptus*), and those of an adult red duiker, or perhaps an infant red duiker track being taken for that of an adult blue duiker, (*Cephalophus monticola*). Never make any judgment on the age class of a track without first being sure that you have correctly identified the species in question. Take note of the habitat type you are in and any other relevant factors to judge the likelihood of a particular species being present.

We have noted that the size difference between males and females can confuse our judgment of age-class but can assist us in judging the sex class of a track. Where sexual dimorphism is absent or very slight, there are other clues that can assist us in putting a sex-class to a track or stretch of trail. When an ungulate urinates, the position of the wet patch on the ground relative to the animal's tracks can indicate its sex. Consider the animal's anatomy; with a male the urine splash should land between its front and hind tracks, beneath its body, whereas with a female, the splash will be positioned between or just behind the rear tracks. Depending on the species concerned, it may be that in adult males, a broad chest will place the front tracks wider apart than the hind tracks whereas in the female, the broader pelvis may produce the opposite effect.

Sex and age classes can occasionally be determined through the examination of an animal's feces, although a good degree of sexual dimorphism will be necessary to determine the former. By carefully measuring the diameter of all feces collected from observed individuals, it is possible to build up a chart of dimensions that roughly conform to age classes. Always take diameter measurements at the widest point of the sample. Once again, certain variable factors can alter the size of the feces and these must be taken into account before any interpretation can be made. The most important of these is diet. The feces of herbivorous animals will often reflect the nature and quality of the plant material available. Rich new growth is likely to be better digested than woody old growth, and will produce softer fecal material that is less dependable for age-classing. With omnivorous animals the relative proportions of fibre to flesh can alter the feces. A chimpanzee that has been feeding primarily on leaves and stems will usually produce drier, firmer dung than one which has been eating meat. Fruit pulp will often affect the consistency of fecal material in a similar manner to flesh, rendering it softer. The fruit of *Irvingia gabonensis* (bush mango) is a prime example, making the feces of chimpanzees, gorillas and elephants particularly slimy and wet. On the whole, the firmer the dung is, the more representative the diameter measurements will be. Other factors that can affect the consistency and therefore the measurements of dung include the health and mental state of an animal. Animals with gut infections or high intestinal parasite loads may pass particularly wet feces, and with some species, diuretic dung may be passed if the animal receives a severe fright (such as coming into contact with humans). Dried dung is likely to have shrunk, so only record data from fresh, moist samples, unless the majority of your samples will be dry, such as in hot, open areas. Clearly, variability in the material concerned can create many pitfalls and in smaller animals the differences in size between samples may be too small to be of any practical value. However, at its best, using fecal measurements can, for instance, indicate the age class of all the individuals in a group of gorillas, from the 1-3cm diameter feces of an infant, through juvenile and adolescent/sub-adult classes to adult female or silverback (Figure 9.1). Bear in mind though, that it is not possible to determine the actual age of an animal using this method once it has reached adulthood.

Figure 10.1: Diameter / age classes of the dung of western lowland gorillas

Feeding Trail

Where the tracks of one species are very similar to those of another, or where tracks are unclear, deformed or even absent, trail (including feeding remains, feces, resting sites and territorial markings) can often provide additional clues to an animal's identity. Many animals leave characteristic feeding remains behind them as they move around the forest. By carefully recording details of these remains, it is possible to establish baseline data as for tracks, which will enable you to learn about species even if they are rarely sighted. Furthermore, if carefully recorded feeding trail can contribute valuable information about the diet of little known forest animals, leading to a better understanding of their ecology and management needs. For example, much of what we know about the feeding ecology of western lowland gorillas (*Gorilla g. gorilla*), and forest elephants (*Loxodonta africana cyclotis*), is the result of information meticulously noted by researchers along feeding trails (see Box 10a) and by sorting through fecal remains (see Boxes 10b and 10c).

Although the diets of many animals in the same habitat may overlap, there are often certain items that are only consumed by one member of a given taxonomic group. For instance, the processed seed pods of the forest tree *Tetrapleura tetraptera* found scattered beneath the canopy, in Gabon, might indicate that black colobus monkeys (*Colobus satanas*), had been in the area, rather than members of the Cercopithecinae family (guenons). Likewise, ape trail including scraps of the fruit *Pentadesma butyracea* might indicate that it was gorillas (*Gorilla g. gorilla*), and not chimpanzees (*Pan t. troglodytes*), that had been present. Be aware, however, that food preferences can differ between different populations of the same species without apparent reason (*Tetrapleura tetraptera* fruit is frequently eaten by gorillas in Congo, but is rare or completely absent in the diets of gorillas from central Gabon), so take care if using a food list from somewhere else to identify feeding remains in your own area.

Even when the same item is eaten by more than one species, the manner in which it is processed can sometimes identify the consumer. For instance, the soft bases of rolled (not yet unfurled) *Megaphrynium* leaves are eaten by both mandrills (*Mandrillus sphinx*), and apes. While their feeding trail looks very similar, a closer inspection will often show the tendency of mandrills to leave an untidy shredded edge to the discarded section of the leaf while both chimpanzees and gorillas will nip off the edible base cleanly.

In a similar way, the remains of grass feeding can tell us several things about the consumers. A clean, knife-like cut to a leaf blade, often at an acute angle, may indicate rodent feeding; a serrated horizontal edge is more likely to indicate feeding by a ruminant, where the blade has been gripped between the lower teeth and the hard upper palate and torn off. A leaf blade that shows signs of having been chewed usually indicates the activity of a carnivore such as a member of the cat family which may have been eating the vegetation to obtain certain vitamins or swallowing grass in order to trigger a vomiting response that helps rid the stomach of fur balls and may also have a function in controlling parasites. Grazing trail can be further delimited by looking at the height of the feeding above the ground.

The manner in which the same prey species is processed by different predators may also differ sufficiently to allow identification down to at least the level of order. For instance, when the carcass of a bird is found, an examination of any feathers removed may indicate the nature of the predator. Birds of prey usually pluck out the feathers of their kill, sometimes damaging the quill with a small hole, whereas the smaller carnivorous mammals tend to chew the feather off low down, leaving the quill rough and frayed.

Box 10a GORILLA AND CHIMPANZEE FEEDING REMAINS

Western lowland gorillas eat a lot more fruit than mountain gorillas and therefore spend a lot of time moving through the forest in search of trees and lianes with ripe fruit. They leave a lot less feeding trail than mountain gorillas but in the forests of central Africa gorillas (and chimpanzees) feed on the pith and young leaves of several species of Marantaceae, leaving characteristic feeding sign. A good tracker can often follow gorilla feeding trail and locate nest sites or even find the animals themselves. Chimpanzee feeding sign tends to be sparse and often consists of little piles of plant remains, but be sure to look carefully for dung or knuckle prints before deciding which ape left the trails you find. Figure B10.1 illustrates some typical Marantaceae feeding trail.

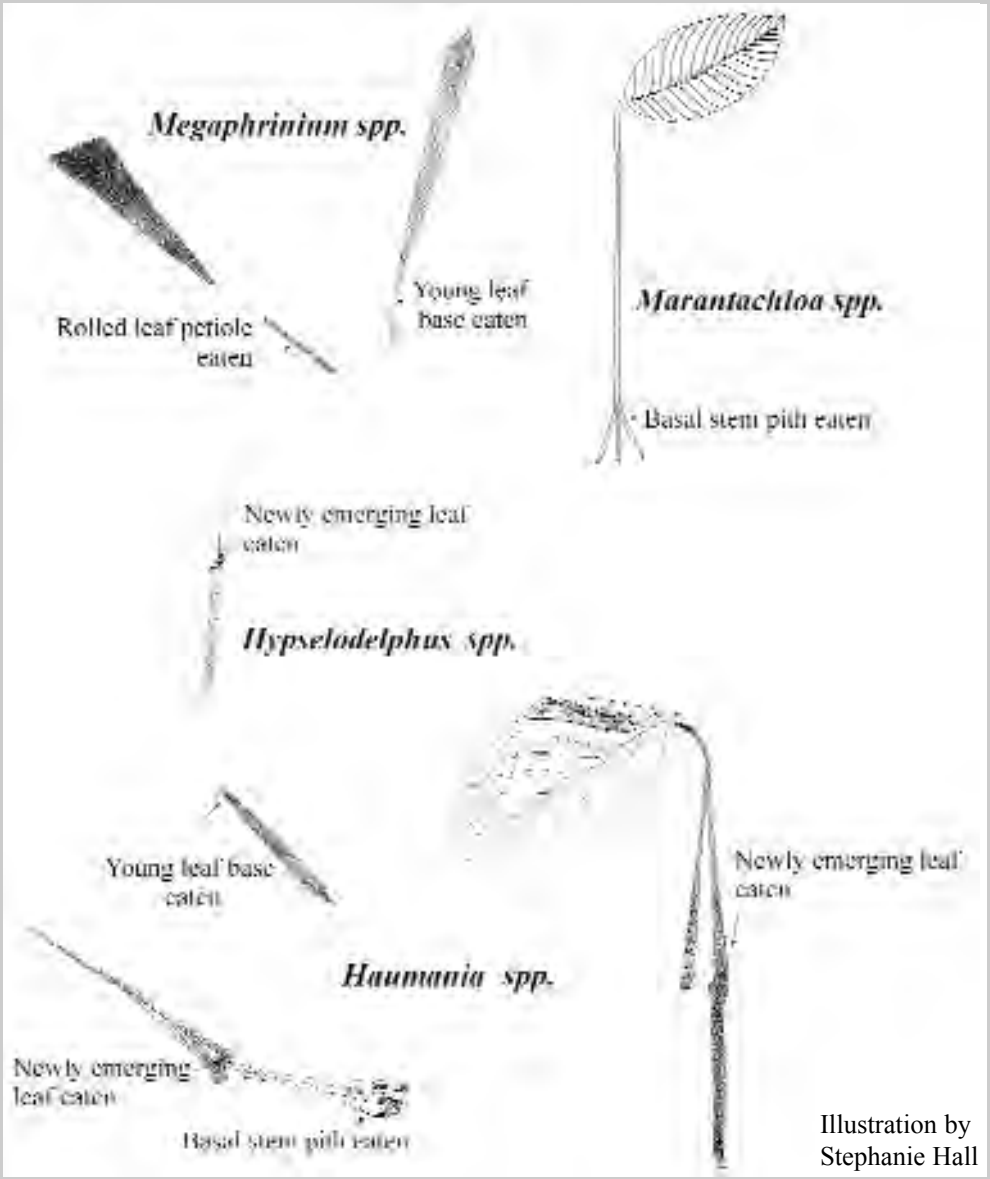


Illustration by Stephanie Hall

Fecal Remains

Feeding behaviour can also be used to identify species through the examination of fecal material; and conversely, correctly identified dung can be extremely useful for the study of the diet of little known species. Furthermore, density of dung is often used as an index of animal abundance or to assess habitat preference (see chapter 12), and dung size classes can be used in the same way as tracks to monitor changes in the age structure of populations.

Fecal shape and size are also good indicators of species. Both may alter slightly due to changes in diet, but some general characteristics usually remain. For instance, the feces of the brush-tailed porcupine (*Atherurus africanus*), are blunt tipped cylinders, 1.5cm x 0.5cm; the feces of the bushpig (*Potamochoerus porcus*), consist of many round or heart shaped pieces of about 2cm diameter that may be scattered loosely or compacted into a cylinder, 4cm in diameter; the feces of the gorilla are comprised of long tri-lobed sections, whereas chimpanzee feces show more compacted round sections. The location in which a fecal sample is found can also help to indicate the species involved. Otters habitually defecate on raised surfaces close to water, such as rocks, in order to advertise their presence. A similar behaviour is exhibited by the African civet (*Viverra civetta*), which frequently sites its dung middens (latrine pits) on flat rocks or areas of bare earth. The leopard (*Panthera pardus*), will often deposit its feces on roads or paths, where they are most likely to be encountered by other leopards, thus serving as visual and olfactory markers.

When studying diet, or searching for clues as to the identity of the depositor, hard dry feces can be soaked in water to soften them, or gently teased apart as they are. With damper material, a wire sieve and running water can be used to wash the matrix from the sample, exposing the larger parts, such as seeds, undigested fruit, leaf and fibre fragments, insect parts, bone, meat and hair (see Boxes 10b and 10c).

Other Signs

Other signs that can assist the field worker in identifying animals include resting and sleeping sites, territorial markings and shed fur or skin. These three categories alone cover a vast array of examples. They not only allow identification of species, but can also sometimes be used for density estimations, or to assess seasonality of reproduction. Consider the different nesting styles adopted by birds, from the narrow tunnels excavated in river cliffs by the shining-blue kingfisher (*Alcedo quadribrachys*), through the pendulous woven baskets of the blue-billed malimbe (*Malimbus nitens*), to the huge roofed structures built in marshland trees by the hammerkop (*Scopus umbretta*). Observation of these nests can indicate the presence of a bird species in a protected area, even if it has not been seen in the flesh; can be used to define breeding seasons when hunting or tourism should be more strictly controlled; or, if observations are made in a systematic way, they can be used as a measure of population size or trends over a number of years.

African ape densities are most often estimated using transects enabling researchers to count nests (see Chapter 13). The tree nests of chimpanzees and gorillas can be easily confused, although chimpanzees only nest in trees (with the exception of occasional day nests on the ground), while gorillas nest either in trees and on the ground. The height of tree nests can be of some help in distinguishing the species, with chimpanzees often nesting higher than gorillas, but the most reliable clues come from a search for ground nests and an investigation of the shape of any feces found.

Other sleeping sites such as burrows and holes may indicate the homes of animals such as the ratel (*Mellivora capensis*), aardvark (*Orycteropus afer*) and others, including snakes and rodents (be aware that confusion can arise when animals adopt the burrows of other species).

BOX 10b OTHER INFORMATION FROM ANIMAL REMAINS: DUNG.

For any frustrated field-mammologists hoping to record observation data on elusive study animals, dung may represent the most readily available and easily collected source of information upon which they may fall back in despair. In fact, a good deal of ecological information can be deduced from analysis of fecal deposits (see "Facts from Feces" - Putman, 1984). Even the animals themselves often are able to glean information from dung. For example, African civets use latrine pits to delineate the boundaries of their territories and in some species the males are able to assess female reproductive status from fresh dung.

What information might you consider obtaining from dung?

- if you know the rate of deposition and the length of time dung remains visible, you can use dung counts to estimate population density or to investigate differential use of habitat types (see Chapter 13).

- larger animals deposit larger and heavier dung piles. For example, the age class of gorillas can sometimes be assessed from the diameter of their dung (Figure 10.1). Even if the relationship between dung diameter and age / size class is not known, information on changes over time of size class distribution of dung in a population can be used to track shifts in age structure, which might result for example, from poaching of large individuals. Such data could be used to show the effects of poaching on elephants, since large males with large tusks are selected by poachers (so the largest diameter classes would become less frequent) and stress within the population results in decreased birth rate (and a corresponding decrease in the smallest diameter classes). However, note that diameter of dung can also vary with season and diet. For ungulates in general there is a good correlation between mean dry weight of 20 randomly selected fecal pellets and body weight (e.g., Coe & Carr, 1982), suggesting that this method could be used to monitor changes in body weight of duikers in hunted areas if the species can be reliably designated.

- dung is often a useful indicator of dietary species composition. For example, gorillas and elephants swallow the seeds of most fruits they eat, and these pass through their digestive systems undamaged and are deposited intact in the dung.

If you take time to search through their dung you can compile a list of fruit species eaten. For example, gorilla and chimpanzee dung is collected in several sites in Africa for analysis. In Lopé (Tutin & Fernandez, 1993) fresh dung of both species is collected in plastic bags on which researchers note: the date; location; species; age class (if possible - see Figure 10.1); and corresponding nest number when dung is found in a nest site. Once a week this information is transferred to a note book, the dung samples are weighed individually and the dung is sieved through a 1mm mesh in a stream near to the field station. Once all of the matrix has been washed out, the quantity of fibre, leaves and seeds are quantified. Seeds □5mm long are counted. Smaller seeds are scored on a four-point abundance scale: abundant, common, few, rare. Non-fruit foods (fibre, green-leaf fragments, insects, meat remains, soil, beeswax etc.) are scored on the same abundance scale relative to the total fecal mass. Various analyses are possible from this information. For example, the number of species of fruits per fecal sample or the total number of fruit species per month can be calculated, to illustrate differences in fruit consumption through the year, or the mean foliage score (summing fibre and green leaf fragment scores; abundant = 4, common = 3, few = 2, rare = 1, giving a maximum of 4 for each quantity) can be calculated to analyse changes in fibre intake in response to fruit scarcity (Figure B10.2).

If you keep a running record of the number of dung piles searched and the number of species of seeds recorded you can plot a species / sample size curve (follow the steps outlined in Chapter 9 for species-area curves, using each dung deposit as your sample or 'area' - see Figure B10.3).

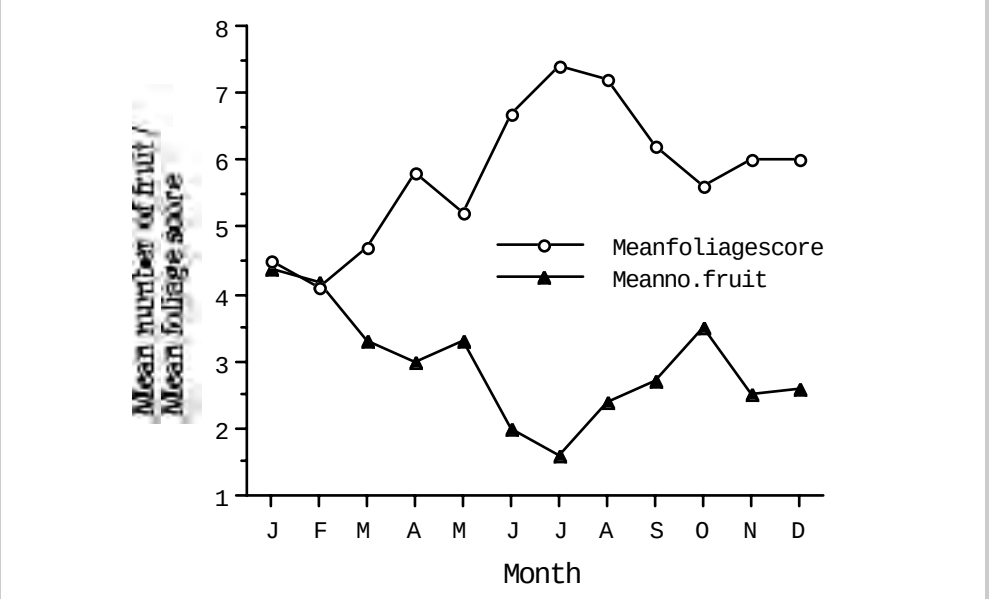


Figure B10.2. Seasonal changes in the diet of gorillas as shown by data collected over four consecutive years (1987-1990) in the Lopé Reserve, Gabon.

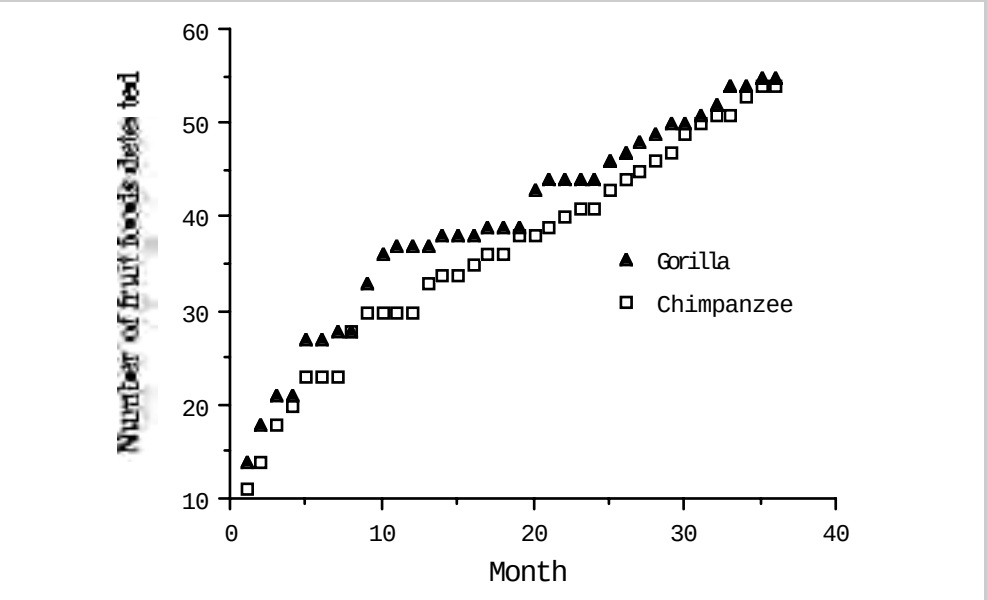


Figure B10.3 A plot of the number of fruit foods detected in ape dung during a three year period in the Lopé Reserve. In order to make a meaningful comparison between the two species, five dung samples were selected at random from a larger (but unequal) sample for each month for each species. Both curves continue to rise showing that the sample size is not yet adequate to fully describe the fruit foods in the diets of these two apes (from Tutin et al., 1991).

Scat analysis is widely used to assess the diet of carnivores such as leopards or civets, since they contain indigestible parts such as bone, hair, finger nails and hooves, teeth, fish scales and insect remains etc. which often can be identified. Similarly, the pellets of raptors, such as crowned hawk eagle, contain fragments of their prey and can easily be collected under their nests. In both cases your field museum will enable you to compare fragments of bone and hair to known specimens for identification, and can also be used to store the dung for future reference. Analysis of the diet of ungulates or identification of leaf fragments in ape or elephant dung is more problematic, but microscopic analysis of plant fragments can be used to identify species if you have a good reference of known plants for your study area (such as your field herbarium - see Chapter 8).

Obviously you are not going to start collecting dung samples indiscriminately in the hope that one day somebody will analyse them. Think carefully about how dung analysis might be of use in answering your research questions. For example, if there are complaints from people who claim that leopards living in a protected area are killing their livestock you might make a collection of leopard scats and assess what proportion contain sheep or goat hair. If, after careful consideration you decide to make a dung collection and analyse its content, treat each deposit as you would a dead animal or plant collection, carefully documenting collector, date, species, location, weight etc. If planning such a study, a good place to start is by reading Rory Putman's article 'Facts from feces' published in the Mammal Review (1984, pp. 79-97).

Box 10c CARNIVORE SCAT ANALYSIS

by Justina Ray

Unlike many of the other medium- and large-sized mammals of the African rain forest, carnivores are rarely seen and weeks or months can pass between sightings. Yet these elusive creatures form rich communities, with as many as 12 species living in an area. Because of their furtive habits, research relies heavily on indirect observations. Luckily, sign, particularly in the form of scats, can be very abundant, especially in areas which constitute travel routes for these species, such as along trails or roads. One can learn a great deal about carnivores from collecting scats and analysing their contents. Scats also yield a tremendous amount of information about the prey base of an area, which can include everything from fruits and insects to fish, birds, herpetofauna and mammals.

Examples of what can be learned about carnivores from their scats:

- diets of species in the community, seasonal changes in the diets, and resource partitioning among species;
- relative abundances of carnivore species in the study area;
- information on carnivore movements and habitat use.

Examples of what scats can reveal about the prey base:

- information on the presence and relative abundance of prey species living in the area that are difficult to trap. For example, a sample of 1,000 carnivore scats in the Dzanga-Sangha Reserve, C.A.R., revealed 16 species of shrews, whereas only two species were captured during extensive small mammal trapping (Ray & Hutterer, 1996);
- data on seasonal abundance patterns of prey groups;

- population monitoring of key prey species. For example, one can measure resource overlap over time and in different areas between leopards and human hunters, who utilise some of the same prey species, such as duikers and porcupines.

A note of caution about identifying scats

The identity of a carnivore that deposited a scat can never be taken for granted. Most forest carnivores are similar in body size, hence the diameters of their scats are generally alike. It is also a common mistake for people to identify a scat on the basis of its contents; for example, believing that the scats of cats (such as leopard or golden cat) contain only bones and hair. Biochemical identification has shown that mongooses and genets are equally likely to eat mammalian prey, even prey as large as duikers. The only carnivore whose scats are easy to distinguish based on size is the leopard (□2.5 cm in diameter), the sole large carnivore found in the majority of the African rain forest. Otherwise, identification must rely on field sign (tracks, sightings), microscopic examination of hairs ingested while grooming, and molecular techniques. DNA analysis and thin-layer chromatography are being increasingly utilised. A small piece of fresh scat should always be saved in a vial filled with ethanol. However, even if the scat-maker cannot be identified, the information gathered from scat analysis can be useful.

Scat collection protocol

When the scat is found in the field, store it in a plastic bag or a big leaf and record the following information:

- location of collection
- collection site description (e.g., on or beside a trail, in or beside a puddle, forest type, on log, etc.)
- accompanying carnivore sign if any (scrape, tracks, view of animal)
- maximum and minimum diameter
- estimated age of scat
- whether or not it was collected from a latrine (communal defecation spot).

Be careful never to touch the scat with your bare hands. When back in camp, assign the scat a number and prepare a card (or log book entry) with the above information and the date. Later you can use the back of the card to record information on scat contents. Place a small piece of fresh scat into a small vial filled with ethanol. Using a flour sieve (available at most markets) wash the scat. Make sure to wear plastic or rubber gloves. Some older scats may have to soak for a while to soften. After the bile has been removed from the scat, place the remaining contents (hair, bone, insect parts, seeds, etc.) on a tray, or a square of flat strong cardboard, labelled with the scat number and place it in a drying oven. Once dried, store the scat contents in a labelled plastic bag until ready for analysis.

Scat analysis

Identification of prey remains can be accomplished through examination of hair, teeth, nails, hooves, scales, and/or quills of mammals, feathers of birds, vertebrae of herpetofauna or fish, exoskeletons of insects or seeds of fruits. Reference collections are essential and visits to museums are helpful. To quantify carnivore diets from scat contents, use presence / absence, frequency of occurrence (percentage of scats in which a particular food item occurs), mean relative volume of a food item in scats, mean number of individuals in each scat, and / or relative biomass ingested. Differential digestibility of different prey types sometimes makes the use of correction factors necessary in the calculation of biomass (see Reynolds & Aebischer, 1991).

Information from animal tracks and trail

We have already noted how some animals use their feces as territorial markers. Other signs in this category that may be useful to the tracker include the scratch/scent marks left by felids such as leopards. Leopard scratch marks are mostly found 2-3m above the ground on thick inclined trunks. These appear to serve the mixed purposes of removing old claw sheaths, possibly also leaving a scent from glands in the foot, and acting as a visual advertisement to reinforce the urine spraying that usually accompanies scratch marking. Bushpigs also habitually scratch mark vegetation using their tusks. These marks can mostly be found at a height of 10 to 50cm on small saplings seldom more than 5cm in diameter. Even where the saplings are not damaged by the tusks, the bark, polished by the pig's cheek and the distinctive smell both point to the same conclusion. It is, of course, impossible to make a reference collection of smells, but the tracker should never neglect this important tool in animal identification. The tangy smell of a bushpig rub, the extraordinary odour of a frightened silverback gorilla or the fruity pineapple scent of Peter's duiker (*Cephalophus callipygus*), can all burn their way into the memory in a way that pages of notes and track diagrams never will.

A final word should be made of signs that include actual parts of the animal concerned. The sloughed skin of a snake can be used to positively identify the species from which it came. Rather less obvious are shed hair and fur. These can be useful aids, for instance, if a gorilla ground nest is suspected but no vegetative construction is evident. Gorilla hairs are hard to see, but they are usually there, accompanying nests, feeding sites and trail, and will become easier to spot with practice. The resting sites of other animals such as duikers can be identified by areas of flattened leaf litter in secluded spots such as between the buttress roots of large forest trees, and can be confirmed by the location of their hairs on the site. It is ironic that the most easily overlooked of trail items can now tell us more about the animal than we could learn even from direct observation of the animal itself. Through the process of DNA extraction and amplification, a single hair can now tell us not only an animal's sex and species but also its parentage (see Box 13l).

To conclude this section, it must be stressed that while a single track, trail item or other factor may point incontrovertibly to a particular species, many field situations will require the tracker to study an area in some detail, using a combination of several different factors to build up a picture of the animal.

Other Information from Tracks and Trail

A track indicates which parts of the foot support an animal's weight. This may seem a fairly obvious statement, but it refers to an important system of classification for all animals. The original, primitive mammal foot showed four basic features that can be used as the basis for this classification system. Figure 10.2 shows a track with five claws on one foot. Beneath each claw is a small pad known as the *digital pad*. Below this is the area corresponding to the palm of the human hand. It is comprised of up to four pads, known as *interdigital pads*, that may be separate or fused together. Finally, at the rear of the foot come one or two *proximal pads*, that correspond to the human heel. Many animals still place the whole of their foot, that is, the digital, interdigital and proximal pads, on the ground and are said to be Plantigrade. These include the porcupine, the ratel, and the otter.

Other animals have evolved to walk up on their toes. These, including cats, dogs and mongooses, show only the digital and interdigital pads in their tracks, and are know as Digitigrade. Finally, some animals have moved up onto the very tips of just two of their toes. These toes have become ‘cleaves’ or the two halves of a hoof. Animals with cloven hooves, such as antelope, buffalo, and pig, are known as Unguligrade. Horse-like animals are in a class of their own, where only the vastly enlarged third toe supports the animal's weight.

This evolution away from the plantigrade foot is associated with the need for greater speed. It should be borne in mind that some animals (e.g., otters) may leave plantigrade style tracks when walking on a soft substrate, but show a more digitigrade configuration on harder surfaces. Also note that some animals, including the apes, walk plantigrade on their hind limbs (feet flat on the ground), but digitigrade on their front limbs (knuckle walking).

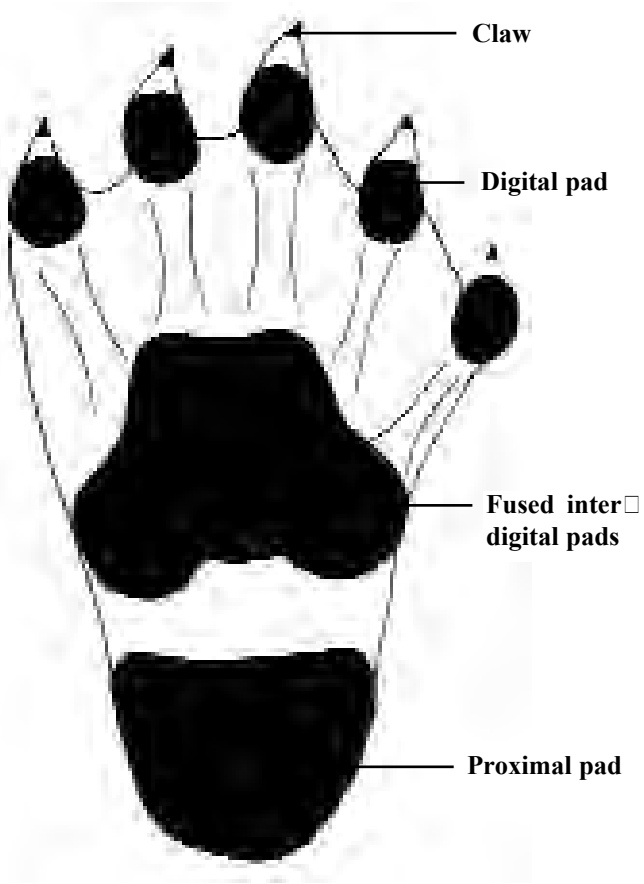


Figure 10.2 A typical plantigrade foot

The direction in which an animal was travelling can be learnt by studying both tracks and trail. Even the most basic knowledge of foot morphology will enable a tracker to tell in which direction an animal was facing when a track was made, enabling him or her to decide whether to follow the trail back towards a feeding site or night resting area, or to pursue the animal itself. In cloven hoofed animals, the narrowest section of the track is normally made by the front of the animal's foot. Exceptions exist where the two halves of the foot are splayed wide apart by a soft substrate or where the foot is normally splayed to provide a large surface area to avoid sinking in soft mud, such as is the case with the sitatunga (*Tragelaphus spekii*). With most other mammals it is necessary merely to locate the prints made by the animal's digits. These are always at the front of the track, thus indicating the direction of travel.

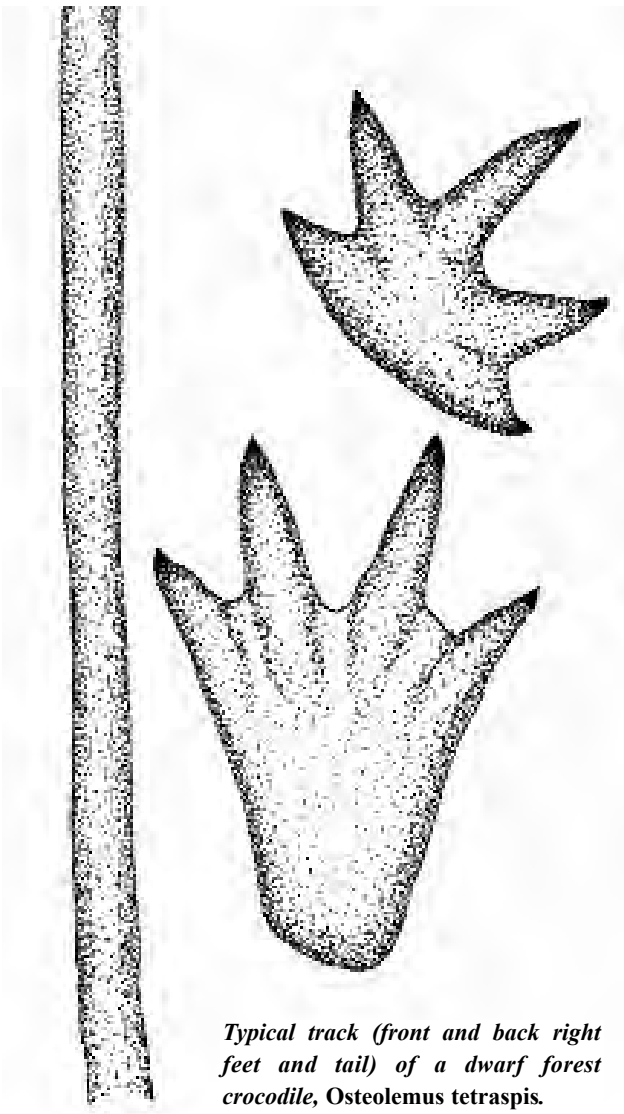
Direction of travel can sometimes be betrayed by an animal's feces. Gorillas and chimpanzees both have a habit of using logs that have fallen across the path they are following as toilet seats. Thus, if you are following the trail of an ape and find its feces on the ground in front of you, just before a log, you can be fairly sure that you are travelling in the same direction as the animal. With apes and some other mammals, it can be useful to study the shape of the feces themselves. Unless the animal turned around to defecate, the thinnest, pinched off end of the sample will point in the direction taken by the animal immediately after having left the sample.

Other direction indicators include vegetation bent over (in the direction of travel) by an animal as it passes through an area. The freshness of any trail items found in one location relative to those found previously can also be used to indicate the direction of a trail.

Information from animal tracks and trail

A highly skilled tracker may be able to estimate the weight of an animal by the depth of its tracks in any given substrate. Hunters, who have the opportunity to check their estimates when an animal is finally killed, are often more skilled at this than other trackers. However, even for a hunter, this technique can prove quite inaccurate due to the multitude of variable factors that may effect the depth of any given track, and its use in scientific survey should, in normal circumstances, be limited to giving trackers a rough guide to the condition of the animal they are following. For instance, if among the tracks of a small group of adult buffalo, a particularly shallow set of adult tracks is found, this may well point to the fact that one of their number is sick or old, and has as a consequence, become unusually thin. Tracks can also indicate other physical conditions such as foot or limb deformities that may show themselves either in strangely shaped tracks or through limping or dragging gait patterns.

Do not forget that tracks are not only made with an animal's feet. In soft substrates, both tail and body can leave prints of their own, telling us more about the activity of the animal in question (see below).



***Typical track (front and back right feet and tail) of a dwarf forest crocodile,* Osteolemus tetraspis.**

Finding tracks and trail

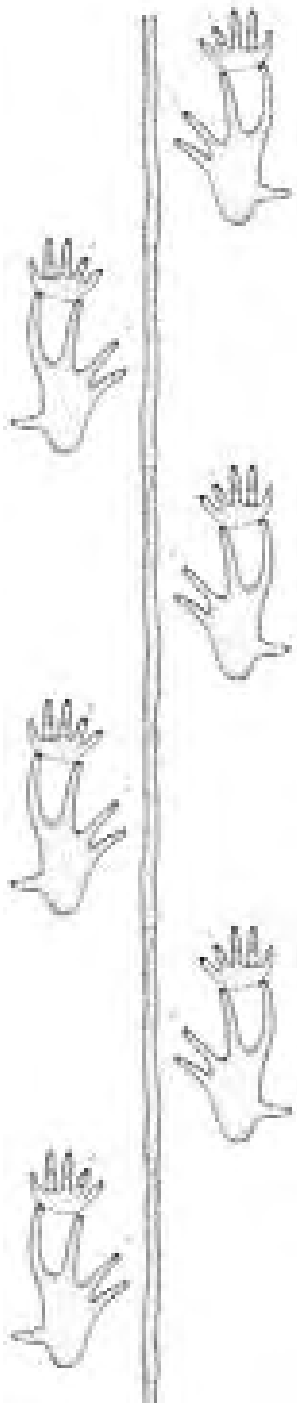
To the uninitiated, the experienced tracker can obtain astonishing results by their ability to decipher meaning from barely visible signs. However, while there may be times when the tracker uses a sense of intuition resulting from a heightened empathy and identification with the animal being followed, most time is spent reading and interpreting a text that is visible to anyone willing to learn its language. This is the language of disturbance. By becoming acutely aware of the natural order of their surroundings, trackers are able to spot where things have been moved, taken away, altered or added to. These disturbances can be obvious, like an elephant footprint in soft mud, or incredibly subtle, like a glimpse of the silvery underside of a leaf contrasting with its undisturbed neighbours, or an upturned pebble showing its darker underside. By becoming totally familiar with the appearance of untrodden leaf litter, the tracker is able to see where an animal has passed, merely in the slight increase in shadow cast by leaves that have been lifted by the animal's feet. An ability to see subtle disturbances takes considerable time to develop. Even seeing more obvious trail can prove difficult at first, so knowing where and how to look for animal sign can save a great deal of frustration and time.

The best way to start is by getting used to seeing and recognising tracks in ideal conditions. This usually means spending time searching stream, lake and river banks for tracks that have been preserved in sand or soft mud. These areas, as well as offering an ideal substrate for preserving tracks, are favourites of a wide variety of animals. They attract animals who come to drink, to feed on vegetation only found near water, to hunt, to use the waterways as routes through otherwise dense vegetation, or to deliberately use thick water's edge vegetation for cover. Be aware that soft, dry sand will not hold any track with precision, and sand that is too waterlogged will tend to produce tracks too deep to show detail and prone to collapse or dissolving. After moist clay, however, firm, damp sand is one of the best substrates for the recording of tracks, and beaches can be veritable treasure troves to the apprentice tracker. For the same reasons, sandy roads are good places to begin searching for tracks, allowing the tracker to cover large areas swiftly. Acting as routeways for a wide range of animals, roads are particularly good places to study extended sets of tracks or 'track patterns'.

Forests are, for the most part, difficult places in which to study tracks. The large quantities of leaf litter usually present act as a barrier between the foot and the soil, and although leaves can sometimes be pulled away to reveal the track impressions beneath, unless the animal was heavy or the leaf litter particularly thin, the likelihood of these tracks being of good quality is slim. It is therefore necessary, at least for the less than expert tracker, to narrow the search. Many terrestrial mammals are creatures of habit, and follow the same routes through their territories over and over again. These routes may delimit an animal's territory, or lead from a burrow or resting site to a favourite feeding area or dung midden. Over time, these routes, or game trails, often become well worn and quite visible. An extreme example of this is the network of wide paths that crisscross any forest where elephants are present. Elephants create and use these paths for general travel, commonly following ridges or valley bottoms, or to get to specific fruiting trees. Many of these paths may be hundreds of years old, and have come to serve not only the elephants, but many other members of the forest community. Such paths are excellent places to search for tracks and trail alike.

The chances of picking up tracks can be increased if the substrate on the path itself is improved. This may be as simple as sweeping away a patch of leaf litter to expose a clean area of soil or sand, or may involve carrying in a few sack-fulls of sand to be spread over an area of the path, creating a 'track trap'. These can be used to good effect on paths and transects, or can be placed beneath fruiting trees so as to monitor terrestrial members of a frugivore community. They should be checked very regularly for tracks, and once notes have been made, should be smoothed over again, ready for the next passerby. In some cases, baiting the 'traps' may increase the chances of getting a result. In one study the feeding behaviour of the yellow-backed duiker (*Cephalophus sylvicultor*), has been monitored by baiting sand traps with *Diospyros mannii* fruit (the tracks indicated whether or not the duiker had carried the fruit away to consume under cover). In another study a researcher attempted to map territories of African civets (*Viverra civetta*), by plotting the location of feces. They used several mixtures of sardines and peanut butter containing tiny discs of differently coloured plastic, placed at different feeding stations. Sand traps were used to check whether the baits had in fact been taken by civets or by another species. Civet dung was then recovered from latrine pits and checked for presence of plastic of different colours.

Whenever animal remains are found, the use of sand traps can help to build a picture of the carrion eating community. Whatever the situation, your sand traps should be made in a manner so as to arouse the least possible suspicion, whether it be visual or olfactory. Animals notice disturbance too).



Typical track of a nile monitor lizard,* Varanus niloticus ornatus *front and back feet and tail tracks are shown

Once you have found a likely location for tracks, or have put out sand traps, you face the biggest hurdle of all .... seeing them! Just because the tracks are there does not guarantee that you will spot them. Just take a walk in a place you thought you knew in the company of a practiced ornithologist, and you will see the area come alive with birds, bird calls, nests and other sign. Tracking, like bird-watching, demands that the participant adopt a certain way of looking at the landscape. Most of this way of seeing can only come with a good deal of practice, but there are guidelines that can be of immediate benefit.

The first and most important is ... **SLOW DOWN!** It sounds so simple, and yet to some people, this may be the hardest task of all. Walk slowly and quietly; pause frequently; do not keep your eyes glued on the path at your feet, but let your gaze wander through the whole scene. If you are too intent on spotting a single track, you are very likely to walk right over a game trail or pass by a feeding area without ever realising it. Look for trampled pathways, tunnels in the vegetation, disturbed leaf litter, feces and feeding remains. Once you have located a trail, whether it be from tracks or other signs, get down on your knees and begin familiarising yourself with the finer details. If there are tracks, feel them gently with your fingertips and try to estimate their age. If you are able to find the next track on, break off a dead twig or grass stem so it measures the distance between the two tracks. Then, if subsequent tracks are not obvious, lay the measured stem out from the front of the last track you can find, and search the area at the forward tip of the stem. Be careful not to step on the tracks you are following, as it may be necessary to go back and check a section of the trail if things become confused. If possible, try to keep the tracks you are following between yourself and the greatest source of light (usually the sun) as this will enhance the shadows within them, making them easier to spot. Tracks are often easiest to follow if you a travelling in the same direction as the animal that made them. For this reason, if you are searching for tracks, turn round frequently and check the area you have just covered from this new perspective. Bent over vegetation is also much easier to spot if you are facing in the right direction. Try lowering the angle between your eyes and the tracks you are following, even to the point of getting level with the ground. In this way, even slight depressions will stand out clearly. By getting close to the ground you are much more likely to find easily overlooked sign such as hair. By approximating the same height as your animal, you will be more likely to follow routes that it may have taken round small obstructions that would not even appear to you if you were standing. Remember that the vast majority of animals are considerably shorter than you and therefore see the world from a perspective quite different to that of a human. Try to imagine yourself being the animal you are following: What are your habits? What do you eat? Is there food nearby? Water? Shelter? Predators? Can you pick easily through dense vegetation or do you prefer to follow a path? Successful tracking hinges on the ability to combine the interpretation of physical clues with an imagination capable of, to some extent, getting 'inside' the mind of the animal in question.

Recording Tracks and Trail

A few people are being brought up in societies where tracking is necessary to their survival. They gain a detailed knowledge of tracks from watching and being taught by the members of their community who are already skilled trackers. For the rest of us, the best way to learn is to spend as much time as possible looking at tracks, and keeping records of everything we find. By sketching and measuring tracks, it is possible to learn what a 'typical' adult track looks like for any given species; how the tracks of different age classes differ from these and how to distinguish the tracks of closely related species. By keeping careful records of the conditions in which a track is found, the effects of different substrates on track morphology can be studied.

Trackers Field Kit

A trainee tracker should never go anywhere without a small kit for recording tracks. The basics of such a kit are as follows:

1) *Notebook* - This should be as large as can be sensibly accommodated by your field bag or pocket. Squared paper is often preferred as it can make the drawing of tracks easier.
2) *Pencils* - Choose one of the softer grades of pencil if you can.
3) *Pencil sharpener and Eraser*
4) *Ruler* - If you can get hold of one, a metal ruler is best as the numbers and gradations do not wear off as easily as those on plastic rulers. Do not carry anything too long as it will invariably get lost or broken. An ample length for your ruler is 20cm (8"). A plastic ruler can easily be cut down to this size so as to fit easily into a pocket.
5) *Tape Measure* - For sheer ease of use, choose a metal, recoiling carpenters tape. A length of 2 or 3 meters is sufficient.
6) *String* - A length of string, about 5m long should be carried to assist you in recording track patterns (see under the section entitled 'Gait').

What to Record

To keep your own track measurements consistent and comparable with other reference collections, it is important that you record certain features in a recognised way. Figure 10.3 shows which measurements to take for two types of track; one from an animal with soft pads (the front track of an African civet), and the other a typical antelope track (the front track of a bushbuck). The general rules shown here can be applied to most of the tracks you are likely to find, although with some you will simply have to use common sense in deciding what to record. Remember that it is best to have more measurements than are necessary than to find yourself lacking one vital dimension.

Measure the width of a track (1) from one side of the track to the other at the widest point. Measure the length of the track (2) (on its own first, not including the claws or dew claws), then measure any other component parts such as the distance from the rear of the hooves to the rear of the dew claws, or from the front of the pads to the tip of the longest claw. When measuring tracks made by hooves, always record the distance between the front points of the cleaves (3) as the degree of splay can occasionally be useful in telling certain species apart, as well as telling front and hind tracks apart from each other.

As a fast moving foot enters a soft substrate, it is liable to leave a hole larger than the foot itself, so make sure that your measurements are from the edges of the track itself, which should be imprinted at the bottom of the entry hole.

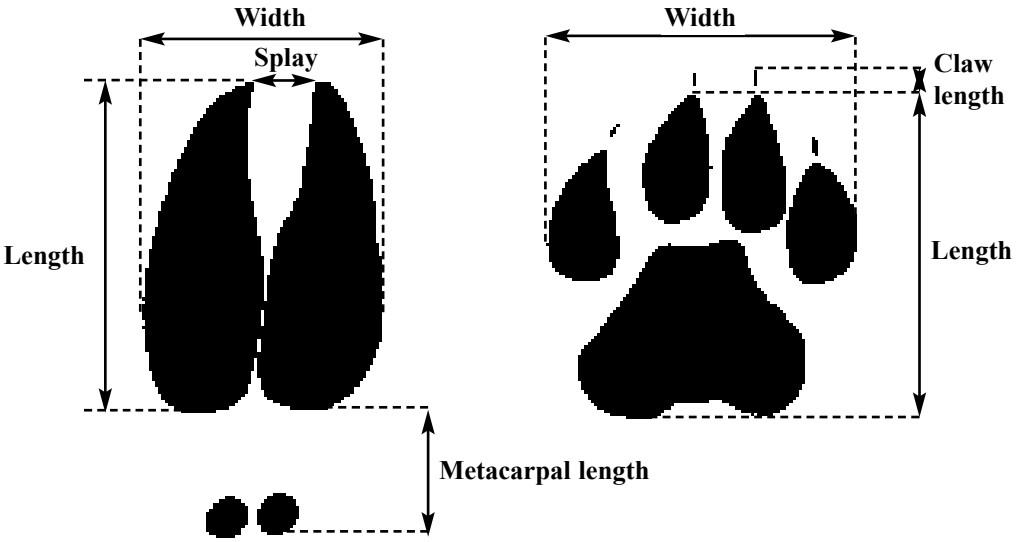


Figure 10.3 Measurements to take from two common types of track.

Recording data

Although your initial measurements will be recorded in your notebook, ideally you should transfer your data to a checksheet as soon as possible. In this form it is less likely to be lost or destroyed. If you use a separate checksheet for each species, you will be able to compare variables such as different age-classes, front and hind tracks and gaits at a glance. The following list suggests information that you should record whenever you measure a track, and also represents the headings you should use on your checksheet (see Figure 10.4).

Date
Location
Substrate - Is the track in earth or sand? Is the ground wet, damp or dry? Soft, firm, hard or crumbling? Is the ground flat or inclined? Be as precise as possible.
Age-class of animal - Unless you can be any more precise, use terms such as adult, sub-adult, juvenile or infant. If you know the sex of the animal then you can record it under this heading on your checksheet as well.
Front or Hind - (F/H) Specify, if possible, whether it is the animal's front or hind print you are measuring.
Left or Right (L/R) Is the track from the animal's left or right side?
Gait - Was the animal walking, trotting, loping, galloping or jumping etc.? (see below)
Track Width - (T. Width)
Track length - (T. Length)
Step, Stride, Straddle, Bound, Distance to Median Line
Other - used for any miscellaneous notes you may wish to add - this category should include, where appropriate, the distance between the front points of two cleaves (splay), the distance to the dew claws, or the distance between the toes and the claws.

To fit more information onto the checksheet, abbreviations can be used, such as Fl. = Flat; I. = Inclined; F. = Front; Ad. = Adult; L. = Left; etc.

These titles and any others appropriate to your research should be laid out across the top of your checksheet. The following is an example of how the initial entries on a checksheet for the leopard might look. By looking at the lower two entries, we see that front and hind measurements were taken for the same animal. We can note the general felid trait of the front paws being larger than the hind and that the difference is most noticeable in terms of width.

Another useful record you might consider keeping is a map showing the location of the tracks you find. If possible, keep one map for each species and mark the location of each set of tracks with a number. This number should be entered alongside the corresponding details on your checksheet. In this way, it may be possible to build up a picture of the distribution and habits of species within your area and possibly even to map the range of individuals.

Figure 10.4 Part of a data checksheet for *Panthera pardus*

Track Drawings

There is no better way of getting to know an animal's tracks than by drawing them. The process demands that a track be studied at length and all its subtleties appreciated. Besides the training in observation that track drawing provides, the practice enables the tracker to build up a reference collection of tracks from as many species as possible. When looking at the tracks of a new species, you will rarely find the ‘perfect’ track straight away, but by gradually upgrading your track drawings as you find more representative examples for a particular species, you will be made aware of the many ways in which variable factors such as substrate and gait can influence a track. An example of how substrate can effect the morphology of a track is given in Figure 10.5. Although the tracks shown were all made by the same forest buffalo, the track to the left was made on firm soil, that in the middle on soft mud, and the track to the right on a hard, compacted surface. The tracks are drawn at a third of their normal size.

Ideally your own track drawings should be life-size. To ensure accuracy, always begin by measuring the track at its widest and longest points and mark these out on the page before beginning to draw in the details of the track itself. In many animals, the front and hind tracks differ in size and occasionally in form, so make sure you get drawings of both. Also, do not limit your collection to adult tracks, but whenever you get the chance, record those of juveniles and infants as well. Your checksheet of track measurements will eventually begin to reveal to you what the ‘average’ track dimensions are for each species. But do not be tempted to use mathematically derived dimensions to create your definitive drawing. Simply choose one of your field drawings that best represents the trends shown by your measurements, and use this. Once you are happy with a drawing in your field notebook, make a neat copy of it into a larger book to be kept out of the field and used for your eventual reference collection. Always remember to record details of the substrate in which the track was found.

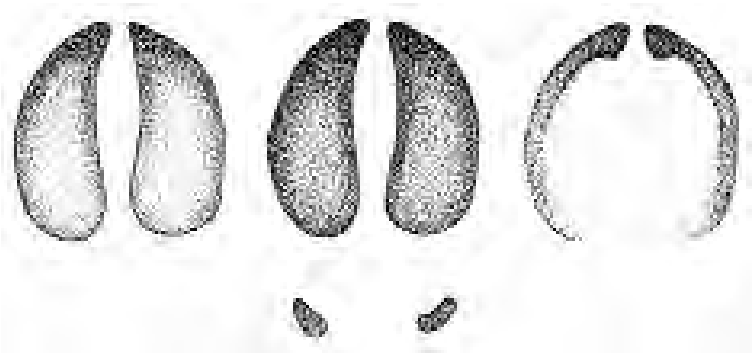


Figure 10.5 Three tracks made by the same forest buffalo but in different substrates (one third life size).

Plaster Casts

It is possible to make exact copies of tracks using a moulding material such as plaster of Paris or paraffin wax. Paraffin wax requires heating, so plaster of Paris is more often the material preferred for use in the field. Once a characteristic track has been found, a collar should be placed around it. This can be made from a strip of cardboard, stapled or joined with a paper-clip to form a rectangular frame. Make sure the frame is slightly larger than the track so that no damage is done when it is pushed gently into the ground. In the absence of a frame, a little sand or soil can be heaped around the track to serve the same purpose. Mix sufficient plaster to fill the frame to a depth of 1 to 2cms depending on the area to be covered. Once the plaster is a thin creamy consistency, it can be poured slowly over the track. Allow at least 20 minutes for the plaster to set, then gently lift the cast and frame out together. Any soil sticking to the cast can be washed off under running water. You should now have what amounts to a model of the bottom of your animal's foot. Either label and store it as it is, or make a further plaster cast so as to recreate the track itself. If choosing the latter, remember to lightly smear the first cast with Vaseline, so that the two casts do not stick together as the fresh plaster hardens.

Casts made with plaster of Paris can be extremely fragile, so consider reinforcing them by laying pieces of wire into the plaster as it sets.

Photography

A camera can be used to good effect in the recording of tracks, but without the appropriate lenses, the results may be distant and unsatisfactory. Try to take track photos shortly after sunrise or just a little before sunset, when the shadows needed to define the track's outline are most likely to be present. Remember to keep the track between you and the sun. Unless you are recording a track pattern over a considerable distance, try to take all your photo’s from directly above. In this way, you will avoid distorting the length of the image being taken. Always place a ruler or other familiar object next to the track to provide a sense of scale, though you should not see this as a substitute for measuring the track before taking the picture. As a general rule, a detailed track drawing can contain more information than most photographs. However, photography comes into its own in the recording of feeding remains, feces and other trail features that either decompose rapidly or, like burrows and scratch marks on trees, are immovable. Remember to include a ruler or other object to give the picture scale.

Recording and preserving feeding remains and other trail items

By carefully recording feeding remains, it is sometimes possible not only to say what an animal eats, but also how important that food is in its diet and when it is most likely to feed on a particular food. This can be hard or impossible with small mammals as much of their feeding sign may be too small to recognise, much less count. Problems can also arise when the animal in question ingests all of the food item, leaving behind no sign at all. However, at its best, with ape trail for instance, the processed and discarded food items can tell us a great deal. As you follow a feeding trail, note down each item as you find it. Record not only the species, but also which part of the item has been eaten. In order to get the most from this information, you will have to take feeding data in this way over a long period of time, covering each season and including samples from all the habitat types used. Your aim is to be able to say that in a particular habitat type, at a particular time of year, the diet of your chosen species is made up of, say 60% one food, 20% a second, 10% a third and 10% a fourth. If you get the chance to follow feeding trail over a considerable distance, your data can be enhanced by recording not only the quantity of different foods eaten, but also the order in which they are selected. Could the choice of some food items be linked to the time of day, due to the habitat type in which an animal chooses to make its bed for the night? Could it be that after feeding extensively on one food type, such as fruit, an animal will seek out a different type of food, such as green leaf material, as its next meal? Is feeding on one food species or type more likely to precede a period of rest than another? Rather than cluttering up your notebook, you might try to design a checksheet for recording this type of data.

If you are able to follow an animal's trail for the whole of its active day (or night), you have the opportunity to plot all its activities against time. When does it drink, feed, mark its territory, travel or rest? Note where on the trail the animal defecates and try to build up a picture of how often it does so during the course of a day, as this is of particular interest for animal censusing (see Chapter 13).

While photography may be the best way to record most trail items (with the exception of tracks), a collection of certain items can be made for reference or display purposes. Most fleshy food items will shrink and change colour considerably when dried so should be preserved in alcohol, but drier woody materials will dry and store well (see Chapter 8). The methods used by squirrels, mice and other rodents to get through the hard protective shell of a nut often differs. When you are able to observe this type of feeding by an animal, the discarded shells should be labelled and stored in the same way as botanical vouchers. Tooth or claw marks on seed pods or bark can often be indicative of the species responsible, and may be collectable. A collection of mammal hair can be useful for determining prey species through the examination of carnivore feces. Whenever you find a dead animal, pull tufts of hairs from several areas of the body (try to get a sample of all the different types of hair from different parts of the body on each specimen) and dry them in the sun or an air conditioned room (but see Box 11b). The same can be done with feathers. Some can be mounted on card for reference whilst the bulk of the sample should be carefully labelled and stored.

Carnivore and omnivore feces, as well as hair, may contain some of the hard parts of the prey they have been eating. These might include both whole and fragmented bones, scales, teeth, hooves, or insect parts such as legs, heads and wing cases. These can all be teased out of the sample once it has been thoroughly soaked in water or alcohol. Given this resource, it may be wise to collect whatever relevant animal parts you can whenever you find an identifiable animal dead (see Chapter 11).

Ageing Tracks and Trail

Tracks

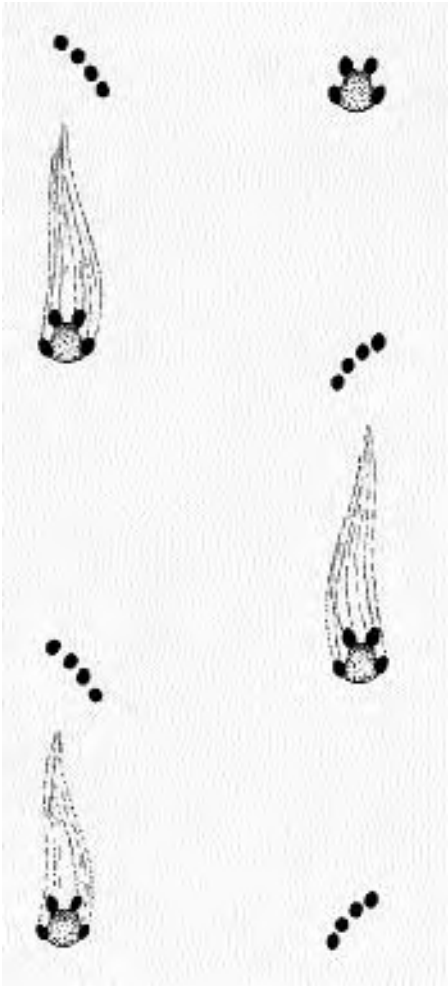
It is possible with a combination of close observation and experience to tell when a particular set of tracks was made. This is, however, one of the hardest tasks the tracker faces. From the moment a track is made, it begins to deteriorate and lose definition, and will eventually disappear. It is primarily by judging the level of deterioration that the tracker aims to date the track. Once again though, a whole host of factors may conspire to confuse the issue. At the extremes, a track made in soft, dry sand on a windy day might last for less than a minute, whereas one left in firm clay in a drying atmosphere might last for many months. Within these extremes lie an array of ground surfaces and atmospheric conditions, the combinations of which can offer endless challenges to the tracker. The only way to speed up a learning process that would otherwise take many years is to make your own tracks using a blunt stick or a finger (a finger will approximate the track of a soft padded animal whereas a stick will give more of the crisp edges made by an ungulate hoof). Choose several different soil types, and on a swept area of each one, make a series of marks at varying depths. Study these carefully and make notes on the crispness of the edges and any other features that are visible. On dry sand, you will probably want to return and repeat the process after no more than an hour as noticeable changes may well have taken place. On more stable soils, the interval can be longer. When you return to the tracks, look closely to determine how, if at all, they have changed: Have the edges begun to wear down due to wind action? Have any cracks appeared around the track? Have any of the fine details disappeared? Make notes on whatever you find, then make a new set of tracks along side the first set. Come back and repeat the process after the same time interval throughout the day, constantly comparing the freshest tracks with the various stages of ageing in the others. Following the first day, it may only be necessary to check the tracks you have made once a day. Continue to do this for as long as you can; preferably until the first tracks are no longer distinguishable. Always keep an accurate record of the weather conditions for the whole period of the trial and note the effects of precipitation, flooding, drying or freezing on the tracks as they happen. The more notes you take on the condition of the soil, air temperature, hours of direct sunlight, amount of rainfall and so on, the more valuable your results will be.

Another clue that can assist the tracker in ageing tracks is the presence and quantity of debris that accumulates in a track (ask yourself when the wind was last strong enough to blow leaves off branches, or take note of the rate at which leaves fall without wind). As an animal walks, grass stalks or other vegetation may be pushed into the track: Look for this as a sign of fresh tracks as the vegetation will frequently recover and pull out of the track with time. Also take note of whether the tracks have been crossed by those of a more recent passerby.

Trail

An ability to age feces allows the field worker to gauge when an animal passed by with considerable precision. In dry, open areas, this method is less precise as the sample is often either baked hard, preserving its shape much longer than had it remained moist, or it swiftly crumbles away to dust. However, in a more humid forest environment, the gradual deterioration of a sample offers many clues to its age, particularly within the first day. Once again, it will be necessary to study the the ageing process in several different sites, such as on rock, at the edge of a marsh, on a sandy path and so on. The effects of sunlight, shade, rain and others climatic influences will also need to be taken into account.

Note the consistency of the sample and, if it is possible to do this without breaking it up, give some idea of its contents (a fecal sample full of seeds usually break apart more quickly than one containing mostly fibre). If you are tracking a particular animal and come across a really fresh fecal sample, the temptation to collect it for analysis is considerable, but it is only by occasionally 'sacrificing' these treasures that the field worker can progress towards reaping the greater riches that accurate tracking offers. Feces can change within minutes of having been deposited. The first change is usually in colour as the outer surface is exposed to the air. This colour change will progress steadily from the surface down through towards the centre, and precision ageing can be achieved by sectioning a sample and seeing how deep the colour change has penetrated. With herbivores and many omnivores, the change is often from a pale green to dark brown, while with some carnivores, although the initial change may be similar, the high calcium content of the feces will eventually turn them white. Note also that the smell of a fecal sample changes over time and can be useful in helping you to age trail.



Track pattern of a hinged tortoise,* Kinixys erosa, *showing how it drags its feet as it moves forwards.

The action of dung beetles should also be noted: How quickly do they begin to excavate holes beneath the sample? How long is it before their tunnelling affects the structure of the dung? Another insect aid is the fly. These often arrive within seconds of the dung being passed, but how long is it before maggots can be found inside the sample?

If a sample is unaffected by the initial onslaught by insects, the next notable sign of change may well be fungal. Note how long it takes for fungus to show and spread on a sample. The time taken for the complete breakdown of a dung sample will depend on all these factors in conjunction with the initial consistency of the dung, the substrate and the climatic conditions.

Urine splashing on vegetation or being caught on leaf litter can be a further aid, which although usually drying swiftly, stains the leaves it touches. As these stains are easily washed away, it is possible for the tracker to gauge whether any accompanying trail was made before or after rain. This can be a particularly useful aid in ageing the tree nests of apes, beneath which a great deal of urine is often splashed.

Partially eaten and discarded trail items can also be of assistance in ageing trail. Plant parts such as cut stems, leaves and fruit are used in a similar manner to feces in that the tracker pays particular attention first to colour changes on the exposed or damaged surface, then monitors these while also noting the gradual drying or rotting processes that follow. Many of the same variable factors that influence the ageing of feces are also relevant to

For instance, rain or high humidity will alter the rate of colour change and slow the process of wilting. Once again short cuts in an otherwise very lengthy learning process can be made by artificially creating trail items and following their deterioration in differing microclimates. It should be noted that plants bearing close resemblance to one another and sharing identical habitats will often age in completely different ways. *Megaphrynium gabonense* and *Haumania liebrechtsiana* are both herbs in the family Marantaceae that grow in dense tangles of under-storey growth in the forests of central Africa. Gorillas, chimpanzees and mandrills all eat the tender bases of the unfurled leaves of these two plants, leaving trail that appears identical when fresh. However, while the white damaged area of *Megaphrynium* will turn yellow after 2-3 days (depending on humidity), the same area of the *Haumania* leaf turns black after just 10-15 minutes exposure to the air. Partially eaten animal remains can also be aged, with a potentially high degree of accuracy, through the study of the blowfly eggs, larvae and pupae that often accompany them.

Measuring Tracks and Track Patterns

All the measuring techniques to be described below rely on the tracker being able to tell right from left tracks and front tracks from rear. This is not always easy though there are some general guidelines that may help.

Right or Left?

Most mammals, with the exception of those with hooves, have five digits, although with some, the first digit (corresponding to the human thumb or big toe) may either be greatly reduced, so as not to normally show in the track, or be absent altogether. Use your own hand as a guide; If the track being considered shows five digits, then the smallest, which often lies at a different angle from the others, is the first digit, corresponding to your thumb. If this appears on the left side of the track, then the track has been made by one of the animal's right feet. After the first digit (or thumb) the next shortest is the fifth, corresponding to the human little finger. This knowledge now gives us a guide to those tracks that only show four digits, such as those made by members of the Cat family. In the absence of a first digit, the shortest is the fifth. If it is found on the left side of a four digit track, then the track in question was made by one of the animal's left feet. Animals with cloven hoofs can prove more troublesome. The two halves of an antelope's hoof are digits three and four. Digits two and five are the dew claws, with the first digit being absent. Close examination may sometimes show that the two sides of the hoof are not completely symmetrical. In this case it is often the fourth, or outer digit that is the longer of the two. Damage or distortion in the hoof form can be responsible for this, but it is often the case that the longer side is the fourth digit, which is always the outer of the two halves. So, if the track you find shows the left side slightly longer than the right, it is likely that the track has been made by one of the animal's left feet. This method is far from foolproof however, and should only be considered if the track's position relative to its neighbours is not conclusive.

Front or Hind?

The best way of telling front from hind feet in animals with cloven hooves is to measure the distance between the front tips of the cleaves. Front feet typically show a greater degree of splay than the hind. For example, where the inter-cleave distance for the front track of an adult bushbuck would be c. 1.4cm, the same measurement for the hind foot gives only c. 0.5cm. For animals with pads, other methods must be used. There is often a difference in overall size which may be useful, as is the case with the leopard, who’s front tracks are broader and generally more massive than the hind. This trait is shared by most members of the Felidae family and can also be seen in the African civet. The inclusion of the first digit is often a help when identifying front and hind tracks in the mongoose as the ‘thumb’, where it shows at all, will be best defined in the front track.

In animals with proximal or 'heel' pads, the hind tracks are often longer than the front, as seen in examples such as the otters, pangolins and the cane rat, *Thryonomys swinderianus*. However, there are exceptions to this and while certain guidelines can assist the tracker, he or she should study tracks of each species until front, hind, left and right become second nature.

Track patterns

Learning to read track patterns is one of the more difficult skills a tracker has to master. Whenever possible, try to observe animals in motion and follow the order in which their feet are set down in various gaits. Do not think that you have to wait for brief glimpses of your study animal to improve your abilities. A great deal can be learnt from watching cattle, donkeys, goats, cats and dogs. Although domestic animals may exhibit some styles not typically seen in the wild, learning the gaits of just these five species will give you a key to those of a large range of wild animals. When looking at a particular species, try to get into a position where you can observe the animal in motion and then study the tracks it has made. Many animals, for instance, will habitually cross roads or rivers at the same places. If you have often found the tracks of a species that interests you passing the same spot, try erecting a hide and being there the next time it passes.

If you have access to a video camera, try filming animals in motion then playing the tape back in slow motion, noting the order in which the feet are set down. One last aid in what is essentially a problem of visualisation, is to make a model animal. This does not need to be at all elaborate, but can simply involve two lengths of flexible wire laid side by side and tied together one third then two thirds of the way along the length. The middle third now comes to represent the animal's body with the other thirds being bent down and separated to form the front and hind legs. Different body types can easily be made to represent, for instance, the long body and short legs of a mongoose. With the model made, it is possible to go through various gaits step by step, or if only a track pattern is available, to try out different options to re-create it.

As with individual tracks, there are particular ways of measuring track patterns to ensure that your data give you as much information as possible and are comparable to measurements from other populations or reference collections.

The terms used in measuring track patterns are sometimes given different meanings depending on which guide book you are using. We suggest that for your measurements you adopt the terms used here and stick to them, but take care when comparing your data with another set, that you fully understand what is meant by the terms it uses. The following is a list of the main features.

Establishing the Median Line

To measure any track pattern, it is first necessary to establish the position of the 'median line'. This is an imaginary line that divides the pattern down the middle, into two halves that usually correspond to the tracks made by the animal's right and left feet. In most registered walking gaits, for instance, both front and hind left feet will fall to the left of the median line, with those of the right side falling to the right. With more complicated track patterns, tracks can fall directly on the line or even cross over to the other side of it. In these cases, it can be quite difficult deciding where the line should be placed. A length of string is very useful. Attach one end of the string to a stone and place this at the head of the set of tracks you wish to measure. Next, stand at the base of the set and gently pull the string tight. Lay it down on top of the track pattern in the position you believe to be most central. It will often be necessary to adjust the position of the anchor stone slightly once you have begun to position the line.

The Step

A Step is the distance from the front edge of one foot to the front edge of the next foot in the track pattern. For this reason, it is primarily of use as a quick measure for track patterns where all the distances involved are the same and the pattern is continuous, such as the walk and the trot.

The Stride

This is perhaps the most important single measure. It should be taken from the front edge of the first track in a set, to the front edge of the same track where it next occurs in the pattern (Figure 10.6). In this way, the stride in a direct register walking pattern, for instance, will simply be twice the distance of the step. However, if the track pattern becomes more strung out and uneven as the animal accelerates, the stride distance will demonstrate the magnitude of the speed increase. For example, a slender mongoose (*Herpestes sanguineus*) speeding up from a trot to a gallop, goes through a brief period of loping. The distance from the front edge of its front left track (the first to be set down in that grouping of tracks) to the same point on the same track in the next set of tracks is generally about 55cm for the lope. The distance between the same two points measured a short way down the trail, once the animal has begun its bounding gallop, is generally about 65cm. By collecting such figures from a number of different individuals of the same species, it is possible to arrive at an average stride for any given gait. This can then be used as an aid to identifying which species is responsible for a track pattern when the tracks themselves are unclear. The stride for the bounding gallop of a marsh mongoose (*Atilax paludinosus*), for instance, is about 82cm.; about 17cm more than that of the smaller slender mongoose.

As a general rule, as the speed of an animal increases, so does the length of its stride.

The Straddle

The straddle is a measure of the width of a track pattern. It is usually taken from the outside edge of the track furthest to the left (as you look down the length of a track pattern) to the outside edge of the track furthest to the right. The straddle gives both an indication of the width of an animal (connected both to its species, age, and sometimes sex), and an idea of its speed. For example, the straddle of the sitatunga may be between 20cm and 28cm, while that of the smaller, more slender bushbuck seldom exceeds 15cm. One African civet moving at a steady walk was recorded with a straddle of 13cm with a stride of 52cm. With an increase in speed, the stride increased to 66cm, but the straddle dropped to 9cm. Therefore, as it speeds up, its feet land more centrally beneath its body.

While the stride and straddle are arguably the most important of the track pattern measurements, there are several others that can assist the tracker. Step measurements should ideally be taken in all situations, covering not only the distance from the first track in a set to the next, but also from each of the others to the tracks ahead of them. In this way, the tracker will become more aware of exactly how the increase in stride, that comes with speed, is achieved. Whenever a bound or jump is involved in a track pattern, this should be measured separately, from the front edge of the last track in a group to the heel of the first track in the set following the leap. In other words, measure the gap between groups. This measurement is often the key to distinguishing species that use a hopping or bounding style as their normal gait. In order to record the lateral position of each track, it is usual to measure from the middle of the front edge of the track (between the two cleaves of a hoof) back to the median line. It can also be helpful to record the angle of an individual track in relation to the median line. This can be done by using string or a grass stalk to mark a line that passes through the centre of the track and continues out behind it until it meets the median line. Measure the angle made by the two lines with a protractor, watch or compass. This measurement can occasionally be of use in determining species. For instance, the tracks of a bushpig and the bushbuck can sometimes be confused. However, a close examination of their track patterns will reveal that the tracks of the bushbuck commonly lie at a wider angle to the median line than do those of the bushpig. Figure 10.6 shows the standard measures to be taken when recording a track pattern. In this example, the animal's gait was a fast gallop.

Gaits

The gait of an animal is the style of locomotion it adopts for a specific purpose. This purpose might be, for instance, to graze, to walk calmly through an area, to move more rapidly, to flee from a predator, chase down prey, or to clear obstacles. To the tracker, the physical evidence of gait is the position of individual tracks relative to one another, forming a 'track pattern'.

While learning to recognise individual tracks can usually tell us the species involved, learning to interpret track patterns gives the tracker the ability to bring tracks to life; to learn how an animal moves; how it adjusts to different terrain or reacts to other animals. In addition to this, in instances where individual tracks are of a quality insufficient to determine species, a knowledge of track patterns can often significantly narrow down the possibilities or be diagnostic of the animal involved.

There are many gaits that the tracker must eventually become familiar with, but a good starting point is to learn three gaits that are commonly used by a large range of animals; walking, trotting and galloping (Figure 10.7).

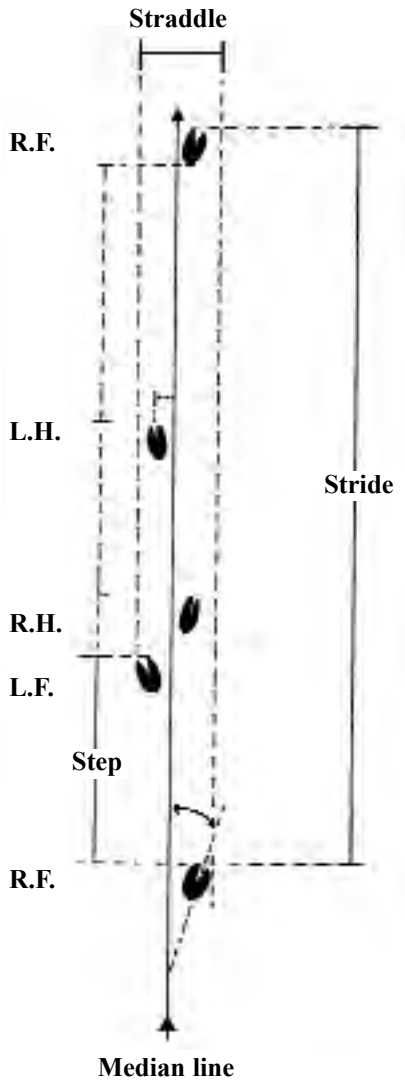


Figure 10.6 Standard measures to take when recording a track pattern. **R.F.** = right front foot; **L.H.** = left hind foot; **R.H.** = right hind foot; **L.H.** = left hind foot (for a galloping ungulate).

The Walk

For a great many mammals, this is the method of locomotion used most of the time. In a typical walking gait all four limbs are lifted and put down at different times. The classic diagonal or 'cross-walk' involves, for example, the initial placing of the animal's front left foot, followed by its hind right foot. This is followed by the front right which is in turn followed by the hind left foot. The visual effect, if you watch one side only, is very much of the animal trying to tread on its own heels, as the rear foot is set down only a fraction after the front foot of the same side has been lifted. With a standard walking gait, the track pattern will show the left and right tracks quite widely separated, and the hind tracks placed directly on top of the front tracks (known as direct register), landing just ahead of the front track (over-registration), or just behind the front track (under-registration). Where direct registering is representative of an animal's normal walking pace, over-registration will denote that the animal was walking quickly, while under registration shows a slower walk than normal. Animals that use the cross-walk as their normal relaxed gait include cats, dogs, duikers, bushbuck, forest buffalo and elephant.

The Trot

If a cross-walker wishes to increase its speed gradually, it moves from a fast walk into a trot. There are exceptions to this such as the elephant, who, due to the limitations of its great size, speeds up by merely walking faster. In the trot, diagonal pairs of feet are lifted and set down at the same time. That is, the front right foot and the hind left foot are lifted and moved forwards at the same time, followed by the front left and hind right feet. The way in which the trail pattern of a trot differs from that of a walk is most noticeably seen in the position of the hind tracks. These will typically become over-registered, or may land well in advance of the front track of the same side. As the animal's speed increases, the hind tracks will continue to move further and further ahead of the front tracks. Another consequence of the acceleration to a trot is that the stride distance will increase and the straddle decrease (see under 'Measuring Track Patterns'). The splay between the toes is likely to increase, and the tracks themselves are often less well defined.

The Gallop

The gallop can be seen as the fastest of the gaits for most of the animals that have the cross-walk as their normal gait. Not only is it fast, but it can be sustained for relatively long periods, as opposed to the jump, which, although fast, is likely to tire the animal sooner. During a gallop, there is a brief phase when an animal's four feet are all off the ground at the same time. It lands on its hind feet, coming down first on one, then, as its weight shifts forward, bringing the other down. By the time the latter hind foot is about to take off, the first of the front feet has come down and briefly takes the animal's weight, followed by the other, as the hind legs both move forward through the air. Thus the thrust for the gallop comes chiefly from the front legs. The track pattern for an average gallop shows first one front track, then, ahead of it, the other. Not far ahead of this, but further out width-ways, comes the first of the hind tracks, followed a little way ahead by the second.

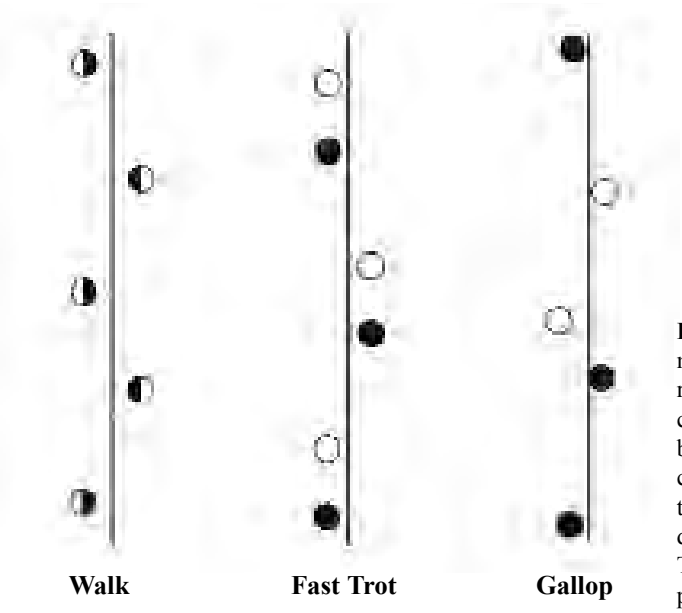


Figure 10.7 Track patterns for three main gaits. The black circles represent front tracks, and clear circles, hind tracks. In the Walk, the black and clear semi-circles correspond to fully registered tracks (where the hind foot lands directly on top of the front track). The solid line in the centre of each pattern is the position of the 'Median Line' (see text for explanation).

Other Gaits

There are other gaits besides the three main groups shown above. For instance, many grazing animals, such as the buffalo, exhibit a slow-walking style as they feed that has one hind foot coming forward and chasing the front foot of the same side ahead of it, with both feet coming down before the process is repeated on the other side of the animal's body.

Between the trot and the gallop, it is not uncommon to see "the lope" (or canter). The track pattern for this is similar to that of the gallop, except that only one of the hind tracks is found ahead of the forward front track. The other is found back between the forward and rearward front tracks.

"The jump" has the animal leaping into the air, propelled by its hind legs. It lands first on one front leg, then the other, but these both leave the ground again as the hind legs come through and past them. The hind legs are set down, usually very close to one another, a short distance ahead of the now vacated front tracks. The main feature of the jump track pattern is the large distance between the hind tracks and the front tracks ahead of them, coinciding with the period when the animal is airborne.

It should be noted that while a great many animals follow the acceleration process of Slow Walk - Walk - Trot - Lope - Gallop, this is not always the case. The main thing is to identify the gait an animal uses as its normal 'relaxed' form of locomotion. Squirrels, for instance, prefer a form of the jump for normal travel, in which the hind feet come down together, just ahead of the front feet, which also land as a pair.

Another gait to be aware of is "the pace", in which both limbs from the same side of the body are lifted and set down at the same time. The action and speed of this gait can vary considerably, from a slow and ponderous shuffling, through a standard walk (as used by the springbok, *Antidorcas marsupialis*) to a style commonly used by species such as the slender mongoose, in which the action and speed can be likened to that of the trot.

"The stot" (or sprone) is a gait adopted by certain antelope in times of stress, such as when being pursued by a predator. The animal propels itself high into the air using all four limbs for thrust. It lands back on all four feet and immediately shoots back into the air.

"The bipedal walk", in which an animal that normally uses all four limbs for locomotion, chooses to move only on its hind legs, is adopted by apes, particularly during aggressive displays, when the gait increases the animal's height. The giant pangolin (*Manis gigantea*), is also able to walk bipedally, though this is most likely due to the difficulties involved in putting weight on its front feet, which are armed with the huge curved claws it uses for digging.

Conclusion

The aim of this chapter has been to illustrate some of the wealth of information available to anyone taking the time to learn something of the art of tracking. It has suggested various techniques that may assist the tracker in that learning process and has highlighted some of the difficulties that may be expected.

As the study of animal ecology advances and biologists tend to place more emphasis on computer modelling and genetic studies, it is easy to understand how inexperienced field workers might view tracking as an old-fashioned skill, not deserving their full attention. However, the reality is that tracking and interpretation of trail represent the bedrock upon which the majority of wild animal studies are built. Successful tracking demands the same approach as any scientific endeavour. In order to find answers to the questions posed by a track or section of trail, the tracker must have the ability to learn from the finest details while at the same time being acutely aware of the broader framework in which they appear.

Information from animal tracks and trail

As we have seen, tracking at its most basic level can tell you of an animal's presence in an area. At the other extreme, experienced trackers are able to choose between following a trail back to the animal's sleeping area, or forward to catch up with the animal itself. They can determine its age and sex, its condition, what it is doing at any given time, its diet and its daily routine. In some circumstances they may be able to estimate how many animals of a particular species use an area. In short, tracking at some level is bound to play a role in whatever animal study you are involved in. The significance of that role will depend on the nature of the study and on the time and effort you are willing to invest in learning the fascinating language of animal tracks, trails and signs.

The following pages show some examples of track drawings made in Lopé, Gabon and Nouabalé-Ndoki, Congo, **all drawn at 50% of actual size**.

Recommended reading

Two guides to African animal tracks reviewed

A Field Guide to the Animal Tracks of Southern Africa.

Louis Liebenberg, 1990. David Philip Publishers.

This book gives an excellent coverage of the tracks of Southern African mammals. It deals not only with a wide variety of mammals, but also with birds, reptiles and invertebrates. The track drawings are of a high quality, with many of them reproduced at full size. For each species shown, there is a very helpful written description of its tracks, habitat, diet and lifestyle, though feces and feeding trail etc. are dealt with only fleetingly in a general section at the beginning of the book. A distribution map is given for each species, covering an area from the southern tip of Africa as far north as southern Malawi and Angola. There are interesting sections on the principals of tracking, gait, types of feet and a rather over-long chapter on dangerous animals. The degree of overlap with rain forest animals is naturally slight, so, while an excellent book, its usefulness is limited.

A Field Guide to the Tracks and Signs of Southern and East African Wildlife.

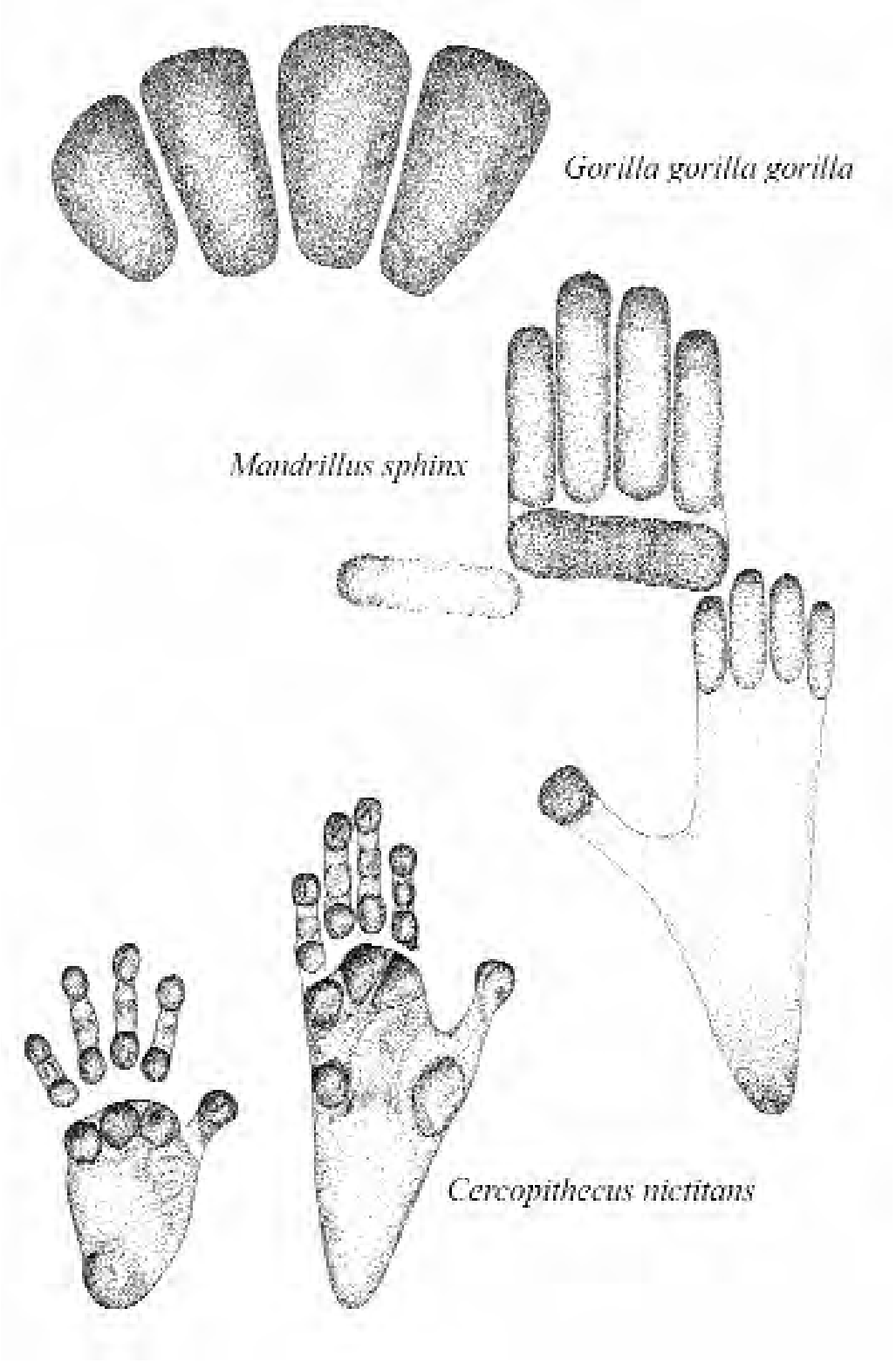
Chris & Tilde Stuart, 1994. Southern Book Publishers.

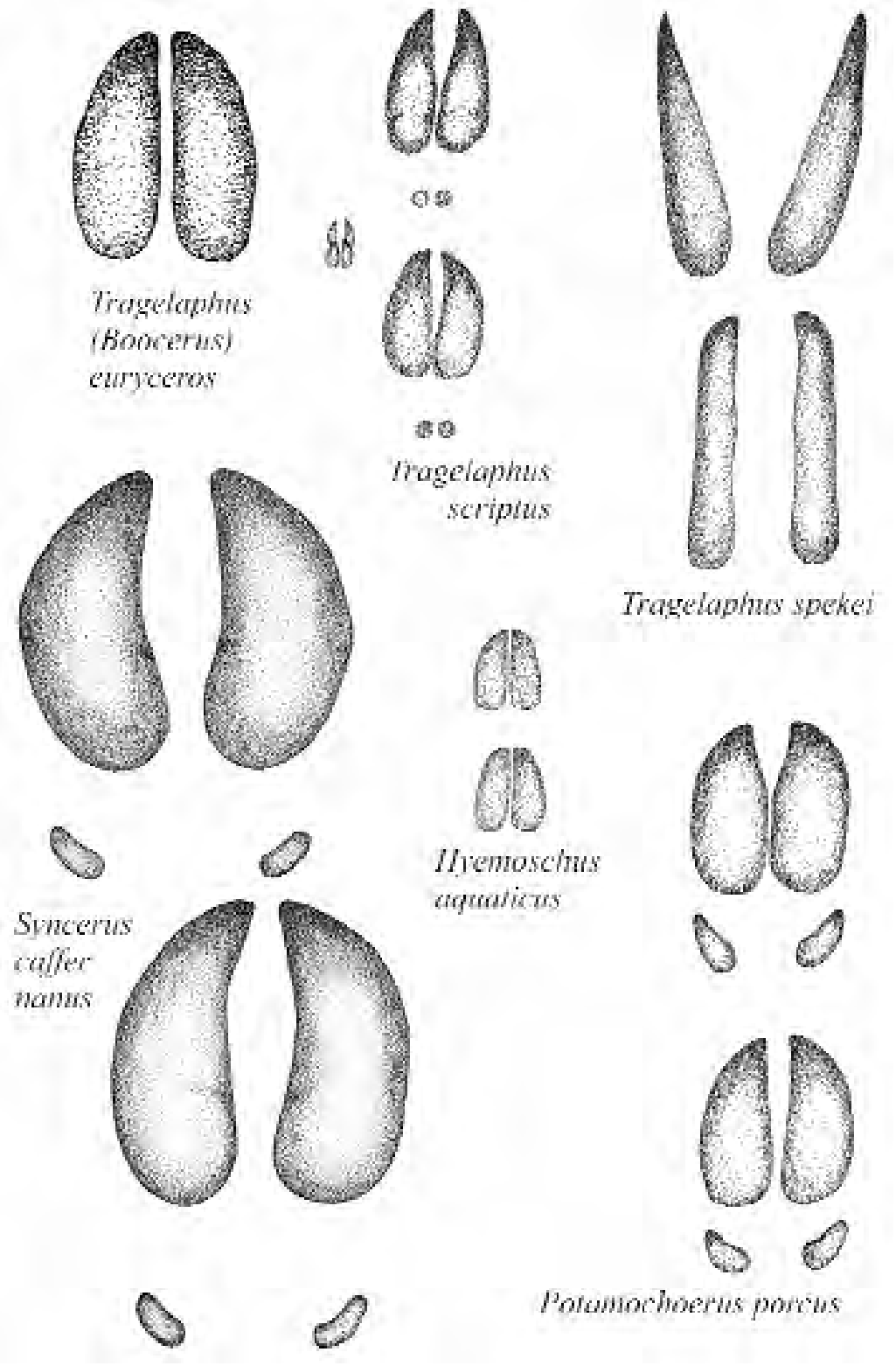
As the title suggests, this book also omits the bulk of Africa's rain forest, but in dealing with East as well as Southern Africa, there is more overlap with forest animals. Where this book really excels is in its scope and presentation. The book is full of excellent photographs which illustrate not only tracks in the field, but also animals' feet, feces, feeding remains and trails, nests and shelters, rubs, wallows, shed skin and hair. There is even a section on mammal skulls. The emphasis is very much on widening the readers perception of what constitutes animal trail; tracks being only one aspect in a wide range of examples. In this respect, even though not dealing directly with rain forest fauna, it is thoroughly recommended.

Other titles of interest

A Field Guide to the Animal Tracks of Southern Africa. 1990. Louis Liebenberg. (David Philip)
The Art of Tracking. (The Origin of Science). 1990. Louis Liebenberg. (David Philip)
Chris and Tilde Stuart's Field Guide to the Mammals of Southern Africa. 1994. (New Holland Ltd)
The Kingdon Field Guide to African Mammals. 1997. Jonathan Kingdon (Academic Press)
Tom Brown's Field Guide to Nature Observation and Tracking. Tom Brown Jnr. and Brandt Morgan 1993. (Berkley Books) *
Tracking and the Art of Seeing. 1992. Paul Rezendes. (Camden House Publishing Inc.) *
Collins Guide to Animal Tracks and Signs. Preben Bang and Preben Dahlstrom. 1974. (Collins) † (No longer available in English).
Animals. Tracks, Trails and Signs. 1992. R.W.Brown. M.J. Lawrence. J. Pope. (Hamlyn Publishing Group) †
Tracks and Signs of the Birds of Britain and Europe. 1987. Brown. Ferguson. Lawrence. Lee. (Christopher Helm Ltd) †

* North American animals.
† European animals.


Gorilla gorilla gorilla
Mandrillus sphinx
Cercopithecus nictitans


Tragelaphus (Boocerus) euryceros
Tragelaphus scriptus
Tragelaphus spekei
Hyemoschus aquaticus
Syncerus caffer nanus
Potamochoerus porcus

a □what is commonly seen; **b** □the whole tracks

Crocuta crocuta
Lutra maculicollis
Aonyx congica
Herpestes sanguinea
Atilax paludinosus
Panthera pardus
Civettictis civetta

CHAPTER 11
Information from dead animals and their curation

Alan Rabinowitz, John Hart, & Lee White

The tropical rain forests of Africa are teeming with many rare and poorly known animals. Anyone who spends time in a forest will encounter dead animals, such as those killed by disease, vehicles, predators or poachers. Meat decays rapidly in hot humid conditions, but, while often unsavoury, dead animals represent an important source of information that can be collected at little cost. Whenever dead animals are found, or live animals confiscated, as much information should be taken from them as possible. This information includes measurements, weights, age, sex, and general condition. Once measurements have been taken on a dead animal, the whole animal, or a part such as the skull, once preserved, can be incorporated into a reference collection. If this reference collection is well maintained it will contribute significantly to what is known about animals found in your area, and will represent the beginning of a small field museum.

The measurements and specimens you collect can contribute to the knowledge and management of a reserve in many ways, for example:

• in the same way as a botanical reference collection, a well documented collection of animals from a region provides indisputable evidence of their presence and acts as an aide to identification for all managers and researchers working in a reserve. By systematically preserving specimens you will gradually compile a species list which includes even some of the more elusive animals which are rarely, if ever, sighted alive;

• the process of collecting and preserving specimens will help field staff become familiar with the animal species present in their reserve, reducing the risk of their mis-identifying species which are observed in the wild;

• the bodies of dead animals can yield information about the status of animal populations in any given area (e.g., if there is a gradual increase in the proportion of juveniles and sub-adults amongst duikers seized from poachers it may indicate that over-hunting is a problem);

• museum specimens provide materials for scientific study. Many rain forest mammals are poorly known and there are still new species waiting to be described;

• bones and hair on museum specimens can be used as a reference to help identify animal remains found, for example, in poachers' camps, or in leopard scats;

• by monitoring the causes and frequency of animal deaths you will be well placed to detect health problems within the populations in your reserve and to react to any dangerous epidemics (see boxes 11a & 11b).

Preserving animal specimens

On finding an animal specimen there are several tasks to complete in order to transform it into a well documented museum specimen of lasting value. Unless all stages in this process are successfully completed the value of the specimen will be minimal:

- The first step is to record all useful details about the specimen in your field book;
- The second step is to preserve the specimen;
- Then a catalogue of specimens should be filled in and the specimen assigned a unique collection number;
- Finally, a ticket should be attached to the specimen and it should be integrated into the collection.

Box 11a FIELD VETERINARY PROGRAM

The Wildlife Conservation Society operates a 'Field Veterinary Program' which can provide immediate and long-term solutions to wildlife health crises. For example, in the late 1980's wildlife managers discovered that thousands of Zambian hippopotamuses were mysteriously dying. Local wildlife experts were unable to identify the cause of the massive die-offs, which threatened to decimate hippo populations in Zambia. They turned to WCS for help and a pathologist found that anthrax, a highly contagious and fatal disease, was the cause of the crisis. A vaccination program was launched immediately to ensure that healthy hippos and other wildlife, including the endangered black rhino, were not lost to the disease. Hence, vigilance on the part of national wildlife managers and rapid response of WCS' veterinarians helped to minimise the impact of this wildlife crisis.

Wildlife managers should monitor the health of the ecosystems in their charge. Unexplained deaths of animals should be investigated and if ever similar crisis situations arise in the future, a rapid response may be the key to minimising the damage. For advice about monitoring wildlife health, or for help in an emergency, contact the Field Veterinary Program at WCS.

Learn more about the WCS field veterinary program on the web at WWW.FIELDVET.ORG

Box 11b HEALTH

In 1995 a freshly dead chimpanzee was found close to a village in north-eastern Gabon. It was butchered, cooked and eaten. Shortly thereafter many of the people who came into contact with uncooked flesh and blood developed haemorrhagic fever, or Ebola. The chimpanzee itself had suffered from and died of Ebola and the disease was able to pass through the skin on contact and infect humans. This rather drastic example serves as a vivid reminder that one should always take care when inspecting and handling dead animals, particularly if the cause of death is unknown or suspicious.

It is important to use rubber gloves when handling dead animals or their tissues, to disinfect any equipment thoroughly after use, and to thoroughly wash hands and other exposed skin once you have finished. If you cut yourself during preparation of specimens disinfect immediately with absolute alcohol. Burn or bury any tissues not preserved.

Step 1: recording information in your field book

When you encounter an animal specimen, be it a dead animal (or its remains) found in the forest, or a carcass confiscated from poachers, you should note in your field book (see Chapter 5) details of the circumstances in which the specimen was obtained, the location, and any information about the animal itself which might be of interest. Note:

- the date the specimen was collected (this should already feature at the beginning of your field notes for the day in question);
- the location where the specimen was collected (you should refer to a place of known geographic location which features on maps of the area and, if possible, the geographic co-ordinates; add local names which may add precision to the location);
- the genus and species, if known, and any local names (note the dialect) for the animal;
- the sex of the animal (if possible);
- body weight, measurements and descriptions, as outlined below;
- notes on the age and body condition of the animal;
- reproductive status (e.g., number of embryos, nipple condition, condition of testes);
- presence of parasites and ecto-parasites (note if collected and cross-references specimens); ectoparasites are best preserved in alcohol.
- details of the circumstances in which it was found / collected (e.g., eagle kill, trap line, poacher's bag, road kill, etc.);
- details of the location where it was found (e.g., habitat type, distance to the nearest stream)
- any significant behaviour observed;

Measuring animals

The following measurements should be taken on any live or dead animals acquired (see Figures 11.1 - 11.5):

Mammals

- General description
- Weight (the weight of the live animal, or the weight just after the animal has died, but before the intestines, skin or any other part is removed).
- Total length (include the tail, follow the curves of the body -A plus B in Fig. 11.1a)
- Head and body length (do not include the tail - A in Figure 11.1a)
- Tail length (base to tip, do not include hair at the tip - B in Figure 11.1a)
- Girth (circumference of the body measured behind the front legs - C in Figure 11.1a)
- Ear length (ungulates - from notch to tip - B in Figure 11.1b)
- Hind foot length (from the heel to the tip of the longest digit - A in Figure 11.1b)
- Height at shoulder (from top of ridge between shoulder bones to the base of the front foot)
- Height at hindquarters (from the backbone to the base of the hind foot)
- Horn or tusk length (A-B in Fig. 11.2); spread at tips (C-D in Fig. 11.2);and maximum spread (E-F in Fig. 11.2).

Figure 11.1. Body measurements on a mammal (ungulate).

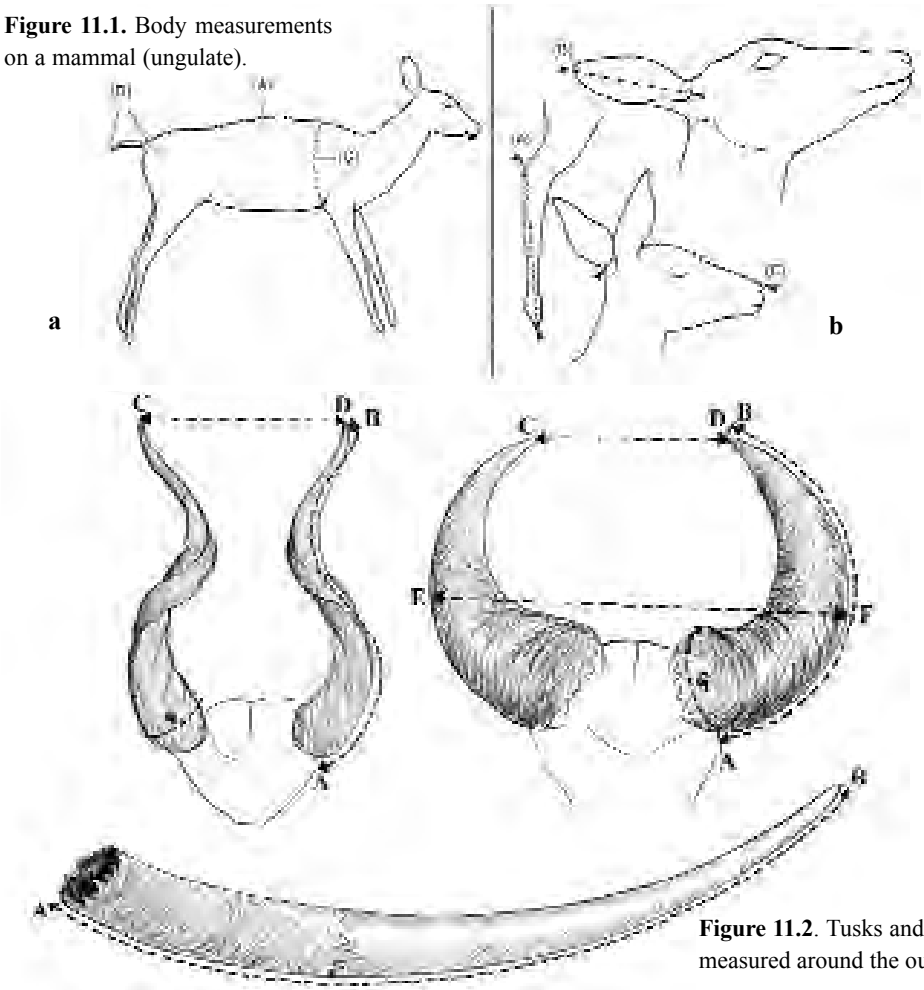


Figure 11.2. Tusks and horns are measured around the outer curve.

Information from dead animals and their curation

Birds

- General description (see Figure 11.3)
- Weight

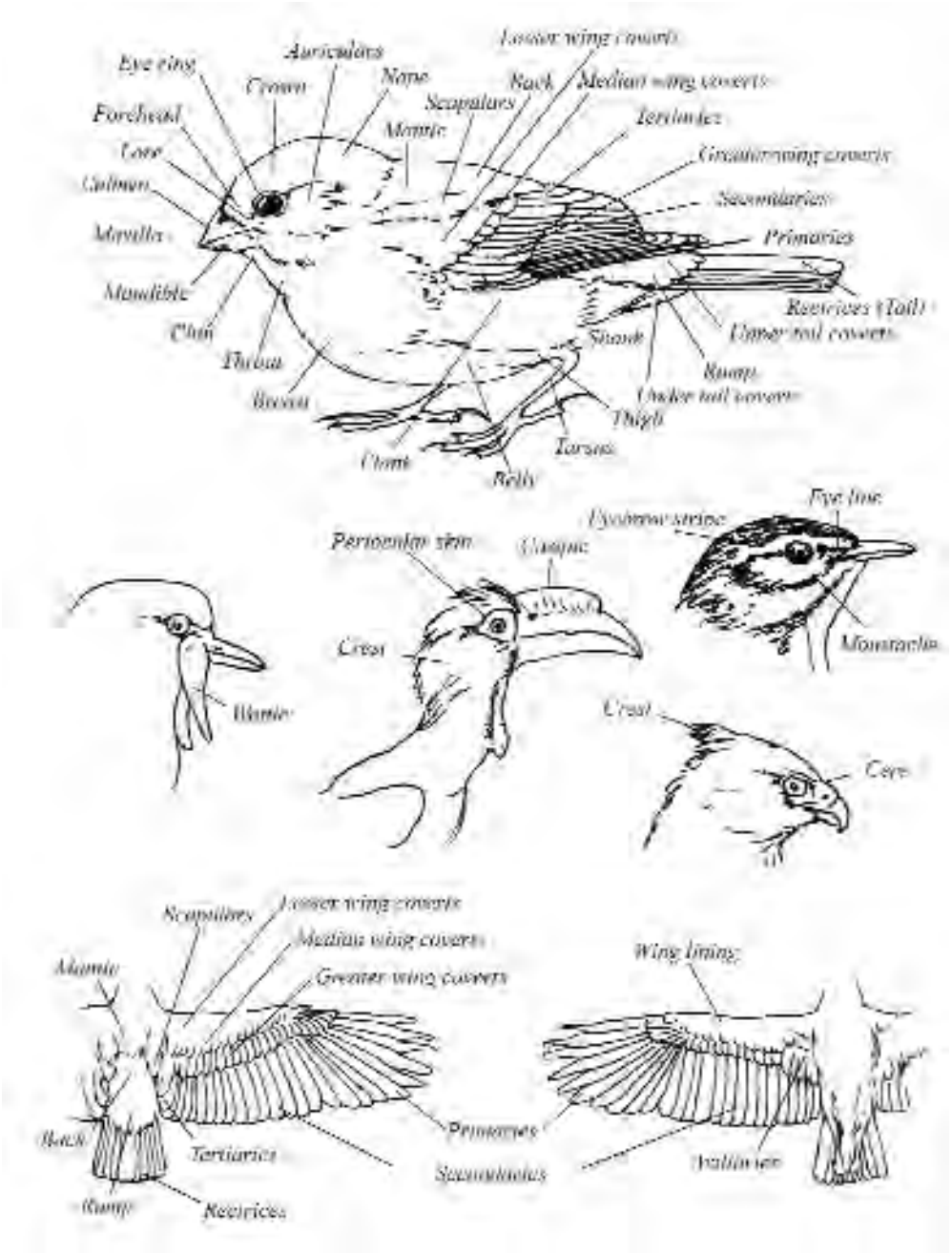


Figure 11.3. Terminology for different parts of a bird

- Total length (from tip of the bill to tip of the tail feathers - Figure 11.4)
- Tail length (from the base of the tail to the tip of the tail feathers - Figure 11.4)
- Wing length (Figure 11.4)
- Bill measurements (Figure 11.4)
- Length of the tarsus or leg bone (from the "heel" to the base of the digits - Figure 11.4)

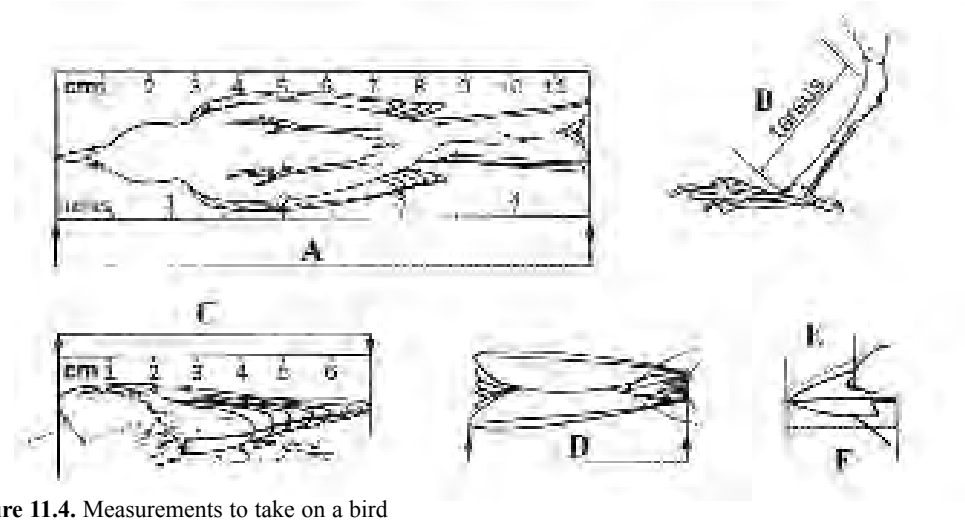


Figure 11.4. Measurements to take on a bird

Reptiles and Amphibians (see Figures 11.5 & 11.6)

- General description
- Weight
- Total length (from nose to tip of tail)
- Tail length
- For turtles: length and width of shell

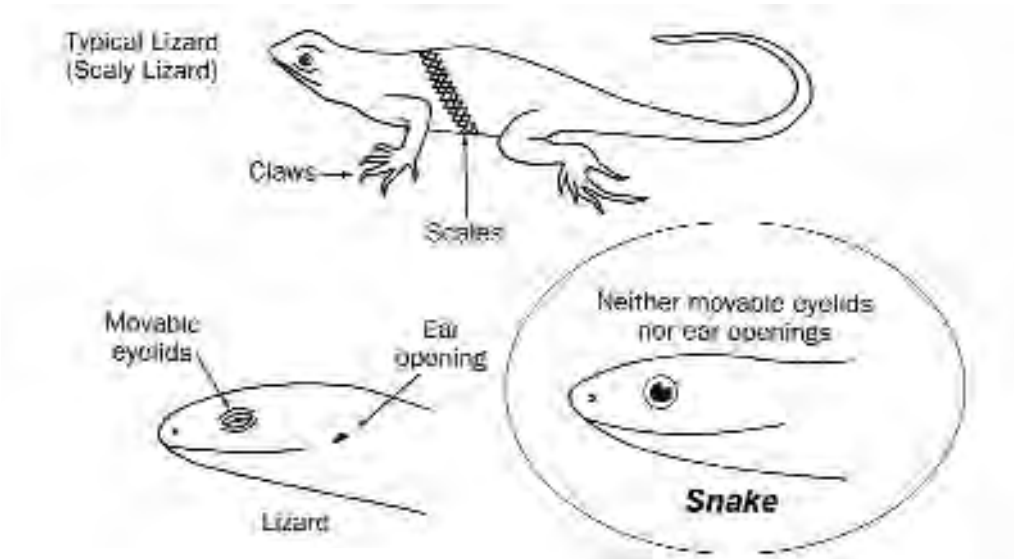


Figure 11.5. Distinguishing snakes and lizards

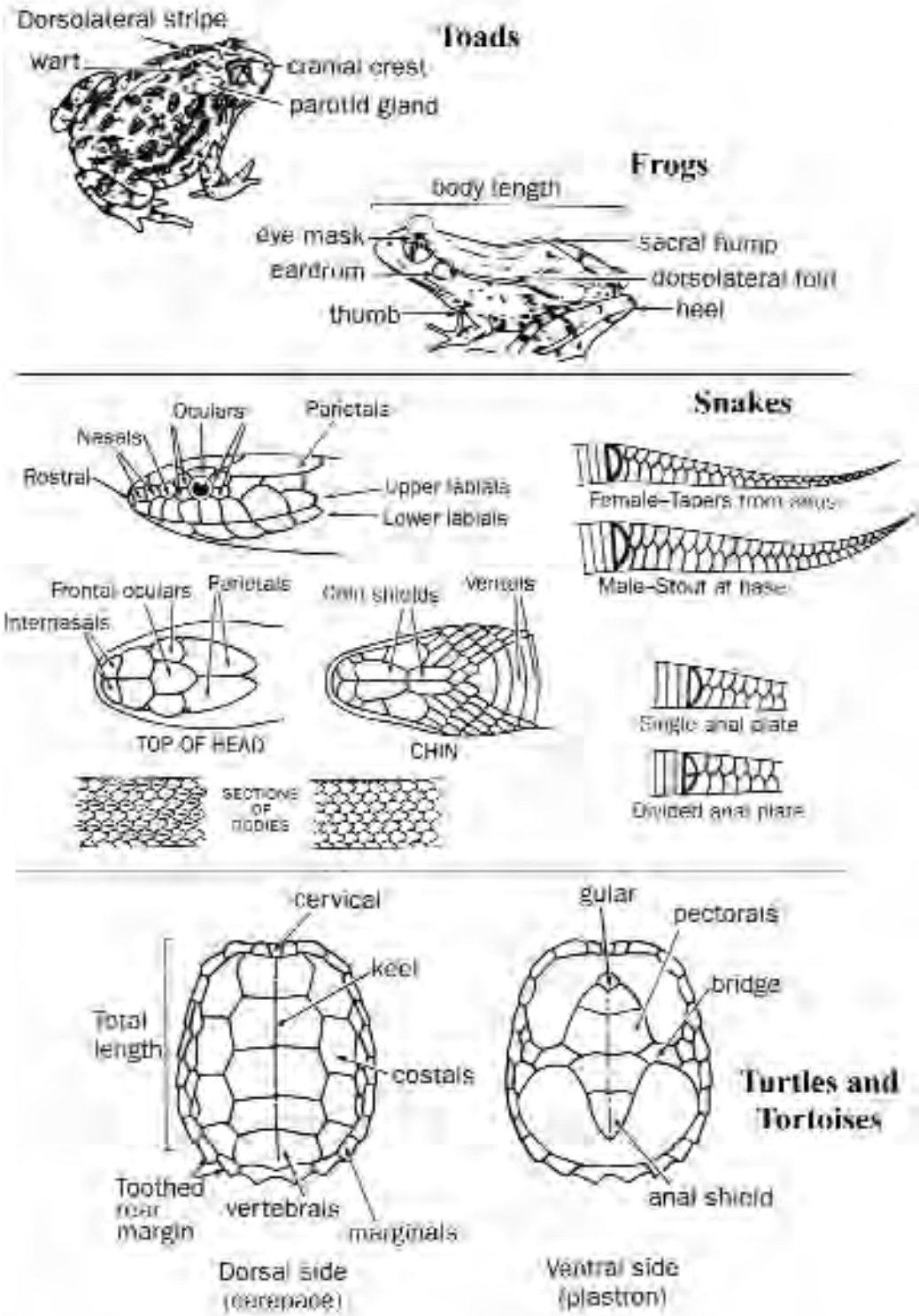


Figure 11.6. Terminology for different parts of amphibians and reptiles

Assessing animal age and condition

Age Classifications

- Infant - not yet weaned
- Juvenile - weaned, but still somewhat dependent on the mother, not sexually mature
- Sub-Adult or Young Adult - independent of the mother, but not sexually mature (either physically or behaviourally), note that this stage usually lasts longer in males than in females
- Mature or Old Adult - sexually mature, fully grown in size

Indications of Age

- Tooth Order - In most species, teeth become visible in a very specific order, and at specific ages throughout an animal's life. Immature animals often have fewer and smaller teeth than mature animals (see Box 11c)
- Tooth Wear - As mature teeth age, they can become darker, and the cusps or points become duller.
- Skull Suture Closure - In very young animals, sutures between the bones at the top of the skull are not fully closed. As the animal becomes an adult, the sutures close, but they remain visible. In older animals, the suture lines often become very thin and nearly invisible.
- Body Colouration - Some species, such as primates, change colour as they age.
- Nipples (females only) - The nipples of a female that has not yet suckled young tend to be pink and smooth. The nipples of a female that has suckled young tend to be brown and wrinkled.
- Horns and tusks - Horns and tusks continue to grow as an animal ages. Therefore, the size and the amount of wear on the horns or tusks can be used as indicators of the animal's age.

Indications of Condition

- Sufficient Food Intake - Look for extra fat on the body and the condition of the fur. In dead animals that are not too decayed the best measure of condition is the fat deposit on the kidneys.
- Signs of Fighting or Falling - Look for scars, broken teeth and evidence of broken bones.

Step 2: preserving the specimen

Mammals

Whole animal (small mammal)

A whole animal can be preserved in a container of alcohol (70-90%) or formalin (10%). Before storing the animal in formalin, inject formalin under the skin and into the organs. Remove the intestines (after the animal is weighed) before freezing the animal or storing it in alcohol.

Note that formalin is highly toxic □take great care when handling it and wash hands thoroughly when finished.

Skulls

Carefully cut the skull away from the vertebrae. Remove as much flesh and brain as possible with a knife. Soak the skull in water for about one week, or simmer it lightly in a pot. Remove the remaining flesh. (Note: Boiling can crack canine and other teeth.) The skull also can be buried in the ground to be cleaned by ants and beetles. Once the skull is clean it can be bleached in the sun or with 3% hydrogen peroxide. Skulls can be de-greased with carbon tetrachloride.

Horns

Boiling can split horns. Therefore, if you boil a head, keep the horns out of the water. If horns can be removed after steaming, clean the horn bone (skull) and the inside of the horn with formalin, then slide the horn back on the skull. If the horns cannot be removed, then after the skull is bleached, inject 30% formalin between the horn and the horn bone (skull).

Box 11c USING TEETH TO AGE DUIKERS AND ELEPHANTS

The progressive eruption of molar teeth can be used to determine the age class of certain animals. Two examples of particular interest to wildlife managers in the African rain forests are elephants and duikers.

Elephants

During their lives elephants progressively develop six molar teeth on both sides of their upper and lower jaws. Molars 1-6 are different in size (becoming progressively larger) and can be distinguished by measuring the length and width. By first identifying the leading molar in the lower jaw and then by studying the patterns of wear on the teeth, one can estimate the age of dead elephants (see Moss, 1996):

- first determine the number of molars in the lower jaw;
- then measure the length and width of each molar using callipers - be sure to note which measures are for the left and right, and the position (front or back) of each tooth measured if there are more than one in the jaw (Figure B11.1). Be sure to make separate measures for each tooth if more than one are present simultaneously on one or both sides of the jaw;
- now compare measurements of the principal tooth (usually the foremost tooth, although a peg of the previous tooth sometimes persists in front of the principal, least worn, fully erupted, tooth) with the information below to determine which molar was present when the elephant died and the corresponding age range;

Molar Number	Molar length (cm)	Molar width (cm)	Approximate age
1	1.0 - 4.0	1.3 - 2.0	0 - 2
2	5.0 - 7.0	2.5 - 4.0	1 - 4
3	9.5 - 14.0	3.9 - 5.2	3 - 9
4	13.0 - 17.5	5.0 - 6.8	9 - 18
5	17.5 - 22.5	5.9 - 8.5	16 - 36
6	22.0 - 31.0	6.4 - 9.4	34 +

- if the molar length falls into two categories, then use width to identify the correct molar number;
- if molar width falls into two categories, then use length to identify the correct molar number;
- if both molar length and width fall between two molar classes assign the tooth to the class whose maximum width or length dimensions are closest to the measured dimensions;
- for further details of how to obtain more precise estimates by studying wear and eruption of the teeth, see Moss, 1996.

Figure B11.1 Measuring the teeth in the lower jaw of an elephant.

Duikers

For duikers the age class can be determined by studying tooth eruption and wear in the lower jaw, particularly for the molars, as shown in Figure B11.2

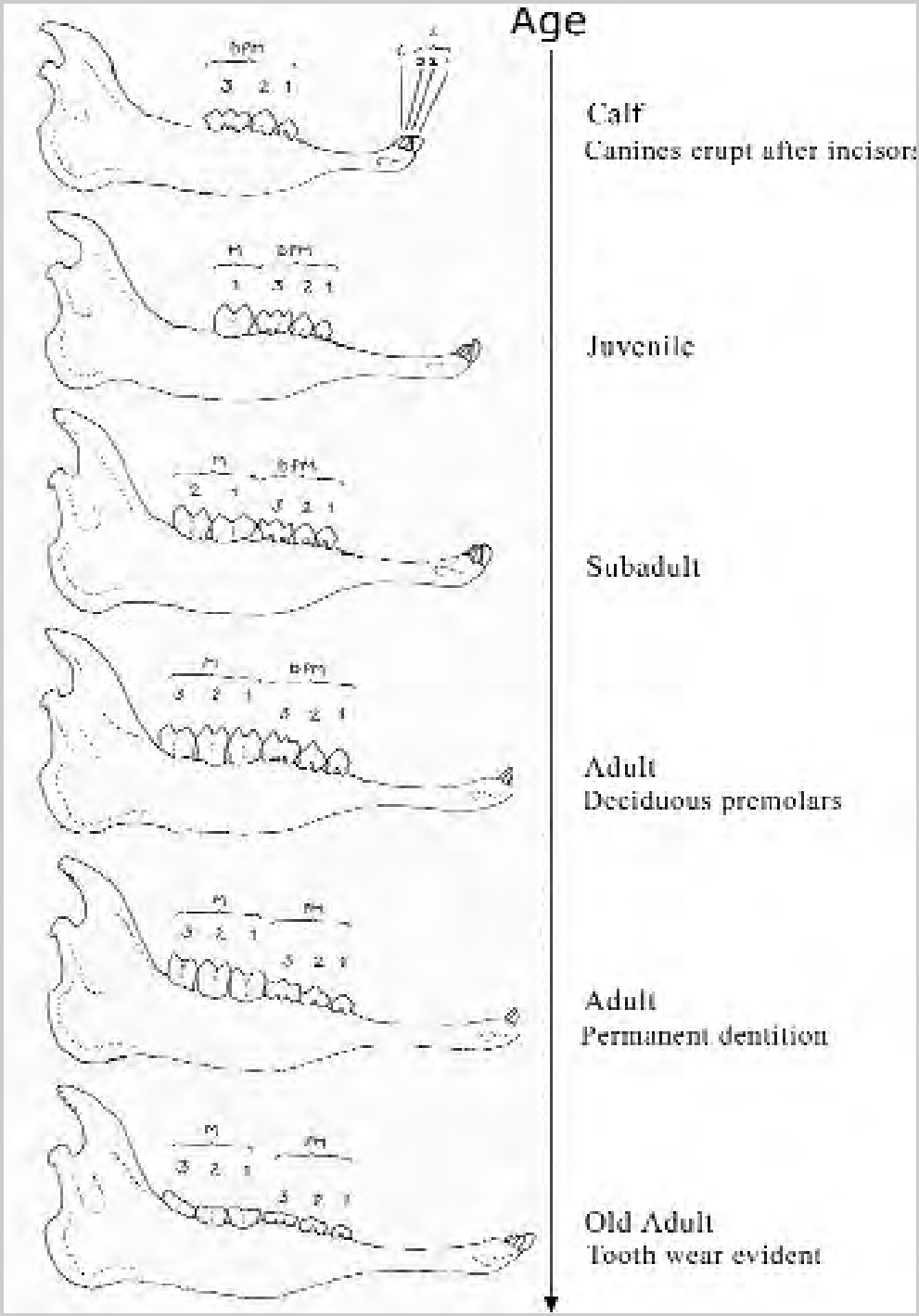


Figure B11.2 tooth eruption and wear in a typical ungulate lower jaw.

Skins

After removing the skin from the animal, clean as much flesh off the skin as possible. Then dry the skin in the sun, or if necessary, high over a fire, either hung on a line or stretched between pegs. Salting the skin will speed the drying process and temporarily preserve the skin. Areas that still have flesh or fat should be salted thoroughly. Powdered borax can be put on the skin to further preserve it - alternatively use cold ashes from a fire. When the skin is nearly dry, fold it with the hairy sides together.

Stomach Contents

Place the stomach contents in 5% formalin or 30-40% alcohol. Wrap large stomachs in cloth before putting them in 10% formalin.

Birds

Whole Bird

Store the bird in 70% alcohol or 5-10% formalin. Inject formalin into the muscles and body cavity. Otherwise, remove the organs (after the bird has been weighed) and freeze the bird.

Skins

After skinning the bird, rub the skin with borax, or a arsenic-borax mixture (if available) or alternatively with salt.

Stomach Contents

Store in 5% formalin or 70% alcohol.

Reptiles and amphibians

Whole Animal

Inject 10% formalin or 95% ethyl alcohol into the body cavity of reptiles. Or, inject 2.5% formalin or 25% ethyl alcohol into the body cavity of amphibians. After the animal hardens in the desired position, store it in 5-10% formalin. After 48 hours the animal can be wrapped in cloth and placed in a sealed plastic bag with just enough formalin to wet the cloth and the animal. The animal can also be stored in 70% ethyl alcohol or 40-50% isopropyl alcohol.

Skins

Either dry the skin carefully, or store it in 70% alcohol. Salt also can be used (for example, for snake skins). Snake skins can be rolled flat and placed in 10% formalin.

Turtle Shells

Put salt on the inside of the shell.

Fish

Store the whole fish in 70% alcohol or 10% formalin. For fish longer than about three centimetres, make a slit along the right side of the abdomen to facilitate penetration of the formalin. Or, after placing it in 10% formalin for 48 hours, the fish can be stored in 70% ethyl alcohol or 40-50% isopropyl alcohol.

Soft-bodied insects or parasites

Store in 70% alcohol - a few drops of glycerol will prevent the specimens becoming brittle.

Dung or feces

Dung can provide a great deal of information about many species (see Boxes 10b & 10c). Dry thoroughly in the sun, then store in plastic bags. If preserved in 10% formalin dung can by studied in the future for parasites.

Step 3: the specimen catalogue

Each collection should have a catalogue of all specimens collected by each collector (this will be a unique number series as for plant specimens, see Page 94, Chapter 8). Each specimen in a collection should be documented in the catalogue. Each specimen should be given a unique collection number, following on in sequence from that of the last specimen added to the collection (this number should be added to your field notes so that you can easily locate all specimens you have collected for future reference). The information noted in your field book (see above) should be reproduced in the catalogue such that there is a permanent record of the information associated with the specimen left in the field museum.

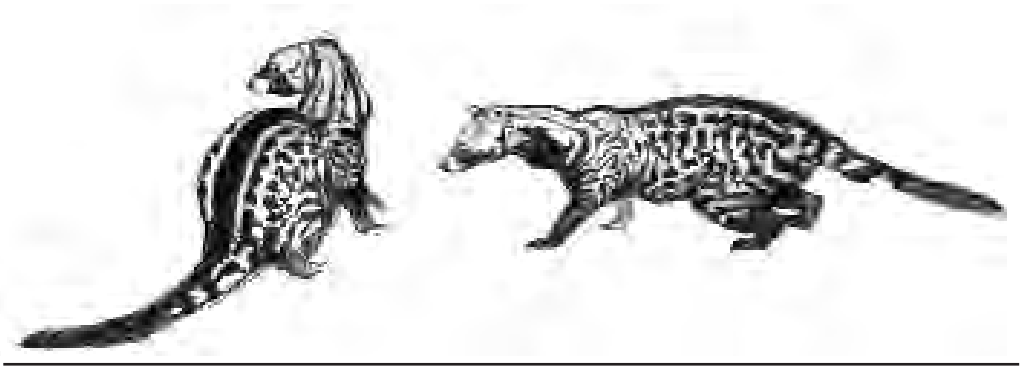
Step 4: labelling the specimen

Every specimen should have a label attached to it, or to its permanent container. A poorly labelled specimen is of little value. Without a label a specimen has no scientific value. The label should be made out of plastic or strong paper. Write in permanent ink or pencil (for specimens preserved in liquid). Be sure that the label is well attached (for samples in liquid include one label in with the specimen and a second stuck to the outside of its container, fixed with varnish if possible).

Note:

- the unique collection number in the collector's series;
- the date the specimen was collected (to avoid confusion write the name of the month and do not forget to include the year);
- the location where the specimen was collected (should refer to a place of known geographic location which features on maps of the area and, if possible, the geographic co-ordinates; add local names which may add precision to the location);
- the genus and species, if known;
- the sex of the animal (if possible); and
- the name of the person(s) who collected the specimen;
- additional information entered in the catalogue can be reproduced on the back of the specimen label if desired.

Attach the specimen label as soon as is convenient in order to avoid possible confusion. It is a good idea to attach a temporary label in the field prior to transporting the specimen back to your base.



CHAPTER 12
Necropsy procedures for wild animals*

Linda Munson

(with input from: W.B. Karesh, M.F. McEntee, L.J. Lowenstine, M.E. Roelke-Parker, E. Williams, M.H. Woodford; Illustrations by D. Haines)

Why necropsy wild animals?

Disease is one of many factors affecting the viability of wild populations. In a balanced ecosystem, most populations survive with low levels of disease or with periodic epidemics. However, as wildlife is impacted more by habitat disturbances the risks of a catastrophic epidemic within wildlife populations increase. Transmission of diseases between wild and domestic animals also becomes more likely.

To determine the disease risks to a population, the causes of morbidity and mortalities in that population must be identified. Risk assessment also includes an understanding of the natural history of infectious diseases in that environment, including the history of previous epidemics. Many wildlife disease epidemics affecting valuable wildlife resources or livestock have gone undetected because appropriate samples were not collected for diagnostic testing from animals that died during the epidemic. When appropriate samples and accurate written and photographic records are taken, the cause of an epidemic can be determined in most cases.

While it is ideal to transport sick or recently dead animals to a pathology laboratory for necropsy by trained personnel, in most circumstances transport is not possible. However, appropriate tissue samples can be obtained by field personnel if trained in necropsy procedures and sample collection. The purpose of this chapter is to provide practical guidelines for performing field necropsies on wild animals and for collecting, storing and shipping samples in the field for diagnostic testing.

General considerations

When performing a necropsy we strongly recommend that you take complete tissue and blood samples from carcasses. If only selected samples are taken because a particular disease is suspected and the animal does not have that disease, these samples may be inadequate to test for other diseases that might be causing the epidemic. Furthermore, selective sampling limits the information that could be procured from a wild animal necropsy that could aid in future population or ecosystem management.

Before performing a necropsy on an animal two important points need to be considered:

1. ZOONOTIC DISEASES:

Could this species have a disease that is transmissible to humans? Diseases such as rabies or echinococcosis (Hydatid disease) in carnivores, anthrax or rabies in ungulates, or psitticosis in birds can cause serious and fatal diseases in humans. Many primate diseases also can cause human illness. FOR THIS REASON, THE PERSON PERFORMING THE NECROPSY SHOULD WEAR A MASK AND PROTECTIVE CLOTHING. Wearing a mask is particularly important when preforming a necropsy on a primate, bird, or a carnivore suspected of rabies or hydatid disease. Also, all samples should be handled with care and unfixed samples should be placed in LEAKPROOF containers so that dangerous infectious materials do not leak during transport

*This chapter is available in English at http://www.vetmed.ucdavies.edu/whc/necropsy/toc.html and in French, Spanish and Portuguese at: http://www.fieldvet.org

2. REPORTABLE AND INFECTIOUS DISEASES:

Could this animal have a disease that is infectious to livestock or other wild animals? Diseases such as anthrax, foot and mouth disease, or tuberculosis can spread to other animals through contamination of the environment during the necropsy procedure. Anyone necropsying wild animals should be aware of the typical lesions of these diseases and take extra precautions when decontaminating a necropsy site. Suspicious carcasses should be deeply buried to prevent scavenger access if anthrax is suspected.

Important diseases to recognise

The following important diseases have lesions that can be recognised at necropsy and should be handled appropriately. Note that animals with rabies do not have any typical gross lesions.

ANTHRAX

Ruminants have a large, dark "tarry" spleen; Carnivores have a swollen head or neck due to edema in the soft tissues. Blood smears contain large bacterial rods (3-10 µm long by 1 µm wide) with blunt ends surrounded by a capsule. Bacteria occur singly or in short chains. Carcasses with anthrax should not be necropsied.

ECHINOCOCCUS ("Hydatid disease")

Clear cysts in the liver of carnivores or rodents

TUBERCULOSIS

Nodules in the lungs, enlarged lymph nodes or thickened intestinal wall.

EBOLA / SEVERE HEMORRHAGIC DISEASES

Bleeding from body orifices - do not touch or necropsy.

Preparing for the Necropsy

Equipment

A basic necropsy kit can be assembled in preparation for transport to a field necropsy site on short notice. The kit should contain the following items:

Protective clothing:

- Rubber Gloves
- Rubber Boots or Plastic Foot Protectors
- Rubber Apron
- Coveralls
- Mask (to cover mouth and nose) and Eye
- Goggles or Face Shield

Necropsy documentation

- Camera and film
- Field notebook

Necropsy equipment

- Sharp knife (including sharpening stone or steel)
- Scissors (small and large)
- Forceps
- String
- Ax or hatchet

- Hack saw or bone saw
- Small and large shears
- Chisel and mallet
- "Come-along" or block and tackle
- Scalpels and razor blades
- Alcohol lamp or gas burner for sterilising instruments
- Plastic ruler or measuring tape

Specimen containers and sampling equipment

- Rigid plastic containers with tight fitting lids (approximately 1 litre)
- Small vials, tissue cassettes, or tags to identify specific samples
- Sterile vials or blood tubes
- Plastic bags with closure tops (zip-lock or whirl-pack)
- Parafilm or sealing tape
- Aluminum foil
- Sterile syringes and needles (20 g)
- Sterile swabs in transport tubes
- Labelling tape or tags, waterproof labelling pens, and pencil
- Microscope slides and slide boxes for transport
- WHO rabies kit (or drinking straw in a small jar of glycerin)
- Whatman-type filter paper

Transport materials

- Ice coolers
- Leak-proof, break-proof containers
- Absorptive packing materials
- Sealing tape
- Sterile buffered glycerin (50%) (see Box 12a for formulation)
- "Easy blood" (see Box 12a for formulation)

Fixatives

- 10% buffered formalin (see Box 12a for formulation)
- 100% acetone for cytologies
- 70% ethyl alcohol for parasites

Disinfecting materials

- Pail and brush
- Disinfectant
- Borax
- Sodium hypoclorite (0.5%) (10% Chlorox)
- 70% ethyl alcohol (for disinfecting instruments)

Equipment

- Microscope: A microscope with a mirror for a light source or adapted for car batteries (a field scope will permit assessment for anthrax before opening a carcass).
- Centrifuge: A portable centrifuge for spinning blood is optimal. (Eg. Mobilespin centrifuge from Vulcon Technologies, 718 Main, Grandview, MO 640330 USA; 816-966-1212 or FAX 816-966-8879)

Safety Precautions

Personal safety

Because some diseases of wildlife can cause serious illness or death in humans, all carcasses should be handled as if they were harbouring potentially dangerous diseases and precautions for personal safety should be exercised. Minimal protective clothing includes coveralls, gloves and a mask that covers the nose and mouth, rubber boots. When necropsying a primate, a full face shield, coveralls, and double gloves should be worn. A washable rubber apron also is recommended.

Carcass handling and disposal

Diseased wildlife also should be handled to minimise exposure of other wild and domestic animals. If ANTHRAX is suspected, make a blood smear by nicking an ear vein or other available vein and check for *Bacillus anthracis* by microscopy before opening the carcass (see Box 12b). Carcasses with anthrax or other infectious diseases should be buried (preferably covered with a disinfectant and buried at least 2 m deep to prevent scavenging).

Shipping samples

Fresh and frozen samples should be packaged so that no leakage occurs.

Preparing Sample Containers

All containers, tubes, slides, and bags should be labelled using a waterproof marker. Placing a second label in a plastic bag that is then attached to the container adds further security. For formalin-fixed tissues, a paper label with the animal identification written in pencil can be submerged in formalin with the tissues.

The following information should be included on the labels:

- Date
- Geographic location (Park name or nearest town, country)
- Species
- Sex and approximate age
- Tissue Identification (this is not necessary for formalin fixed tissue samples)
- Person taking sample
- Animal ID (if available)

Handling Decomposed Carcasses

Most carcasses will have some AUTOLYSIS, but diagnostic tests can still be performed if tissues are properly handled. Handle autolysed tissues for histopathology very gently Hold tissues at the edges only. Cut with a sharp knife or scalpel. Quickly place in formalin Freeze or refrigerate samples as soon as possible for infectious disease or toxicology testing.

Autolysis can cause many artifacts in tissues that can be confused with a disease process. However, it is always best to take a sample from an area that looks abnormal rather than assume that the change was caused by autolysis. Histopathology will be able to distinguish between true lesions and post-mortem changes.

Freezing of carcasses or tissues will severely diminish the ability to recognise most changes microscopically, though other laboratory test procedures may still be possible.

BOX 12a FORMULATIONS FOR SAMPLE PRESERVATIVES

Sterile Buffered Glycerin (50%) (for transporting tissues for culture when refrigeration is not available).

To make sterile buffered glycerin, mix glycerin with an equal amount of buffer composed of:

- A. 21 g citric acid mixed in 1000 distilled water
- B. 28.4 g anhydrous sodium phosphate in 1000 distilled water

Mix 9.15 ml of A and 90.85 ml of B. Mix 100 ml of buffer with 100 ml of glycerin. Then sterilise in small tubes to take into the field

"Easy Blood" (for transporting DNA from blood cells for genetic studies when refrigeration is not available - also can be used to preserve DNA for longer periods of time if refrigerated or frozen).

- 1.2 g Tris HCl
- 3.7 g Na2 EDTA
- 2 g sodium dodecyl sulfate (SDS)

Add water to 100 ml

10% Buffered Formalin (for fixation of tissues for histology)

To make one litre mix:

- 100 ml formalin (38-40% formaldehyde)
- 900 ml distilled water
- 4 g sodium chloride (table salt) [OR 4.5 gm sodium phosphate (monobasic) and 3.6 gm sodium hydroxide].

BOX 12b CHECKING FOR ANTHRAX

If you suspect that an animal may have died of anthrax take blood smears from peripheral areas (in ungulates) or from areas of swelling (in carnivores).

IF ANTHRAX BACTERIA ARE SEEN ON BLOOD SMEARS, THE CARCASS SHOULD NOT BE OPENED.

Opening a carcass with anthrax will cause the bacteria to sporulate and disperse the spores throughout the environment.

To screen for anthrax before a carcass is opened, a small amount of blood should be obtained from an ear vein or coronary band. In carnivores, blood should be obtained from areas on the face and neck that are swollen.

Take a single small drop of whole blood and place it near one end of the microscope slide (A in Figure B12.1). Bring the end of a second slide (held at a 45 degree angle) up to the drop until the drop disperses along the edge of the second slide (B in Figure B12.1). Then with the first slide placed on a flat surface, push the second slide quickly and evenly toward the opposite end of the first slide (C in Figure B12.1). The smears should also be made by spreading a drop of blood in a small circle on the slides. When the smears are dry, label one end of the slide (with a solvent resistant pen or pencil) with the date, species, animal ID, and location. Slides can then be transported to a laboratory or stained in the field with New Methylene Blue or a Wright's stain kit.

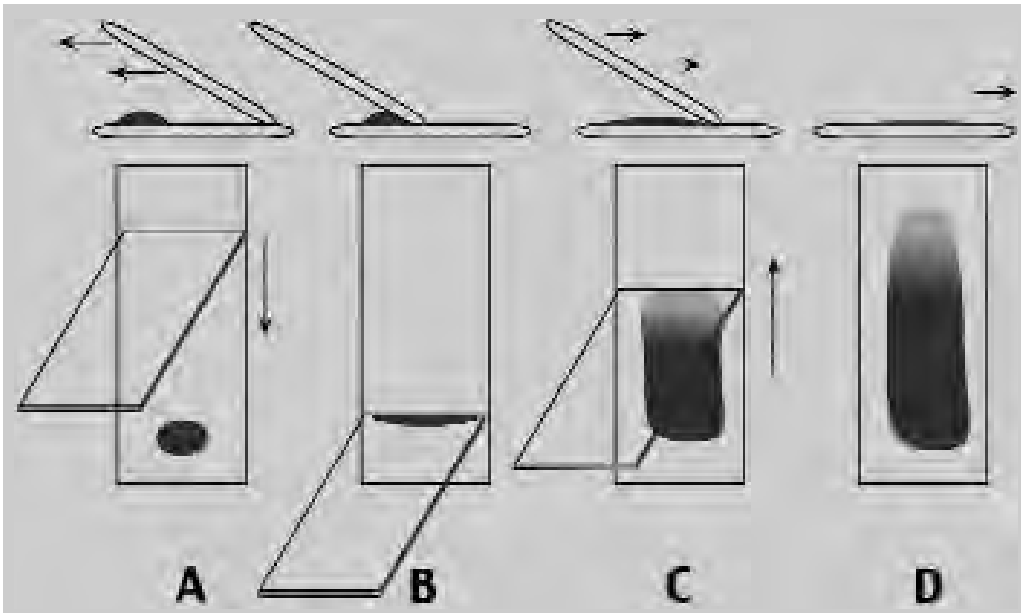


Figure B12.1 Making a blood smear.

Anthrax bacteria are commonly confused with Clostridium bacteria (see Figure B12.2). Anthrax bacteria appear as large rectangular rods (up to 10 μm long), alone or in chains and surrounded by a capsule. Anthrax bacilli obtained from a recently dead carcass usually do not form spores.

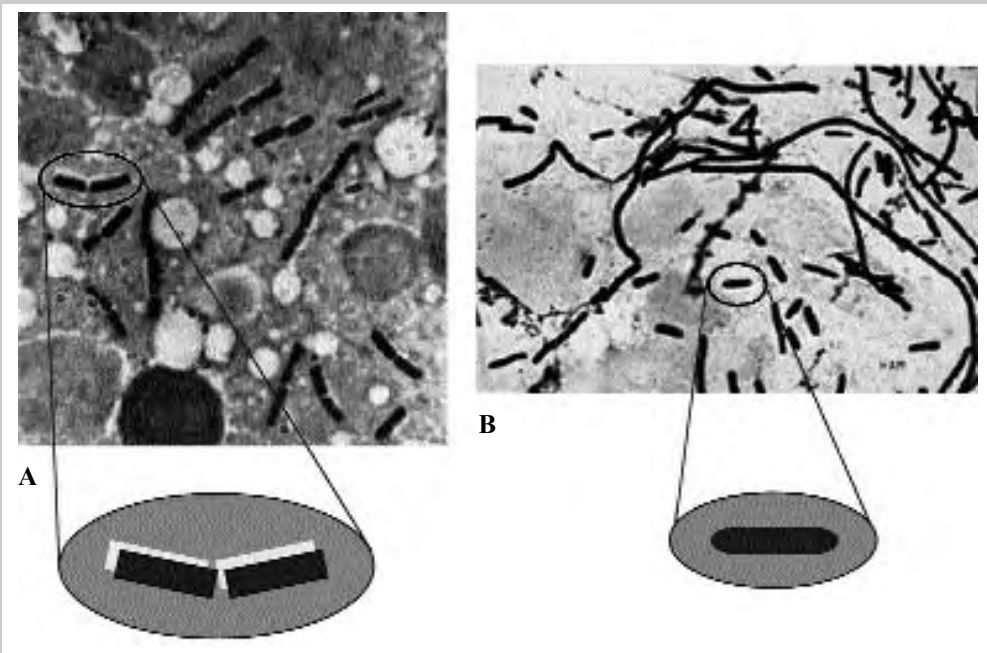


Figure B12.2 A. *Bacillus anthracis*, x2000. Note two main characteristics: 1) sharp, square ends; 2) pale capsule around bacteria. **B.** *Clostridium septicum*, x2250. Note three characteristics: 1) smaller size; 2) no capsule; 3) rounded ends.

Performing for the Necropsy

Examining the Carcass and its Environment

Assess the Condition of the Environment

- Note recent weather conditions that could have caused animal deaths (drought, floods, electrical storm, etc)
- Note the ambient temperature
- Note signs of struggle

Assess the Condition of the Animal

- Note any bite WOUNDS or other signs of predation. If wounds are present, look for bruising and bleeding in the tissues near the wounds which would indicate that they occurred before the animal died. Otherwise these wound most likely were caused from the carcass being scavenged.
- Look for broken bones, missing hair, broken or missing teeth or other signs of trauma.
- Look for and preserve any external parasites.

Determine Nutritional Status of the Animal

- Take weight (if possible) and/or body length and girth (see Chapter 11). Assess fat stores under the skin and in body cavities.
- Note the amount of fat around the heart and kidneys
- Note the muscle mass of the animal
- Note the amount of food in the digestive tract
- Note the condition of the teeth

Tissue Sampling Procedures

Histology

Box 12c gives a tissue check list to act as a guideline as to what samples to collect.

- Samples should be taken from all major organs and ANY ABNORMAL AREAS
- All samples should be placed in a common container of 10% buffered formalin (Box 12a)
- SUBMERGE TISSUES IN 10 TIMES THE VOLUME OF FORMALIN AS THE VOLUME OF TISSUE
- Samples should be no thicker than 1 cm so that they can fix, but long and wide enough to represent the different areas of a tissue as well as any abnormalities. Samples that include abnormal areas and surrounding normal areas are best.
- Samples should be handled carefully by grasping at the edges. Do not scrape surfaces of tissues or compress them with forceps.
- Most tissues do not need individual labelling. If a tissue needs special labelling (eg. a specific lymph node), place it in a different container (or tissue cassette) or attach a piece of paper to the tissue with string or a pin and label the paper or container with pencil or waterproof marking pen.
- **Never freeze tissues to be used for histology either before or after storing in formalin.**

Microbiology (Bacteriology and Virology)

To take samples without contaminating them, the samples need to be taken BEFORE tissues are touched and the instruments need to be sterilised. These samples also should be placed in sterile containers.To sterilise instruments: dip the tips in alcohol and then flame them or flame the tips until they are red and then let them cool. Samples also can be taken with a sterile swab, sterile syringe, or by placing a large (3 cm2) section of tissue in a sterile container (the centre of the tissue will be uncontaminated).

Take samples that contain abnormal areas. Appropriate samples include: whole blood, pus, areas with abscesses or nodules, or intestinal contents (within a loop of intestines).When taking samples from infected tissues, select an area near the edge of the affected tissue where live organisms are most likely to be found. If no abnormal areas are present, take standard tissue samples of lung, liver, kidney, spleen, tonsil, and intestines (see Check List in Box 12c)

Keep samples moist with sterile transport media, sealed in a sterile container and cold. If refrigeration is not available, samples can be placed buffered glycerin.

Smears of pus or infected tissues also are useful and can be air-dried and shipped with cultures.

Box 12c CHECKLIST OF TISSUE SAMPLES FOR HISTOLOGY

Fixed Tissue Checklist for Histology

Preserve the following tissues in 10 % buffered formalin at a ratio of 1 part tissue to 10 parts formalin. Tissues should be no thicker than 1 cm. INCLUDE SECTIONS OF ALL LESIONS AND SAMPLES OF ALL TISSUES ON THE TISSUE LIST.

- Salivary gland
- Oral/pharyngeal mucosa and tonsil -plus any areas with erosions, ulcerations or other lesions.
- Tongue - cross section near tip including both mucosal surfaces.
- Lung - sections from several lobes including a major bronchus
- Trachea
- Thyroid/parathyroids
- Lymph nodes - cervical, mediastinal, bronchial, mesentericand lumbar. Cut transversely.
- Thymus
- Heart - Sections from both sides including valves
- Liver - sections from 3 different areas including gall bladder
- Spleen - Cross sections including capsule.
- GI Tract - 3 cm long sections of:
- Esophagus
- Stomach - multiple sections from all regions of the lining
- Intestines - multiple sections from different areas
- Omentum - ~3 cm square
- Pancreas - sections from two areas
- Adrenal - entire gland with transverse incision.
- Kidney -cortex and medulla from each kidney
- Urinary bladder, ureters, urethra - cross section of bladder and 2 cm sections of ureter and urethra.
- Reproductive tract - Entire uterus and ovaries with longitudinal cuts into lumens of uterine horns.
- Both testes (transversely cut) with epididymis. Entire prostate, transversely cut.
- Eye
- Brain - cut longitudinally along midline.
- Spinal cord (if neurologic disease) - sections from cervical, thoracic and lumbar cord.
- Diaphragm and Skeletal muscle - cross section of thigh muscles
- Opened rib or longitudinally sectioned femur - marrow must be exposed for proper fixation
- Skin - full thickness of abdominal skin, lip and ear pinna.
- Neonates: umbilical stump - include surrounding tissues

Box 12d CHECKLIST OF TISSUE SAMPLES FOR MICROBIOLOGY AND TOXICOLOGY

Tissue	Microbiology	Toxicology
Brain	*	*
Fat		*
Kidney	*	*
Stomach contents		*
Hair		*
Liver	*	*
Whole blood	*	*
Lymph nodes	*	*
Tonsils	*	*
Spleen	*	*
Abscesses, granulomas	*	

Serology

Serum should be placed in sterile tubes then stored and shipped frozen. In recently dead animals, the right heart usually contains plasma clots (yellow/tan material), unclotted blood, or clotted blood. Plasma or blood should be removed and left undisturbed for approximately 30 min to encourage clot formation, then centrifuged at approximately 2000 X G for 20 min. The Mobilespin centrifuge from Vulcon Technologies, 718 Main, Grandview, MO 640330 USA;816-966-1212 or FAX 816-966-8879 is portable and easily adapted for field use. When a centrifuge is not available, serum can still be obtained by letting the clot or blood cells settle. The serum (clear/yellow fluid or red-tinged fluid if the animal is autolysed) or plasma should be separated for the blood cells, divided into at least two aliquots, transferred to vials, and then refrigerated or frozen (-20° or -70°) until transported to a laboratory. Serum vials should be labelled with the species, animal ID, date, and owner (e.g., country and park) using a waterproof marker.

If a centrifuge is not available and blood is obtained from a live animal or a dead animal whose blood has not yet clotted, remove whole blood into a blood tube, let the blood clot with the tube inverted (rubber stopper down), then turn the tube right side up and very carefully remove the stopper with the blot clot attached, leaving the serum in the tube.

Toxicology

Take samples and place half of each sample in aluminum foil and half in plastic bags or containers (aluminum or plastic interfere with the testing of some toxins). Samples should be stored frozen (if possible) until shipped to a laboratory (see check list in Box 12d).

Parasitology

Make at least 3 blood smears on clean glass slides (see Box 12b). Fix approximately 2 g feces in 70% ethyl alcohol or formalin.

Making Slides for Cytology

- Make a clean cut with a scalpel blade across the surface of the abnormal area of the tissue you wish to examine .
- Grasp the sample firmly with forceps, placing the cut surface down.
- Blot the cut surface of the sample across a paper towel or other absorbent surface until no blood or fluids are evident.
- Then gently touch the blotted surface in several locations on clean slides.
- Air dry slides.
- Fix slides in 95% alcohol

Filter paper

Small amounts of blood can be collected on filter paper and used in testing for some viral, bacterial and blood parasite DNA or RNA. Antibodies against some pathogens may also be detected using these samples.

- 1-2cm spots of whole blood are dripped on a thick Whatman-type filter paper. A prepared filter paper and cover is Fitzco/Whatman - FTA #8504, available from Life Technologies.
- Spots of blood can also be made by touching the filter paper to an organ or muscle.
- Before using, or after blood spots are dried, filter paper can be labelled with the species, ID and date using a pencil.
- After drying ar room temperature, store filter papers in individual plastic bags in a dry place. Adding a small silica gel pack to each plastic bag is recommended.

General steps to performing the necropsy: carcass dissection using a carnivore as an example

Introduction

The necropsy method outlined below provides a simple consistent method to examine a carcass and its body organs. A CARNIVORE has been used to demonstrate carcass dissection and tissue sampling procedures. Procedures for necropsy of UNGULATES, BIRDS, and REPTILES also are demonstrated. Very small animals (less than 100g) can be fixed whole by opening the body cavity and submerging the entire animal in formalin.

All carnivores and ungulates are placed ON THE LEFT SIDE so that the right side of the carcass is opened. All birds, reptiles, and primates are placed ON THEIR BACK.

After the body cavities are opened, the general nutritional condition of the animal and location of all organs should be assessed (to determine if any organs are displaced) before organs are removed. At this time, a sterile blood sample for culture can be removed from the heart (the right atrium is the best location), then additional blood can be taken to obtain serum for serological tests. Also sterile samples of other organs should be taken for culture before organs are handled.

After the general condition of the animal has been recorded, individual organs can be removed, examined, and sampled in a systematic manner. Any abnormal findings (lesions) should be described. Photographs of abnormal findings provide the best documentation for records. Box 12e gives guidelines on what information to note during the necropsy. These forms should be filed and completed when results of laboratory tests are known.

Describing Abnormalities Found at Necropsy

Any abnormality should be described by the following criteria:

- Location
- Number and distribution
- Colour
- Size
- Shape
- Consistency and texture

For example: “The liver contains multiple tan, firm nodules ranging from 1 to 3 cm in diameter that are distributed throughout all liver lobes. The nodules are gritty on cut surface.”

Box 12e NECROPSY PROTOCOL EXAMINATION WORKSHEET

Name of prosector _____

Address of prosector _____

_____

Location of carcass _____

SPECIES _____

ANY ID # _____ WEIGHT _____ SEX _____

DATE OF DEATH DATE OF NECROPSY

HISTORY: (briefly summarise clinical signs, circumstances of death):

GENERAL CONDITION: (nutritional condition, physical condition)
Neonates: examine for malformations (cleft palate, deformed limbs, etc).

SKIN: (including pinna, feet)

MUSCULOSKELETAL SYSTEM: (bones, joints, muscles)

BODY CAVITIES: (fat stores, abnormal fluids)
Neonates: assess hydration (tissue moistness)

HEMOLYMPHATIC: (Spleen, lymph nodes, thymus)

RESPIRATORY SYSTEM: (nasal cavity, larynx, trachea, lungs, regional lymph nodes)
Neonates: determine if breathing occurred (do the lungs float in formalin?)

CARDIOVASCULAR SYSTEM: (heart, pericardium, great vessels)

DIGESTIVE SYSTEM: (mouth, teeth, esophagus, stomach, intestines, liver, pancreas, mesenteric lymph nodes). Neonates: is milk present in stomach?

URINARY SYSTEM: (kidneys, ureters, urinary bladder, urethra)

REPRODUCTIVE SYSTEM: (testis/ovary, uterus, vagina, penis, prepuce, prostate, mammary glands, placenta)

ENDOCRINE SYSTEM: (adrenals, thyroid, parathyroids, pituitary)

NERVOUS SYSTEM: (brain, spinal cord, peripheral nerves)

SENSORY ORGANS (eyes, ears)

PRELIMINARY DIAGNOSES:

LABORATORY STUDIES: (list bacterial and viral cultures submitted and results, if available)

SHIPPING TISSUES: OBTAIN CITES IMPORT AND EXPORT PERMITS BEFORE SHIPPING TISSUES BETWEEN COUNTRIES. After 72 hrs in fixative, ship tissues in a leak-proof container in adequate formalin to keep tissues moist.

Procedure for Dissecting Carnivores (See check lists in Box 12c for tissues to sample)

1. Examine the carcass for wounds and not the general condition of the fur.
2. Place the carcass on its left side.
3. Cut the skin along the ventral midline from the chin to the tail (Step 1, Figure 12.1). On females, examine the mammary glands. On males, examine the prepuce and penis. On neonates, examine the umbilicus.
4. On the right side, reflect the skin to the level of the backbone.
5. Reflect the right limbs by cutting through the muscles and the hip and shoulder joints.
6. Open all three body cavities (abdomen, chest, heart): Remove the flap of muscle and other tissues that cover the right side of the abdominal cavity. Open the right side of the chest cavity by cutting the ribs along the sternum and backbone (Step 2, Figure 12.1). Open the sac surrounding the heart.
7. Record any abnormal locations or size of organs.
8. Record the quantity, colour, and contents of any fluids in the body cavities. Look for abnormal attachments of organs to the body cavity and determine if these attachments are easy to break.

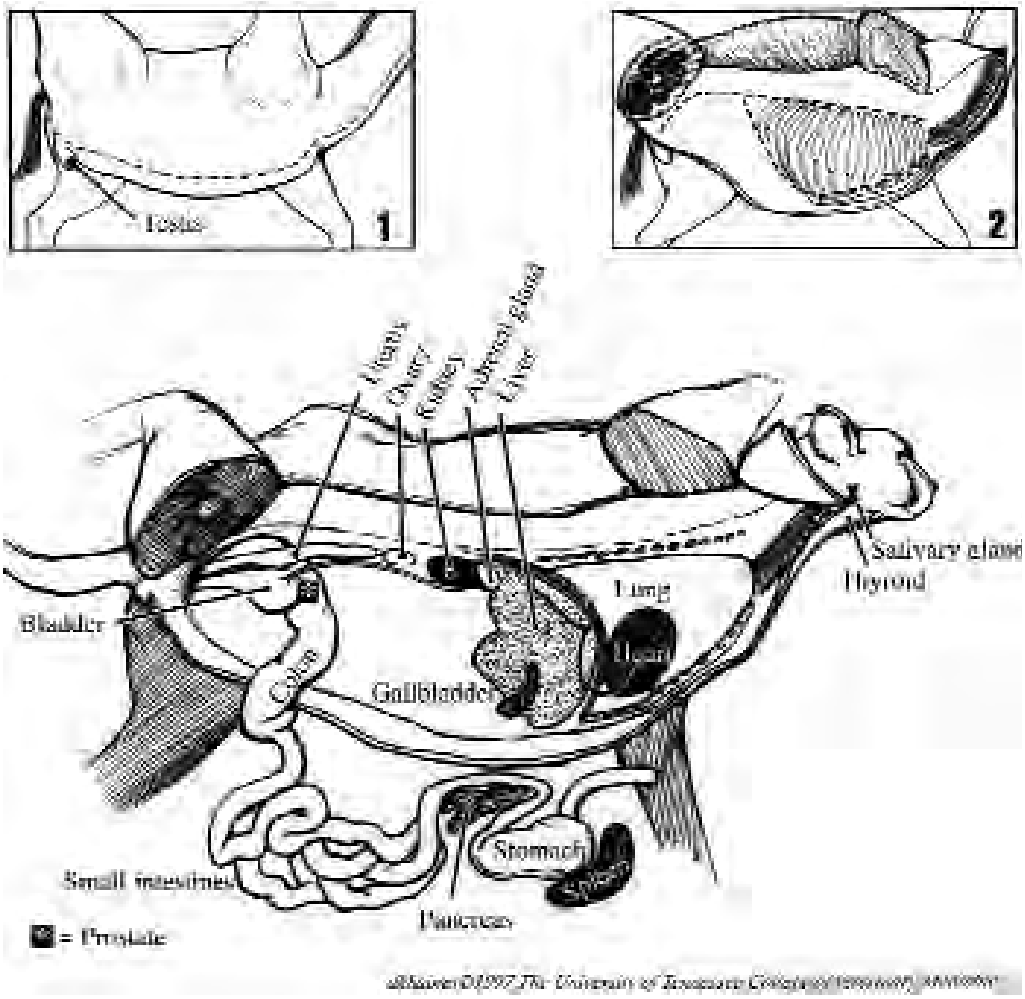


Figure 12.1. Opening a carnivore carcass

General Organ Sampling Procedures

Stomach and intestines

Remove stomach and intestines first but open them last to prevent contamination of the necropsy site.

Find where the esophagus enters the stomach and cut across the esophagus while holding the entry to the stomach closed to keep any food inside. Remove the stomach and intestines as a unit by cutting the mesentery where they attach to the intestines. Sample several lymph nodes along the attachments of the intestines. Leave the pancreas attached to the intestines and the spleen attached to the stomach (Figure 12.2). Cut across the rectum while holding it closed to prevent feces from escaping. Open the intestines along their length (intestines are best opened at the end of the necropsy to prevent contamination of other organs with food and fecal material). Note the content of the intestines (amount of food, presence of abnormal materials such as poisonous plants). Take stomach contents for toxicology. Take tissue samples from all areas of the GI tract and the pancreas.

Spleen, liver, pancreas

Remove the spleen from the stomach, examine it for abnormalities by slicing it across in multiple sites and then remove samples for histology. Remove the liver; open the gall bladder. Examine the liver by cutting it across in multiple sites. Take samples for histology and toxicology.

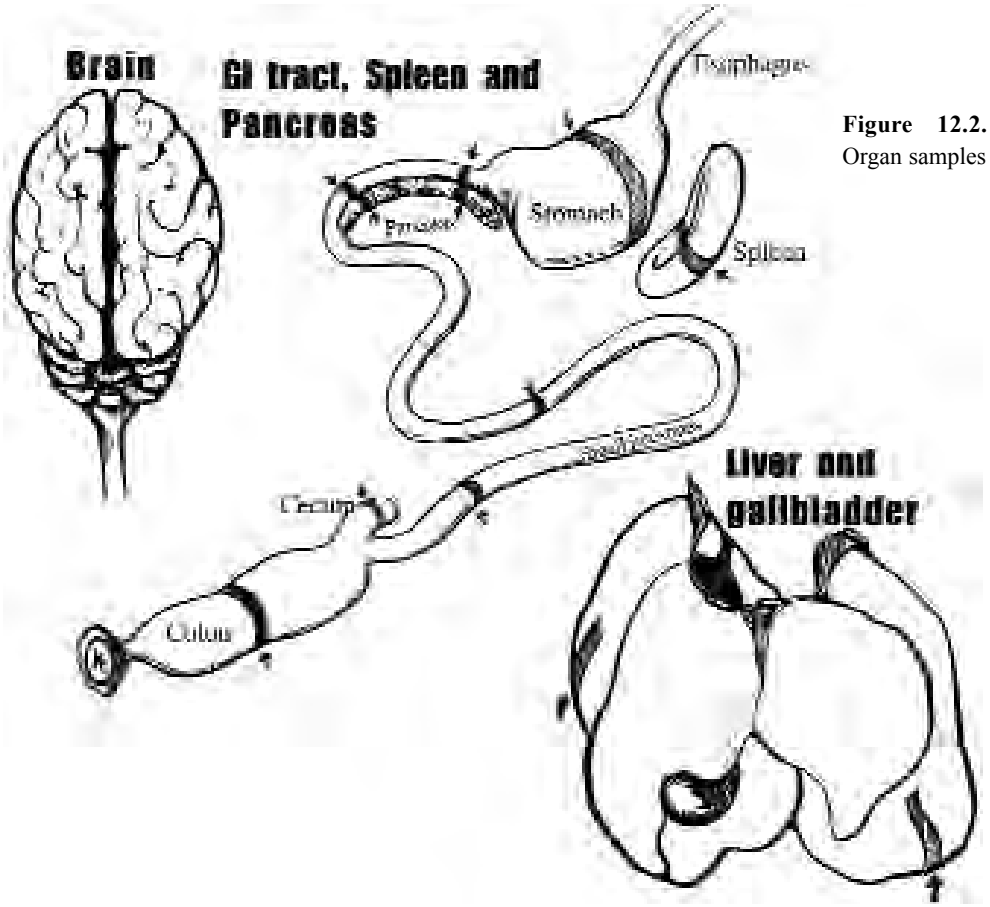


Figure 12.2. Organ samples

Reproductive tract

Remove the reproductive tract (testis if male; uterus and ovaries if female) and make a cut across the gonads and into the uterine lumen before placing in fixative for histopathology.

Kidneys and adrenals

Remove and examine the adrenal glands and take transverse samples for histology (Figure 12.3). Remove the kidneys, examine them for abnormalities and take samples for histology that include the cortex, medulla, and pelvis and take samples for toxicology. Remove the bladder and take a section for histology.

Heart and lungs

Separate the bones of the larynx behind the tongue and dissect out the trachea with esophagus attached. Continue into the chest including the lungs, heart, and large blood vessels. Take a sample from the thymus if present.

Open, examine and sample the trachea and other airways and the esophagus. Take samples from lymph nodes surrounding the airways.

Examine the lungs for lumps and areas of firmness. Take samples of any abnormal areas, as well as areas that look normal.

Open the chambers of the heart and examine the heart valves between the chambers. Take samples of the heart including valve. Open the great vessels and take a sample.

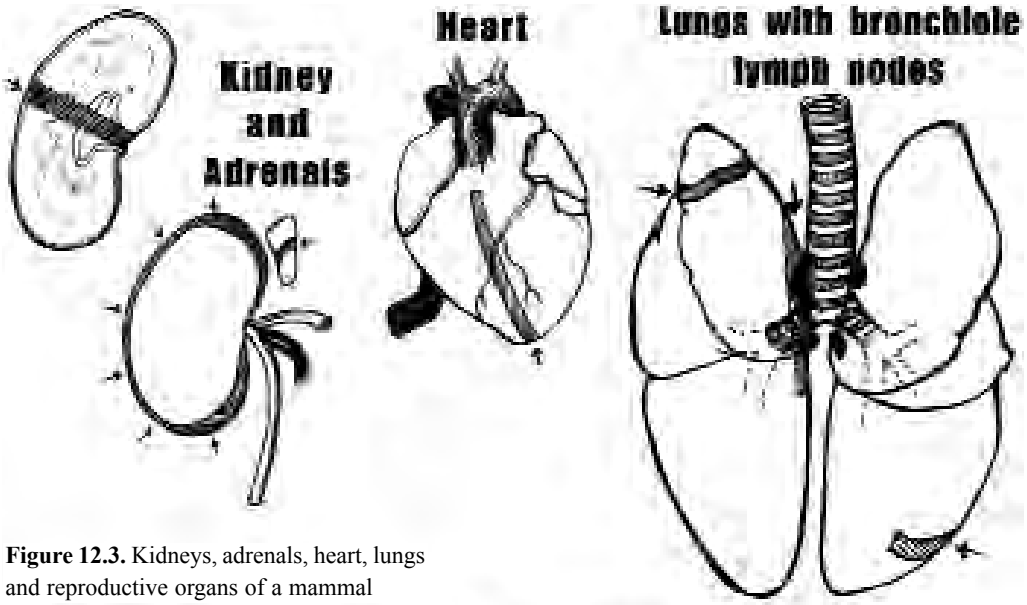
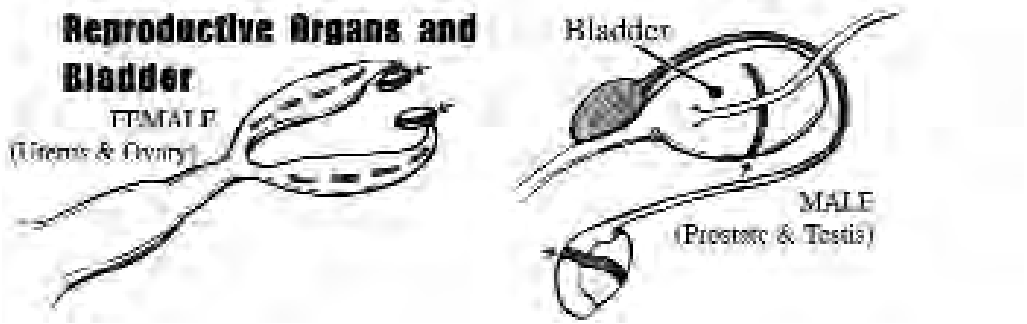


Figure 12.3. Kidneys, adrenals, heart, lungs and reproductive organs of a mammal



Head and oral cavity

Examine the eyes, mouth and nostrils for ulcers and abnormal discharges. Remove an eye for histology by cutting the muscles around the eyeball. Cut between the lower jaw bone and tongue and remove the tongue from below. Examine the inside of the mouth, tonsils, and teeth. Remove the other tonsil and several lymph nodes under the skin at the angle of the jaw and above the larynx for histology.

Brain

To remove the entire brain: Separate the skull from the neck at the junction with the vertebra. Remove the skin from the top of the head then the top of the skull (Figure 12.4).

Remove the brain and cut it in half. Preserve one half in formalin and split the other half into containers for virology and toxicology.

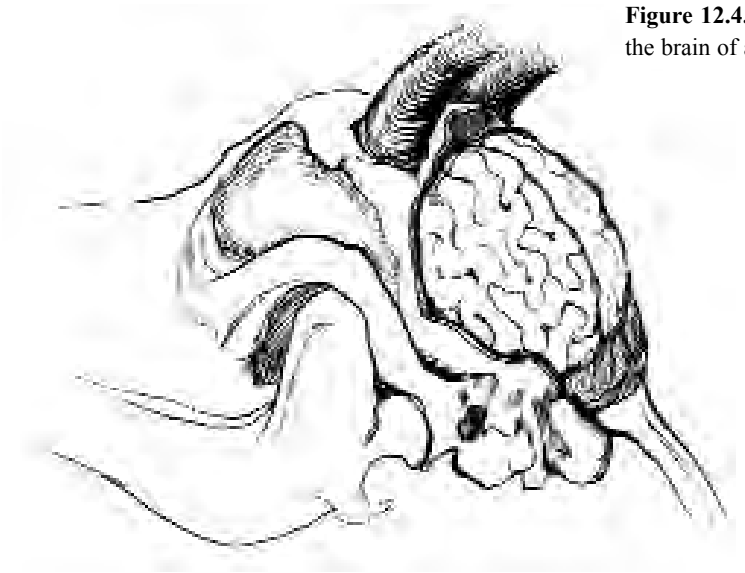


Figure 12.4. Removal of the brain of a mammal

Removing a brain if rabies is suspected.

If rabies is suspected, extra precaution should be used when removing the brain. The person removing the brain should wear a face shield or mask and eye goggles. The safest procedure is to remove the head and insert a drinking straw through the hole at the base of the skull where it attached to the neck. The straw should be inserted in the direction of the eye. Pinch the base of the straw and remove the straw with the brain sample. Then cut the straw (with the brain sample still inside) into 1 cm lengths and drop the sample in either glycerin or formalin. Although this procedure is very safe, it only allows testing for rabies. So if the samples are negative for rabies, no other brain tissues are available to test for other diseases.

The best procedure is to remove the entire brain, then cut it lengthwise down the middle. Send one half to a laboratory to test for rabies and fix one half for histology.

Skeletal Muscles and Nerves

Take samples of the diaphragm and several leg muscles. Take a sample of the large nerve between the muscle bundles of the back leg.

Bone Marrow

Remove a long bone from the leg and crack it open in the middle. Fix one half for histology and store half for microbiology.

Ungulates

The procedures for ungulates are the same as for carnivores, except that all forestomach chambers should be opened (Figure 12.5).

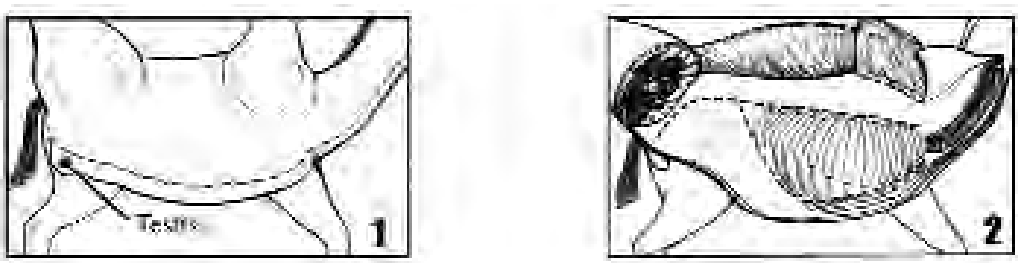


Figure 12.5. Opening an ungulate carcass

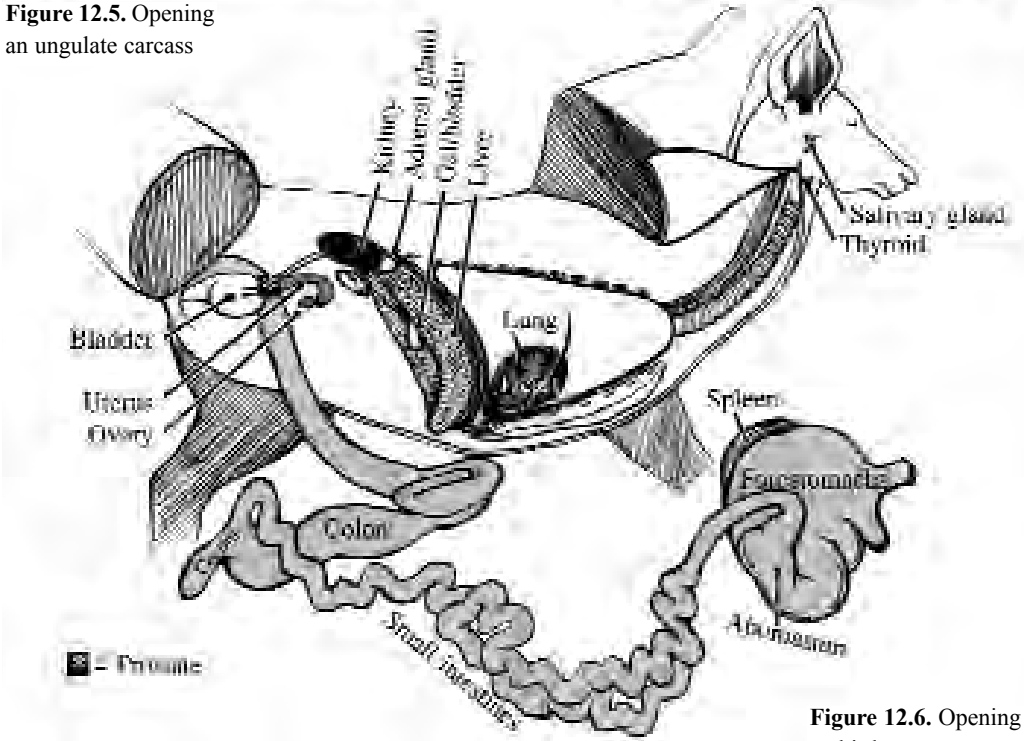
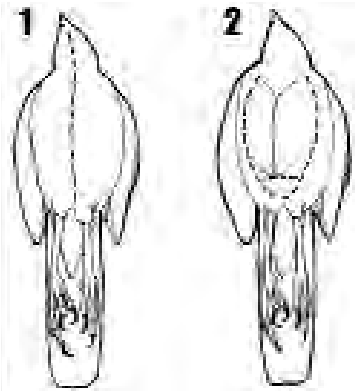


Figure 12.6. Opening an bird carcass



Birds

- THE PROSECTOR SHOULD WEAR A MASK because humans can acquire psitticosis, tuberculosis, and fungal diseases from birds.
- Dip the carcass in water containing a disinfectant or spray it to wet the feathers.
- Examine the carcass for evidence of trauma and ectoparasites.
- Place the bird on its back and open the skin from the beak to the vent (Figure 12.6-1).

- Retract the skin to expose the keel and breast muscles, ribs, and muscles over the lower celomic cavity. Assess the amount of body fat under the skin and in the body cavity. Assess the amount of musculature over the keel (Figure 12.6-2).
- Open the celomic cavity by making a horizontal cut at the bottom edge of the keel extending on each side through the pectoral muscles and then lifting the sternum. Cut the ribs and clavicle near the attachment to the sternum. Open the caudal part of the sternum long the midline (Figure 12.7).

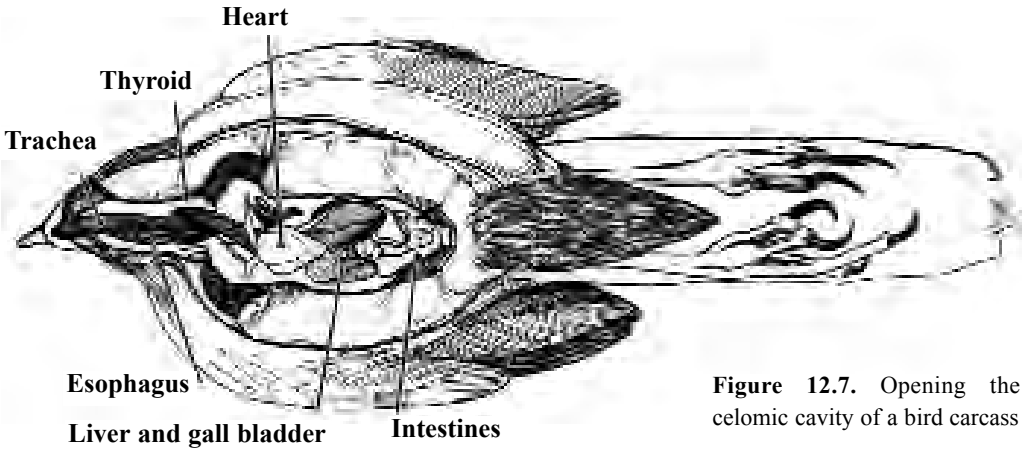


Figure 12.7. Opening the celomic cavity of a bird carcass

- Inspect the location and size of all organs. Examine the air sacs for transparency and note any plaques or opaque areas. Note any abnormal fluids
- Using sterile instruments, take sterile samples of any visible lesions as well as of spleen, lung, and liver.
- Remove the tongue, trachea, esophagus and heart as a unit. Take the thyroid glands for histology (thyroids are located at the thoracic inlet where the major blood vessels above the heart branch). Open the trachea and heart and take samples for histology (Figure 12.8).
- Gastrointestinal tract, liver, and spleen: Remove the liver and take samples for histology and toxicology. Remove the proventriculus and ventriculus (gizzard) and intestines, including the cloaca and bursa of Fabricius. Note the spleen at the junction of the proventriculus and ventriculus. Fix what is remaining of the spleen (after taking a sample for culture) for histology. Open the intestinal tract along its length, noting the content and taking samples for toxicology. Leave the pancreas attached to intestines and take samples
- Lungs: Dissect the lungs away from the body wall (Figure 12.9), examine them for firmness or lumps, and take samples for histology.
- Reproductive tract, adrenal glands, and kidneys (Figure 12.9): The gonads (testes in the male or left ovary in the female) are located in front of the kidneys along the backbone. The adrenal glands are located just in front of the gonads and are also attached to the body below the spine. The female also should have an oviduct visible. Bluntly dissect the kidneys from the body wall, leaving the gonads and adrenals attached. Fix for histology.
- Brain: Remove a section of skull covering the brain, remove a sterile sample (half of the brain, if possible) for microbiology and then fix the rest in the skull (in small birds) or remove the brain from skull before fixing (in larger birds).
- Bone marrow: Remove a tibiotarsal bone and crack it open before fixing.
- Nerves: Take samples from the large nerves between the wing or the leg and the body wall.

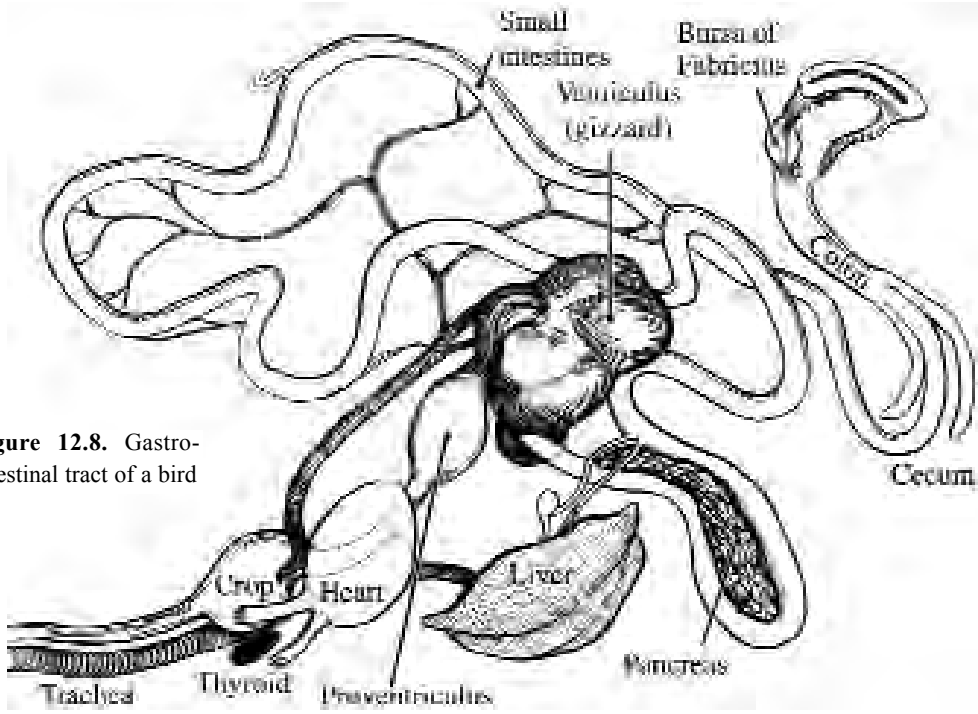


Figure 12.8. Gastro-intestinal tract of a bird

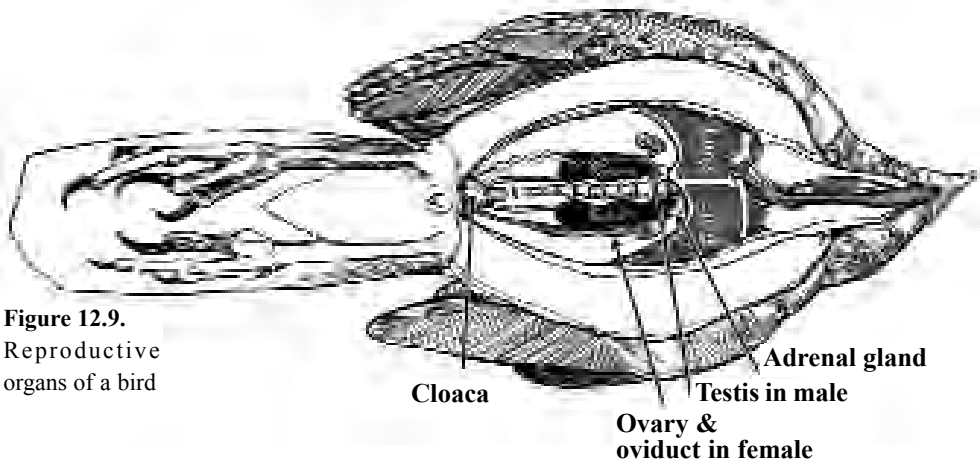


Figure 12.9. Reproductive organs of a bird

Reptiles

The procedure for reptiles is similar to birds. A snake is used as an example (Figure 12.10). Other reptiles have similar organ anatomy, although the kidneys of lizards are further back in the pelvis.

- Place the animal on its back and examine for evidence of trauma.
- Open the body along the midline.
- For turtles, the plastron is removed at the junction with the carapace with a saw.
- Assess the amount of body fat and condition of the musculature and check for any abnormal fluid.
- Take sterile samples of lungs, liver and spleen for culture before handling the tissues.
- Find the thyroid(s) anterior to the heart (single midline organ in some species and paired

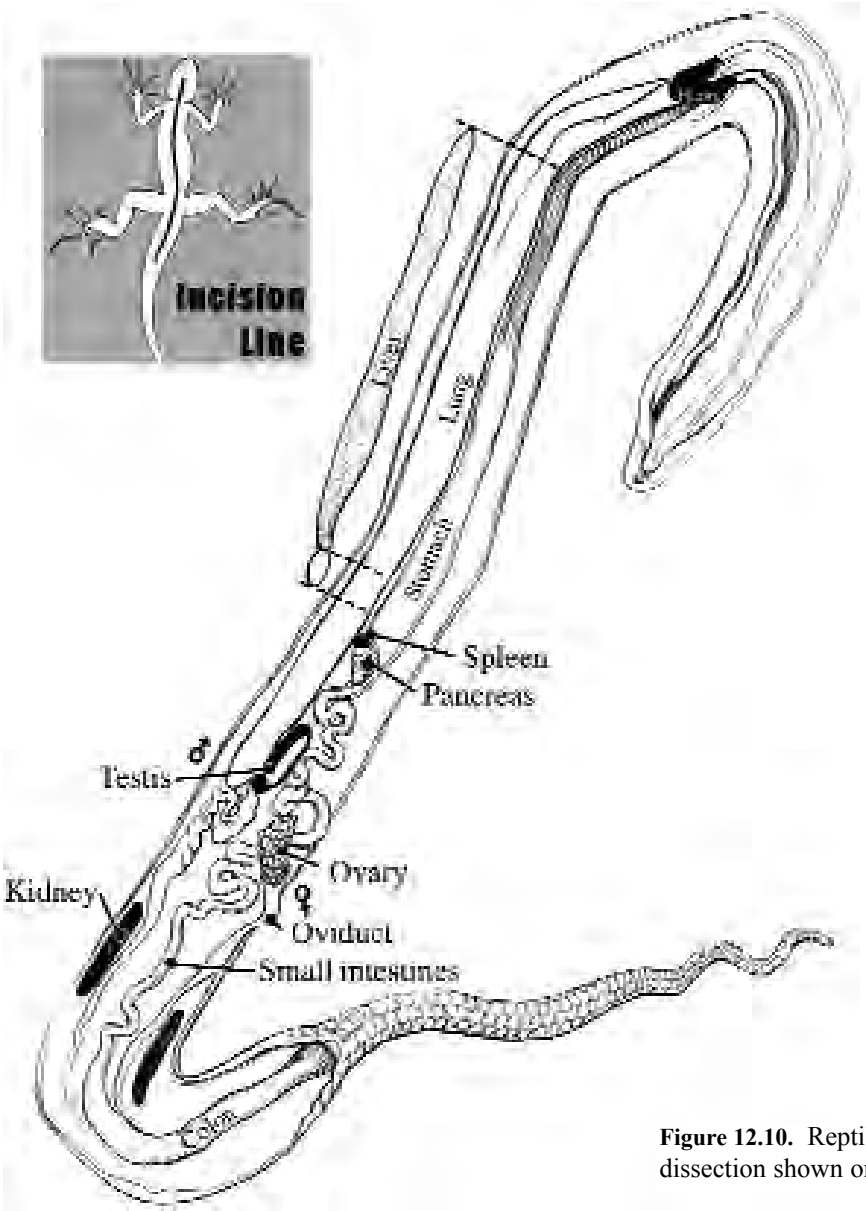


Figure 12.10. Reptile dissection shown on snake

- Beginning at the mouth, remove the trachea, heart and lungs.
- Open the trachea and examine the lungs for firmness or lumps.
- Open the heart. Take samples of all organs for histology.
- Remove the intestinal tract as a unit, beginning in the oral cavity.
- Open the esophagus, stomach and intestines along their length and take samples for histology
- Remove the liver, spleen and pancreas.
- Examine and sample for histology
- Remove the gonads and adrenals (along the midline in front of the gonads). In females, remove the oviduct with the ovaries.
- Dissect the kidneys from the body wall. Take samples for histology.

Post-necropsy

Disinfecting the necropsy site

The carcass and all tissues from the carcass including blood soaked dirt should be buried or incinerated. All contaminated paper or plastic materials should be either thoroughly disinfected or incinerated. All blood and residual tissues should be removed from the instruments and tools with soap and water. Then the instruments should be disinfected. Necropsy boots and apron should be cleaned and any contaminated clothing thoroughly washed. The external surfaces of any containers with samples should also be washed.

Storage or submission of samples

Formalin-fixed samples can be kept at a cool room temperature until shipped. NEVER FREEZE FORMALIN FIXED SAMPLES.

Any samples for culture should be kept refrigerated (for parasitology or bacterial cultures) or frozen (for toxicology or virus cultures). Freezing at -70°C is preferable to -20°C (standard freezers).

TO SHIP SAMPLES:

1. Contact the laboratory before shipping.
2. Check regulations for shipping tissue samples. Get proper permits and use the correct containers.
3. Frozen samples must be shipped in insulated containers and by express carrier. Pack specimens in dry ice or with ice blocks. Seal container to prevent leakage. Include proper permits and animal identification.
4. Formalin-fixed tissues should be fixed for at least a week in 10 times as much formalin as tissue. Then most of the formalin can be removed and tissues can be wrapped in towelling soaked in formalin for shipping. The tissues soaked in formalin can then be placed in leakproof containers for shipping.

IT IS BEST TO SHIP FROZEN AND FIXED SAMPLES SEPARATELY. IF THEY MUST BE SHIPPED TOGETHER, then insulate the fixed tissues from freezing by wrapping in newspapers. Assure that there is no spillage of formalin, because fixation of frozen samples will make culturing for bacteria or viruses impossible and will alter cells on blood smears or cytology slides.

Dealing with an Epidemic

- Complete the FIELD REPORT OF WILDLIFE DEATH form found in Box 12f.
- Contact appropriate local or national governmental personnel.
- Collect samples from as many dead or affected animals as possible.
- Collect the following information on the epidemic:
  1) Species and approximate number affected
  2) Signs the animals are showing
  3) Location of affected animals

Box 12f FIELD REPORT OF WILDLIFE DEATH

Date: _____

Person reporting: _____

Affiliation: _____

Address _____

_____

_____

Telephone #/Fax/E-mail _____

Species _____ Age/Sex _____

Clinical signs noted before death:

Number affected: _____

Other species in the region/Number affected:

Location where carcass(es) found: _____

Environmental conditions:

Other information / observations:

CHAPTER 13
Methods for assessing the status of animal populations

Lee White & Ann Edwards

Conservationists working in the rain forests of central Africa (and to a lesser extent in east and west Africa) are generally dealing with large, remote, little known protected areas. Conditions of work and observation are difficult, and hence research and monitoring is relatively expensive. The most efficient survey method for large areas of tropical rain forest is using line-transects (e.g., Barnes & Jensen, 1987; Buckland et al., 1993; Tutin & Fernandez, 1984), or the reconnaissance modification of line-transect sampling (Hall et al., 1998; Walsh & White, 1999). We want to identify major patterns of abundance, distribution and seasonal movements of key species, in relation to human presence and habitat; evaluate effectiveness of protection in forest parks; identify areas of conservation concern (e. g., zones of high human impact or faunal concentrations); and provide a basis for monitoring ecological changes and patterns of human use. This chapter will describe the use of the line-transect and other related methodologies for animal surveys and describe how to collect complementary information on vegetation and human sign that will help in interpretation of results. Our emphasis is on larger-bodied animals, since these groups are a preoccupation for many wildlife managers and researchers in African rain forests. However, the techniques described are equally appropriate for other taxonomic groups, such as frogs or butterflies. The chapter is divided into two main sections, first describing general surveys in little known areas and then presenting a number of methods of use for more detailed studies.

General surveys of little known areas

Why focus on large mammals?

Surveys undertaken by wildlife managers working in the African rain forests tend to concentrate on large mammals, particularly elephants, apes, duikers and monkeys. These focal animal groups are chosen because:

- these animals and their sign are most readily visible;
- people locally, nationally and internationally are especially interested in large, charismatic, mammal species;
- they tend to be the most heavily hunted animals and are therefore of special conservation concern;
- they tend to be a good index of the overall integrity and conservation status of a region;
- there may be conflict between large mammals such as elephants, gorillas, leopards or bush pigs and people living in and around protected areas;
- methods have been developed and widely field tested to assess large/medium mammal abundance and distribution.

The pioneering work of Tutin & Fernandez (1984 - gorillas and chimpanzees) and Barnes et al. (e.g., 1991 - elephants) concentrated on one or two focal species and documented their distribution and abundance on a national scale. More recent surveys have tended to try to collect data on a diverse array of species. You should aim to follow this trend when possible. Once you have defined your research question and methodology you will need to undertake a preliminary survey. However, before you begin a preliminary survey think carefully about how to stratify your study area.

Stratification.

Once you have compiled relevant background information about your study area and species, stratify your study area (see Chapter 3 for more details). For example, elephant densities are greatly influenced by human activity (e.g., Barnes et al., 1991; Fay, 1991; Hall et al., 1997 - see Box 3.2) and it is likely that other large mammals will be similarly influenced. Hence the location and intensity of human activity will be one of the main features of any stratification. The following factors should be considered:

- locations of human settlements (note that these are not always near roads);
- areas around extractive activities such as gold mining, commercial meat trading, logging;
- existing or past roads or trails used by humans for movement between centres of population;
- areas of past settlement where old secondary vegetation may result in increased densities of gorillas, elephants and some other species (Barnes et al., 1991; Tutin & Fernandez, 1984);

Also consider major forest types when stratifying your study area (for example, monodominant stands, mixed forests, hill regions, savannas, extensive swamps, major river valleys) since these naturally occurring features affect animal densities, distribution and seasonal movements.

As a rule of thumb you should aim to:

- stratify according to ***human density*** by sampling randomly within strips or concentric bands 10 km wide moving away from settlements, human activities and access routes.
- sample each vegetation type, including secondary vegetation, in each stratum.
- ensure thorough geographic coverage by sampling in all parts of the area of interest as far as possible.
- sample each stratum or block in proportion to the expected number of animals (a product of density, determined during a pilot study and the surface area of the zone). That is, if there are two strata in your study area, one with double the expected population, you should plan to cut twice as many transects in the stratum with the larger population.

A good deal of background knowledge is required in order to classify and stratify an area using the criteria. If available, maps, air photographs and satellite images should be consulted. Older maps may indicate the locations of trails and villages long since abandoned. If these aids are of poor quality or unavailable then your preliminary survey will need to collect the relevant information. In addition, survey methods should include records of features, such as vegetation type or frequency of human sign, which will allow refinement of the stratification (see below).

Once a (preliminary) stratification has been undertaken, transects should be located from a randomly selected starting point within each stratum. They should be oriented to cross major drainage features in order to sample a representative proportion of all vegetation types (Norton-Grifiths, 1978).

Preliminary survey

Before embarking on a large scale or intensive line-transect sampling programme you should first gather as much existing information as possible on your study area and species (see Chapter 2). Next you will need to undertake a preliminary survey to investigate your study area and to test and familiarise yourself with the methods you propose to use. This generally involves a combination of talking to people who have a good knowledge of the area or species and reconnaissance trips into the forest, often accompanied by a guide, following existing roads or trails (see Box 13a).

Box 13a A SURVEY FOR THE RARE *CERCOPITHECUS SCLATERI* IN THE NIGER DELTA AREA: USING INTERVIEWS TO DETERMINE DISTRIBUTIONS.

In 1902 a young male monkey of unusual appearance, said to come from Benin, was given to the London Zoo. It was recognised as a new species and described as *Cercopithecus sclateri*. Until recently, the only information on this species' natural habitat was a museum specimen collected in 1934, said to come from 'orchard bush country' in Okigwi Division, Owerri Province, Nigeria, between the Niger and Cross rivers. In 1982 John Oates and Pius Anadu noticed a monkey fitting the description of *C. sclateri* in Port Harcourt Zoo. In 1987 an attempt was made to locate a surviving population of this species (Oates et al., 1992). Oates, Anadu and Lee White travelled to Okigwe (as Okigwi is now known) but found a landscape densely populated by humans with no rain forest vegetation, the presumed habitat of *C. sclateri*. They gradually expanded the area of their search and ventured into the delta of the River Niger.

Once in the forest zone John Oates and his colleagues used an interview technique to assess if local people, particularly experienced hunters, were familiar with *C. sclateri*. One has to be careful when interviewing people since they often give misleading information, either because they are eager to please and hence provide answers they think you wish to hear, or because they are suspicious of your motives and intentionally try to mislead you (see Chapter 15). In each village John first made contact with the chief and elders and offered some cola nuts as a sign of respect and friendship. Then he explained his general interest in monkeys and asked if any hunters in the village could describe the different kinds of monkeys which occurred in the region. Once the hunters had been called to the meeting John would begin to question them. In a small village near Oguta Lake in Imo State the interview went something like this:

First John asked the hunters to name all the monkeys they knew in their local language, Igbo. Both John and Pius had a long experience of working in the area (Pius is, in fact, an Igbo speaker) and they were familiar with the local names for different monkey species. However, this time a new name, *amina*, was cited, which hightened their interest. Having made a list of names they asked further questions about the monkeys. Which were the biggest and the smallest? What colour were their coats? Were there any distinctive features about their faces? All of these pieces of information are known to observant hunters, so they can be used to evaluate their knowledge as well as to decide which name in the local dialect corresponds to each species. To the amusement of everybody, John then asked a hunter to imitate the calls of the different monkeys. The hunter with the best knowledge was shy, so to break the ice John imitated the call of the commonest species, which he was already convinced had been well described. The meeting roared with mirth, but confirmed that he had indeed imitated a mona monkey. The hunter, happy in the knowledge that he could imitate the monkeys better than John, then made the calls of all the species. For the species which had not been convincingly described John began to ask more pointed questions, until he had decided the identity of all species except amina. He asked many questions about this species, which it seemed was quite rare, but could be observed in swamp forest a few hours walk from the village.

John concluded the interview by asking if the monkeys had changed in abundance in recent years, which were preferred as food and if there were any that people did not eat. Finally, once the interview was complete he showed the assembled villagers pictures of monkeys from a mammal guide and asked if they could recognise any of the ones they had named. Some hunters are good at this, but many have had little experience of looking at pictures or photographs and they often identify species which cannot possibly be found in their region as being common around the village. You would be surprised how many hunters identify lemurs from Madagascar as occurring in the Niger Delta!

John and Pius later returned to the village in question and were taken to see amina, which proved to be *C. sclateri*, as they had guessed. Since then they and their colleagues have located a number of populations and Lodewijk Werre, one of John's PhD students, is now using line-transect methods to estimate densities of *C. sclateri*, in parts of the delta.

Transect vs. patrol based surveys

Traditionally mammal surveys in the African rain forest have been conducted using line-transect methods. These methods are generally considered to be the most efficient means of sampling large areas. However, use of line-transect methods in the rain forest involves cutting lines through the vegetation following compass bearings. Progress tends to be slow. As a result line-transect surveys are time-consuming and relatively expensive. Furthermore, there are dangers associated with cutting trails into isolated protected areas (Box 13b).

In the last few years trials of a new methodology, reconnaissance (or "recce") surveys have been undertaken. During a recce survey the observers follow pre-existing trails whenever possible and when necessary cut a path of least resistance through forest vegetation. The recce method allows observers to work in smaller teams (hence it is cheaper), to travel faster (hence to cover more ground) and to minimise the potential impact of their activities in the area. Furthermore, the recce method can also be used during anti-poaching patrols, which tend to proceed in roughly the same manner, permitting data collection for monitoring key animal species alongside routine protection activities.

Recce surveys have the disadvantage that they produce biased data because following existing human paths and animal trails will not give a representative sample of habitat types and human impact in the area. For elephants it has been shown that correction factors can be calculated to account for the bias and that densities can be most efficiently calculated using a combination of line-transect and recce data (Walsh & White, 1999). As further trials of the recce method are undertaken and the methodology is refined it may become more widely used than transects. For the moment it has to be considered to be a method which is under development and in this chapter we will concentrate on line-transect sampling. However, data collection on recces is very similar to that undertaken during line-transect sampling and most of the aspects of line-transect sampling we will describe can equally be applied to the recce method. The recce method is described in detail below.

Box 13b THE DANGERS OF LINE-TRANSECT SURVEYS

Whilst transects are an efficient way to gather data you must bear in mind that they can also have a negative impact on the system you are trying to study or protect. *These considerations should be taken into account when planning a survey and choosing to use line-transects or recce walks.* Negative consequences of line-transect surveys can include:

- penetration of remote and little known regions may show local hunters the way into areas where they have never hunted before;
- the trails may be adopted by hunters after you have left, providing access which results in the death of the animals you were surveying;
- in liane forest the cutting of a narrow transect can have a disproportionate effect on the forest, since cutting a single liane stem may kill a large area of canopy above. Be sure that your transect cutters appreciate that they should penetrate with a minimum of cutting and that they should avoid cutting young trees and lianes whenever possible;
- untrained transect cutters often cut big blaze marks on trees, which sometimes allow infections to become established, eventually causing the death of trees. This should be avoided, particularly if you plan to undertake botanical monitoring along the transect.

In a study undertaken in Nouabalé-Ndoki National Park in northern Congo, a team of researchers cut a set of 20 parallel transects and recces. They then counted the number of stems cut in four different vegetation types along the transects and recces and found that transects consistently resulted in at least four to five times the damage than recces, irrespective of vegetation type or class of plant. In dense vegetation 12.9 stems were cut for every metre of transect, but only 1.5 stems were cut on the same length of recce. Hence recces resulted in less damage to vegetation and and were much less visible, reducing the chances that they would be used by poachers.

Practical details

How to cut a transect

Two people are needed to cut a transect, one of whom can use a sighting compass. Choose a starting point. Cut a stake which, when planted into the ground, is at eye level. Place the sighting compass on the stake and orient it to the correct bearing balanced so you can sight through it without touching it (Figure 13.1). One assistant then traces a path away from the compass person. The assistant should cut a minimum amount of vegetation to mark his or her path. Untrained transect cutters generally cut far too much vegetation, slowing the rate of progress and increasing the ecological impact of the transect sampling (see Box 13b).

Figure 13.1. Using a sighting compass to direct an assistant

Cutters must be carefully monitored by the compass person. Each time the cutter deviates off the path the compass person should immediately call out a correction (left or right). The compass person has to be very strict since cutters often try to deviate along animal trails running in approximately the same direction as the transect. This will result in your collecting biased data (see Box 13c). When it becomes difficult to see the cutter tell them to stop, check that they are standing in the centre of the line, and move forwards with compass and stake to where they are waiting (they must not move in the meantime).

A well cut, straight transect is crucial for good data collection. The method described here has been tried and tested in many differing circumstances and gives consistently good results when the compass person is well trained. Other methods, such as choosing a target tree which falls on the bearing and then cutting towards it without sighting down the compass, invariably result in numerous slight deviations.

Permanent or one-off transects?

When planning your transect sampling you will be faced with the choice of undertaking many one-off (used only once) transects or establishing one or more permanent transects which will be used repeatedly over time. Your decision will depend upon the animals you are planning to survey and the aims of your study. For example, Tutin & Fernandez (1984) used many one-off transects on their nationwide survey of gorillas and chimpanzees using nest counts, because the wanted to obtain an overall number for the whole of Gabon and they neither needed to nor were logistically able to repeat transects. George Whitesides (1987) on the other hand, chose to use a permanent transect to census monkeys in a small study area on Tiwai Island in Sierra Leone, because one line was sufficient to sample all the habitat types in his study area. In addition, had he chosen to use one-off transects, the noise of transect-cutting would have scared away the monkeys he was hoping to census. His one transect was kept clear, such that he could move along it stealthily and count monkeys before they detected him and moved away.

As a rule, if you are planning to undertake direct counts of animals such as monkeys or hornbills, well-maintained permanent transects in restricted areas are probably of most use; whilst if you will be counting sign such as dung or nests (which will not flee as you approach) and particularly if you want to survey a large area, one-off transects are probably the best option. Of course, it may be worthwhile to adopt both strategies, undertaking repeat counts on one or more transects easily accessible from the base camp, in addition to a series of one-off transects in more remote areas. If you choose to use permanent transects you might use the same lines to undertake botanical studies, in order to look for correlations between vegetation type and animal density or movements.

What type of information to collect?

The type of information you choose to collect will obviously depend on the research question you plan to answer. However, for any given question there may be several types of information that can be collected. For any given animal species several census methods may be possible. For example chimpanzee numbers can be evaluated using direct observation of groups of chimpanzees, by counting nests, by counting dung, and / or by noting the frequency of feeding trail or foot and knuckle prints. Furthermore, the frequency of human sign, or the vegetation type, may have a direct bearing on chimpanzee numbers, so it may be important to collect this information at the same time, or to conduct a second study on the same transects at a later date. It is difficult to give detailed guidelines about what information to collect, since this will vary as a function of the aims of your study and the density of animals. For example, in an area where hunting is intense you may never see red river hogs (*Potamochoerus porcus*) but you may occasionally see dung or sign, which can be used as an index of abundance; in an area where hunting pressure is low you may see groups of red river hog often enough to use sightings to estimate density, and there may be so much sign that it would take far to long to record each footprint or dung pile.

How large a sample is enough?

As a rule of thumb, in order to make a density estimate, you should obtain at least 40 and preferably 60-80 or more encounters on your transects. A short pilot study can help you to assess what length of transect is likely to give this minimum.

How long should transects be?

There is no all-encompassing answer to this question. Inevitably there is a compromise between increasing the number of transects and increasing the number of encounters on each transect. By increasing the number of transects you increase the chances that your sample will be representative of the real density (i.e., increased accuracy - see Chapter 4). By cutting longer transects, thereby increasing the number of encounters per transect, the variance of your sample will be reduced. If working in a remote area it is more convenient to do fewer longer transects, because less time is lost travelling between them.

Who should walk the transects?

To collect reliable transect data a researcher or team of researchers need to be able to:

- reliably identify animals by sight and sound;
- reliably identify animal sign;
- follow a transect, and know locations along that transect;
- identify habitat types;
- measure or estimate distances accurately;
- record information reliably; and
- do all of the above while walking slowly, quietly and carefully over long distances.

How many people should walk the transects at one time?

The number of people who should walk a transect at any one time depends on whether permanent or one-off transects are being used, how many animal species are being censused at the same time, and what additional information is being collected.

Permanent transects, once established, are best walked by one or two observers. If possible, data should be collected by the same individual or team each time the transect is repeated.

On one-off transects cutters pass first in order to create the transect. If logistically possible, it is ideal for the data collection team to pass a few days later, when the disturbance caused by transect cutting has passed. Then data can be collected both for sign and sightings. In this case send a sighting person (or team) first, followed by a person (or team) collecting data on sign. This is not generally possible and transect cutting usually takes place as the whole team advances. Porters should travel last - they can be left to tidy up the camp after breakfast and rejoin the research team towards the end of the day to make the next camp.

The number of observers will depend on the number of sets of data you plan to collect. If you are undertaking an elephant survey using dung counts a single observer may be able to collect all the necessary information. Alternatively, if you are planning a large mammal survey, recording data for many different species, several observers may be necessary, each with a separate task.

For example, three observers might divide tasks as follows:

- Observer 1 - dung and sign;
- Observer 2 - ape nests;
- Observer 3 - human sign, topography and vegetation and any observations of animals.

Local trackers tend to be good at spotting sign, but have short concentration spans resulting in data of variable quality. Therefore all data recorders should be experienced at looking for sign. Working in such a team is a good training exercise. Some human traces are best spotted by the transect cutter. Therefore, this person should be trained to draw researchers' attention to old cuts or other sign.

Time of day or night

It is best to walk transects when animals are the most active because when they are active they are more easily seen or heard. It is best to census monkeys, birds and duikers in the early morning, from the time it is light enough to see animals until the heat of mid-day makes animals more quiet. Animals also can be censused in the late afternoon when they become active again. When counting animals (not animal sign) walk the transects at the same times each day.

The activity schedules of nocturnal animals are more variable than diurnal animals because they can be highly affected by the phases of the moon, as well as by time. Therefore, in an attempt to observe animals during all possible hours of the night, night censuses should be walked at each of the following times: early night, middle of the night, and early morning before sunrise, and during different phases of the moon.

Seasonal effects

Transects walked in different seasons can give different results. This is particularly true of animals that migrate from one area to another in search of food or water during different seasons. When animals are known to move seasonally it is important to walk transects during different seasons. If you want to compare results between different areas, the transects in all the areas should be walked during the same season (see Box 13d).

Changes between the wet and dry seasons can have a strong effect on the quantity and quality of animal sign that are visible. For example, dung piles, nests and burrows may disappear more rapidly in the wet season than in the dry season, but falling leaves in the dry season can cover animal sign. Tracks are usually more visible in the wet mud of the wet season, but they may disappear daily with each strong rain.

If you are going to census animals or animal sign only once a year, you should always do it in the same season.

Weather conditions

It is impossible to census animals efficiently while it is raining or at a time when there are high winds, since the noise made by falling water or swaying branches conceals that of animal movements. Always note weather conditions at the beginning of the census as well as any changes during data collection.

Guidelines for Observing Animals While Walking Transects

- Transects should be walked at a speed of 1 km/hour for sighting censuses and about half this for dung and nest counts.
- Stop to listen for animals every 50 to 100m. This will help you to hear the animals before they hear you.
- Do not smoke or talk too loudly while walking transects.
- Do not leave the trail to follow any animal or to count animals beyond your view. Leaving the trail violates the assumptions of transect sampling and may alter the results.
- It is acceptable to leave the trail to measure perpendicular distances to animals, or to investigate rare sightings, such as a leopard kill, but it is best first to advance along the transect 10-20m collecting data, and then to return to make supplementary observations. If you collect information off the trail, clearly record in your notes that it was collected off the trail. Return to the trail to continue the census as soon as possible.
- The same animal (or group of animals) must not be recorded twice on the same transect, even if the animal runs ahead of the observer on the transect. If you are not sure it is the same animal, then record your observations, and make a note of your uncertainty.

Equipment needed

Equipment needs for line transect surveys are quite simple:

- for transects away from a base camp you will need standard camping and medical supplies and sufficient food;
- maps of the area;
- compass;
- wrist watch;
- altimeter;
- clinometer;
- notebook and checksheets;
- binoculars;
- pens, pencils, erasers etc.;
- hip chain and topofil thread;
- tape measure;
- GPS unit;
- secateurs or machete;
- light plant press;
- torch;
- assorted specimen bags and bottles;
- permanent marker pen;
- flagging tape;

Box 13c HOW DO LINE-TRANSECTS WORK? THEORY AND COMMON PROBLEMS

Line transect theory: fall-off distances and effective strip widths

Line-transect sampling is based on the assumption that the probability of detecting an animal, nest or dung pile decreases with increased distance from the line of travel (Figure B13.1). Put simply, you are more likely to see a pile of elephant dung that is underfoot than one that is 12 m from the transect hidden in lush vegetation. The probability of seeing objects at different distances will depend on the density of the vegetation - on a logging road devoid of vegetation you may see an elephant 300m away, whereas in dense Marantaceae Forest you might walk past a resting elephant which is only 5m from you.

Line-transect analyses calculate animal densities (or densities of sign) by using perpendicular distance measurements to estimate a function describing the probability of seeing animals (or their signs, such as dung or nests) at any given distance from the transect line. Different researchers have used different assumptions to estimate this function. One could estimate

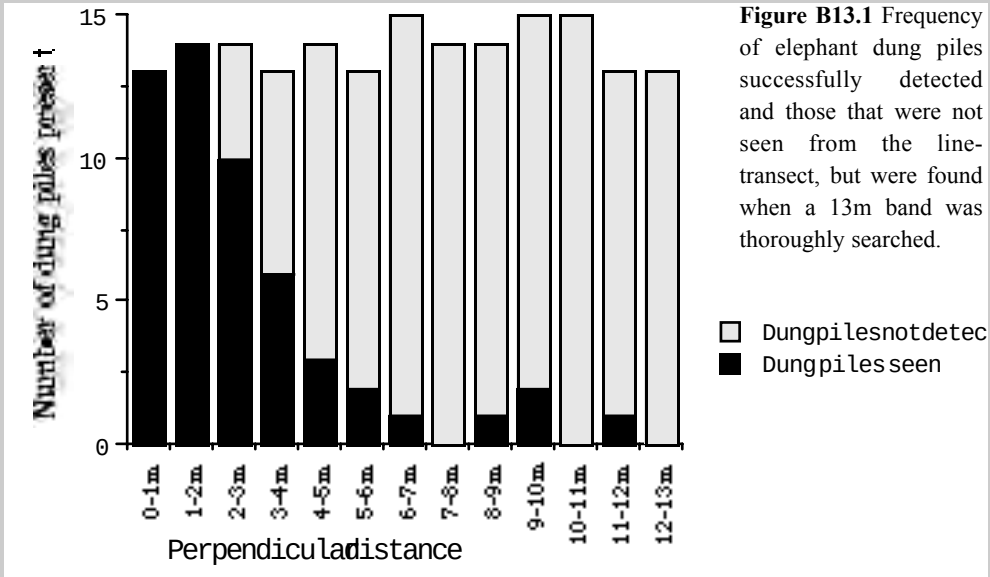


Figure B13.1 Frequency of elephant dung piles successfully detected and those that were not seen from the line-transect, but were found when a 13m band was thoroughly searched.

density by considering only observations from the region close to the transect where all objects are detected (Kelker, 1945). The width of this region can be estimated from a histogram such as that in Figure B13.1 where the fall-off distance would be 2m (the distance beyond which the number of objects detected per distance category declines, called the fall-off point). However, this results in a significant proportion of the data collected being discarded (all observations beyond the fall-off point), and is an inefficient way to use hard-earned information. Other methods make use of data beyond the fall-off point to calculate an 'effective strip width', where the number of objects missed near the transect is balanced by those seen further away (Gates, 1979).

Line-transect analyses rely on the distribution of perpendicular distance measures along the transect to calculate a detection probability (e.g., Buckland et al., 1993). Testing of numerous data sets has shown that it is most efficient to discard the 5% of points furthest from the centre-line. The width sampled on either side of the transect is then taken to be the distance of the last remaining measure - known as the 'critical width'. A mathematical model is fitted to the data to calculate the 'detection probability' - the probability of seeing an object dropped randomly into the area sampled.

Once the detection probability has been calculated the density is calculated by dividing the number of objects detected (N) by the area of the detection zone and the detection probability. The area of the detection zone on each side of the transect is just the critical width (w) multiplied by transect length (l). This gives a density of:

$$D = N / (2lwp),$$

where the denominator is multiplied by two because there is a detection zone on each side of the transect.

A simple method for calculating density from line-transect data, which does not require a computer to make calculations, is described in Box 13f. Other more rigorous statistical methods require complex calculations and are described in detail by Buckland et al. (1993).

A computer program called "*Lopes*" written by Peter Walsh has been developed to enable users of this manual to undertake these analyses. If it was not included with your manual it is available free of charge from the Africa Program of the Wildlife Conservation Society. A second, more complex computer program, called *DISTANCE*, is also available to undertake these analyses (Laake et al., 1994). This program can be obtained free of charge from The Colorado Cooperative Fish & Wildlife Research Unit, 201 Wagar Building, Colorado State University, Fort Collins, Colarado 80523, U. S. A.

Assumptions of line-transect sampling

Three assumptions are critical to achieving reliable estimates of density from line-transect sampling:

- *Objects on the line are detected with certainty.* That is, no objects falling on or over the centre line are missed. This assumption is generally fine for objects on the ground, but be sure to look carefully for monkeys or nests in trees directly above your head as it is easy to miss these individuals and this will bias your data (see Figure B13.2).
- *Objects are detected at their initial location.* This is fine for sign, but if your study animal either moves towards you to investigate you, or if it avoids you by moving away, your data may be biased (see Figures B13.2 & B13.3)

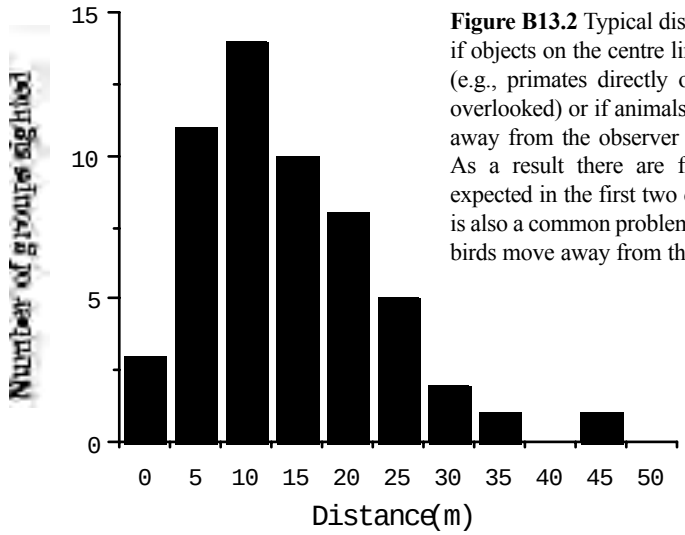


Figure B13.2 Typical distribution of observations if objects on the centre line are frequently missed (e.g., primates directly overhead freeze and are overlooked) or if animals on the centre line move away from the observer before they are spotted. As a result there are fewer observations than expected in the first two distance categories. This is also a common problem for spot counts because birds move away from the observer.

- *Measurements are exact.* Unless measurements are recorded precisely using a measuring tape or some other means of measurement, your estimates may be biased and will lack precision. Always record data as precisely as possible. Although data are lumped into distance categories for display purposes and in the simple analysis described in Box 13f, statistical models are less powerful if data are lumped when recorded. For example, if an elephant dung pile is 92 cm from the centre line be sure to record 92cm not 1m as the perpendicular distance.

A common error of inexperienced observers is to record all distances within about 50-100cm of the mid-line of the transect as 0m (because they fall in the area affected by cutting) and only to measure distances beyond the cut area. *This is wrong and can have a very strong, negative impact on the accuracy of density estimates.*

When distances are estimated (and often even when measured) a common problem is 'heaping'. Observers tend to estimate distances as convenient rounded numbers such as 0, 5, 10, 50, 100 and rarely record numbers such as 3, 4, 7, 8, 11, 19, 49, 101 etc. One way to avoid this is to decide where the mid-line of the transect and the centre of the object to be measured lie before looking at your tape measure.

Another problem with estimated distances is that observers often systematically under- or over-estimate, thus introducing a consistent bias. *Always measure distances if possible. Otherwise, test your precision at regular intervals by estimating distances and then measuring them.* This can be a very useful team exercise with a small prize for the best observer. Make a plot of your estimated distances against the actual measures and check for systematic bias.

Poorly cut transects can bias density estimates

Another common problem is that when tracing a transect through dense undergrowth untrained cutters tend to deviate from the straight line on game trails running conveniently close to the direction of travel. Compass people have to be trained to correct cutters immediately they start to deviate and to bring them back to where they left the line. If cutters deviate frequently from the line along game trails this results in a greater proportion of objects being recorded on the centre line than would be the case if the line was true (see Figure B13.3). This will not be serious for primate censuses where a deviation of 1m or so will have little influence on distances recorded, but for dung counts this can lead to serious bias. The result is the same if permanent transects are adopted by animals.

Hints to help you avoid falling into these traps

- Always measure perpendicular distances to the nearest centimetre.
- Check your or your teams measurements by looking at the last digit of the perpendicular distances recorded. If there are a disproportionate number of measurements ending in 0 or 5 observers have not been reading the tape measure accurately (see Figure B13.4).
- Inspect values between 0 - 1m for dung counts. If there are many more observations recorded as zero (0cm) than other values between 1 - 99, particularly if there are no values recorded between 1 - 50cm, observers are lumping all objects falling in the cut area of the transect as being on the centre-line.• Regularly plot frequency distributions of your or your team's perpendicular distance measurements as described in Box 13f, to check that they do not have follow either of the patterns shown in Figures B13.2 and B13.3.

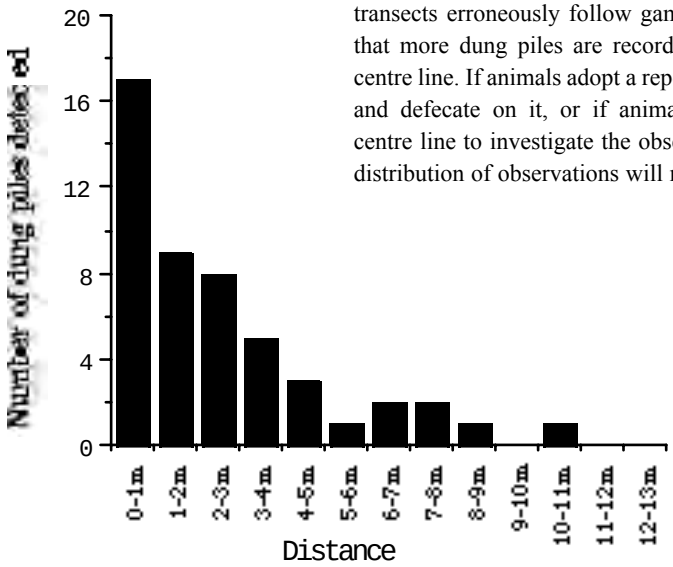


Figure B13.3 Typical distribution of observations if transects erroneously follow game trails. The result is that more dung piles are recorded on or close to the centre line. If animals adopt a repeated transect as a trail and defecate on it, or if animals move towards the centre line to investigate the observer, the same biased distribution of observations will result.

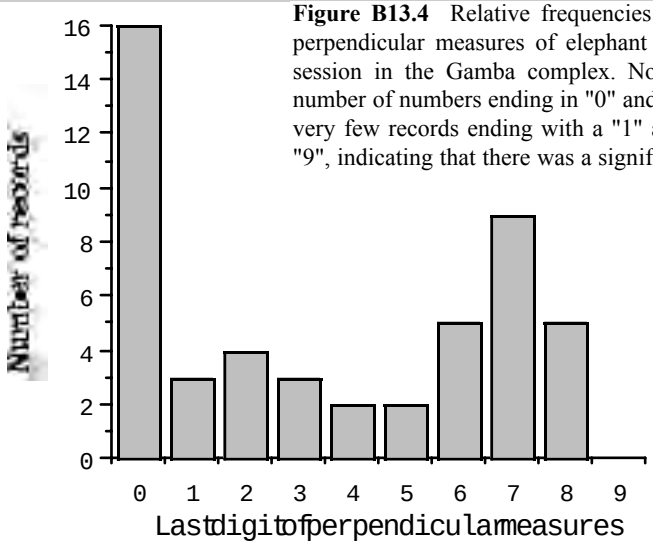
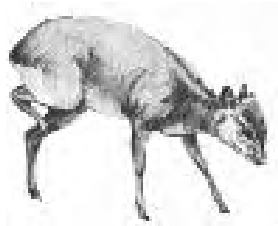


Figure B13.4 Relative frequencies of the last digit of 49 perpendicular measures of elephant dung during a training session in the Gamba complex. Note the disproportionate number of numbers ending in "0" and the fact that there were very few records ending with a "1" and none ending with a "9", indicating that there was a significant heaping error.



Blue duiker
Cepholophus monticola

Box 13d SEASONAL CHANGES IN ELEPHANT DENSITY: LESSONS FOR DESIGNING AND INTERPRETING LARGE MAMMAL CENSUSES

During a study of the effects of logging on wildlife, White (1992) established a 5km line-transect in an area that had recently been logged and a second line in an adjacent unlogged area. The logged forest happened to fall in an area where a large forest tree, *Sacoglottis gabonensis*, was common, but did not occur in the unlogged forest. Elephant dung counts were undertaken monthly on each transect. During the fruiting season of *Sacoglottis* (September to November) elephant densities increased dramatically in the logged forest, peaking at above 5 per km^2 (see Figure B13.5). During the rest of the year densities were relatively low and were comparable to those on the adjacent unlogged transect where *Sacoglottis* did not occur.

The distribution of *Sacoglottis* Forest was subsequently mapped and the majority of this important forest type was included

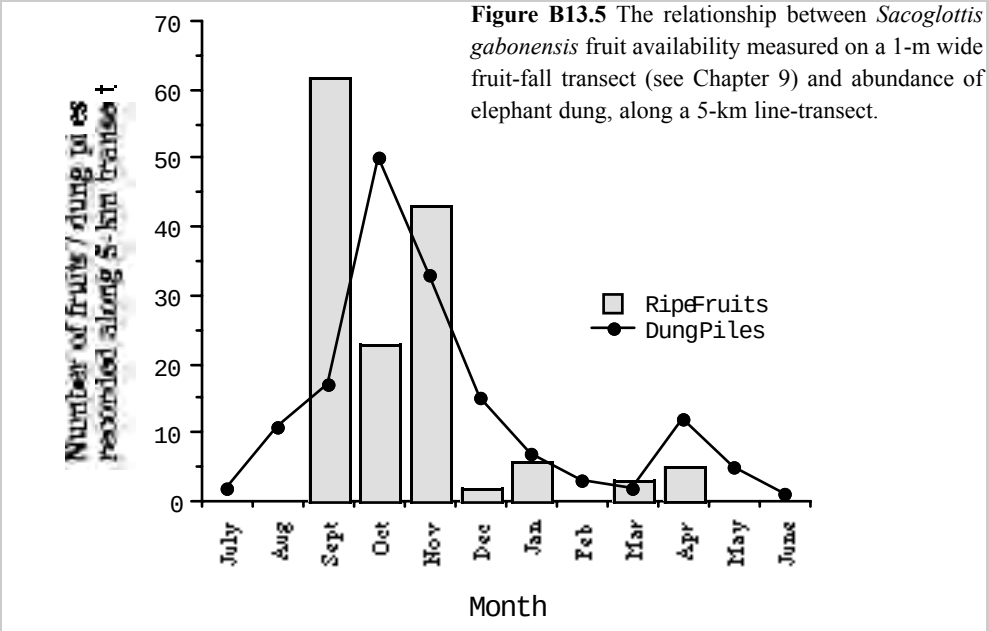


Figure B13.5 The relationship between *Sacoglottis gabonensis* fruit availability measured on a 1-m wide fruit-fall transect (see Chapter 9) and abundance of elephant dung, along a 5-km line-transect.

within a core protected area gazetted by the Ministry of Eaux et Forêts in 1996. Researchers planning animal censuses can learn a number of lessons from this study:

- It shows that extreme seasonal variations in animal density can occur within relatively large areas, highlighting the dangers of one-off surveys.
- It shows that in addition to background variation between transects there may be additional variation due to clumped seasonal food sources. Therefore, when doing a survey of an area without clear boundaries (as is often the case in the central African forests) you must ensure that transects are distributed across all potential sources of variation and cover a large enough area.
- It shows that great variation is possible even in areas which are geographically close. Had White not analysed forest composition and phenology patterns he might have concluded that elephant density were higher in logged forest, whereas in fact logging had no detectable effect on elephant density.

METHODOLOGIES FOR CENSUSING LARGE MAMMALS AND BIRDS

Elephants

Elephants are often a research priority because:

- they are targeted by poachers due to the value of their ivory (and to a lesser extent their meat) and are often one of the first species of mammal to be hunted to local extinction; hence they are a good indicator of hunting pressure, and of human pressure in general (Barnes *et al.*, 1991);
- they are a prime attraction for tourists, so improved knowledge of relative densities in different parts of a protected area, or of seasonal movements, may promote elephant viewing and generate income for the park;
- they are said to be one of the principal crop raiding species in many parts of forested Africa, resulting in conflict between protected area management staff and peoples living in and around protected areas;
- they play a key ecological role in African forests, dispersing the seeds of many plant species, opening clearings in the forest around salt licks, or killing trees and lianes by feeding on their bark, or pushing them over to eat their leaves.

Dung counts are the standard method for censusing elephants in forests (Barnes & Jensen, 1987). However, if you are censusing repeatedly on permanent transects in areas with high elephant densities, it may be possible to use encounter rate to estimate density (White, 1994 b - see below). Dung counts are undertaken along line-transects. As a rule of thumb, the optimal length of each transect is the length required to obtain about 10 dung pile observations. In areas of high elephant density this will be 1km, but in areas of low density this will have to be extended to 5km. The optimal number of transects in any one area is 20 - calculated to minimise effort (and hence cost) and maximise resolution (Richard Barnes, pers. comm.). As the transect is cut an observer walks along concentrating on the ground, searching left and right for all elephant dung piles (this observer may in fact record all dung and sign - see below). For each dung pile (set of boli) record:

- The distance from the start of the transect;
- Perpendicular distance from the transect line to the middle of the dung pile (see below)
- Vegetation type (see Chapter 9);
- Age of dung (see below);
- Diameter of dung, if intact;
- Other notes, such as what seeds it contains or what plants are germinating in it.

Measuring perpendicular distance

Perpendicular distance should be measured from the centre of the transect line to the centre of the dung pile:

- First, sight along the transect and estimate where the centre line lies. This should be imagined as a very thin line indicating the exact centre of the cut passage. A common mistake is to count all dung piles which lie on the cut area, which is often about 1m wide, as being on the transect and scoring these as zero (0m). This is wrong and greatly reduces the value of your data;
- Next, decide where the centre of the dung pile is. If the dung pile is in one amorphous mass this is straightforward. However, bear in mind that elephants sometimes defecate whilst moving, so boli may be spread over several (occasionally tens of!) meters. Look carefully at tracks, size, age and composition / consistency, to try to determine if dung piles close to one another (elephants often defecate soon after one another!) are part of the same 'dung event' or are the result of two elephant 'dung events' in the same area. If you decide that several boli spread out over a number of meters are part of the same defecation measure the perpendicular distance to the nearest and furthest edges of the dung - the perpendicular distance is then the point mid-way between these two points (Figure 13.2);

- Next identify the point from which the perpendicular distance should be measured. This can be done by using a compass set at 90° to the direction of the transect and then moving until the dung pile is on this line, or by standing with your arms out in the line of the transect, and moving until the dung pile is directly in your line of sight;
- Once you have located the two points between which the perpendicular distance should be taken, use a tape measure to measure it and record the measure to the nearest centimetre.

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Figure 13.2 Measuring the perpendicular distance to elephant dung piles.
X is the approximate centre of the dung pile at which the distance along the transect should be recorded.
The perpendicular distance(for both the amorphous dung pile and the dispersed dung pile) = $\frac{a + b}{2}$

If a dispersed dung pile lies on both sides of the transect measure to the most distant point on either side, subtract the smaller value from the larger value and divide the result by two: $\frac{a - b}{2}$

Estimating the age of a dung pile

To standardise between many observers it is best to use a simple system to age dung:

- *Fresh* - sometimes still warm!, with shiny fatty acid sheen glistening on exterior and strong smell;
- *Recent* - odour present (break the boli), there may be flies, but the fatty acid sheen has disappeared;
- *Old* - overall form still present although boli may be partly or completely broken down into an amorphous mass, no odour;
- *Very Old* - flattened, dispersed, tending to disappear;
- *Fossilised* - the dung matrix has become soil, but the presence of a few resistant fibres or seeds still marks the location of a very very old dung pile;
- *Mummified* - baked by exposure to sun, visibly once an elephant dung pile but with no substance remaining.

Estimating the abundance of elephant dung piles and elephants

Because visibility varies in different habitats, the dung encounter rate is of little use for comparing between areas, since a difference in the number of dung piles may occur in areas where dung density is the same but visibility differs. Dung encounter rates can be used as an index of abundance for monitoring trends in any given area, but even in this case, overall elephant density should be calculated in order to assess the reliability of your measure.

In order to estimate the density of elephants using dung piles, the following information is needed (see Box 13e):

- density of the dung piles;
- decay rate of the dung piles; and
- defecation rate of the elephant.

Dung densities can be calculated using the "Lopes Program" (a computer program included with this manual, written to enable complex mathematical models to be used to convert line-transect data to densities) in conjunction with Buckland et al. (1993). However, to run this program you need a good computer and a working knowledge of transect theory. A simple approximation which usually gives reasonable results and which can be done by hand in the field is described in Box 13f.

Dung decay rate is obtained by monitoring marked dung piles for many weeks, both before and during the time the elephant dung is censused (Boxes 13g & 13h). If you are unable to do this decay rates previously recorded elsewhere can be used as an approximation of decay in your site.

Defecation rates can vary widely depending on the types of food eaten and the season. Therefore, the only way to accurately determine the daily defecation rate of a wild elephant is to follow one. This is extremely difficult. If you are unable to do this you can use the figure of 19.8 observed by Tchamba (1992) in forests in Cameroon. However, this number may not be reliable for the elephants you are studying (see Box 13i).

Monitoring

For monitoring purposes it is often argued that dung density is the only measure necessary and that calculation of elephant density is unnecessary. However, if the variance of the sample is to be assessed, differences in decay rate or defecation rate within and between years need to be assessed (Barnes et al., 1994) and the variance of these measures needs to be included in the overall variance of the density index.

Other elephant sign

In addition to dung counts, feeding sign, tracks, rubbing trees, scars on trees due to bark feeding, wallows or trails can be noted. In areas of high elephant density this can be very time consuming and if you are finding sufficient dung it is probably not worth doing this. However, in areas were elephants are uncommon, or where they are only present at a certain time of year, these signs may be the only evidence of their presence. In places where elephants have been hunted to local extinction in recent years, old scars on trees from bark feeding, or overgrown elephant trails, testify to their presence in the past. You will have to decide on the appropriate sets of information to be collected in your area after a preliminary study, but be sure always to note dung and the locations and directions of major elephant trails.

BOX 13e CALCULATING ANIMAL DENSITIES FROM DUNG OR NESTS

In order to estimate animal densities using dung or nests, three pieces of information are needed:

- the density of the dung piles or nests (see Box 13f);
- the rate at which the dung piles or nests are made (creation rate); and
- the rate at which dung piles or nests disappear (decay rate).

How fast are dung piles or nests made?

Chimpanzee and gorilla night nests are made at the rate of approximately one night nest per day per animal (except for infants who sleep with adults). Gorilla day nests can generally be distinguished from night nests, which are slept in, but this may be more difficult for chimpanzees. In Budongo, Uganda, Plumptre & Reynolds (1996) found that 15.8% of chimpanzees reused old night nests, 17.5% slept in their mothers' nests and that a daily mean of 18.8% constructed day nests - resulting in 1.03 nests per weaned individual per day (but only 0.88 nests overall per individual per day).

The number of dung piles produced by one animal in one day (defecation rate) is more difficult to determine. Rates vary widely between species and depend on the food eaten and the season. If you are unable to obtain sufficient hours of observation to calculate defecation rates you will have to use figures reported from elsewhere (see Box 13i).

How fast do dung piles or nests disappear?

The rate at which dung or nests disappear, or decay, depends on many factors (Box 13g). If you are unable to undertake decay studies you will have to use figures from the literature, but be aware that this may result in significant errors being incorporated into your density calculation.

How is the density of animals determined from the density of dung or nests?

To get the density of animals (animals/km^2), divide the density of dung or nests (#/km^2) by the product of the mean duration (the mean number of days it takes dung or nests to disappear from the time they were first created) and the creation rate (the number of dung piles or nests produced by one animal in one day) - see example B13.1:

$$\text{Density of Animals} = \frac{\text{Dung or Nest Density}}{(\text{Mean Duration}) \times (\text{Creation Rate})}$$

EXAMPLE B13.1

The following information was collected for blue duikers in Ituri Forest, RDC:

Average Defecation Rate = 4.9 piles/day

Average Dung Decay Rate = 18 days/pile

Average Density of Piles = 1000 piles/km^2

Therefore, the estimated density of blue duikers is calculated as follows:

$$\text{Density of Animals (animals/km}^2) = \frac{1000}{18 \times 4.9} = 11.3 \text{ animals/km}^2$$

Box 13f A METHOD FOR CALCULATING DUNG DENSITY FROM TRANSECT DATA WHEN YOU DO NOT HAVE ACCESS TO A COMPUTER

by Peter Walsh

A rough estimate of object detection probability can be made by assuming that object detection distances are normally distributed. The normal distribution provides just the right shape for representing how detection probability decreases with increasing distance from the transect line. It has a value of 1 at zero distance which corresponds to the assumption that detection probability is equal to 1 on the transect line. And it falls off to the right, just as detection probability falls off as objects get farther from the transect line. The speed with which the distribution drops off is determined by the variance of the distribution: the larger the variance, the more slowly the distribution drops off. The trick, then is to figure out which variance produces a shape that best approximates your sampling data. Luckily, this variance can be estimated from perpendicular distance data using a very simple calculation that can be carried out by hand or with the help of a pocket calculator (see below).

Once the variance has been estimated we can then choose some critical point on the distribution where detection probability falls very close to zero. For example, let us choose the critical point to be the distance at which 95% of objects are expected to be detected at distances smaller than the critical distance. We can call the critical width "w' and the interval between distance zero (the transect line) and w the "detection zone." The width of the detection zone for variances ranging from zero to 100 is plotted in Figure B13.6.

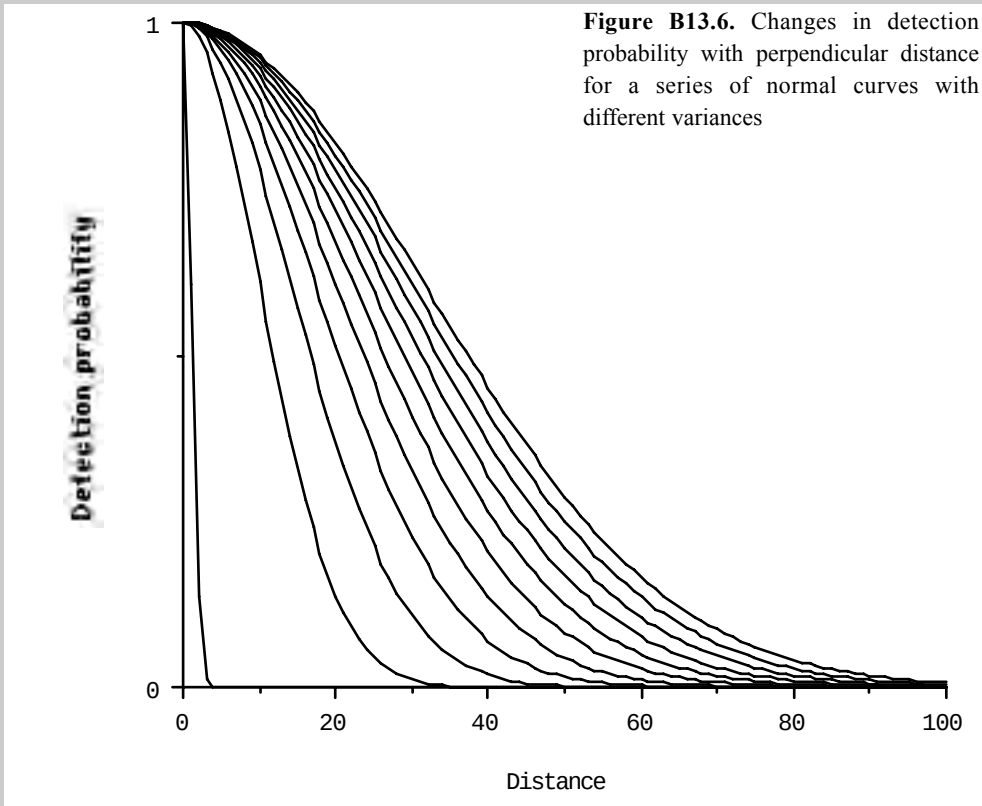


Figure B13.6. Changes in detection probability with perpendicular distance for a series of normal curves with different variances

The final problem is to find the average detection probability (p) in the detection zone. Conveniently enough, the normal distribution has a rather peculiar property. Given a particular critical point, all normal distributions have the same average detection probability (p) no matter what the variance of the distribution. For example, if we choose a detection zone which is expected to include 95% of object detections then the average detection probability is always 0.607, no matter how wide the detection zone is.

We can now calculate density by dividing the number of objects detected (N) by the area of the detection zone and the detection probability. The area of the detection zone on each side of the transect is just the critical width (w) multiplied by transect length (l). This gives a density of:

$$D = N / (2lwp),$$

where the denominator is multiplied by two because there is a detection zone on each side of the transect.

Steps for estimating detection zone width (w).

1) Take the square of the perpendicular distance for each object detected.
2) Sum the squares.
3) Divide the sum of squares by the sample size minus one. This is the variance of your distance data.
4) Locate the variance estimated in step three on the horizontal axis of Figure B13.7.
5) Move vertically up from the horizontal axis until you intersect the curve.
6) Move horizontally from the curve to the corresponding point on the vertical axis. This is the estimated width of the detection zone (w).
7) Detection probability (p) is equal to 0.607.

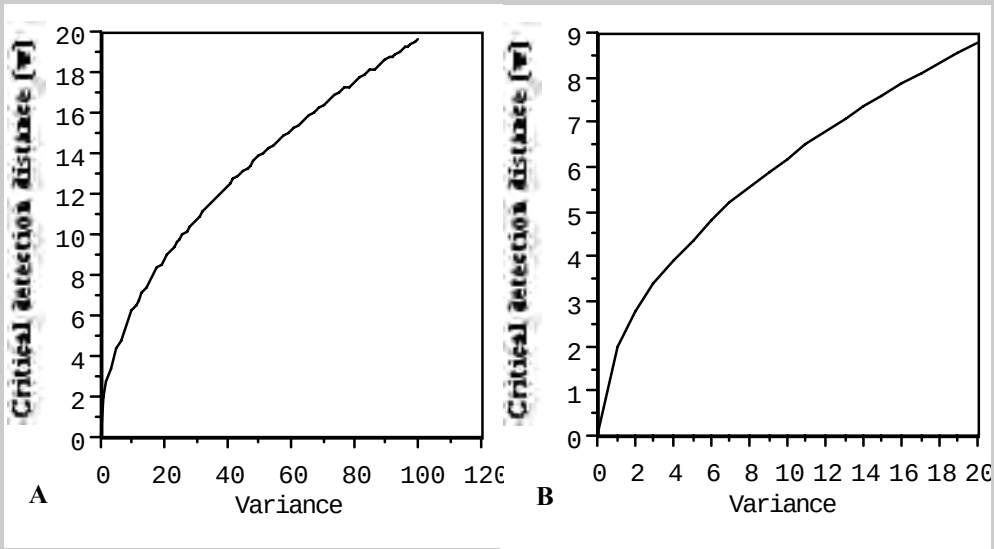


Figure B13.7 Relationship of the variance of perpendicular measures obtained on a line-transect and the critical detection distance.
A is for variances up to 100 and **B** is the same curve for values below 20.

Box 13g HOW TO MEASURE DECAY RATE OF DUNG AND NESTS

When using line-transect methods to assess density, one of the critical assumptions is that all objects on the centre-line are detected with certainty (see Box 13d). With increased distance from the centre-line the probability of seeing an object decreases. When censusing animals directly, the fall-off in visibility is due to the fact that more distant objects are more likely to be obscured by vegetation (or in open habitats because they are so far away they are too small to see). If censusing groups of animals, the fall-off in visibility will be less rapid for larger groups, because as the probability of seeing objects becomes smaller, the more objects there are, the more likely it is that at least one will be visible.

When censusing dung piles or nests one has to consider decay rate if one is to calculate animal density (see Box 13e). The perceived decay rate will differ as a function of the distance from the centre-line. To understand why, consider an elephant dung pile made up of 11 boli. If it is on the transect you will be able to see all 11 boli and you will score the dung is present until all 11 have decayed. If the dung pile is 7m from your transect, all but one or two boli may be obscured by vegetation. The first time you pass your attention may be drawn to it by its odour. The next time there will be no odour. If another elephant has passed in the meantime and moved the foliage around the dung pile, the two boli that were visible may now be concealed - as far as your transect count goes they have decayed, although if you approach the place where the dung pile is all 11 boli may still be intact. Hence, since distance from the transect will affect the number of boli you see, it will have a comparable effect on the perceived decay rate. The same applies to any study of duration. For example, a gorilla nest site may have 11 nests. If your transect passes through the middle of the nest site you may see all 11 nests. That site will be scored as being present until the last nest is no longer visible, even though some nests may disappear relatively quickly and others last a long time. If your transect passes 30 m from the nest site you may only see one or two nests. If these are fast decaying nests the site will appear to decay quickly, even though other longer lasting nests which are not visible from the transect are still present.

When you analyse transect data you assess the probability of detecting objects at different distances from the centre-line. If decay rate increases with distance, this will result in the probability of observing objects falling off more quickly than would be the case if visibility were the only factor involved. However, your analysis of effective strip width will take this into account; any measures of decay rate should therefore be undertaken by observing objects on the centre-line (or within a band either side of the centre line where all objects are detected).

Decay rates of elephant dung piles found on major elephant paths and old logging roads have been found to be biased, because conditions are not representative of those in the forest where dung counts are usually undertaken (White, 1995). If you plan to monitor dung (or nest) decay you should avoid monitoring on paths and roads and instead monitor dung piles encountered in vegetation representative of the area to be censused. This is best achieved on a permanent line-transect that can be walked repeatedly - at one or two week intervals (Barnes et al., 1994).

Dung decay studies

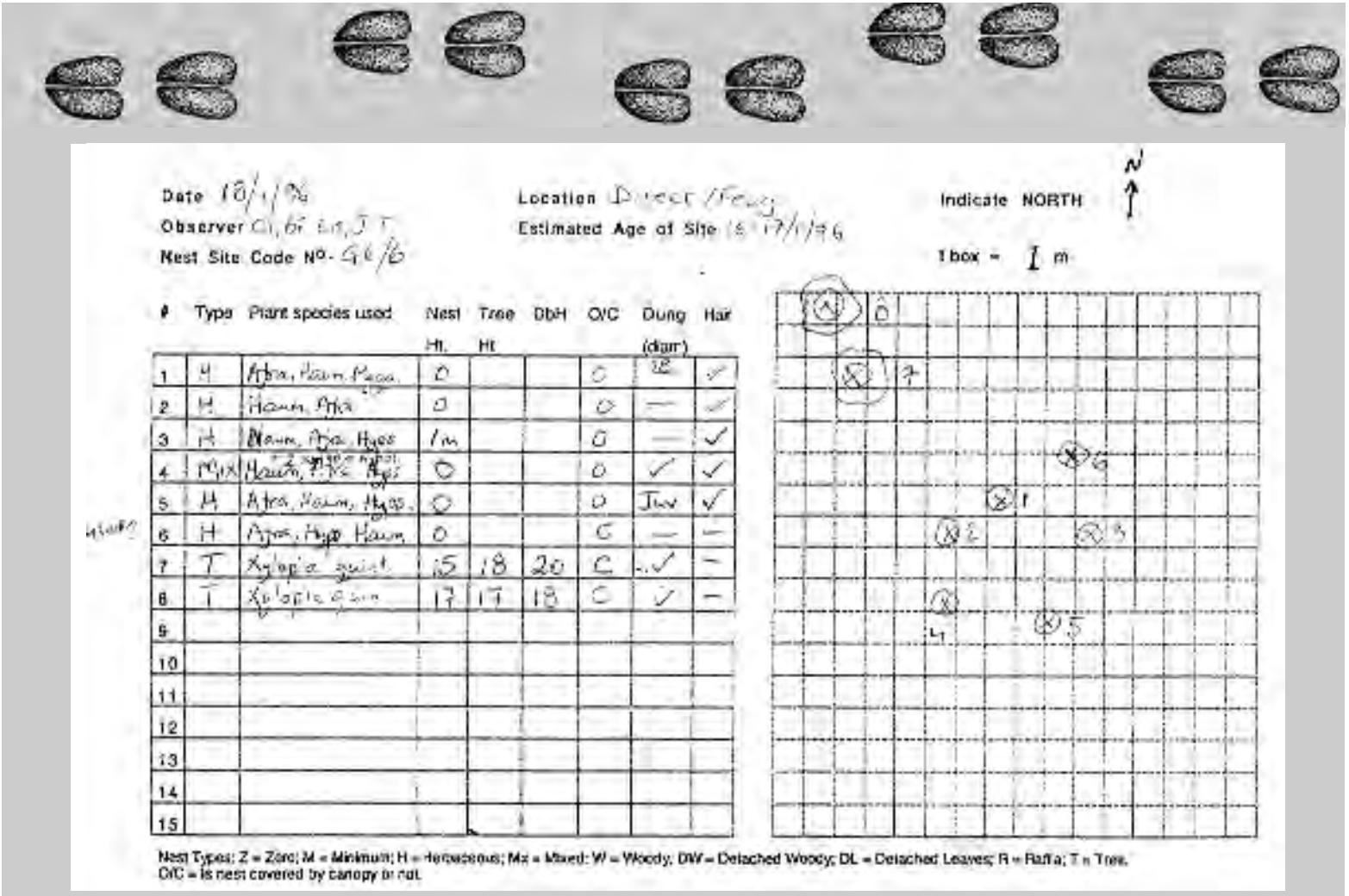
To monitor decay rate for elephant dung, establish a transect in a convenient place where you know there is a high density of elephants. Walk the transect at least once every two weeks, recording all new elephant dung piles you see. When you see a dung pile for the first time mark it in a fashion that does not draw your attention to it. If you attach an orange tape that will catch your eye and increase your chances of seeing a barely visible dung pile you will bias your data. One method that has proved effective is to note with an indelible marker on the underside of leaves overhanging the dung pile a code number and the date it was first seen.

Take standard transect data on the state of the dung, its location along the transect and the perpendicular distance from the transect. Each time you visit the transect you should prepare a check-sheet of all dung piles being monitored, with their location along the transect. As you arrive level with each monitored dung pile you should approach it until it is underfoot. Leave the transect line if necessary. Once you are standing over the location of the dung pile assess whether or not it is still visible and score its age.In order to analyse decay you have to follow all dung piles in a sample until they have disappeared. You should aim for a sample of about 30 - 50 dung piles for each study site or season. Most elephant dung piles will disappear within 60-70 days, but some will last over 100 days and it is important not to abandon these inconvenient samples. The mean duration of a dung pile can be calculated by dividing the total duration of all dung piles by the total number of dung piles. The same methodology can be applied to other dung decay studies and to nests.

Nest decay studies

In the case of nest sites decay should be monitored from the geometrical centre of the nest site. Sample sizes of 50 nest sites in Lopé for both gorillas and chimpanzees were sufficient to calculate decay rates with reasonable accuracies (White & Barnes, unpublished), so this should be regarded as your target sample size. Different gorilla nest types decay at different rates (Tutin & Fernandez, 1984). Proportions of different nest types differ between populations of eastern gorillas (Schaller, 1963) and in different parts of the western lowland subspecies' range (Fay, 1997; Remis, 1994; Tutin et al., 1995). In some sites a great proportion of nests have zero construction (see page 243). For example, at Ndakan, in southwestern Central African Republic, Mike Fay (1997) found 35% of gorilla nests were zero construction, lasting a maximum of 48 hours. This may not have an extreme effect on nest decay studies, because most nest sites also contained well constructed nests. However, a large number of weakly constructed nests would result in an underestimate of group sizes if 1-2 week old nest sites are analysed. Decay experiments will not be possible in all areas, but if data on mean duration of different nest types is collected, in addition to information on nest sites as a whole, it will be possible to correct for ratios of different nest types in areas where no decay experiments have been undertaken (cf. Tutin & Fernandez, 1984). Therefore, in addition to scoring whether the site is visible, it is a good idea to then approach each nest in the site and to score whether or not it is visible. If you plan to undertake such a study you should collect data on nest types as described for transect surveys and make a map of the nest site, to enable you to efficiently check each nest each time you visit the site (see Figure B13.8)

If nest decay studies are not possible you can use figures from Lopé: chimpanzee nest sites lasted on average 106 days and gorilla nest sites lasted an average of 78 days. All chimpanzee nests are in trees. For gorillas the common nest types in Lopé were herb nests (46%), mixed nests (18%) and tree nests (33%). If nest composition is very different in your site you should be cautious about using the Lopé data. For example, at Lopé 22 of the 57 nest sites monitored contained only herb nests. If these are excluded from the sample the proportion of nests becomes 25% herb, 26% mixed and 47% tree and mean duration is 85 days. Excluding a further 9 nest sites with >50% herb nests, the proportions become 11% herb, 34% mixed and 53% tree, with mean duration 93 days. The mean duration of 10 nest sites monitored which had only tree nests was 120 days. You could choose the decay rate which most resembles the nest composition in your site until you have your own data.



Date 18/1/96 Location Dzeer/Feng Indicate NORTH

Observer G, bi En, J T Estimated Age of Site 15 17/1/96

Nest Site Code No. 96/6 1 box = 1 m

#	Type	Plant species used	Nest Ht.	Tree Ht	DbH	O/C	Dung (diam)	Hair
1	H	Afra, Haum, Mess	0			O	12	✓
2	H	Haum, Afra	0			O	—	✓
3	H	Haum, Afra, Hyes	1m			O	—	✓
4	Mix	Haum, Afra, Hyes	0			O	✓	✓
5	H	Afra, Haum, Hyes	0			O	Jw	✓
6	H	Afra, Hyp, Haum	0			C	—	—
7	T	Xylopia quint	15	18	20	C	✓	—
8	T	Xylopia quin	17	17	18	C	✓	—
9								
10								
11								
12								
13								
14								
15								

Nest Types: Z = Zero; M = Minimum; H = Herbaceous; Mx = Mixed; W = Woody; DW = Detached Woody; DL = Detached Leaves; R = Raffia; T = Tree.
O/C = is nest covered by canopy or not.

Figure B13.8 A filled out nest checksheet.
The observer notes: nest type (see page 245) ; plant species used; height of nest; height and dbh of tree for tree nests; whether the nest is under closed canopy or the canopy is open to the sky (O/C); age class of dung associated with the nest (see Figure 10.1); and whether a hair sample was collected for genetic analysis (see Box 13l). In addition a scale map is drawn of the nest site indicating the relative positions of each nest. Note that the observers were not sure that nest 6 had been used.

Box 13h ELEPHANT DUNG DECAY RATES

Dung decay is a complex process. Dung piles deposited on stream banks or in gullies can be washed away overnight by rain, but dung that remains moist, due to contact with marshy ground, can remain apparently fresh for long periods. Dung exposed to direct sunshine can be baked dry, becoming "fossilised" and maintaining its form for a year or more. Dung beetles (Scarabeidae) and termites (Termitidae) are important decomposers of dung piles, but show seasonal activity patterns (e.g., Wiles, 1980; Wing & Buss, 1970). Red river hogs, mandrills and other monkeys, African civets and squirrels forage in elephant dung piles for seeds and insects, breaking and dispersing the boli, and this behaviour may also be seasonal, depending on the species of seeds present in dung, or fruit availability elsewhere. Another factor which could affect dung duration is diet, since this will affect the consistency of the dung (Koster & Hart, 1988; Wiles, 1980). Hence, in any sample of dung piles monitored to determine decay rate there will be a certain degree of variability. Some dung deposits will disappear rapidly and others will last several months. Figure B13.9 shows a typical decay curve.

During a study conducted in Lopé 1282 fresh elephant dung piles were monitored regularly over a two year period. Statistically significant differences in dung duration were found in different months, but this was not a simple dry season / wet season relationship as found in other studies (Wiles, 1980; Wing & Buss, 1970). In another study the diet of elephants in Lopé

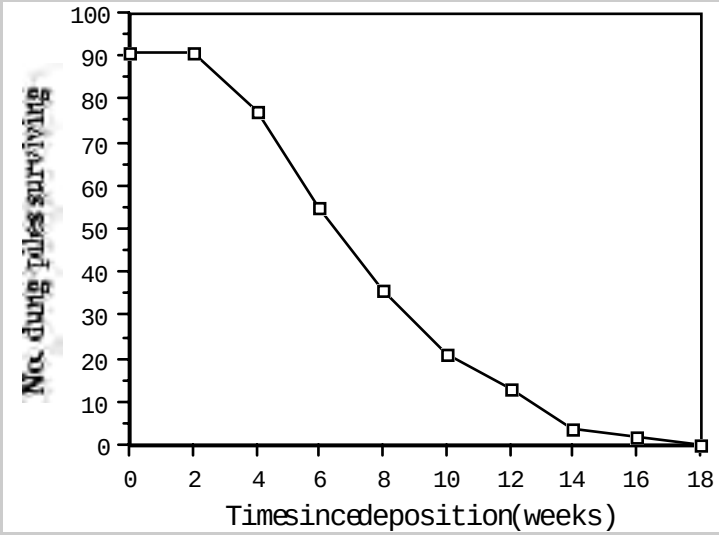


Figure B13.9 Survival of a sample of 91 dung piles deposited in November 1989 in the Lopé Reserve (data from White, 1995).

was assessed between June 1990 to May 1991, by searching through 311 fresh elephant dung piles for fruit remains, principally seeds. The amount of fruit found in elephant dung was found to be directly related to seasonal availability of fruit in the forest. Furthermore, as the amount of fruit in dung piles increased the rate at which they decayed showed a corresponding increase. Therefore, fruit content is at least one of the principal determinants of elephant dung decay rates in the tropical rain forest environment at Lopé. In fact, fruit content was more important than weather in determining decay rates in this sample.

Barnes et al. (1994) undertook studies of dung decay in several sites in Ghana and Cameroon. They were particularly interested in how decay varied with weather patterns. They found that rainfall and temperature in the month of deposition explained 81% of the variation in decay rate across sites. They suggested that in the future it may be possible to develop an equation which can be used to predict theoretical dung decay rates from data on local weather patterns. For this to become a reality there need to be more studies of dung decay and weather patterns in sites spread across the African rain forest.

BOX 13i
DEFECATION RATES

To calculate animal densities from dung counts one has to know both the rate of deposition (number of times an animal defecates per day) and the rate at which the dung disappears (decay rate - See Boxes 13g & 13h). Defecation rates are different for different species (see Table B13.1). Even closely related species can have quite different defecation rates. For example, gorillas defecate about 4-5 times per day but humans tend only to go to the toilet once. Defecation rates can vary with season, diet, age, health, and levels of stress. For example, Barnes (1982) found that elephant defecation rates in Ruaha, Tanzania (in savanna vegetation) were 9.6 times per day in the dry season and 32 per day in the wet season!

Measuring defecation rates is difficult and time consuming and for animals such as elephants involves a certain degree of risk, as one has to track individuals or groups through dense vegetation. However, such data is extremely valuable and if collected well will be a significant contribution to animal censusing. Over time you may be able to opportunistically build up defecation data by systematically noting the time when observations begin and end and the number of defecations in that period. However, bear in mind that when startled or scared, many species defecate involuntarily, so only take data for individuals which are not aware of your presence. Also note the age and sex of the animal(s) you watch, their activity and location. Andrea Turkalo and Mike Fay have been monitoring elephants for several years in a the saline in the Dzanga-Sangha National Park, Central African Republic. Some days there are over 100 elephants in the saline for several hours but it is rare to see even one defecate - it seems that the elephants avoid defecating in this communal area! *For measured defecation rates to be truly representative of your study species, samples should be taken from a random cross section of ages, daily activities and locations.*

Table B13.1 Defecation rates of selected African rain forest animals.

Species	Rate #/day	SE	Comments	Source
Elephant	19.77	0.23	Lowland rain forest, Cameroon. No seasonal differences.	Tchamba, 1992
Elephant	17		Wet season, Kibale, Uganda.	Wing & Buss, 1970
	11		Dry season.	
Elephant	14.4		Follow of collared animal	Powell, 1998
Blue Duiker	4.9	0.66	Ituri forest, RDC,	Koster & Hart, 1988
Bay Duiker	4.4	0.66	Captive animals fed wild foods.	
Buffalo	5.1	0.25	Karisoke, Rwanda	Plumptre, 1991
Gorilla	4.5		Best guess from Lopé	Tutin, pers. comm.
Gorilla	>5		Virunga, RDC/Rwanda	Schaller, 1963.

Other ungulates

When censusing other ungulates besides elephants, such as duikers, bongo, okapi, buffalo or chevrotain, once again dung counts are probably the most appropriate method for large scale survey work. Data should be recorded as for elephant dung. Where species cannot be accurately determined, as for medium sized duikers, the species group (small, medium or large) should be noted instead.

Again, it should be borne in mind that seasonal differences in decay rate are significant (Koster & Hart, 1988; White, 1994b) so comparisons should only be made for data collected in the same season and preferably only if decay rates have also been monitored (see Box 13j). Decay rate can be analysed in the same way as for elephant dung, but dung piles are likely to disintegrate more rapidly (except, perhaps, in the dry season) so deposits should be checked every 1-2 days.

Dung pellets should be classified into the following age classes:

- Fresh - odour and a sheen (fatty acids) still present;
- Recent - pellets with dulled surface, but no mould;
- Old - may have a patina of mould, pellets beginning to break down;
- Very Old - pellets crumbling, dispersed and covered by leaf fall.

Buffalo dung should be classified as for elephants.

Again, other sign of these species, such as duiker prints, or browsing by okapi, can also be noted if appropriate.

Gorillas and chimpanzees

Gorilla and chimpanzee population densities tend to be low, so the frequency of direct sightings will rarely provide sufficient data for density calculations. In most areas, apes will be wary of human observers and will flee on sight. Nest-counts conducted along line transects are a simple alternative. Nests not only remain stationary but are many times more common than individual apes, as a new nest is built each night. Thus, they are a potentially powerful tool for census work. Line-transect sampling is the most robust and practical method to estimate nest density (Buckland et al., 1993). This method has been used to estimate gorilla and chimpanzee nest density in several areas of lowland rain forest in central Africa (Carroll, 1988; Fay, 1989; Fay et al., 1989; Tutin & Fernandez, 1984 - see also Box 13k).

Nest counts

Gorilla and chimpanzee nest counts are undertaken along line-transects in the same way as dung counts (see above). Each gorilla or chimpanzee in a group, with the exception of infants which sleep with their mothers, makes a nest each night in which to sleep. Nests made by the members of a group of gorillas or chimpanzees on any particular night tend to be located quite close to one another, and constitute a nest site. When walking a transect you should be constantly on the look out for ape nests. The chances of seeing other nests in a nest site increase once you have seen the first nest. For this reason transect analyses for ape nests are done on data for nest sites as a whole, rather than for individual nests, just as measurements of elephant dung piles are taken to the centre of a group of boli.

Box 13j
SEASONAL VARIATION IN NUMBERS OF DUIKER PILES FOUND ON TRANSECTS

In the Lopé Reserve, Gabon a sample of 15 piles of medium-sized duiker dung (*Cephalophus dorsalis, C. callypigus, C. ogylbyi, C. leucogaster*) was monitored daily in the wet season. Mean duration was 4.3 days (White, 1992). One pile of dung was watched whilst a large dung beetle collected every bolus individually and rolled them down its burrow, removing the whole deposit in just under two hours. No data were collected for the dry season but dung counts showed a marked peak in dry months (Figure B13.10).

As you can see in the figure, dung counts in the dry season were 10 times higher than in the wet season. Duikers are

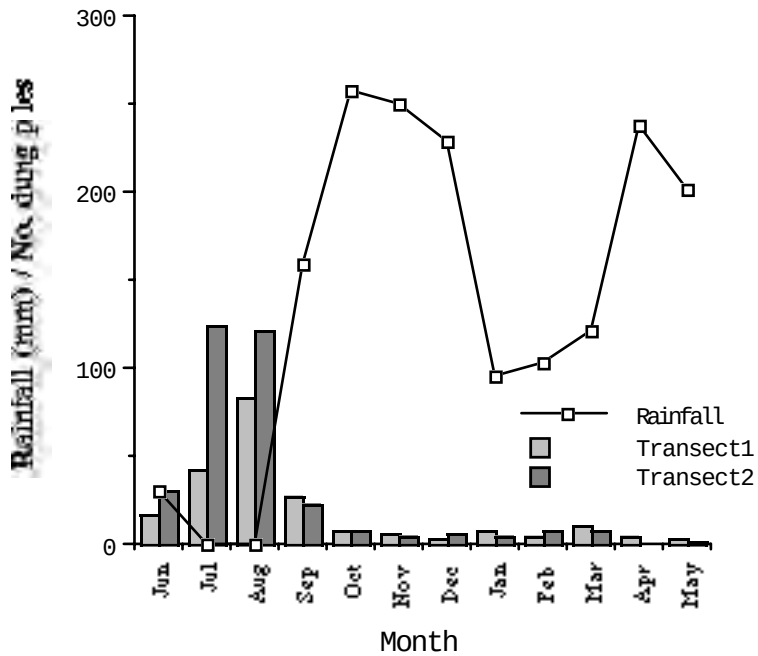


Figure B13.10 Number of medium sized duiker dung piles recorded on two 5km line transects in the Lopé Reserve, Gabon and the rainfall in the previous month.

territorial and do not show large scale seasonal migration. It seems therefore that the difference in dung densities was due to different decay rates (and perhaps different defecation rates - see Box 13i). Decay rates probably differed due principally to changes in rainfall or to a combination of factors, such as decreased fruit availability, insect activity, humidity and rainfall in the dry season.

Whatever the reason for the change in decay rate, the implications for duiker surveys using dung counts should be obvious. If you base hunting quotas on dung counts made in the dry season but use a decay rate calculated in the wet season your sustainable hunting will in fact be a massacre. Seasonal variation is so extreme that you will be obliged to undertake decay studies at the same time as your dung counts if you hope to estimate duiker densities in this way. If you undertake such studies year-round you may be able to establish a correlation between dung decay and weather patterns. This will simplify your monitoring activities thereafter.

For each gorilla and chimpanzee nest site sighted along the transect, note:

- location along the transect;
- the species that made the nest(s), if known (note how the conclusion was reached)- see below.
- estimated age (fresh - odour, and usually dung, present; recent - vegetation still green for the most part but no odour, dung often still present; old - intact, but all vegetation dead and dung absent; very old- decomposition advanced);
- vegetation type (see below); and
- the perpendicular distance to all nests in the nest group, including any which are not visible from the transect - for each nest note on which side of the transect it fell and if it was visible from the transect. **Before approaching the nest site carefully search for all nests visible from the transect**.

Record the following information for each nest:

- nest type (see below);
- nest height;
- plant species used to make the nest;
- age class (diameter of dung) of occupant if fresh dung is present.
- optional - collect hair samples (if present) for genetic tests from each nest using forceps sterilised before each nest - see Box 13l.

Walking off the trail to count and examine the nests

Unlike most of the other observations taken while walking transects, you should leave the transect in order to get an accurate count of the number of nests found in each group, and to look for dung. Gorilla nests tend to be grouped quite closely, although there are sometimes outliers and you should search carefully all around. Chimpanzee nests are generally more spread out than those of gorillas and you must be prepared to search up to 50m from where you see the first nest. Nests high in trees are often hard to spot and you should look up from several different angles.

Box 13k CENSUSING GORILLAS IN MONTANE HABITATS

Nest counts and the diameter of feces associated with fresh nests (which indicates the age of nest builders) have been used to census and to follow demographic trends of gorillas living in montane vegetation (Aveling & Harcourt, 1984; Harcourt & Fossey, 1981; Harcourt & Groom, 1972; Inogwabini et al., in press; Murnyak, 1981; Schaller, 1963; Weber & Vedder, 1983; Yamagiwa et al., 1993). Working in a relatively small area of montane forest it is possible to track gorilla groups from one nest site to the next. To be sure that all individuals in a group have been detected, most surveys try to track each group detected and inspect nest sites from three consecutive nights. This allows accurate determination of group size and age-class structure, except for infants less than a year old which do not make nests.

Surveys of this type are appropriate if you want to get to know the population using a study area, particularly in montane vegetation, such as in the Virunga mountains of eastern Africa. Systematic application of these methods to gorillas in lowland tropical forest habitats is more difficult, because of differences in vegetation, (tracking is more difficult in lowland forests) diet, ranging and nest building behaviour. The frugivorous diet of gorillas in lowland tropical forests results in: 1) wide ranging (making tracking and the finding of three successive nest-sites difficult); 2) soft feces (making it impossible to assess age-class consistently from fecal diameter); and 3) a high proportion of tree nests, at least in some habitat-types (intact feces are rarely found under tree nests) [see Tutin et al., 1995]. Hence, surveys of gorillas in lowland vegetation are almost always done using line-transects.

Determining which species made the nests

Since gorillas and chimpanzees are sympatric in many places you may have to be sure you can distinguish between their nests. Chimpanzees make night nests only in trees (although they occasionally make day nests on the ground). In contrast, most, but not all groups of gorilla night nests will contain at least one nest on the ground (but see Box 13m). Tutin and Fernandez (1984) reported that while some members of a gorilla group might nest in trees, whole groups would not do so and thus the presence of one, or more, ground nest(s) at a site allowed nests of gorillas to be distinguished from those of sympatric chimpanzees. A larger data set of 2435 nests in 373 gorilla nest sites at Lopé shows this statement to be inaccurate: 8% of fresh gorilla nest groups contained only tree nests. In fact, on rare occasions, all members of a gorilla group, including the adult male 'silverback', choose to nest at heights above 20m. The common misconception that a nest above 5m is that of a chimpanzee is wrong.

During the aging process a proportion of gorilla nest-sites ‘convert’ to chimpanzee nest-sites after all herb nests have disappeared and only tree nests remain visible. This is because of the longer lifespan and better visibility of tree nests. Such sites, that would have been identified as having been built by chimpanzees on a one-off census, accounted for 26% of all nest-sites in Lopé. This mis-identification of nest sites would lead to over-estimation of chimpanzee densities and under-estimation of gorilla densities (see Box 13m). In fresh nest sites presence of dung and urine can be used to identify which species slept there, once you are familiar with the dung of each species (see Chapter 10).

If there is no sure indication, such as dung or a ground nest, to allow certain identification of a nest site, it should be classified as an ape nest site, although if circumstantial evidence suggests one species or another, this should be noted. Whilst inspecting nests check carefully for presence of both small and large diameter dung in the same nest, which would indicate a female and her infant slept together. You also need to be able to distinguish between day and night nests, which is generally easy when fresh due to night nests’ flattened appearance and the presence of feces and urine, hair, and odour. In most places day nests are rarely encountered on censuses, but this may not be the case everywhere.

Nest types are defined as follows:

- Tree: Nests built in trees, constructed by bending and breaking branches to form a sleeping platform. This is the only type of sleeping nest built by chimpanzees, although they occasionally build day nests on the ground.
- Zero: No nest structure exists and the gorilla has slept on the ground. The sleeping site appears as a flattened patch, usually with scattered leaves or small flattened plants. The presence of feces, hair, and sometimes the smell of gorilla, aids in identification of the ‘nest’.
- Minimum: The nest consists of one or two stems of herbaceous plants that have been bent (sometimes several times) to form a rudimentary pad on which the gorilla has slept.
- Herbaceous: The nest consists of 3-20+ stems of herbaceous plants that have been bent, and sometimes interwoven, to form a substantial platform with a roughly circular depression where the gorilla has slept.
- Mixed: Similar to the above but woody vegetation (lianes, shrubs, saplings or detached branches of small trees) have been incorporated into the nest.
- Woody: Nests built on the ground made entirely of woody vegetation from bent lianes, shrubs or saplings.
- Detached woody: Similar to the above but built entirely from detached leafy branches that have been carried to the site and assembled into a nest.
- Palm: Nests built entirely out of palm, particularly *Raffia* spp.
- Other nest types may need to be defined as you encounter them.

Box 131 STUDYING GORILLA GENETICS USING HAIR AND DUNG

More and more field workers today are making use of the sort of genetic analyses used in forensic medicine to identify murderers and rapists. Genetic analyses can be used to assess the degree of relatedness of different populations of the same species, or of different species and can also be used to identify new species. Furthermore, genetic fingerprints can sometimes be used to identify individuals, and to determine paternity (the father) of an individual who's mother is known or maternity if the mother is unknown.

DNA (genetic material) can be obtained easily from fresh blood or tissue, but extraction is now becoming possible from naturally shed hair or even from fresh dung. At Lopé the silver-back gorilla called Porthos had been followed by researchers for 12 years when in 1994 he was attacked and killed by another gorilla. His group disappeared. It is likely that the females in Porthos' group joined the group of his aggressor and that his young sons became solitary or formed a small batchelor group. The Lopé researchers had been collecting hairs out of all gorilla night nests found since 1990 and they are now analysing the samples at their laboratory in Franceville in the hope that they can identify Porthos' killer - they are using forensic science in the rain forest!

The extraction and analysis of DNA requires sophisticated equipment and laboratory facilities. However, samples for extraction are easily stored. If you frequently have access to dead animals, such as those seized from poachers, collect a small sample of kidney, liver or heart and store in a freezer (if possible) or in absolute alcohol (or, if this is not available, in strong salt solution). Be sure to note the species, sex, age class, location killed/found (if possible - or point of confiscation) and date (see Chapter 11). Alternatively, or in addition, a piece of skin should be removed and dried and a tuft of about 300 hairs collected and dried (this last sample may prove the most convenient and be the only sample you choose to collect).

Hair samples left in night nests or from dead animals are easy to collect and store. If collecting from an animal pull rather than cut the hairs as DNA extraction is from the root of the hair. The DNA is stable for several days (and for much longer in the right conditions), so that nests or pelts of up to a week old can still be useful. Hair samples should not be handled directly, to avoid DNA contamination from human skin, but should be collected with tweezers (Swiss army penknife type is adequate). The tweezers should be cleaned, either with an alcohol-impregnated swab (do not reuse swabs) or by sterilising in a flame, before each sample. Hairs from each nest or animal should be collected into paper envelopes large enough to take the hairs without bending them. The envelopes should be sealed, but not by licking them (contamination with DNA in saliva!), and stored at base camp in a box containing silica gel to keep the hairs dry (or in an air conditioned room); DNA will degenerate if the hairs are kept in a moist environment. Such hair collections can also be useful for identifying remains in scat samples.

If you are able to making collections for DNA extraction but have no means to analyse them, contact the WCS Africa program and we will try to put you in contact with potential collaborators.

Box 13m GORILLAS MAKE TREE NESTS TOO!
HOW TO CORRECT FOR MIS-CLASSIFIED NESTS IN AREAS WHERE BOTH GORILLAS AND CHIMPANZEES OCCUR.

Early studies on gorillas concentrated in montane forests of Uganda, Rwanda and RDC, where gorillas live in a lush salad bowl and obtain most of their food with little effort. Whilst gorillas will sometimes make tree nests in these forests, it is relatively uncommon and is almost unheard of for large adult male silver-back gorillas to do so. However, eastern and western lowland gorillas living in lowland rain forests spend a lot more time in trees. Fruit is a major part of the diet of lowland gorillas and even silverbacks will climb high into the canopy of emergent trees to reach sweet, juicy food. Furthermore, gorillas in lowland forests often nest in trees. In the Lopé reserve, Gabon, where gorillas have been studied for 14 years, 35% of all nests are in trees. On two occasions all eight members of a known group (including the silverback) nested above 25m, and silverback nests have been recorded as high as 34m above the ground. In fact, in 8 % of gorilla nest sites there were no ground nests.

Hence, in lowland forests the height of tree nests is of little value when trying to decide whether an old nest site was made by gorillas or chimpanzees (Fig. B13.11). The proportion of nests of any given type varies between group, habitat, season and site. An Lopé about 70% of nests in areas with open under-storey are built in trees, but in other habitats only about 20% are in trees (Fig. B13.12). Furthermore, gorillas are more likely to nest in trees when there is a strong likelihood that they will be disturbed in the night by elephants, and in areas where hunting pressure is high, nests are often found in inaccessible places, such as on steep slopes or cliffs.

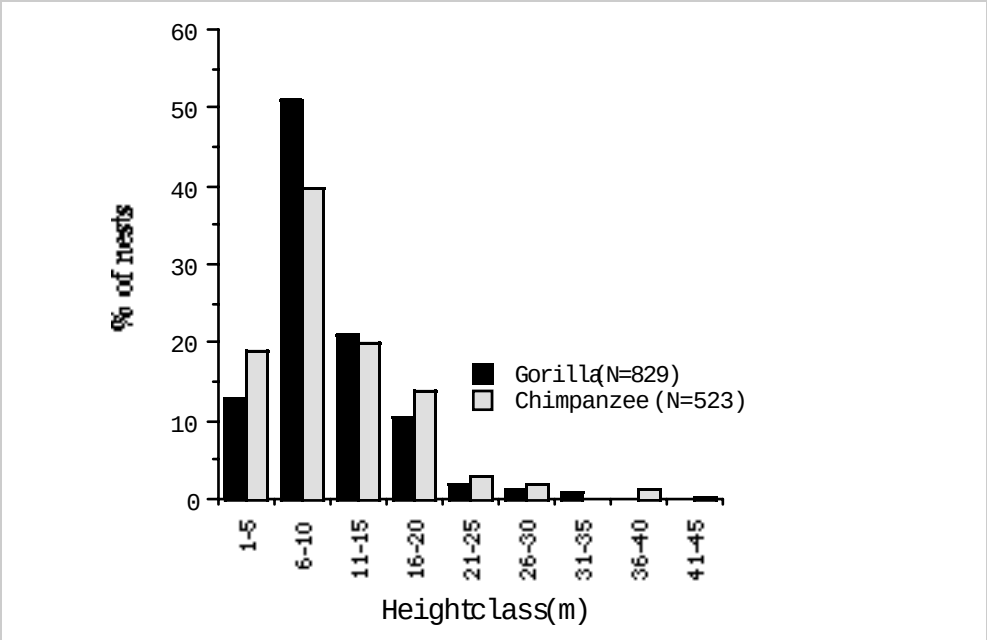


Figure B13.11 Proportion of gorilla and chimpanzee tree nests in different height classes in the Lopé Reserve, Gabon (data from Tutin *et al.*, 1995; Wrogemann, 1992).

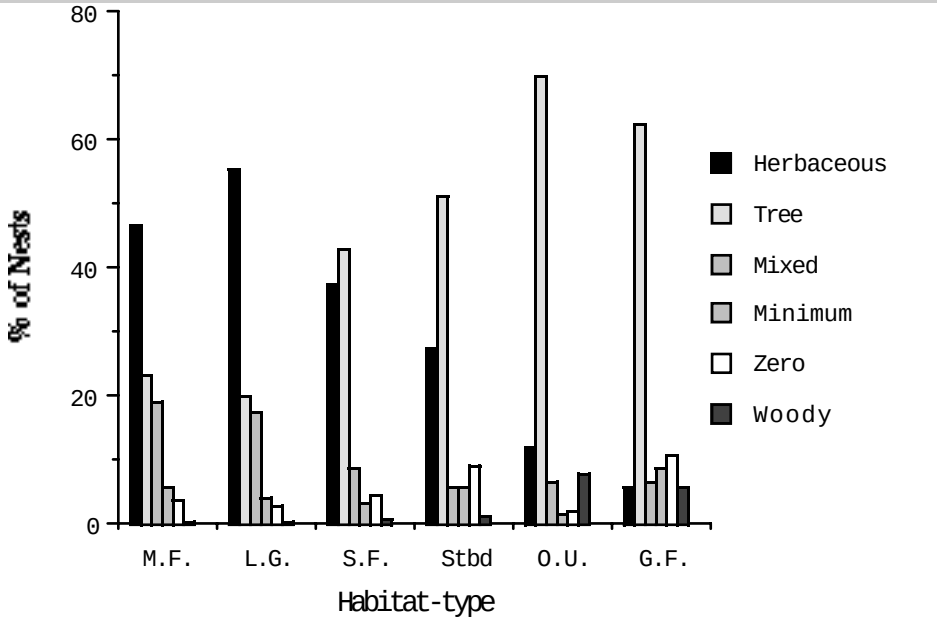


Figure B13.12 Percentage of different nest-types found in six habitat-types in Lopé**.** (M.F.=Marantaceae forest; L.G.=light gaps; S.F.=secondary forest; Stbd = stream bed; O.U.=open under-storey; G.F.=gallery forest)

With experience, one gets a 'feel' for whether gorillas or chimpanzees made a particular nest site. For example, when a group of eight gorillas all nest in trees their nests tend to be quite close together, whilst the nests of eight chimpanzees might be spread over 50m or more. Such feelings should be noted, but you have to bear in mind that there are frequent exceptions to every rule (even to the rule that chimpanzees always nest in trees - there are occasional records of ground nests, and chimpanzees do make day nests on the ground). Only by building up good data on fresh nest sites will you be able to calculate a correction factor for your study area. This will enable you to estimate the number of sites which will be mis-classified. If you have time, the best way to do this would be to undertake a nest decay study (see Box 13g).

Correction factor

If you plan to undertake nest counts in an area where both gorillas and chimpanzees occur you will inevitably mis-classify some gorilla nest sites as chimpanzee sites. You have to follow rules when classifying sites which do not have dung or other sign which allow you to associate them with gorillas or chimpanzees. Sites with ground nests are classified as gorilla nest sites, because chimpanzees almost never make ground night nests. Sites with all nests in trees are classified as chimpanzee, even though some of these will have been made by gorillas.

When calculating nest site densities first do so as if all your classifications are correct. Then check the composition of nests in fresh gorilla nest sites. In Lopé 46% of nest sites have only ground nests and 54% have at least 1 tree nest (8% contain only tree nests). Gorilla dung in Lopé lasts on average 16.3 days (N=30) and 23.5 days (N=28) in wet and dry seasons respectively. One can assume that for about 2-3 weeks (say 20 days) the site will be correctly identified from dung. However, gorilla nest sites last a lot longer than this. Sites with only ground nests last on average 68 days, sites with at least one tree nest last 85 days and those with only tree nests last about 120 days on average. Knowing the proportion of fresh gorilla nest sites that contain just tree nests you can calculate the number of old sites that are mis-classified:

Let,
dg = duration of gorilla nest groups starting with only ground nests
dtg = duration of gorilla groups starting with both tree and ground nests
Ng = number of gorilla nest groups with both tree and ground nests at the time of the census
Nt- = number of gorilla nest groups that have lost their ground nests
Ntg = total number of gorilla nest groups that started out with tree and ground nests
Nt = total number of gorilla nest groups that always had only tree nests
N = estimated total number of gorilla nest groups starting with tree nests that were gorilla sites
P = % of gorilla nest groups starting with at least one tree nest
Pt = % of gorilla nest groups starting with alle tree nests
Ptg = % of gorilla nest groups starting with both tree and ground nests

Consider an example where there are 100 old gorilla nests sites on a survey in Lopé, of which 40 have at least one tree nest, and 125 nest groups that were classified as chimpanzee sites because there were no ground nests (fresh sites where species is known are not included in this calculation). The number of gorilla nest groups with tree and ground nests at the time of the census is the product of the number of groups starting out with tree and ground nests and the ratio of the duration of sites with only ground nests to the duration of nest groups with both tree and ground nests:

(dg / dtg) Ntg = Ng

We can therefore get an estimate of the number of nest groups which started out with both tree and ground nests by inverting and multiplying the equation:

Ntg = Ng (dtg / dg)

We know (see above) that
dg = 68
dtg = 85
Ng = 40
and we get
(68 / 85) Ntg = 40 or, Ntg = 40 (85 / 68) = 50
Therefore, the number of additional groups that have lost their ground nests is
Ntg - Ng = Nt-
50 - 40 = 10

We now need to correct for gorilla nest groups which started with only tree nests and which were mis-classified as chimpanzee nest groups. We know that 54% of gorilla nest groups start out with with at least one tree nest (P) and that 8% start out with only tree nests (Pt). Therefore
Ptg = 54 - 8 = 46%
The ratio of all nest groups with tree and ground nests to all groups with tree nests is therefore Ptg / P. Therefore, the number of gorilla nest groups with tree and ground nests is this ratio multiplied by the total number of groups starting out with a tree nest,
N (Ptg / P) = Ntg
N = (54 / 46) 50 = 58.69
Hence, we estimate that a further 8.69 gorilla nest groups started out with only tree nests and were mis-classified as chimpanzee nests.
Therefore, the ratio of gorilla to chimpanzee nest groups changes from 100:125 to 118.69:106.31
Hence, the estimate of gorilla numbers was wrong by a factor of 100/118.69 (the estimate was 84.3% of the actual number) and the estimate for chimpanzees was wrong by a factor of 125/106.31 (117.6% of the actual figure).

Measuring the perpendicular distances to nests and nest groups

There is some debate about the relative merits of estimating densities of individual nests and nest sites. In that nests are clustered together in sites one can argue that it is nest sites, not individual nests which are detected from the transect (just as we think of whole dung piles rather than the different boli when censusing elephant dung). However, it seems that using individual nests to calculate density does not result in a large bias and in some ways is simpler as one does not have to factor in nest group size. For now it would seem sensible to record **both** the perpendicular distances to the nest group centre and the perpendicular distances to all nests (clearly distinguishing in your notes those visible from the transect from those only seen once you left the line).

Measure the perpendicular distance from the centre of the transect to the centre of each nest (Fig. 13.3). For tree nests, estimate where a vertical projection from the centre of the nest would hit the ground and measure from this point to the transect (not from the base of the tree it is in). If the trail passes through the centre of the group of nests, count the nests on one side of the transect as positive and those on the other side as negative. To find the nest group centre measure to the near edge of the nearest nest and the far edge of the furthest nest and divide by two - see Figs 13.2 & 13.3).

Figure 13.3 Measuring the perpendicular distance from the centre of the transect to the centre of each nest. To find the nest group centre, as shown in Figure 13.2, measure the distance to the far edge of the furthest nest and the near edge of the closest nest and divide the sum by 2 (here, a-b / 2).

Estimating the density of nests and nest groups

In order to calculate the density of individual nests estimate strip width and hence density as for elephant dung, either using a computer program such as Lopes or Distance, or the simplified method described in Box 13f. To then go on and calculate gorilla or chimpanzee density you need to know the mean duration of nests. In order to estimate the density of nest groups, first calculate the distance from the transect to the centre of each nest group as detailed above and use these distances to estimate density.To then go on and calculate gorilla or chimpanzee density you need to know the mean group size and the mean duration of nest sites.

Mean group size

Mean group size should be calculated from sites where you are sure all nests were still visible (fresh nest sites). Data on nests built by a known gorilla group (Porthos' group) over a four year period at Lopé show that extrapolation from the number of nests per site to the number of gorillas should be based on many nest sites because only 30% of nest counts accurately reflected group size (Tutin et al., 1995). In some sites many gorilla nests (• 35%) will be of zero construction and that these only remain visible for 2-4 days (Fay, 1997; Remis, 1994). Therefore, if this is the case in your area only fresh nest sites can be used to calculate mean group size. Furthermore, infants and some young juveniles will share their mothers' nests and will only be included in group size calculations if their dung is seen and distinguished from that of their mothers. In Ndakan, southwestern Central African Republic 15.5% of the gorilla population were not nest builders (Fay, 1997).

Decay rate

To interpret census nest counts it is essential to know how many days nest-sites remain recognisable. Nest decay is complex as the lifespan will be influenced by nest group size and by the types of nest construction. Nest-type varies with habitat-type and season and possibly other factors.

Mean duration of gorilla nest-sites during a study conducted in Lopé was 78 days (see Tutin et al., 1995). This is 32% longer than the figure given by Tutin & Fernandez (1984) who followed 223 gorilla nests at Belinga, in NE Gabon. At Belinga, the mean duration varied from 4.3 to 61.7 days depending on the type of nest. The overall mean nest duration, calculated from the frequency of different nest-types in their sample, was 53.6 days . The difference between the Lopé and Belinga figures in part reflects the fact that in the former, nest-sites were aged, whereas in the latter, nests were considered individually, but other factors such as climate and habitat-type also varied. In particular, the species used for tree nest construction were different: At Belinga most were in *Musanga*, a fast growing secondary tree species with soft wood, and decay was rapid. The difference between these two figures for decay rate illustrates the danger of extrapolating results between areas. Nest decay studies are laborious and time consuming, but essential if the census is to give reliable results.

Studies of relative proportions of different nest types and mean duration of nest sites need to be undertaken in as many areas as possible. Any long-term projects currently underway in the ranges of gorillas and chimpanzees could be undertaking such studies since it need not take up too much of a researcher's (or team's) time, whilst they go about their other interests (see Box 13h). Once you have calculated nest site density follow the steps in Box 12.6 to calculate the density of individual gorillas or chimpanzees.

Other information on animals that can be collected during one-off transects

A number of specific examples of what information to collect on one-off transects have been given here. However, the line-transect methodology can be applied to a wide variety of taxonomic groups and animal signs. Some groups are discussed below in the section on repeat transects, because the noise of cutting a transect is likely to disturb them. Even if the noise you make whilst cutting a transect is likely to chase away animals such as elephants, gorillas, monkeys, duikers or hornbills, it is likely that you will see some animals and if they are considered to be target species you should still note these observations as described below. In addition to observations of animals you may choose to note some of the following sets of information:

• Tracks - in practice it will not be possible to note all tracks of duikers and pigs encountered. This should be done however for other medium-sized and large mammal species of particular interest, such as bongo or gorillas. Where tracks are so abundant that this is not feasible, either note topofil distances for the start and end of the area of saturation or make the decision to ignore tracks in areas of high density where other data (e.g., dung counts) will suffice.

• Excavations - rooting by pigs, soil scraping by gorillas, tusk marks of elephants, burrows of giant pangolin and aardvark , scrapes made by leopards on the ground or on vegetation, resting places flattened by duikers - if any of these species are a priority and other signs are scarce record excavations systematically. However, it is often difficult to be sure which species was responsible for scrapes and to estimate their age, so other indices are preferable.

• Feeding trails - particularly apes and elephants (distinguish chimpanzee and gorilla whenever possible using prints, dung and odour, but be sure to indicate when the species could not be reliably determined).

• Marks on vegetation - elephant bark feeding, elephant rubbings in areas of low densities, duiker scent marks.

• Note the location, size (major, large, medium, small), state (is it swept clear by passage of animals - active; are leaves accumulating - recent use; or is it abandoned) and direction of all elephant trails.

• Note all species of ripe fruit observed and note evidence of feeding by large mammals - keep a daily list of all species of fruit found and tally the number of fruiting trees seen for each species daily.

Supplementary information to record on one-off transects

As noted above, by collecting information on vegetation changes or human activity, in addition to data on animal abundance, you will be in a better position to explain any differences detected in animal density in different areas, or to check that your stratification was valid (see Chapter 3). The two most important sets of information are human sign and vegetation changes.

Human activity

Observations of human sign from transects should be put in a socio-economic context for the entire region surveyed. This context will come from information on settlement locations, economic activity, ethnic associations and history of land use. Gathering this information will demand ancillary or complimentary studies as outlined in Chapter 15.

Information on human activity that can be gained from transects provides an index of the type and intensity of human use of an area. These indices may be directly correlated with changes in faunal abundance over time. They also provide a measure to relate to people's own perceptions of their use of and impact on forest resources.

For each encounter of human sign, the type of evidence, (including a detailed description if necessary) the estimated age and the distance along the transect should be recorded. Common signs are:

- Roads - used or disused;
- Old village sites;
- Machete cuts or broken stems resulting from a single passage - count the number of cuts visible;
- Regularly used human trails, or where old cuts suggest that humans once passed frequently;
- Snare line - active or abandoned (count the number of snares and take data on any animals in snares);
- Shotgun shells;
- Honey extraction;
- Camp sites - active or abandoned (note the presence of drying racks or animal remains, the number of shelters and check for any signs which might indicate whose camp it was);

- Fire places;
- Current or past agricultural activity;
- Mining activity;
- Bark stripping for construction, mining sluices, chords, medicines etc.;
- Large mammal carcasses, especially elephant;
- Rubber tapping;
- Fishing activities (e.g., traps);
- Sites where nuts such as *Panda oleosa* have been cracked open (but note that chimpanzees in West Africa do this, as do red river hogs);
- Sites of canoe construction.
- Tree cutting / timber extraction

Note also signs of changes (increases) in human pressure such as:

- Abundant elephant sign dating from several years before (in areas where there is no recent sign), such as barking of trees since healed or long abandoned elephant trails;
- Overgrown bais, which may indicate that large mammals, which once maintained these habitats, have decreased in density, perhaps due to hunting pressure.

Ask guides with a local knowledge for further information of who lived in old camps and villages or of impressions of areas which may have undergone change in recent years.

Habitat description.

A running *relevé* of vegetation type, slope, altitude etc. should be kept. All physical features of note should be recorded, such as streams and rivers (note size and direction of flow), marshes (size), major tree falls (particularly if you are interested in browsing species such as the okapi), valley bottoms (note altitude), ridge tops (note altitude). A schematic representation of the transect has proved useful to record these data (Figure 13.4). In addition to these features grade each 50m segment by angle of slope, both in the line of travel and along the perpendicular: 1) zero or gentle slope; 2) medium slope (< or > - arrows pointing uphill); 3) steep slope (<< or >>).

Forest types generally have to be simplified if records taken by different observers are to be comparable (see Chapter 9 for more details):

- Mixed Forest with open under-storey;
- Mixed Forest with closed under-storey - may be similar to old secondary vegetation (note if under-storey is dominated by lianes, Marantaceae, or other vegetation);
- Monodominant Forest (note dominant species e.g., *Gilbertiodendron, Garcinia, Berlinia, Lophira, Aucoumea, Julbernardia, Sacoglottis*,etc.);
- Swamps;
- Seasonally Inundated Forest;
- Coastal scrub;
- Mangroves;
- Marantaceae Forest (note the dominant Marantaceae species);
- Montane or semi-montane forest
- Liana Forest;
- Gallery Forest;
- Low Closed Scrub - often with *Ancistrophyllum* palms present;
- Clearings (elephant bais & swamp bais);
- Major light gaps;
- Savannas;
- Mature Secondary;
- Young Secondary;
- Logged (how many years ago?);
- Plantations - recent or long abandoned - note particularly presence of oil palm or mangoes.

More experienced observers can make supplementary notes to add detail to these categories. If you are not planning to undertake a full botanical inventory (see Chapter 8) a very useful addition to these vegetation data is to record all tree species with dbh of • 50 cm within 10m (or 15m) of a point on the transect at 100m intervals - a total of c. 75 (or 175) trees per 5 km. Note species, dbh and height. This will give a measure of the density and frequency of large tree species. From more detailed botanical work you may be able to classify canopy trees into indicator groups. The value of these data will become more important as you learn how some tree species can be used as indicators of forest history or animal habitat quality (see Table 9.1).

Collection of these data will slow the rate of travel (unless you have an additional botanical observer), but once you are familiar with the common species, or if you have a competent assistant who knows scientific or local names, this could be achieved efficiently.

Locating transects in space

If surveys are to be repeated in the future as part of a monitoring process one needs to be able to locate where they were undertaken. Furthermore, much of the environmental data taken has possible application in ground truthing of satellite images and this is important if we are to improve the capacity to use satellite images to monitor large scale changes in forest cover (see Box 9l). With access to GPS equipment this should prove relatively easy. The location of the start and end of each transect should be recorded (from the nearest convenient open area) and waypoints should be taken at regular intervals along the line as opportunities arise, particularly at prominent points likely to feature on maps, photographs or satellite images, such as river confluences. It would be worthwhile leaving the transect to get such fixes if a suitable location is found close by (see Box 6g).

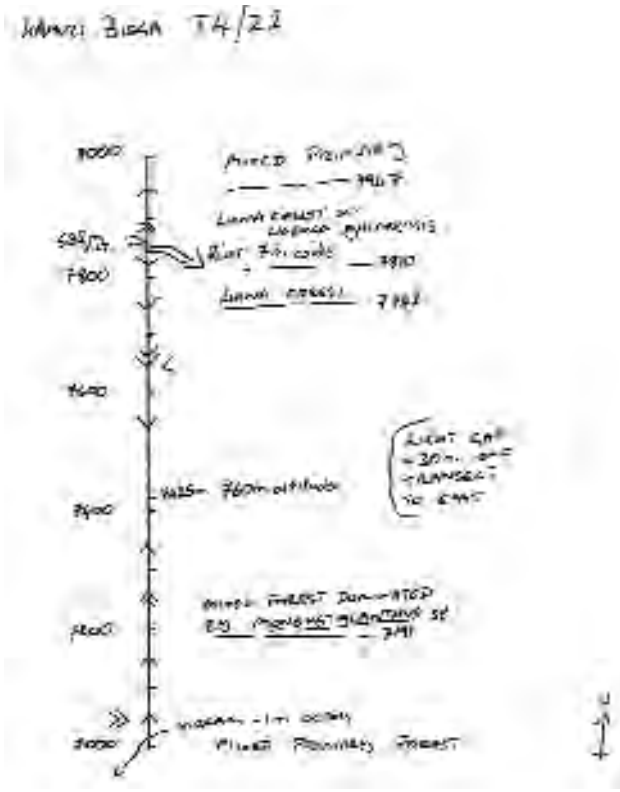


Figure 13.4 A section from a schematic representation of vegetation and topography along a transect. If vegetation differs on either side or the transect this should be noted.

USING ANIMAL SIGHTINGS TO ESTIMATE DENSITY

1) Line-transects

Whilst many species or groups of animals can be censused using dung, nests or sign, these methodologies are not always appropriate. For example, monkeys are one of the dominant groups of mammals in tropical forests but, being predominantly arboreal, they rarely leave sign at ground level and their dung is so small it is extremely difficult to detect and decays rapidly. Furthermore, whilst it may be easier to count the abundance of dung, nests or footprints, converting these measures to reliable density estimates is not always simple or convincing. Some people scoff at the idea of using elephant dung counts to estimate densities. Therefore, estimating densities from sightings of the animals themselves is generally more satisfactory, and sometimes the only method possible. However, this method is generally less well adapted to the job of covering large remote areas for a number of reasons:

- as we have already discussed, animal sign tends to be much more abundant and easy to detect than the animals themselves. For example, one expects to see of the order of 150 elephant dung piles and about 100 chimpanzee nests for every elephant or chimpanzee actually sighted. Therefore, one can get good sample sizes for sign with less time and expense than for sightings;
- animals are very wary of humans, whereas dung and nests do not flee. Therefore you can collect data on sign as you cut a transect in a remote area. In order to census animals using sightings you have to be able to move quietly along pre-cut transects. Therefore, two teams have to access a remote area, one to cut transects and one to collect data, leaving a sufficiently long interval of time between visits to avoid the possibility that disturbance caused during cutting influences animal observations;
- transects opened in order to allow silent passage of observers hoping to observe animals have to be better cut that one-off transects used to census animal signs, increasing the damage caused to vegetation and the possibility that they will be adopted by poachers (see Box 13b);
- in general, data collection for animal observation censuses tends to be more complicated than for sign, and therefore requires a greater level of training.

Hence, animal sightings tend only to be used to census certain groups of animals, or for detailed studies in restricted study areas. In African rain forests they have most often been used to estimate primate densities, but they are equally applicable to other types of animal, such as duikers, squirrels, many bird species, butterflies, etc. If you are interested in obtaining additional data for an area you have identified as having high mammal densities, or if you want to monitor the effects of hunting or logging on mammals in a community reserve, hunting concession or logging permit, you may choose to use sightings to assess population trends. As explained in Chapter 14, there is a great deal of important information (other than densities) that can only be obtained by observing animals, and this may justify the additional effort of undertaking this type of census. If you do choose to census animals directly you will probably do so along a limited number of (semi-) permanent transects, which are traced once to allow quiet passage through the forest, and then are maintained regularly. An approach that has proved useful in several sites is to combine regular animal censuses with botanical research (see Chapter 9) along permanent 5-km transects in areas of particular interest.

Since the method of estimating animal densities from animal sightings along line-transects is often applied to primates, estimation of monkey density is described in detail here. Following that we discuss how to apply the same methods to other taxonomic groups.

Primate censuses along line-transects

Primate censuses are conducted along pre-cut transects. Many researchers have chosen to use 5-km transects since this can be completed in 5-7 hours and tends to give sightings of several groups of monkeys. However, as with dung or nest counts, if you are able to conveniently undertake a number of shorter, randomly located transects, say of 1-km each, your sample is likely to be more representative of your study area as a whole (see Chapter 3).

If access allows, censuses to record animal sightings should begin as early as visibility is sufficiently good to distinguish species high in the canopy. If rain falls continuously for more than 15 minutes the census should be abandoned, because rain and subsequent noise of dripping water may affect animal behaviour and/or observer efficiency, altering the probability of detecting animals. Successive censuses should be conducted in opposite directions if possible. Censuses should be conducted at regular intervals throughout a period of at least a year, say once every week or fortnight, depending on time available. You should aim to obtain 60-80 observations for each species you intend to census - if using five kilometres of transect in an area of reasonable primate density this will probably entail 40-50 repetitions.

Whitesides *et al*. (1988) describe methodology for primate censuses, which is equally applicable to other mammals and some large birds (but which has been slightly modified here):

- move slowly and quietly along the transect at a rate of about 1 km/ h;
- never leave the transect to improve your view since this may result in your spotting monkeys you would not have seen from the transect, and violates the assumptions of line-transect sampling;
- stop periodically to watch and listen for animals.
- upon detecting a group or individual animal, spend up to 10 minutes in situ recording data (you may move up to 25m in reverse to obtain a better view).

Groups of monkeys near transects are usually easy to detect. However, getting detailed information about how many monkeys are in each group is difficult. Furthermore, you are generally detecting groups not individuals. Therefore, when censusing monkeys, as with ape nests (see above), data are collected for each group encountered. A group is defined as an association of monkeys 50m or more from any other association. When you encounter a single monkey travelling alone it is called a "solitary", not a group.

Information should be recorded on a pre-prepared checksheet. This should have the date, name(s) of observer(s), transect name and direction of travel, start time, and end time noted clearly at the top. When a group of monkeys is encountered the following information should be recorded for each species observed (there may be two or more species associated in one group - known as a "polyspecific association"):

- Encounter number - each single species or mixed species group seen during a census is given a number which indicates which observations were part of the same encounter and which were alone. For example, if the first observation on a census was of a mixed group of *Cercopithecus pogonias* and *Cercocebus albigena*, data for both of these species would be assigned to encounter 1.
- Time of day
- Location along the transect
- Habitat type in which the monkeys are observed
- Species of monkey
- How the group was detected (noise of movement or falling fruit, alarm call, contact call, vision etc.)
- Height of first individual seen

- Perpendicular distance from the trail to the estimated centre of the group, measured using a rangefinder, or using a tape measure after data collection is complete (Figure 13.5).
- Distance from the observer to the estimated centre of the group (the "sighting distance")
- Compass bearing to estimated centre of the group (this is used to calculate the "sighting angle" - see Figure 13.5)
- Number of individual monkeys detected for each species in the group (number of individuals actually seen or heard calling from that species)
- Estimated total number of individual monkeys. Estimate the total number of individuals you think are in the group. You can use cues like branches moving and other indirect signs. When more than one species are encountered together, the total estimated number of individuals includes all the species together. If there is more than one observer one person can quietly follow the group in order to obtain a better estimate of group size, whilst their colleague(s) continue along the transect.
- Each time a monkey vocalisation (particularly male loud calls) is heard note the time, your location, the species, the estimated distance and the angle;
- Make additional notes on behaviour, any observed foods, presence of young (ventral) infants, etc.

If you do not have access to a rangefinder and it is not possible to have a team of at least two observers, so that one observer can record distances with a tape measure whilst the other observer continues along the transect, distances will have to be estimated by eye. If this is the case you should test your accuracy periodically since it is easy to make significant errors.

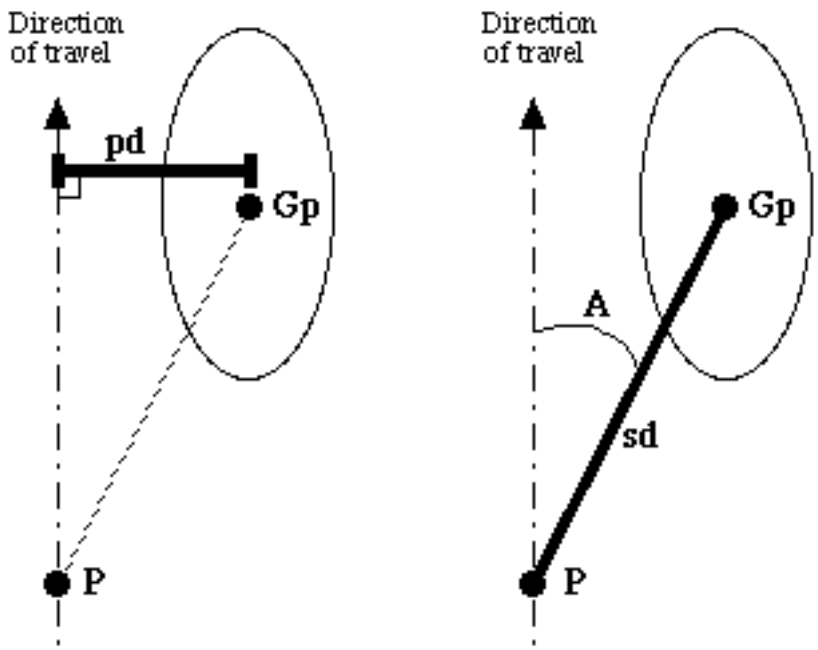


Figure 13.5 The perpendicular distance (pd) is measured from the centre of the transect to the estimated group centre (Gp), at a 90° angle from the transect. The sighting distance (sd) is measured from the person (P) to the estimated group centre (Gp). The sighting angle (A) is measured between an imaginary line along the centre of the transect to an imaginary line connecting the observer and the estimated group centre.

Estimating the abundance of monkey groups

In order to estimate the density of monkey groups you will have to analyse your data to calculate the area you have censused for each species. Computer programs such as Lopes and DISTANCE (see above) are available to do this, but if you want to analyse data in the field by hand or do not have access to computer facilities, follow the steps in Box 13f.

The average number of times monkey groups were encountered per kilometre (i.e., the total number of groups encountered divided by the total number of kilometres walked) can be used as a simple index of abundance, particularly for species which are rare and for which you do not have sufficient data to calculate actual density.

Estimating the abundance of individual monkeys

To estimate the density of individual monkeys you will need to collect data on mean group size (since you will not be able to count all individuals in each group encountered on transects). Estimates of group size made during censuses are unlikely to be accurate, unless there are several observers and one leaves the transect and follows the monkeys after data collection is complete, in order to assess group size (if this observer sees any animals they cannot be recorded unless also seen by the observer who continues along the transect). If this is not possible an effort should be made to make estimates of group size at other times.This can be done opportunistically, whenever an entire group of monkeys is seen crossing a road or river. However, such opportunistic counts are likely to be biased towards small groups, since you are more likely to get a full count of a smaller group by chance. Therefore, if possible make a separate study of group size by attempting to follow and observe a number of different groups. Later, multiply the density of monkey groups by the average number of individuals found in each group (see Chapter 14). This should be done separately for each species.

Censusing other species along line-transects

By taking similar information to that described for primates on other species one can efficiently census a wide variety of animals, including elephants, butterflies, hornbills, frogs and lizards. When censusing species which are encountered as solitary individuals measurements should be taken to the animal itself; and whenever groups are encountered, distances should be measured to the group centre as for primates.

Night surveys on line-transects

Line-transect observations need not be restricted to daylight hours. Many animal species that are not visible during the day can be censused at night. These animals include the nocturnal primates, such as the galagos or bush babies, pottos and angwantibos; small carnivores, such as genets, civets and mongooses; and, giant rats, porcupines, pangolins, water chevrotains, owls, bats, hyraxes, flying squirrels, and duikers. In fact, night counts are particularly effective for duikers, giving encounter frequencies which are much higher than diurnal censuses on the same transects.

Move quietly along the transect searching all around with a headlamp for eye shine and movement. If a powerful headlamp is used, it is easy to see reflections from the animals' eyes. Usually an animal will stand still when the light approaches, allowing the animal to be identified to species. In order to allow for differences in activity patterns, night censuses should be walked shortly after dusk, in the middle of the night, and in the early morning before sunrise, and during different phases of the moon.

Record standard line-transect data for individuals or groups of animals seen on night transects. Density calculations are again as described in Box 13f.

2) Point counts

Point counts (sometimes called point-transects) are a variation on line-transect sampling often applied to songbird surveys. Instead of proceeding along a line transect the observer(s) make observations from points. These points can be arranged at intervals along a transect, or randomly in different habitats. The observer measures the distance from the point to any animal seen or heard calling, or to the centre of any group, and notes the species, height, time etc. as for line transect data. In fact, this method can be envisaged as a modified type of line-transect in which the transect length has been reduced to zero. The point from which observations are made is considered as the centre-line of the transect and the same assumptions described in Box 13c apply to this point. As discussed above, reducing transect length to zero and maximising the number of samples will make the sample more representative of the study area, but time will be lost moving between transects. This methodology has many advantages over line-transect sampling:

- once the observer is in place he or she can concentrate solely on detecting, locating and identifying birds, without the need to traverse difficult or noisy terrain;
- he or she can take the easiest route in and out of the pre-selected points;
- patchy habitats can be sampled more easily and representatively, facilitating density estimation by habitat;
- describing vegetation structure and composition associated with a point is easier than for a line;
- only the observer - object distance needs to be measured, which is easier than measuring perpendicular distances from a transect;

The disadvantages of this method are that:

- objects may be disturbed by the observer as he or she approaches the point. If they are ignored density will be under-estimated;
- many objects may be detected, but cannot contribute to the data set, whilst the observer is moving between points;
- the presence of the observer at the point will probably affect the distribution of objects, resulting in fewer values close to the point than would be expected, hence complicating analysis.

Ideally the point count should represent a point both in space and time. Practically this is not possible as it will take time for the observer to detect and record data for all objects. If you are working on the entire bird community, which may include more than 400 species, this is a daunting task. Data collection from each point should be undertaken in a specified length of time. This should be optimised to maximise the proportion of objects detected and minimise the chances of objects moving into sight from outside, or moving far enough such that they are counted twice by mistake. Typically a period between 2 - 20 minutes is chosen, depending on the habitat and the number of species included in the study.

As for line-transects, you should aim to collect a minimum of 60-80 observations for each species of interest. It will be necessary to undertake some pilot samples to assess the number of point counts necessary to obtain a sample size of this magnitude. Having done so, sample points should be located randomly or in some systematic fashion in each area or habitat to be sampled (see Chapter 3). The DISTANCE program can be used to analyse data, but once again, the simplified analysis in Box 13f can be used whilst in the field as a first approximation.

This method is unlikely to be chosen for surveys in large, remote areas, due to the amount of time that would be lost moving between points (unless it could be combined with related work which necessitated such travel). However, it may prove particularly useful for assessing densities of different bird species in complex vegetation mosaics (such as areas of mixed forest and savanna, or in disturbed and undisturbed patches in logged forest), or for detailed studies in restricted areas of particular interest.

3) Recce walks

The aim of recce walks is to assess animal populations and evaluate human activities during surveys in remote parts of various protected areas / project zones, and to map vegetation types. This methodology was developed for data collection on reconnaissance walks, such as those undertaken during preliminary surveys; but recent studies comparing recce data with data collected along line-transects suggests that this methodology has good potential for monitoring trends in animal populations in remote areas. The basic principal is to take the path of least resistance to move across a remote area, by intentionally walking through forest types with clear under-storey, along elephant or human trails, up water-courses, etc. and avoiding dense vegetation where passage is inevitably slow. Data collection is similar to that for line transects, but this method is intended to allow one to cover ground more rapidly; you are not restricted to movement in a straight line, and fewer measurements are taken.

The disadvantages of this method compared to line-transects are that:

- the sample is not representative of the study area as a whole because certain vegetation types are avoided and others selected preferentially;
- since animal or human trails are often followed, signs are concentrated along the trail;
- recce analyses cannot be used to calculate density.

The advantages of this method are that:

- observers are not slowed down by the need to cut a straight transect and will therefore be able to cover 10-15 km per day compared to 2-3 km on transects;
- because the speed is increased greater distances can be covered in the same amount of time. Hence a smaller team can cover a greater area, decreasing cost and increasing efficiency;
- because observers tend to follow existing trails they have little need to cut vegetation. Therefore recce paths are less likely to be adopted by poachers after the passage of researchers, and damage to vegetation is minimised (see Box 13b);
- recce data can be collected during the course of other activities. For example, during an anti-poaching patrol into a protected area certain recce data could be collected to permit trends in human activities or animal numbers to be monitored over time. If such data are collected routinely on missions it will give you a means to assess your effectiveness and to identify priority areas for future anti-poaching action. Once incorporated into the routine, data collection will not slow your team down significantly, and any slight delays are far outweighed by the gains.

The results generated by recces will be indices of abundance of mammal species and human activities, and information on distribution of vegetation types. Unfortunately these indices are biased and are of very limited use for comparison between different areas. However, recent studies in several sites have shown that, at least for elephant dung and gorilla nests, there is a good correlation between data collected during recce walks and line-transect data collected in the same area. Therefore, it is strongly recommended that you undertake a comparative transect survey at intervals along your recce walks in order to evaluate the relationship between recce data and animal densities. This will enable you to evaluate the bias in the recce method and it may be possible to actually estimate animal abundances from data collected on recce walks (see Boxes 13n).

Practical details

- Data collection during recce walks is very similar to that undertaken on transects, with the exception that perpendicular distances are not measured (this in itself saves a fair amount of time).
- Researchers require the same degree of training and knowledge.

- Efficiency of data collection will vary with speed of travel and state of mind. If short of food, with a long way to go, after an arduous survey, you will miss sign you would otherwise detect. Keep a careful note of the circumstances and note time at each 1 km covered. If you have two trained assistants one should walk close to a local guide who opens up a passage. This first assistant should ensure that the guide remains on the correct approximate bearing and should check for terrestrial signs of animal presence. Deviations up to 60° are acceptable but an overall course should be respected as far as is practical; any deviations greater than 60°, for example to follow a particularly large elephant trail you suspect may lead to a bai, should be clearly noted. The second assistant should concentrate their attention in the trees, looking for nests and primates. If the team is larger a number of observers can check both on the ground for dung and sign, and in the trees for nests.
- Note the time you start moving in the morning, the time you stop at the end of the day and distances and times of any rest periods.
- You will generally be spending several nights in the forest so be sure to take standard camping, catering and medical supplies. Equipment needs are fairly basic: a compass for each researcher; GPS; topofil and sufficient string (best to take a spare topofil in case of disaster); altimeter; binoculars; tape measure; bags for collecting any objects of interest; checksheets; a small, light plant press.

What data to collect?

Route, navigation and physical features

With access to GPS equipment it should be relatively easy to accurately plot the route of travel, but if you do not have a GPS, compass navigation works well if you are careful. The location of the start of the walk should be noted (GPS point in the nearest convenient open area if necessary - see Box 6g) and waypoints should be taken along the walk as opportunities arise, particularly at prominent points likely to feature on maps, photographs or satellite images, such as river confluences.

The census route should be measured using topofil. At 100m intervals note the direction (compass bearing) of travel.

A running relevé of vegetation type, slope, altitude and physical features of note should be recorded, as for line-transects (see Figure 13.4).

Note if you are travelling on a human trail, on a major elephant path, on minor game trails or cross-country on a compass bearing. In addition note the location, size (major, large, medium, small), state (is it swept clear by passage of animals - active; are leaves accumulating - recent use; or is it abandoned) and direction of elephant trails.

Other information

Information on animals (sightings, dung, nest, tracks etc.), vegetation types and human sign is recorded as for line-transect data, with the exception of perpendicular distances, which are not recorded. As for line-transect sampling a great many different sets of information can be collected, and what you actually decide to record will depend upon your specific objectives. Once you have decided upon what research question you want to ask you should think carefully about what information will be necessary to answer your question. Once you have done this, you will need to undertake a pilot study to see if it is feasible to collect this information, given your financial and time constraints and the conditions on the ground. In particular you will have to decide what level of detail to collect. Will you record all human sign, or just camps, fires, cartridges, snares etc.; will you record all elephant sign, or just sightings, dung and major paths; will you record all birds sighted, or just a few rare or important species?

It is difficult to make firm recommendations, since each situation will be different. Furthermore, this methodology is still in the development phase. Trials to date have shown good correlations between line-transect data for elephant dung and gorilla nests, and a weaker relationship for duiker dung. If these relationships hold up and can be extended by further studies, this methodology will become more widely used.

Box 13n COMPARING DATA FROM RECCE WALKS AND LINE-TRANSECTS

During a large mammal survey in and around Kahuzi-Biega National Park in RDC, Jeff Hall and colleagues undertook line-transect and recce counts of gorilla nests in the same areas. Line-transects were located from randomly chosen start points and crossed major drainage features in a number of zones defined by distance from centres of human population. Several parallel transects were cut in each zone. Recce walks were undertaken prior to line-transects, to establish baselines from which transects could be located, to identify sites for base camps and to check that assumptions made during stratification were valid. Recce data were also collected by teams returning from the end of transects to the base camp.

Recce routes were plotted on a map of line-transects. A hypothetical line was drawn half way between each line-transect. Each reconnaissance was then divided into segments corresponding to areas either side of each transect, as delimited by the lines between transects. The relationship between gorilla nest site encounter rates on line-transects and recce walks was then plotted (Figure B13.13). There was a correlation between the two sets of data. The relative encounter rates on line-transects and recce walks were related by the equation describing the straight line fitted through the points on Figure B13.13. Hence, in this case recce walks gave results which correlated with line-transect gorilla density estimates.

In a study of the value of recces for assessing elephant densities, a series of 20 1-km transects were cut in three study areas in the Gamba complex in southwestern Gabon (Walsh & White, 1999). The number of elephant dung piles visible from each transect were counted and a recce walk was undertaken, starting from a point displaced 50m from one end of the transect. The dung counts for each transect-recce pair were then compared (Figure B13.14). There was a strong correlation

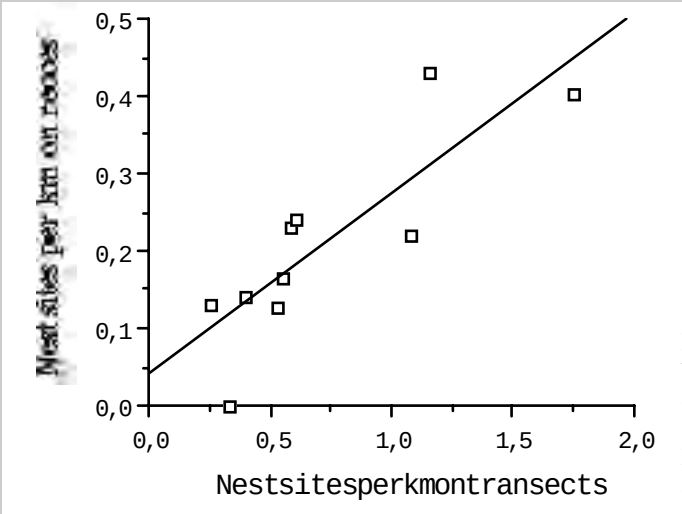


Figure B13.13 Relationship between gorilla nest site counts on transects and recce surveys in Kahuzu Biega National Park, eastern Zaire (from Hall *et al*., 1998).

between the two sets of data, once again suggesting that recce walks have potential for providing accurate density estimates.

New studies are now underway to assess the relative advantages and disadvantages of the recce and transect methodologies. It is possible that the additional distance that can be covered per unit time / effort on recce walks will result in density estimates with similar confidence limits to line-transect surveys. The Lopes computer program will soon include a module to allow researchers to undertake their own recce-transect comparisons and as more such comparisons become available we will be better able to assess which methodology is best suited to large scale survey work.

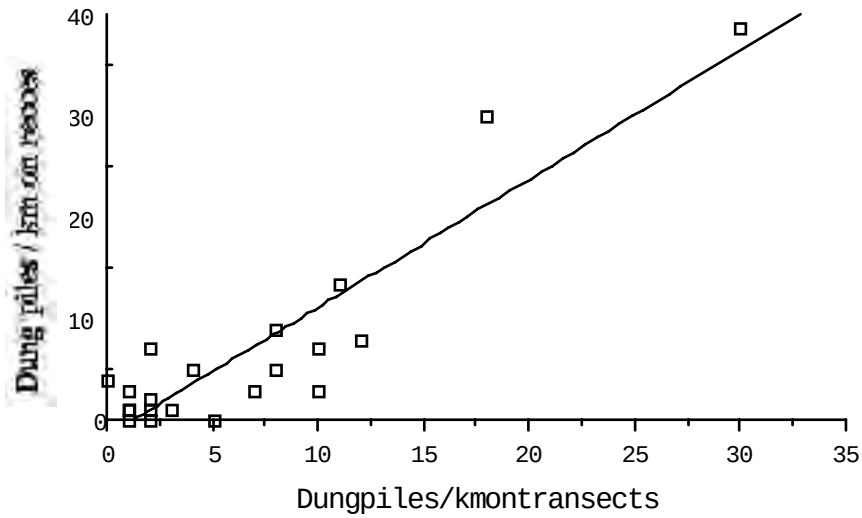


Figure B13.14 Relationship between elephant dung counts on transects and recce surveys in the Gamba complex, Gabon.

Recce-transect surveys: Density calculation using transects to convert recce encounter rates.

In the last few years a new method referred to as the "recce-transect" has been developed and tested for surveying large mammals, particularly elephants (see Walsh & White, 1999). This method involves walking a path of least resistance following a general compass bearing, with short transects cut perpendicular to the direction of travel at regular intervals (often 1km). This is represented schematically in Figure 13.6.

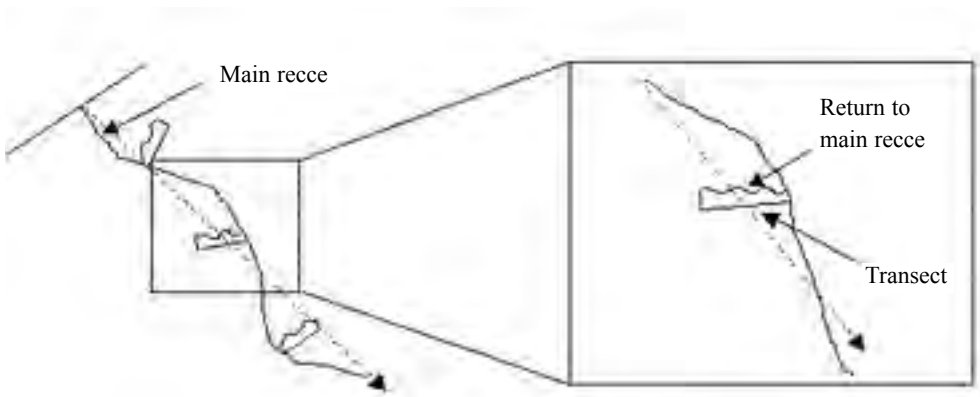


Figure 13.6 Layout of a recce-transect survey

The recce-transect method provides much of the efficiency of recces, but includes some transects which are used to calibrate the biased recce data. Using the data from the transects you can assess and correct the bias. This is particularly important when comparing different areas because the bias will probably change as a function of vegetation, topography, season and even observer. The great advantage of transect analyses is that they take all these factors into account when the detection function is calculated. For example, if only recce data is collected for two zones by two different teams, and there is a three-fold difference in elephant dung encounter rate, it is impossible to say if this is because the density was three times higher in one zone, or because visibility was three times better due to a difference in vegetation, or because one team was three times more observant - all are possible! Using the recce-transect method confidence limits can be attached to the index and you know that any differences detected between zones are real.

When using the combination of recces and transects the following guidelines apply:

- Do at least 10 and preferably 15-20 recce-transect pairs per stratum. They should be located randomly or systematically from a random start point.
- 30% of effort (in hours) should be spent on transects; 70% on recces.
- Transect length is dependant on the size of the deviations around thick, steep or swampy patches. Short, frequent transects are appropriate where the habitat is open (and therefore deviations are small); longer transects are appropriate where the habitat is heterogenous (with thick patches of lianes, for example) or mountainous, and deviations are therefore larger.
- The formulae for estimating density on recces which have associated transects are as follows:

Density of objects on transects:

$$D = \frac{N_t}{2L_t WPTF}$$

where,

N_t = number of objects on transects
L_t = length of transect
W = width of strip surveyed on transects (minus the furthest 5% of observations)
P = detection probability on transects
T = dung decay rate
F = defecation rate

Density of objects on recces:

The number of objects on recces is a function of the relation between the encounter rate on recces and the encounter rate on transects, as in Figures B13.13 & B13.14.

Thus,

$$N_t / km = \mu N_r / km$$

where,

N_r = number of objects on recces

Thus,

density of objects on recces, D =

$$D = \frac{\mu N_r}{2L_r WPTF}$$

W and P are calculated from the transect data.

Density of objects on recces plus transects for each segment:

$$D = \frac{N_t + \mu N_r}{2(L_t + L_r)WPTF}$$

The confidence intervals should be determined by bootstrapping, because the recce and transect in each pair are not independent.

These calculations, which are relatively complex, can be undertaken using the computer program "*Lopes*" written by Peter Walsh, available free of charge from the Wildlife Conservation Society if you did not receive a copy with this manual.

4) Sweep censuses of diurnal animals

Sweep censuses (e.g., Whitesides et al., 1988) are a modification of the line-transect method. Several parallel transects are cut either 50m or 100m apart (depending on visibility and study species). Each transect is then walked by a different observer (or team), with the aim of detecting all animals in the area to be surveyed. Data collection is as for line-transect censuses, but observers need to advance at the same rate and a system to ensure that progress is uniform has to be agreed upon. For example, watches can be synchronised (this is good practice anyway) and times at which observers will reach certain distances can be set. Observers ahead of the pace wait for their colleagues to catch up.

Using this method adjacent observers will often encounter the same group, or hear movements of animals which are observed by their colleagues. At the end of the survey all observers sit down together and go through their notes in order to work out how many groups there were in the area 'swept'. In addition, groups heard but not seen can be located by triangulation (see Chapter 6) if two or more observers detected them.

The area censused is calculated by summing the total distance between the outer two observers, adding a distance equivalent to the distance between trails (to allow for observations by the outer observers away from their colleagues, and multiplying by the length of the lines (Figure 13.6). However, perpendicular sighting distances should be analysed for each species sampled in order to check that it is reasonable to expect that all individuals or groups of each species have been sampled. In this case inspection of a bar chart of perpendicular sighting distances will suffice (it is not necessary to do a complex analysis of the detection function using Lopes or DISTANCE). If the sighting frequency does not fall off significantly before a distance equal to half the distance between observers (50m if transects are 100m apart), it is reasonable to expect that the observers will indeed manage a complete count. Animal densities can then be calculated easily since the area sampled and the number of groups detected are known.

This method can be used for many different taxonomic groups, although it has most often been applied to primates and other large mammals. It is poorly adapted for survey work in large remote areas, since it is very labour intensive, but can be used to study areas of particular interest in detail. It is also particularly well adapted for undertaking surveys in forest-savanna mosaics, where animals live in narrow forest galleries and isolated forest patches, in which it would be difficult to locate line-transects of any length.

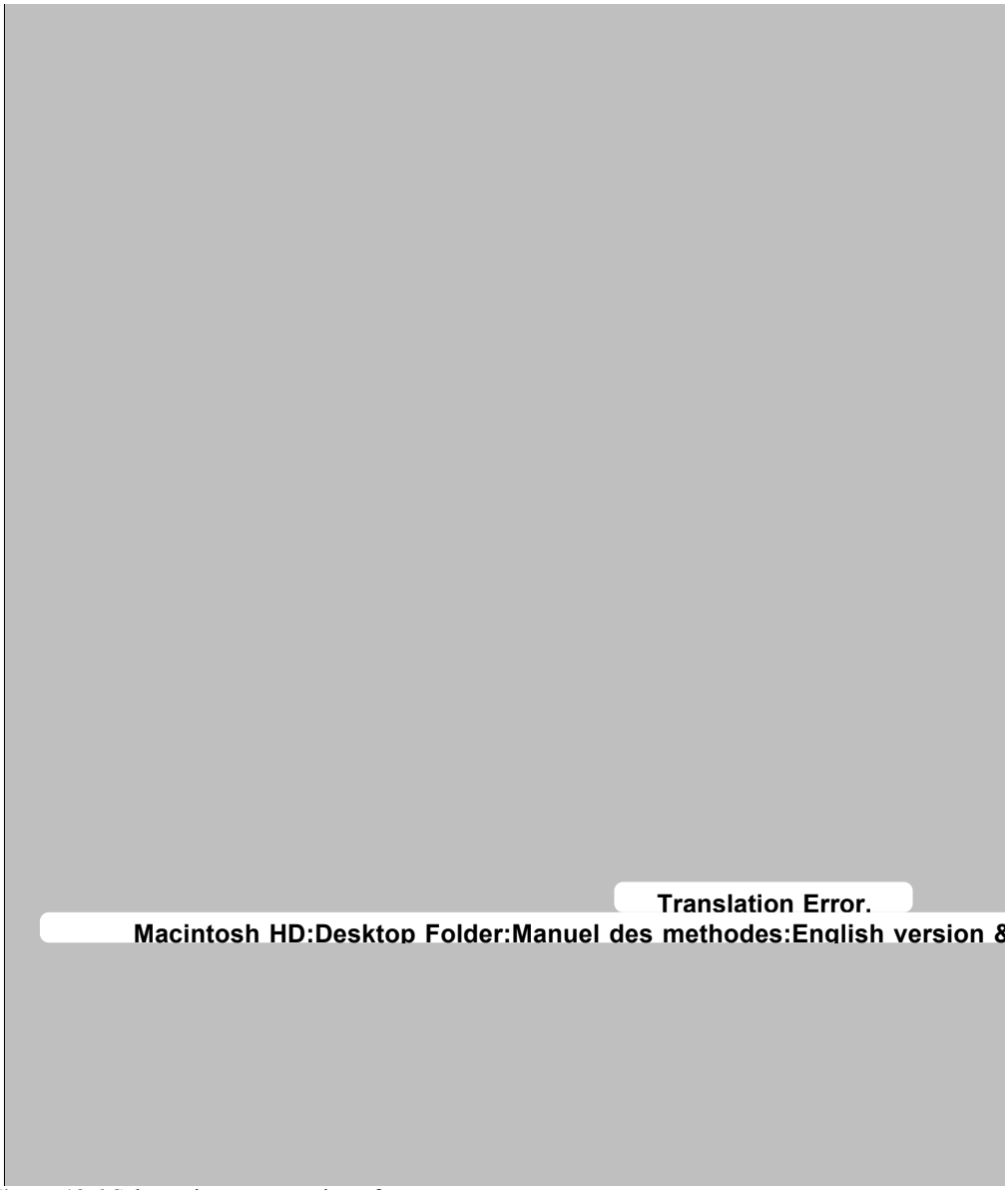


Figure 13.6 Schematic representation of a sweep census.
Three groups of primates were observed and their group spreads mapped. Groups more than 50m to either side of the outer observers were recorded but not included in the analysis. Three observers took bearings on a male vocalisation to the northeast and were able to use triangulation to plot its position.
Area sampled = 400m x 500m, plus 2 x (50m x 500m) = 25 ha (half the distance between observers is added to account for observations on either side of the outermost transects).

5) Monitoring animals concentrated in forest clearings ("bais")

A method which is proving extremely useful and much less labour intensive than line transect surveys is 'bai' monitoring. A bai is a forest clearing, generally located on a water course, where large numbers of mammals (particularly elephants, gorillas, buffalo, bongo, sitatunga and forest pigs) concentrate to feed on mineral salts or aquatic vegetation. These occur across the African forest system from Nigeria to eastern RDC, and in places are an important feature of the vegetation. In that bais concentrate animals in an open area they are a favourite hunting ground for poachers, big game hunters and local communities. Frequency and timing of animal visits into bais changes in response to this hunting pressure - if hunting occurs animals enter the bais less frequently and generally at night. However, if protected from hunting, bais represent an ideal situation for making observations on some little known species. Hence, by observing animals in forest clearings you can both protect key areas from poachers whilst collecting data of use for long term monitoring of the state of your ecosystem (and at the same time develop a resource which may attract tourists).

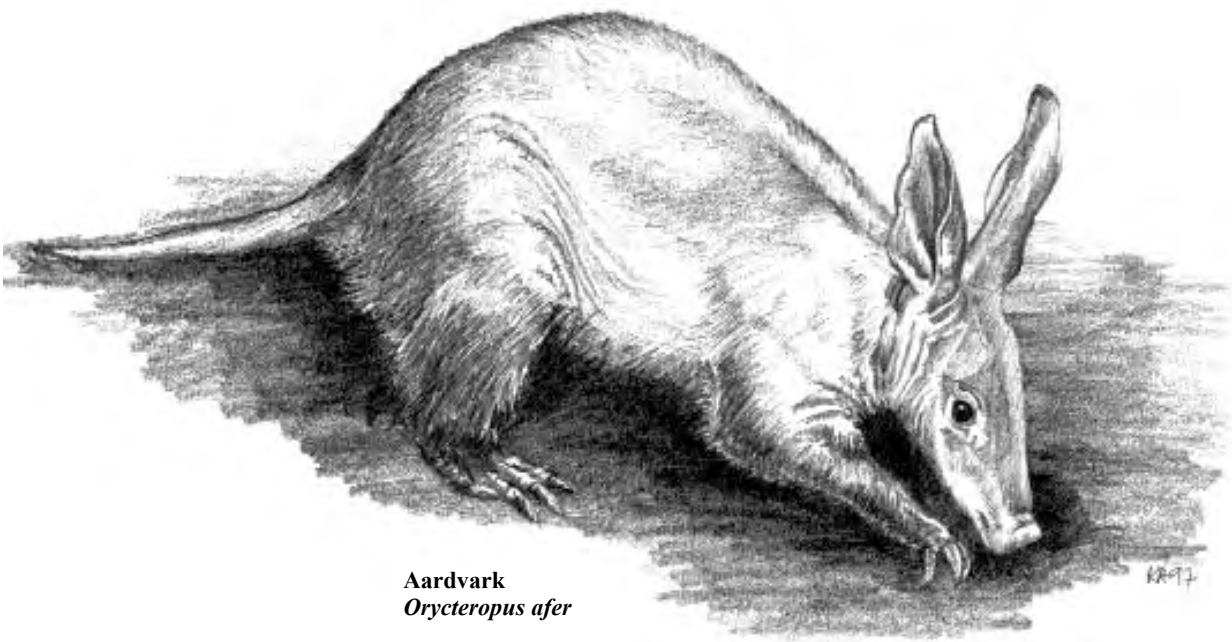
The most spectacular example of this method to date is the Dzanga saline in Central African Republic (Figure 13.7), where Andrea Turkalo has recorded visits from 2215 different, individually recognised elephants (Turkalo & Fay, 2000). During a 5-year study Turkalo has begun to piece together many aspects of the life of the little known forest elephant based on observations from a platform located at the edge of the Dzanga clearing. With so many elephants visiting the clearing it is evident that they travel long distances to feed on the mineral rich soil at Dzanga. Therefore, by monitoring the elephant population from her platform Andrea Turkalo should be able detect the effects of large scale poaching anywhere in the Dzanga elephant populations' range and warn protection staff in the Dzanga-Sangha National Park of any worrying trends.

Methods for sampling in bais are simple. First ensure that the area is protected from hunting so that animals visit during the day and behave naturally. If the bai was heavily hunted before you began your study it will probably take about a year before animals start coming in in any numbers during the day. Set up a platform in a strategically located tree with a clear view of the bai. The platform can be manned permanently, or at times when there is most activity in the bai (which you will be able to evaluate after a pilot study). Make a large scale map of the bai and its major features, such as the water course, large elephant trails and other landmarks (see Chapter 6).

Figure 13.7 Elephants in Lobéké (left) and Dzanga Bai (right)

Record the locations of all mammals (and other species, such as African grey parrots, if possible and useful) in the bai every 15 or 30 minutes (see scan sampling, Chapter 14). If possible record also the time and location of entry and exit for all mammals. For certain key species, such as elephants, gorillas and bongos, you may choose to undertake more intensive observations (see Chapter 14). It is useful to build up notes, sketches and photographs of known individual animals: elephants can be identified using features such as tusk size and shape, cuts or prominent veins in the ears, as well as by association with other members of their group (see Kangwana, 1996); gorillas need to be observed for quite long periods but, once experienced, an observer is able to recognise them quite easily; and bongo can be recognised using stripe patterns on their flanks (Paul Elkan, personal communication).

As your study and protection continue, frequency of animal visits is likely to increase and plots of the number of new individuals visiting your bai per unit time will give some idea of the percentage of the local population that you have recorded (see Chapter 9 & Box 11a). With time you will accumulate enough data to evaluate social organisation and calculate inter-birth intervals, age of sexual maturity, growth rates etc. for little known forest mammals (see Chapter 14). Such information is of great biological interest and is needed for setting hunting quotas for subsistence or sport hunting and predicting the responses of these key species to disturbance.



Aardvark
Orycteropus afer

CHAPTER 14
Behavioural ecology data and its relevance for management

Kate Abernethy

Why behavioural ecology data is useful for animal population management

This manual focuses on the management of large protected areas, habitats or ecosystems and their populations of animals and plants. Data on animal densities are often collected without direct observation of animals, through counting of their tracks, nest sites or dung piles (see Chapter 13). Although such data can provide valuable insights into the species that exist in an area, the density of each species and its distribution over different habitat types, they cannot tell you about how animals behave, or the details of the life of an individual animal. In order to understand which factors affect the behaviour of an animal it is necessary to spend time observing the animal and recording its interactions with its habitat, the other members of its society and other animal species. This is the study of behavioural ecology.

Observing animal behaviour in their natural habitat can provide information which will directly affect management of the population by enabling you to understand the animal's needs.

To assess the type of habitat animals need, you may want to know:
- what they eat and how they find their food;
- whether their diet changes seasonally;
- where they sleep or shelter;
- whether special 'breeding grounds' are used for mating or giving birth.

To decide how many individuals of a species live in an area, you may also ask:
- what is the size and composition of the group the animal lives in (how many males, females and young) and does this change through the year, or in different habitats;
- whether the animal defends a territory or interacts non-aggressively with members of its own or other species.

Then to decide whether the population you have can survive and breed in this area in the long term, you may want to add to your knowledge:
- what time of year they breed or how often they breed;
- how many young are produced;
- whether they need special resources (like nest materials or sites) during the breeding season, which are not used during the rest of the year;
- how they find and choose mates;
- whether they establish breeding territories;
- what age males and females start to breed.

And to decide how important individual animals are in this population (and so predict how other members may be affected by their loss):
- which animals defend or lead a group;
- how long young are cared for by the mother or other members of the group.

Finally, if you are a protected area manager, you may need to resolve conflicts between animals and humans in your area, or (better) to predict where problems may arise and adjust your management plans accordingly. You will be more able to do this if you know:

- what animals do that bring them into contact or conflict with humans in your area;
- whether all members of the species behave like this or only some individuals;
- whether these behaviours are seasonal (for example linked to migrations or to breeding seasons) or whether they occur all the time.

Good data on the behavioural ecology of a species provides important information for good management. Think about what you would need to know in order to decide whether an animal population is able to support some hunting in your area, perhaps for sport hunting permits or to set quotas for local villagers' needs. You need first to calculate how many animals could be killed in a year without the population declining, then to decide whether hunting will account for less or more than this number. To make these decisions you need (at least) to know how big the existing population of animals is and also how fast it reproduces (how many young are born each year) and how many animals die naturally, without hunting, in a year, i.e. the population turnover.

You may be able to estimate the size of your current population from a census, using some of the methods discussed in Chapter 13, but without reference to a study of the behavioural ecology of the species, you will be unable to estimate the breeding potential of this population; how old these existing animals are, whether all of them can breed or only a few, whether they will produce many young per year or only one or how long they may be expected to live and continue to breed, and so you will not yet be able to predict how hunting will affect the population. Nor will you know what roles various individuals play in the animal's society and how their deaths may affect other members. For some species, like elephants, if a large female who defends and leads the group is killed, her offspring will be very vulnerable and may also die later. If it is a juvenile that is killed, the group may be able to survive afterwards without problem. A hunting bag of five animals may really mean a large loss of several groups of animals if all five deaths are large, group-leading females, but it may represent a much smaller impact on the population if all the dead animals are young, non-breeding individuals.

Without both the data on population sizes, densities and distributions that you can gain from censuses, and a good knowledge of the species' behavioural ecology and life history, you are not in a position to properly manage the animals in your area, or to understand how changes in their environment, like increased hunting pressure or habitat loss (or even your own management decisions) are affecting them.

The collection of many aspects of behavioural ecology data requires patient observation and study of animals over long periods of time, in order to fully understand seasonal changes in behaviour and behavioural changes through the lifetime of the animal. Unfortunately, because collection of the data requires such a large investment of time (though not necessarily large expense), the behavioural ecology of most animal species in the African rain forest is still poorly understood and the effectiveness of our management of them remains guesswork. However, several long term studies of large mammals do exist and can provide some data that can be used in your area, if you have no locally collected data (see the section below on where to look for existing information). Results from specific studies may not always be applicable to different areas, but in the absence of any other knowledge, they are better than wild guesses.

This chapter will discuss some of the sorts of information that behavioural ecology studies in the African rain forest can provide and how this information has been or could be relevant to management. The text goes on to explain how you might start collecting data on an unknown population in order to answer some of your own management questions, and how, if you find you have the resources, you could develop longer term studies. Behavioural studies of single species, especially those which are to date relatively unknown, are not necessarily costly or difficult to carry out alongside other management activities and can provide essential data for management.

The types of information you may want

There are several aspects of an animal's life that may interest you. This section discusses a few important areas of behavioural ecology and why they are relevant to management issues. A lot of data can be collected on these subjects in the course of your daily activities, using opportunistic sampling (see below). If you want to pursue a more detailed behavioural study, creating the conditions for observing animals and recording their behaviour on a regular basis are discussed in a methods section below. The methods (both simple and detailed) most appropriate for studying each aspect of an animal's behavioural ecology are outlined in Figure 14.1.

PRIOR KNOWLEDGE	SUBJECT OF INTEREST					
	Diet	**Activity rhythms**	**Group sizes & compositions**	**Territoriality and ranging**	**Reproduction**	**Rare behaviours or interactions**
Unknown animals	Dung analysis Opportunistic observation	Opportunistic observation	Opportunistic observation	Camera traps Opportunistic observation	Post mortem data Opportunistic observation	Opportunistic observation
Regularly observable animals	*Ad libitum* observations Scan sampling Focal animal watches	*Ad libitum* observations Scan sampling Focal animal watches	*Ad libitum* observations Opportunistic observation	Focal animal watches Opportunistic observation	*Ad libitum* observations Focal animal watches Opportunistic observation	*Ad libitum* observations Focal animal watches Opportunistic observation
Habituated or known, identifiable individuals	*Ad libitum* observations Scan sampling Focal animal watches	*Ad libitum* observations Scan sampling Focal animal watches	*Ad libitum* observations Opportunistic observation	Radiotelemetry Focal animal watches Opportunistic observation	*Ad libitum* observations Focal animal watches Opportunistic observation	*Ad libitum* observations Focal animal watches Opportunistic observation

Figure 14.1 Behavioural ecology subjects you may want to study and methods to choose for different levels of recognition of the study animals

What the animal eats

One of the most important things for an animal population is its food supply. Animals must be able to find food and water throughout the year if they are to remain resident in an area. If they cannot find sufficient food or water, they will migrate to different areas in search of them, or if they cannot do this, perhaps because they are too small or weak to travel to areas where food is available, or because they are prevented by fences or other barriers, they will die of starvation. If you have a good knowledge of the dietary needs and preferences of the animal populations you study or manage, you can then monitor the availability of their food supply, which in turn enables you to identify key food species or areas to protect and predict times of food scarcity for the animals (see Box 14a). Times of hardship may occur naturally in dry seasons or during cold periods, but they may also be artificially created by human influence - like the felling of certain fruit-producing trees, or the tilling of herb rich grasslands for agriculture. Times of abundance in the year are also important as these are likely to be times when animals will breed.

If you understand the diet of an animal, you will be better able to interpret its ranging behaviour in relation to food availability, and to decide if a protected area provides sufficient resources for the whole year (see also box 13d). If it does not, your animals may migrate out of the park and be vulnerable to hunting at some time every year. In this case you are not fully protecting them.

Understanding diet will also show you some of the ecological relationships between plants and animals that are crucial to the survival of both. For example, the relationship between the forest elephant and the moabi tree (*Baillonella toxisperma*). Moabi fruits contain seeds so large that the only animals that can swallow them are forest elephants. Elephants find large amounts of food at moabi trees, because the fruits fall to the ground when ripe (abscise), rather than staying on the tree for primates, squirrels or birds to find. For the elephant, moabi fruit are important in its diet and influence its ranging, as it moves from one fruiting tree to the next. For the moabi tree, elephants are vital to the survival of its offspring, as they carry the seeds away from the parent tree to new, favourable places to grow, and deposit them in a dung heap of natural fertiliser to help them get started.

Collecting data on diet can tell you:

- what an animal needs to eat;
- whether its diet changes seasonally;
- how much of each food is eaten.

With this information you can look at the food supply available in your area and decide whether it is likely to be able to support the animals you have. To decide how to collect data on diet, look in the sections below on collecting behavioural ecology data of your own.

Terms used to describe diet

There is some commonly used terminology to describe an animal's diet; knowing these terms will help you describe what you see to other researchers and to look up information in published references.

Animals that eat plants are called *herbivores*. This can be further classified:

- animals that eat only grass are grazers; e.g. zebra or savanna buffalo;
- animals that eat woody material from shrubs and bushes are browsers; e.g. elephants or bushbuck;
- animals that eat leaves (other than grass blades) are folivores; e.g. some species of colobus or caterpillars;
- animals that eat fruits are frugivores; e.g. chimpanzees or fruit bats;
- animals that eat seeds are granivores; or seed predators e.g. parrots or black colobus;

- animals that eat insects are insectivores; e.g. pangolins or galagos;
- animals that eat meat (including fish and birds) are carnivores or predators e.g. golden cats or pythons;
- animals that eat a mixed diet are omnivores; e.g. African civets or rats

Often an animal has one type of food that it eats most of the time, but may sometimes eat another type and it is difficult to decide which category or niche, it fits into. In fact this is more often the case than a very specialised diet. Many species also change their diet at different times of the year, depending on food availability. Within a species, diet can change between local populations or subspecies, depending on food availability in their area. The diets of the forest and savanna subspecies of elephants or buffalo are different, as are the diets of mountain and lowland gorillas. Western lowland gorillas eat mostly fruit, but sometimes they also eat insects or leaves, especially at times of low fruit availability (see Box 14a). Even though their whole diet is not fruit, they are still called fruit-eaters (frugivores), because fruit is the most important part of their diet. Mountain gorillas, however, who have little fruit available to them, are folivores, eating mostly leaves and stem pith.

It is acceptable to describe animals using two categories; you could, for instance, call forest elephants browsers/frugivores as both fruit and shrubs are important to them. The terms and examples below that are not fixed rules, they are meant to help you see patterns in the ecology of different species and to think of them in groups.

When the animal is active

Animals do many different things during the course of a day. On a daily basis they need to eat and sleep, and therefore also to travel from safe places to sleep to places where they find food. During the course of a year they may also need to travel to find mates or migrate to new food sources. If you want to see animals, and to be able to show them to visitors, it helps to understand their activity patterns and predict where they will be and what they will be doing at certain times of the day, or seasons of the year (see Box 14b).

For animals, travelling to find food or mates is tiring. If changes in the environment force them to travel further or for longer than they normally would, they can become exhausted and even die. If the habitat in an area is changing, perhaps through human colonisation or logging, drought, erosion, hunting or climate change, animals may be forced to travel further and search harder for food. If you know the usual activity patterns of animals, you will notice if their behaviour changes and can try to find out if there is a problem.

Terms used to describe activity patterns

Animals are generally either active by day (diurnal), or at night (nocturnal), or in two periods at dawn and dusk (crepuscular). Some animals, particularly large herbivores like elephants, buffalo or pigs, are active both day and night. In order to take data on activity patterns, watch the animals at all times of day and night to establish which of these categories best describes them. Do this either by following individual animals or groups all the time and noting all of their activities, or by searching for animals at different times of the day and noting the activity of several different groups during one time period. For better studied species, like most primates, the general activity pattern (continuous, nocturnal, diurnal or crepuscular) is already known and you can look up the information in guide books and specialist references (see below). However, animals can change their activity patterns in different areas. For example, otters are nocturnal in areas where there is lots of human activity in their habitat, but can be completely diurnal if they are undisturbed.

Behavioural ecology data and its relevance for management

When you see group-living animals, members of a group are not always doing the same thing; some may be sleeping whilst others are feeding or grooming each other. In this case, you may want to take data that will allow you to calculate the most common activity at a certain time (to let you predict what you are most likely to find the group doing at a given time), or the average time one individual spends in each of these activities (for example, to see if they are spending less time resting and more time searching for food in a drought year). In order to do this, you need to record the behaviour with one of the observational sampling methods below (*opportunistic sampling, scan sampling, focal animal watches*).

Box 14a 'KEYSTONE' FOODS

The tropical rain forests of Africa are a seasonal environment where patterns of flowering, fruiting and production of young leaves are tied to periodic changes in rainfall and temperature. Consequently the amount of food available for rain forest animals changes with the seasons.There is usually a season in the year when fruit becomes scarce and many frugivorous species of large birds and mammals have either to change their diet or spend long periods searching for what little fruit there is.

Plant species which provide a dependable food source during periods of scarcity are known as 'keystone' species (e.g., Terborgh, 1986). These might be species whose leaves and bark are eaten, or they might be plants which consistently fruit when little food is available. This is true of palms, figs and trees in the Myristicaceae (nutmeg) family in many tropical forests. Such species can be far more important for rain forest birds and mammals than their relative abundance might suggest, and if they are harmed by logging or other forms of forest exploitation, there may be grave consequences for forest frugivores.

Keystone resources differ across the African forest system (e.g., Gautier-Hion & Michaloud, 1989; Tutin et al., 1991; Wranghan et al., 1991) and if you are planning to manage an area for timber production or other forest products as well as for wildlife, it is important that the key foods in your area be identified. This will involve your undertaking analyses of the diets of the animal species you intend to protect or manage, using observational methods to study feeding behaviour and / or dung analysis methods as well as analysing fruiting and flowering patterns (see Chapter 8).

Fruit is very scarce in the long dry season from June-September in Gabon, and during this period gorillas change their diet, spending long periods feeding on aquatic plants in marshes and swamps as well as consuming large amounts of bark and leaves from several tree and liana species. One tree which is particularly important is *Milicia* (*Chlorophora*) *excelsa* (Iroko). In the dry season it is not unusual to see whole groups of gorillas, including the adult male silverback, 30m up in the canopy of an Iroko feeding on leaves and the sugary inner bark of the terminal branches. However, the wood of this tree is valuable as timber and it is frequently cut by foresters. If all of the Iroko trees in the area used by a group of gorillas are cut, even if the remainder of the forest is untouched and it seems superficially that logging has been of low impact, gorillas might suffer unexpectedly because they have lost a key food upon which they depend during a period of scarcity.

Iroko
Milicia excelsa

Box 14b DAILY ACTIVITY PATTERNS IN MOUNTAIN GORILLAS

During his 1959-1961 study of mountain gorillas, George Schaller recorded the activity of groups of gorillas at the time when he first met them on each encounter. The data was taken *only* at the time when he first met the gorillas to be sure that his presence had not affected their behaviour. He recorded one activity for the whole group; either 1) feeding and/or moving along 2) resting or 3) feeding slowly and resting (this category was used when roughly half the animals were in each activity).

He recorded the activity of a group on 279 occasions between the hours 07:00 and 18:00. To interpret the data he lumped together observations taken in the same 60 minute time period on different days and calculated the percentage of all his observations in that hour that showed each activity. The percentages are shown in Table B14.1 and plotted against time in Figure B14.1.

Although he standardised his single observation to include only the occasions he met the gorillas between 07:00 and 18:00, he also took opportunistic data when he observed them earlier or later. He made notes on their nesting and rising times into a field book. Here are two extracts from those notes, taken from:

«Nov 17, 1959: Group VI.	All nests were built between 1805 and 1810.
Jan 3, 1960: Group VII	The whole group built its nests between 1800 and 1810, with the male constructing his first. The group was aware of my presence but seemed undisturbed.
Feb 7, 1960: Group VII.	The male built the first nest at 1805, but the whole group became aware of me after that.»
« Sept 8, 1959: Group IV	Some of the animals apparently awakened to the distant chest beating of a lone silverback male at 0530. The dominant silverback male was the first to leave its nest at 0600.
Sept 26, 1959: Group IV	A peripheral silverback male was the first animal to leave its nest at 0650. the last animal left at 0710.
Feb 8, 1960: Group VII	All animals left their nests between 0700 and 0705.»

Although there was variation in nesting and rising times, he did not have enough data to calculate a reliable 'mean rising time' or 'mean nesting time', so he used 07:00 as the rising time and 18:00 as the nesting time and assumed the gorillas were usually all awake by 07:00 and all in the nest by 18:00. For the hours for which he had more data, he could calculate a more precise activity pattern.

Table B14.1:

Time of encounter	Feeding intensively and /or moving rapidly		Some feeding leisurely; others resting		Resting		Total No. Obs.
	No Obs	Per cent	No Obs	Per cent	No Obs	Per cent	
07:00 - 07:59	9	100	0	0	0	0	9
08:00 - 0859	35	85.7	4	14.3	2	0	41
09:00 - 09:59	39	44.3	25	28.4	24	27.3	88
10:00 - 10:59	9	16.4	11	20	35	63.6	55
11:00 - 11:59	5	20.8	2	8.3	17	70.8	24
12:00 - 12:59	5	20.8	2	8.3	17	70.8	24
13:00 - 13:59	5	38.5	0	0	8	61.5	13
14.00 - 14:59	4	57.1	0	0	3	42.9	7
15:00 - 15:59	3	100	0	0	0	0	3
16:00 - 16:59	8	61.5	1	7.8	4	30.8	13
17:00 - 17:59	2	100	0	0	0	0	2
Totals	124	44.4	45	16.1	110	39.4	279

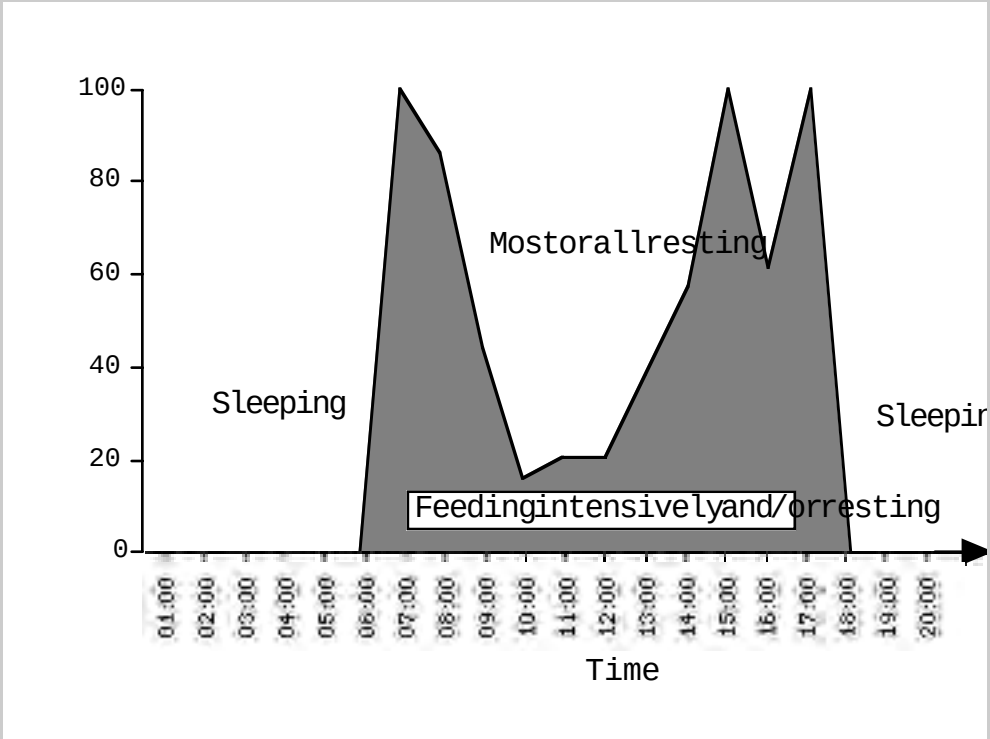


Figure B14.1. Mountain gorilla group activity at various times of day.
Data, graphic and field note extracts taken from SCHALLER, G.B. 1963. The Mountain Gorilla; pages 138 & 146; Figure 41, page 147; Table 26, page 360.

How many and what ages and sexes of animals live together in a group

The social needs of animals are important for the health of the population. Some animals live alone, others live in groups all the time. For group-living animals the structure of the group is very important; the ratio of males to females in a population (called the sex ratio) is not always equal, because one sex naturally dies younger than the other (generally there are fewer males than females). Also the proportion of young in the population changes if the population is growing (more young born than old animals die) or declining (few young born and many animals dying). The proportion of the population that is in each age class; i.e. infant, juvenile, subadult, adult is called the age structure of the population. The age structure you expect to see in a population (or a group within a population) depends on the species, its social system, whether it breeds every year and how many offspring are produced, and on the condition of growth or decline of the population.

Often for mammals, the most common group is a mother and her offspring, but several mothers and offspring may stay together to form a larger group. In some species, generally those that are relatively short-lived and breed quickly, like rodents, the offspring become independent very quickly and leave the group they were born in. In other species, especially long-lived ones like elephants or apes, the members of a family stay together for many years, perhaps throughout their lives.

The males of some species live separately from the females (except during the mating season); in others they stay with the females year round. For species in which the males and females live separately, it is important to know if both sexes are resident in a protected area, and if their needs are the same. To find this out, it is not enough just to count the number of individuals there are in a group, you need to make counts at differing times of the year and you need to identify the sexes of all the members of the group.

Knowing the 'normal' group sizes and compositions found in your population can help you to see changes and interpret them (see Box 14c). You will be able, for example, to detect if large male elephants are seen less frequently than before, from which you might deduce that they are suffering more from poaching than others. Or to see that the population has less infants than normal surviving, and so discover that it is no longer breeding successfully because of a drought year. You may also detect seasonal changes in group size or composition, for example during mating seasons, or birth seasons.

Why mean group sizes are needed to calculate animal densities from transect censuses

In order to census animals, it is often crucial to know the average group size. For example, during transect censuses of monkeys, what you actually count is the number of groups you see (see methods in Chapter 13). In order to then calculate the actual number of individual monkeys in the area, you need to know how many there were in each of those groups. It is usually impossible to count the individual monkeys from the transect (which is why you count groups), so you use an average group size to estimate the numbers in the area. If you spend time when you are not on a transect study, observing different monkey groups and counting them as they cross open areas (roads, rivers, savanna stretches or canopy gaps), you can build up a sample of real group sizes for different species. You can then calculate a statistically reliable 'mean group size' (see Chapter 4) to use in interpreting your transect census data. As collecting this sort of data takes time, people often use mean group size figures collected in other areas to calculate densities from transect data. This is often the 'best guess' but can lead to large errors. For example, in south Cameroon mandrills live in groups of 15-95, a mean size of about 55 (Hoshino, 1985). However, in central Gabon, mean group size is around 700 [excluding lone males](SEGC unpublished data); choosing one figure or the other will change your density estimate for mandrills by a factor of 14! It is always better to collect your own data, but if you have to use group sizes from other sites, try to match the animal species, geographic area, vegetation type, and hunting history as closely as you can to your area.

If you see animals often, or are on a long term study and can take group counts regularly, you may also be able to detect differences in group size in different habitats or in different seasons or years. This will not only improve your census accuracy, but will allow you to better understand how animals use and respond to changes in their environment.

Data on group sizes and compositions can tell you:

- what the mean group size is;
- if group size changes seasonally;
- if group size is different in different habitats;
- if males and females live together;
- what the sex ratio of the population is;
- what the age structure of the population is.

To find out how to collect this data, see the section below on starting to collect your own behavioural ecology data.

Where an animal goes

For all animals you can only make sensible decisions about population management if you know whether they are resident in the area you study or work to protect. Resident animals need to be able to find all their needs within the area, whereas migratory animals, or far-ranging species can move to other places. To understand the needs of an animal you must know how far it travels daily to find food, water or mates, whether it migrates seasonally and to where, whether it always sleeps in the same place and whether it is territorial or not. The size of the area an animal uses will be different in different habitats. For example, guenons in a forest with lots of plant species may have food available all the year round, whereas the same guenon species in a less diverse forest with fewer fruit trees may have a larger range area (see Box 14d).

Studying ranging is actually quite difficult. Just seeing animals in different locations tells you nothing, unless you can recognise that the same animal (or group) has moved from one place to another. When you can do this by following the animals constantly; so you know that they are the same ones, even if you can't recognise them individually. This is, however, usually extremely difficult in a low-visibility, forested habitat, and in any case, sooner or later you will need to stop to eat and sleep and may lose your study group.

If you can't follow the same animals constantly, you need to be able to reliably recognise them so that you can find them again in new locations as they range. This you can do most simply by knowing them as individuals, as you would other people. Occasionally you can also recognise individuals by the trail they leave; a particularly long footprint, or one with a missing toe for example are valid ways of recognising individual tracks. For some species where individuals are very variable in appearance and which you can often see, like bonobos, elephants or forest buffalo, this is possible (see Box 14e). For others where individuals are highly similar in appearance, or more difficult to see, like civets, this is very difficult. If you can't recognise individuals using their natural features, you may want to put identification (ID) marks onto them, so that you can be sure of correct recognition. Identification marks have the advantage that even inexperienced observers who don't 'know' the animals can still collect reliable data on their locations, using a catalogue of the marks; however, the problems associated with capturing and marking the animals shouldn't be underestimated. Natural marks by which you can recognise animals are invaluable, and you should make every effort to identify your animals without artificial marks before you decide to try to capture and mark them artificially.

Box 14c ELEPHANT GROUP COMPOSITION IN TWO PARKS IN KENYA

During long term research at the Amboseli Elephant Research Project in Kenya, a team of biologists has carefully recorded the behavioural ecology of a population of savanna elephants (Moss, 1988; Poole, 1996). The Amboseli elephants suffer some hunting, but are largely a well-protected population from which 'baseline' data on natural ecology in open savanna habitat can be inferred. The data from the Amboseli elephants has been used to deduce how other elephant populations have been affected by poaching and has also shown how natural changes in the environment can affect population structure.

Cynthia Moss and Joyce Poole have carried out most of the long term research on the Amboseli elephants since 1973. They learned to individually recognise all the elephants resident in the Amboseli study area, in order to study their behaviour over many years. By carefully observing and photographing animals of known age, they were able to estimate the ages of unknown animals and to document the age structure of the population, the compositions of groups of elephants and the way they were related to each other. They also recorded the dates of birth and death of animals, and intervals between births to particular females. In addition, they recorded data on the weather patterns, human influence and food availability which were to be invaluable in interpreting their data.

Natural fluctuations in age structure

The age structure of the Amboseli elephant population <15 years old looked like this in 1983 (Figure B14.2): Clearly the distribution is not uniform. There are fewer animals in the 6-9 year old class than would be expected, and more in the 0-3 year olds. The ecological data from Amboseli showed that in 1975 and 1976 (7 years previously), there had been a severe drought, during which 48% of the elephants born in 1976, along with several older calves, died. No females conceived in 1976 and there were only 2 births between late early 1977 and late 1978. This crisis accounts for the low numbers in the 6-9 and 9-12 year classes. In 1976 the drought broke and grass became plentiful. The elephants began to

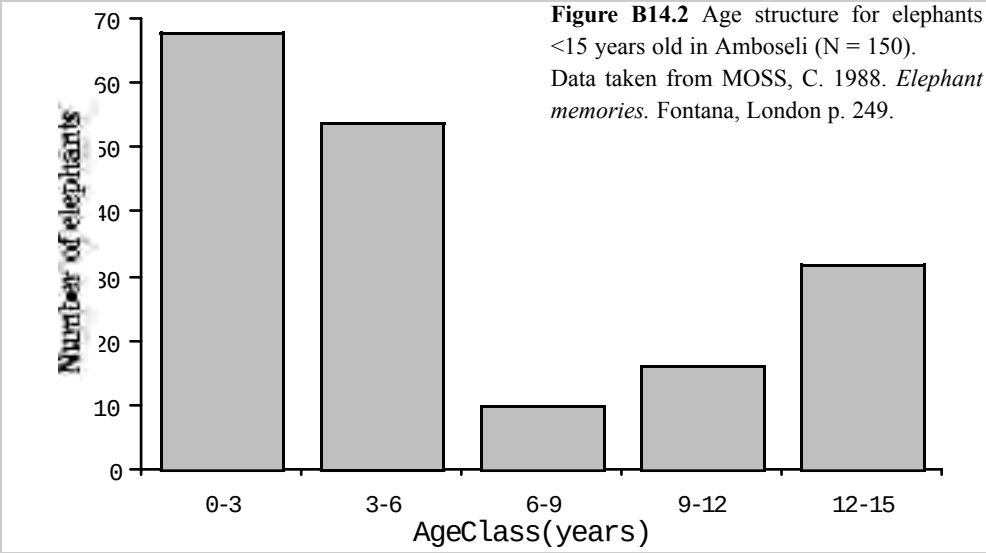


Figure B14.2 Age structure for elephants <15 years old in Amboseli (N = 150). Data taken from MOSS, C. 1988. *Elephant memories.* Fontana, London p. 249.

breed again and the calves conceived in 1977 and 1978 were born in 1979 & 1980, increasing the number in the 3-6 class. The abundance of food also led to a high survivorship (98%) of calves from this cohort. The mothers of these calves then conceived again in 1981 and gave birth in 1983, producing a large cohort in the 0-3 age class.

The effect of the drought was not just to suppress breeding and kill young elephants during the dry period. It also meant that the females who started to breed after the crisis all produced calves at the same time, for at least two generations after the drought. This may later mean that all the young females born together will mature and start to breed synchronously as well. Periods of abundance or hardship can be detected in the age structure of long lived animals many years after the event.

Poaching pressure assessed by group composition changes

During the 1980's many surveys of east African savanna elephant populations were undertaken as the increasing ivory trade led biologists to fear for the survival of the remaining elephant populations in Kenya in and Tanzania.

The Amboseli Elephant Research Project had amassed a pool of data on the composition of groups and this was used as baseline data to compare with the groups seen in other parks. The Amboseli dataset had shown that male elephants over 10-15 years old are usually solitary and that groups are composed of females and young. Female group sizes ranged from a mean of 15.1 during the drought of 1976, to a mean of 45.9 in 1979 when food was abundant. Groups are made up of family units, comprising a matriarch and all her surviving offspring up to 10 years old, when males leave the natal group. Female offspring may stay with the matriarch and give birth to their first calf at around 13 years old. Sometimes the birth of a new calf can cause the group to split and the new mother to leave.

In 1989, Joyce Poole undertook a survey of the elephants in Tsavo National Park (Poole, 1996). She counted and classified 842 elephants; 503 in Tsavo East and 338 in Tsavo West. The data showed a very skewed sex ratio in the breeding adults (males over 25 and females over 10 years); 82% females and 18% males in the east part of the park, and 97% female and 3% male in the west. Data from Amboseli show that there is an even sex ratio at birth, and so a skewed adult sex ratio is evidence of unequal mortality. In the Tsavo case, it showed that poaching was strongly biased towards the older adult males. Her behavioural research at Amboseli had shown that females prefer to mate with older, musth males, rather than younger males, but the Tsavo elephants had very few animals over 40 years old surviving. The prediction was that the remaining young males may not breed as well as older males and the capacity of the population to recover from the hunting would be weak

She then compared the compositions of the groups she saw in Tsavo to the 'normal' group from Amboseli of a matriarch and her offspring, including adult daughters (assumed 100%).The results are shown in Table B14.2.

Table B14.2 Group compositions in poached elephant populations in Tsavo compared to the protected population in Amboseli.

Data from POOLE, J. 1996. Coming of Age with Elephants. Hodder & Stroughton, London.

	National Park		
Group composition	**Tsavo East**	**Tsavo West**	**Amboseli**
Matriarch over 30 yrs and offspring including adult daughters	37	29	100
Younger matriarch and offspring including adult daughters	32	25	0
Matriarch and offspring, but missing one or more adult females	28	32	0
Orphans	3	14	0

The comparison of the populations of the two parks showed that Tsavo had not only lost many males, but had also lost a large proportion of the adult females. As females carry less ivory than males, this showed that the poachers were continuing to hunt even for smaller amounts of ivory. Sixty-three per cent of Tsavo East and 71% of Tsavo West's female juvenile elephants were surviving in families disrupted by poaching.

Poole then looked at the survival data for families in Amboseli that had lost adult females. She found that even without hunting, if a calf loses its mother before the age of 2 it will die. For those between 2 and 5 there is a 30% chance of survival and for those between 5 and10 only a 50% chance that they will survive to adulthood. She also found that calves are less likely to survive in families where there are no other females besides the mother, and that if the matriarch dies, a family unit will often break up. The chances for the calves in the disrupted families in Tsavo seemed very low.

The interpretation of the data on group compositions at Tsavo was much stronger evidence for the collapse of the elephant population than the simple counts. Together the behavioural and census data showing the decline of Kenya's elephants under severe poaching pressure provided the impetus for the Kenyan government to revise its policies on elephant protection and develop an effective anti-poaching campaign in its National Parks.

Box 14d RANGING BEHAVIOUR OF BLUE MONKEYS IN TWO DIFFERENT FOREST TYPES IN UGANDA

During a study in Budongo Forest, Uganda, Chris Fairgrieve (1995) used a 100m x 100m trail grid system to map the home ranges of four blue monkey (*Cercopithecus mitis*) groups, two in areas logged about 35 years before the study and two in unlogged forest. His intention was to investigate the effects of logging on feeding ecology, ranging and density of blue monkeys. He used a 15 minute scan sampling regime to record both feeding and activity data. Data were recorded from approximately 07:00 to 19:00. In each 15 minute period the scan lasted 10 minutes or until 5 individuals had been sampled, whichever came first. He scanned his groups from left to right and noted the first discernable activity lasting for at least 5 seconds. The movements of each group were recorded on a 1:3,000 scale map of the trail system at the end of every 15 minute scan.

Fairgrieve found that blue monkey densities were higher in logged forest compared to unlogged areas and that the home ranges of the two groups in logged forest were about half those of the groups in unlogged forest. Figure B14.3 summarises the ranging data for two of his study groups, one in logged forest (Group L2) and one in unlogged forest (Group UL1). These differences were thought to be due to the fact that tree species diversity increased following logging, due to the growth of a number of secondary species (see Box 9c) not found in unlogged forest, including several species of fig. This resulted in greater and more even food availability in logged forest, enabling blue monkey groups to obtain sufficient food from a smaller area, and hence to occur at higher density. This information is interesting for forest managers in Budongo and elsewhere, but bear in mind that such results must only be generalised to other sites with extreme caution, since local conditions vary greatly and logging is not always 'good' for primates.

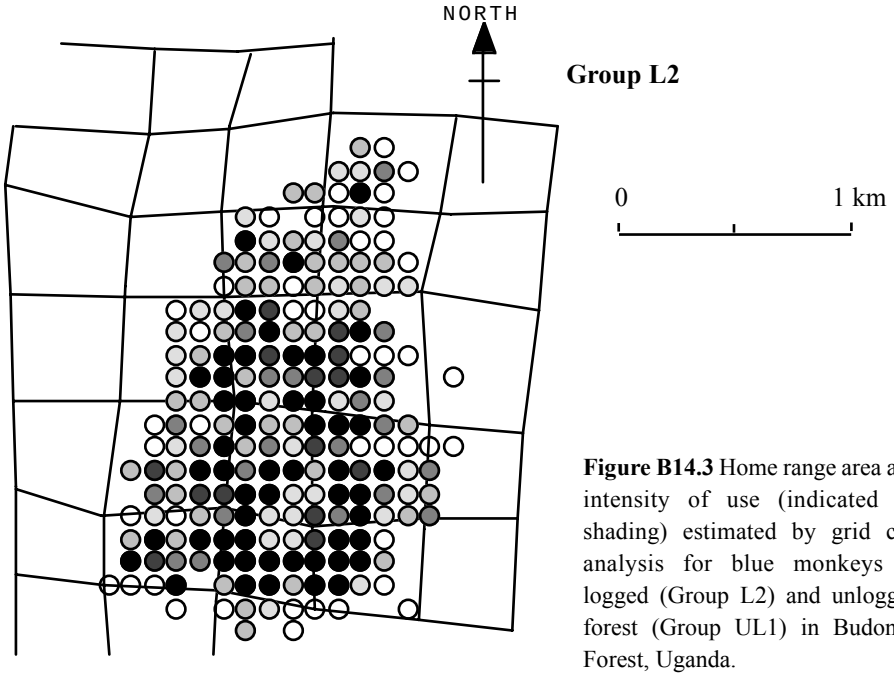


Figure B14.3 Home range area and intensity of use (indicated by shading) estimated by grid cell analysis for blue monkeys in logged (Group L2) and unlogged forest (Group UL1) in Budongo Forest, Uganda.

Group UL1

1-2 2-4 4-6 6-8 8-10 10- 54

Box 14e KEEPING AN INDIVIDUAL RECOGNITION FILE FOR FOREST BUFFALO

Recognising individuals in a population can give you insights that you cannot get from 'anonymous' animals and provides very powerful data for population management. However, in order to be sure you correctly identify individuals, you must keep a careful record of the distinguishing features of each animal and systematically confirm your identifications. If you see animals in the same place often, it is easy to think that they are the same ones and that you 'know' them as individual animals. But how can you be sure you are not confusing two similar individuals? Could you still recognise these animals if you saw them in a different place? Unless you are careful to check your identifications against notes and photographs and so learn to recognise individuals no matter where you see them, your data are likely to contain errors!.

An individual recognition guide has been created for a population of forest buffalo that range in the savannas in the Lopé Reserve, Gabon.

As many different buffalo as possible were observed before starting to build the file, and the 10 most variable characters were chosen as recognition aids. These were:

1. sex
2. age class (calf < 1 year, subadult, adult)
3. body size
4. horn size and shape
5. coat colour
6. eye colour
7. hair tufts on the ears
8. tail length and tail brush
9. scars on the skin.
10. any other rare feature

A 'blank' buffalo was then drawn on a card (see Figure B14.4), so that the details of these characters could be drawn in for each individual as well as described in writing. Recognition details were taken whenever buffalo were seen. As each new animal's features were recorded, it was given a name or code to locate it in the library. The name also allowed a cross referencing system so that other notes on it's behaviour and life history could be kept sensibly together.

The card records in the file were grouped according to the most obvious characters, so that after observing a buffalo, the possible cards could be quickly searched to see if the animal was already known. The cards were grouped first by sex, then coat colour, then horn presence, then ear characters. This was enough to reduce the choice of possible cards to 2 or 3. As the records were collected, the details of each 'new' buffalo were compared to the existing records, before a new card was compiled for it.

The notes system allowed a library to be developed quickly in the field, but over time, both colour photographs and digital video footage of each individual were taken when good opportunities arose and added to their library file.

Similar individual recognition libraries have been developed for elephants using their sex, size, tusks, head shape, ear shapes and ear venation patterns as distinguishing features. references for their work are in:

KANGWANA, K. 1996. *Studying Elephants.* African Wildlife Foundation, Nairobi.
(available from the AWF, P.O. box 48177, Nairobi, Kenya);
TURKALO, A. & FAY, J.M. 1995. Studying Forest Elephants by Direct Observation. *Pachyderm*, **20**: 45-54.

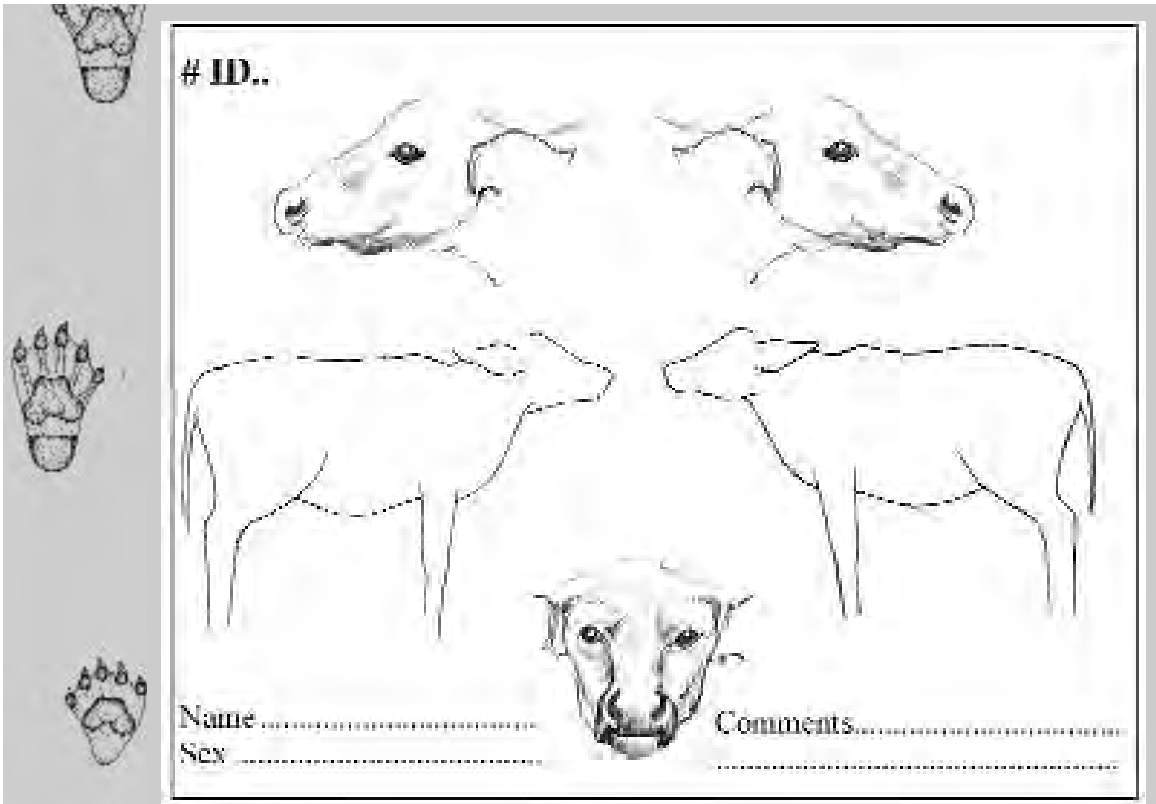


Figure B14.4 Card onto which buffalo features are drawn to facilitate individual ID.

Using artificial marks to identify animals

It is often important to work out the ranging patterns of your animals. For example, if you suspect that the protected area is too small, but you need to prove that your animals are moving outside and into areas where there is a risk of poaching before you can persuade the authorities to expand it, you need to document ranging patterns. If you cannot recognise the animals individually, fitting identification marks is a good first step to being able to follow their movements. ID marks may be tattoos, painted marks, coloured tags in the ears, or dyed patches on the skin or fur, anything that is visible from the distance you will observe the animal at, and will not harm the animal, make it more vulnerable to predation or change its behaviour. After choosing a viable, harmless marking system the big problem is getting the marks onto the animals.

If animals have a moment in their lives when they are naturally vulnerable to capture, you may be able to use this to mark the animal. However, the animals are often also very vulnerable to stress at these times and you should be careful to be as sensitive to their fear as possible. Biologists studying turtles can get close to the animals without needing to capture them when they come onto land to lay eggs. They can then paint on ID marks, or attach small metal tags to the shell, so that they (or others) will recognise the animals again. Using these ID marks, biologists have discovered that marine turtles use the same beaches where they were born to lay their eggs. Despite the fact that they travel right across the ocean from west Africa to America, 'African' turtles are dependent for their survival on the protection of these nesting sites. This is information which is vital to the protection of turtle species, but would have been very difficult to find out without the use of ID marks.

If you cannot identify a naturally convenient moment to mark animals, you will have to think about trapping them to mark them. There are many ways of doing this and no standard method. You should keep in mind that your priority is the animals' safety (both during the capture and and after its release), and then your own. Seek the advice of local trappers and of qualified veterinarians before you embark on live capturing animals.

A classic ID marking scheme is the world-wide system for ringing birds. This is a method of fitting very lightweight coloured plastic or metal rings to the legs of captured birds, in order that they can be recognised later. The rings have a number and a code, identifying who captured and ringed the bird and where, so that when the bird is seen later, the observer can find out where it came from. Birds are highly migratory; they often move between countries or even continents, but the system allows ornithologists everywhere to contribute to knowledge of their ranging patterns (see Box 14f).

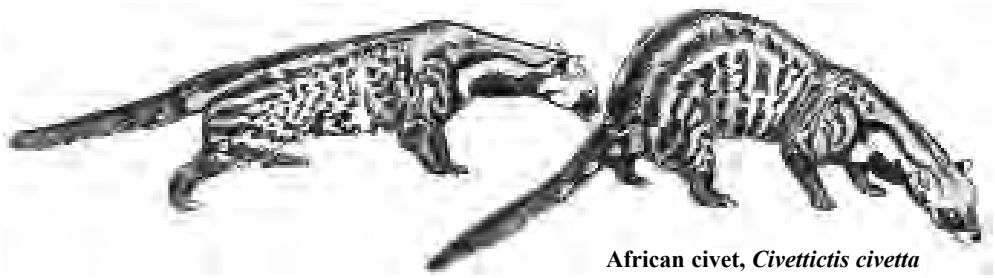
Keeping log records of sightings

For many studies of ranging, you will only need a few ID marked or individually recognisable animals and can work with a system developed locally for your needs. If you do use an artificial identification system, *you must be very careful to keep a catalogue* of the ID marks that you put onto each animal, along with its sex, age, where it was caught and marked and when. If you can't match the ID marks you see with these details of the animal, you are not much better off than before you started. You must also rigorously keep a log book of all the times and places the animal is subsequently seen, so that you are actually using the ID marks to study its ranging (see opportunistic sampling and focal animal sampling below).

It is only worth the effort of fitting ID marks to animals if you then put sufficient time into following these animals and documenting their movements. Generally, this is best done by a combination of opportunistic sightings and planned surveys. If you happen to see a marked animal in the course of other activities, you should note that observation. However, you should also plan regular searches for the animals, so that you are sure you find them wherever they are. If you only use occasional sightings, you will only see the animals in areas you visit often, like along roads, near your house or by the river you use. This will not give you a clear picture of the animal's true ranging behaviour.

If you manage to recognise individual animals (either with natural markings or artificial ID marks) and make the effort to study ranging, you can collect information on:

- how far animals travel daily;
- whether they rely on a particular place to sleep or shelter;
- whether they have a year-round home range or migrate seasonally;
- whether the same individuals stay in your area or if they come and go (they may be territorial);
- how big an area they cover in a year;
- which habitats they use in which seasons.



African civet, *Civettictis civetta*

BOX 14f INTERNATIONAL BIRD MARKING SYSTEMS

Capturing and marking birds which can be later recognised is a useful technique for bird census, studies of local ranging and habitat use, migration patterns, dispersal from colonies and behaviour and life histories of individuals. However, capturing birds and then attaching rings to them necessarily implies a risk to the bird and so most countries regulate the practice to protect their birdlife.

The techniques used to capture birds are regulated, as are the marking systems that are then applied. Bird are ringed in almost every country in the world, so for species that migrate, international colour coding agreements have been drawn up to prevent confusion between birds marked in different countries. Obtaining a ringing license in most countries required biologists to comply with these agreements for the good of everyone using the system.

Before you embark on ringing birds, you need first to contact the wildlife authorities in your country to find out what laws govern capturing birds, how to obtain a ringing license and which marks you can use on which species. If you have difficulty getting this information, two British organisations may also be able to help you: the British Trust for Ornithology (BTO), The Nunnery, Nunnery Place, Thetford, Norfolk, UK., and the Royal Society for the Protection of Birds (RSPB), The Lodge, Sandy, Bedfordshire, UK. Information is also available in the 'Ringers Manual', 1984, published by the BTO in the UK.

Small passerine birds are most often captured in mist nets, but larger species (like flamingoes or raptors) can be trapped or caught by hand at nest or roost sites. You should choose a capture technique that is legal and will minimise the risk of injury to the birds. However, you should also think about how many birds you can capture and ensure that you are likely to be able to capture a big enough sample size to gain good results from your study. Once captured, there are several methods available for artificially marking birds:

- *Metal rings*. These are individually coded rings, usually with a contact number or address for the person or organisation who ringed the bird (if you find one send details of where, when and in what circumstances it was obtained). This is the best system for birds likely to be recaptured (or found dead) far away from the ringing site. However, the codes are very hard to see on the wild bird, so they are not useful for individual recognition in a local study.

- *Coloured plastic leg rings and flags*. For birds that need to be recognised at a distance, colour rings, or coloured plastic flags attached to a metal ring can be used. Combinations of rings can be used to increase the codes available, but the colours may be restricted in birds that could migrate to other study areas and disrupt data collection there if they use the same colours. The weight of rings is carefully controlled so that small species are not harmed by carrying too much extra weight.

- *Wing flags*. Coloured plastic or material flags can also be pinned to the wings, though preening them off can be a problem. These tags are not allowed for diving birds.

- *Plastic collars*. On large, long necked birds like storks, geese or flamingoes, coloured plastic collars can be used, but they must be flexible and extreme care needs to be taken to ensure they can't choke the bird, or become caught on things and trap or kill it.

- *Feather dyes*. Colour dying feathers is a simple way to mark birds and unlikely to cause harm to the bird if dark colours are used. However, the marks will be lost when the bird moults its feathers.

Whether the animals are territorial or otherwise aggressive

Some animals live in territories which they do not leave and which they defend from other members of their species. Territories can be individual in solitary living species like duikers, or group territories in social species like chimpanzees. Group territories are sometimes defended by all members of the group, sometimes by only one class - usually the males. Some species establish permanent territories, where they live throughout their lives and offspring may even inherit these areas from their parents. Others may only establish territories at certain times of the year; a classic example would be the breeding behaviour of many bird species, whose males fight over nesting sites to attract females, but are not aggressive outwith the breeding season.

Animals like gorillas, which are not territorial (do not defend an exclusive area), but nonetheless remain in the same area throughout the year and are not nomadic or migratory, are said to live in home ranges. These are the areas they spend most of their time in, but home ranges do not have actively defended boundaries and are often not exclusive to one group or another, i.e. the home ranges of two groups can overlap.

If an animal species is territorial, members of different groups will be naturally aggressive to each other. This can cause intense problems in places where habitat loss, usually through human encroachment, is causing members of one group to live too closely to members of another group. If members of one social unit are pushed into the territory of another, they may be attacked and even killed (see Box 14g).

Even animals that are not territorial may behave aggressively at some times of their lives or in some seasons. An obvious time when you may see aggressive behaviour is when an animal is under threat, or thinks it is being attacked. It is important to view an animal's behaviour in terms of what it itself understands; a gorilla will not understand that a tourist merely wants to take a photograph of it - it will be scared of a human approaching and may charge at the photographer. This does not mean that it is an aggressive animal, merely that it felt threatened in that situation. Other reasons for aggression in animals may be competitiveness between males during the breeding season, conflicts within a group, (like a change of group leader), sickness or injury of the animal or the presence of young, vulnerable infants in the group. Most animals do not behave aggressively to each other very often. If animals in your area do frequently behave aggressively towards each other or towards humans it is important to understand why, so that you can try to resolve the problem.

Taking notes on aggressive behaviour

It is very difficult to build up a picture of the reasons for aggressive behaviour. You need to study animals very carefully to understand fully the reasons for their behaviour, but if you see aggression often in a population, you should try to find out why this is. You should note every time you see an animal attack or threaten another. You can do this by noting interactions between animals during any behavioural sampling plan.

In this way you will build up a file of incidents from which you may be able to see a pattern to the behaviour. You may find that fights between sitatunga males always occur in a particular season and relate that to their breeding season. Or, you may see that buffaloes that charge at tourists are always lone animals. You could then advise guides only to let tourists approach groups, which could immediately alleviate the problem.

To find out how to plan a detailed study of this sort of behaviour, see the methods section below; *opportunistic sampling, focal animal sampling, behavioural sampling*.

Box 14g WHY DO CHIMPANZEE DENSITIES DECLINE IN TROPICAL FORESTS SUBJECT TO LARGE SCALE, MECHANISED LOGGING?

Amongst the African apes chimpanzees (*Pan troglodytes*) appear to be more ecologically adaptable than either gorillas (*Gorilla gorilla*) or bonobos (*Pan paniscus*): their geographical range is much wider; they occur in a greater variety of habitats; a single chimpanzee community may range over hundreds of square kilometres; and they fashion a variety of tools to overcome diverse practical problems in their daily routine. During a study of the impact of logging on vegetation and wildlife in Lopé, Gabon (White & Tutin, 2000), chimpanzee and gorilla densities were monitored using direct observation and nest counts along line-transects. Logging involved extraction of a mean of two trees ha^{-1} resulting in 10% loss of both canopy cover and basal area. This damage level is low compared to the pan-tropical average of 50% and logging did not actively target plant species which provide important foods for chimpanzees, which could result in a disproportionate effect on their population numbers. All sites were located in a Reserve where hunting is effectively controlled.

Chimpanzee densities were found to be reduced following logging. In a site monitored before and after logging, the effect was immediate. Chimpanzee density declined to about 20% of pre-logging levels and remained low for at least a year after logging. In other sites, logged up to 15 years previously, chimpanzee numbers were low. Given that damage levels were low and food trees were not selectively killed, the abruptness of the response suggests that the reason is not ecological. Furthermore, there was no comparable change in gorilla densities after logging and since the diets of gorillas and chimpanzees in Lopé are similar this supports the belief that chimpanzees were not ecologically stressed following logging. It seems likely therefore that the explanation is a social one.

Chimpanzees live in stable communities with large home ranges which partially overlap. They behave territorially, patrolling and defending boundaries aggressively. Long-term studies at Gombe and Mahale in Tanzania have documented occasional extreme inter-community aggression, during which individuals from a large community systematically tracked and killed adult males of another smaller group, resulting in its extinction. Adult females and immature individuals were absorbed into other communities but any unweaned infants were vulnerable to infanticide by unfamiliar males.

Logging operations in Lopé advanced on a continuous front 5-10km wide from north to south. Noise from active areas can be heard from about 5km and is likely to cause stress and disruption to chimpanzees with no experience of such disturbance. It is not unlikely that the advancing chaos could displace entire chimpanzee communities. Chimpanzee density at the beginning of this study was high and might have reflected displacement of individuals by the activities of loggers to the north. During this period chimpanzees were often encountered in large, excited groups. The overall impression was that they were experiencing high levels of stress, perhaps because two communities were coming into contact. The displacement of an entire community of chimpanzees would probably rarely occur naturally. However, observations from Gombe and Mahale suggest that violent conflict and high mortality would follow. This rather disturbing scenario could explain the immediate, long lasting decrease in chimpanzee density caused by logging in Lopé. Studies elsewhere in Gabon, Cameroon, Congo and Uganda found decreased densities of chimpanzees in logged compared to unlogged forest. Western lowland gorillas would not be expected to suffer a similar fate because home ranges of adjacent groups overlap almost entirely and inter-group encounters rarely involve aggression.

Hence it is thought that chimpanzees' response to logging is due to their highly territorial behaviour. If driven into the territories of their neighbours by disruption of their habitat they are forced into a mortal conflict which results in a population crash.

How animals in a group interact with each other

As discussed in the sections on group sizes and territoriality, the way animals relate to each other is very important to the well-being of the population. Within a group the individual animals have different roles to play. In a human society, there will be many men in the village, but they are not all treated equally by each other or by the women and children. The chief has authority and respect from everyone, other men may be respected for their age or knowledge of medicine or religion, but some are younger and are less respected than the older women in the village. Animals that live in groups also have social structure and patterns of behaviour. Some animals are dominant to others and can take food and resources from them; some individuals lead the group to food sources. In some animal societies, like elephants, it is the females that lead the groups. In others, like mangabeys, it is the males.

Once you can recognise the sexes and ages of animals, you can already take some data on the composition of the groups they live in. This will give you a lot of information about the age structure of the population, how long infants stay with their mothers, whether males stay with females all year and whether there are times in the year (breeding, periods of food abundance) when animals get together in larger groups than normal and also about the way different individuals interact.

Clearly if a population is being hunted, or suffers from disease, or other stress, some individuals may be more vulnerable than others. If you understand the behaviour of the individuals in a group, you can predict how they may respond to these events. For example, you may see infant gorillas for sale in your local market, but think that one or two gorillas is not too much of an impact on the population. Studies of gorilla behaviour, however, have shown that adult male "silverback" gorillas will protect the young in their group from attack, as will the mother. Therefore it is likely that the hunters had to kill the silverback and the mother gorilla in order to capture the baby. Furthermore, studies of gorilla behaviour also show that the silverback leads, unites and protects his group. So even if some of the group escaped, without the silverback they are likely to be vulnerable, lost and perhaps even unable to find each other, reducing their chance of survival. Now the appearance of the two baby gorillas at the market seems much more serious, as it represents the loss of at least one, possibly two separate groups of gorillas from the area. You are only able to interpret the true consequences of the babies' capture through a knowledge of gorilla social behaviour. It is only with this level of understanding of animal populations that we can truly hope to manage protected species sensitively and rationally.

The tree pangolin, *Manis tricuspis.*

The breeding behaviour of the population

In order for any animal population to survive in the long term, it must be able to breed and raise young to maturity. This means that they must be able to find and attract mates, perhaps build nests or dens for giving birth, find enough food to support the growing offspring and be able to protect them from predators, including humans. Many animals, like mandrills, breed seasonally, but for some species, like forest buffalo, young can be born at any time of the year. For many less well-known species, like marsh mongooses or forest genets, the breeding pattern is unknown.

Why understanding breeding is particularly important for good management

If you study ranging behaviour, group sizes and compositions, social structure and territoriality of the animals through a year, you will already be able to answer many questions about breeding behaviour. However, there are some questions about an animal's breeding biology which will not be covered unless you search carefully for the information and concentrate on the breeding behaviour of individual animals. These include:

- at what age do the males and females start to breed?
- how many young does each female give birth to?
- how many survive to adulthood?
- how many are fathered by the same male (and so are full brothers and sisters)?
- do other animals help to raise the young?
- at what age are the young independent?
- how long between births to an individual female?

The answers to these questions are important to the understanding of both the relatedness of the population, which can affect its breeding success, and to estimating the time it takes for a population to grow. The *generation time* is the average time it takes from the birth of a female animal for it to grow up and breed itself. If you need to calculate how fast a population could expand (or decline), you need this information. Suppose you have a bushpig population that has been hunted to near extinction (2 males and 3 females left) and you succeed in enforcing a ban on hunting them. How long will it take before the population has grown back to 100 pigs? And how many can people hunt each year after that, without driving the pigs to extinction again? Before you can answer these questions, you need to know if the remaining animals are old enough to breed, how many will be born, how many will survive and how long it will take for these first piglets to become members of the breeding population.

The relatedness of animals in a breeding group is important for the long term health of the population. If closely related animals (fathers and daughters, mothers and sons or brothers and sisters) breed together, their offspring stand a higher chance of being born with abnormalities, or of being less strong and healthy than the offspring of unrelated parents. Breeding between closely related animals is called *inbreeding*, breeding between unrelated parents is called *outbreeding*. Most animals have a social system that prevents inbreeding. Often, this is achieved by the young moving into a different group from their parents (and opposite sex siblings) between the ages of independence and sexual maturity, but sometimes even though related animals stay in the same group, they avoid breeding with each other. If you see both lone animals and groups of the same species, you will often find that the lone animals are young males in the process of finding a new group. Many primate species behave like this. If these lone males cannot move to new groups, because they are prevented by barriers, or because they become very vulnerable to predators, the population as a whole will suffer gradually from inbreeding. Generally, this is a very long term process and other pressures on the populations will need to be addressed before the issue of inbreeding, but if you can plan for long term management, you should bear in mind the need for exchange of breeding animals between populations.

Terms used to describe breeding behaviour

There are many recognised patterns of breeding behaviour in animals, knowing some of the frequently used terms will help you to describe the behaviour you see and read about it in other references.

If animals live together all the time as well as breed together in a stable group, there are three terms that describe this:

Monogamy

If two animals stay together throughout the breeding season and mate only with each other, they are said to be monogamous (e.g., the Blue duiker, *Cephalophus monticola*). Some monogamous pairs stay together all their lives, others stay with the same partner throughout one breeding season, but may change partners between years.

Polygamy

If a male animal lives with and mates with several females, in a group around him (e.g., giant forest hog, *Hylochoerus meinertzhageni*) or because his territory overlaps with those of several females (e.g., Royal antelope, *Neotragus batesii*), the relationship is polygamous. Polygamous animals may live in groups with only one male, or groups with several males, each of which mates with several females. However, in groups with several males, there is usually strong competition between the males for access to the females.

Polyandry

If a female mates with several males (though for each infant only one male can be the father), the system is polyandrous. This is a much rarer breeding pattern in mammals than polygamy.

If male and female animals do not live together in stable groups, but search for mates only during a period when they are sexually active they are said to be *promiscuous* breeders.

Sexual activity may occur seasonally when all adults become sexually active around the same time, or each individual may have an independent breeding cycle.

For seasonal breeding promiscuous species, two terms can be used to describe the mating pattern:

Lekking

If animals come together in large groups to mate, and males display to attract females who choose whom to mate with, the gathering is called a lek and the species is a lekking species. Mandrills (*Mandrillus sphinx*), have a moving lek system - males enter large groups consisting of females and their young only in the breeding system, when they display constantly to attract receptive mates, as the group moves through the forest in search of food.

Harem holding

If a male chases receptive females into a group and mates with them whilst preventing other males reaching them, he is holding a harem. After the mating season, he will no longer keep the females in this group.

For species like chimpanzees or elephants, females (and also males in elephants) have individual cycles when they are fertile. In this case courtship and mating behaviour may be seen between a breeding pair of animals, yet others in the population do not change their behaviour.

When a male animal searches for and stays with a female only at the time when she is fertile (he often uses scent to follow the females who give off a special smell when they are fertile or in oestrus), the term *consortship* is used. Often males will guard their consort from other males, but this is different from a harem, as only one female is guarded at a time.

How animals of different species interact

No animal in any ecosystem on earth lives in isolation. All animals and plants are interconnected by complex ecological relationships that have evolved over many millions of years. If one part of an ecosystem is changed, many other animal and plant species will also be affected, often in ways that are not easy at first to see. For instance, imagine that all the leopards in the forest are killed to sell their fur, so there are none left to eat bushpigs. This will allow the bushpig population to grow more rapidly than normal, because more survive. This may mean that they start to eat more *Coula edulis* nuts than before, because there are more pigs to feed. Which will lead to a shortage of *Coula* nuts for other animals, like mandrills, which may then start to search elsewhere for their food and come into villages to raid crops. It would be very hard to understand immediately that the increase in crop-raiding by mandrills was an effect of leopard poaching sometime beforehand, but it could be the case. The more knowledge you have of the interrelationships between animals and plants, the more likely you are to be able to detect changes in the system and predict their effect on other species.

Terms commonly used to describe interspecific relationships

Some of the most obvious relationships between species are based directly on feeding behaviour and these are described at the end of the section above on finding out what animals eat. Other relationships have evolved because one species uses the feeding behaviour of another to its advantage. Examples of these interspecific relationships would be *seed dispersers*, which carry the seeds of the fruits they feed on to new places to grow; or *pollinators*, which pick up pollen on their bodies as they feed on the nectar of flowers, and transfer it to other flowers when they move on to feed. In this way the flowers of the second plant are fertilised and fruits can grow. Common pollinators are insects, birds and nectar eating bats.

Another type of relationship, often based on feeding, is *parasitism*. If one species relies on another for its survival, but is harmful to the species it relies on, it is a parasite. The species a parasite lives on and harms is its host. An example of this type of relationship is an tapeworm living in a pig. The tapeworm (parasite) gets food and shelter from the pig's body, but the pig (host) loses nutrition and is harmed by the worm. Mistletoes living on trees are also parasites.

If two species live in the same area and use the same resources they are *competitors*.

Rare events

Some things that happen in the lives of animals are rare events, yet have important consequences. Things like fights, copulations, births and deaths, emigration of adolescent animals from their natal group, predation and other interactions with different species can be very important to the animals, yet are rare, difficult to predict, so hard to plan to take data on. Because these are important events that can influence the behaviour of the animals during the rest of their lives, you should *always* take notes opportunistically when you witness these events.

The 'rarity 'of an event can depend on how often you see the species of animals in question. For animals that are very rarely seen, like fishing genets, otters, leopards or galagos, to name but a few, every observation is rare and should be recorded. As our knowledge of the ecology and behaviour of such species is so scant, every contribution is significant.

Even brief notes on important events will often help you to interpret how changes in the ecosystem can affect all the different parts of it. When you see interspecific interactions, you should try to work out the relationships between species you see by asking why each species should need or want to interact with the other. Studies of interspecific relationships are often difficult to plan as it is hard to know when the two species will come together, so data on interspecific interactions are often gathered entirely opportunistically (see *opportunistic sampling* below). Even if you study a species which is observed regularly, opportunistic observations of rare events can contribute significantly to your understanding of the other behaviours you see. As you cannot plan for observations of rare events, you should prioritise them and stop other activities to take notes if you are lucky enough to be in a position to witness some 'special' event.

Where to look for existing information on behavioural ecology

If you need to study an animal population, either for ecological research, or management purposes, or , as is most frequently the case, a combination of the two, you will probably want to ask some of the questions discussed above. Often, there is some information about your study species already available, which will help you plan the best way of spending your own observation time. However, you should be aware that results from other studies, may not be representative of what happens in your area.

First try looking in general field guide books or natural history accounts of your area. When looking up field guides (or other sources), try both the common name and the Latin name of the species, genera and family.

For African mammals try:

KINGDON, J. 1997. *The Kingdon Field Guide to African mammals*. Academic Press, London.

HALTENORTH, T. & DILLER, D. 1980. *A Field Guide to the Mammals of Africa including* Madagascar. Collins, London.

DORST, J. & DANDELOT, P. 1970. A Field Guide to the Larger Mammals of Africa. Collins London.

For birds try

SERLE, W. & MOREL, G. J. 1979. *Birds of West Africa.* Delacroix & Niestle, Paris.

MACKWORTH-PRAED, C. & GRANT, G. 1970. *Handbook of African Birds: West central and Western Africa.* Longman, London.

These will give you a general description of the animal and may tell you how to recognise the different sexes and the juveniles from the adults. They will usually tell you how to distinguish the species you are interested in from closely related species. Often guide books also give brief details of the animal's behaviour; most commonly these include its type of diet (carnivore, insectivore, herbivore etc.), general activity pattern (nocturnal, diurnal, etc.), social system (group living or solitary), territoriality and breeding behaviour. They should provide you with a map showing where the species is known to exist, though these maps are often not perfectly accurate; either because existing surveys are not exhaustive, or because the current rate of habitat change in Africa is altering species distributions faster than books are updated. You may find species in your area that were not previously known to exist there, or find that species which were thought to be there are now locally extinct.

Field guides sometimes suggest other references that will give you more detail about certain species, or you can read general texts about the subject you want to study, rather than the species. If you have access to a library, look under headings like 'Animal behaviour', 'Behaviour', 'Behavioural Ecology, 'Ecology', 'Feeding Ecology', 'Reproductive Behaviour' and 'Social Behaviour' for more information.

One excellent general reference for behaviour is

ESTES, R. D. 1991. *The Behaviour Guide to African Mammals*. University of California Press, Los Angeles.

Information on the behavioural ecology of individual species

There are many long term projects in African sites that have provided data crucial for the management of the species they study. Information from these sites can help you in your site, but you must be careful to compare the conditions in the two areas and remember that animals can behave very differently in different environments. Listed in box 14h are some of the general texts about single species ecology that will be most easy to find. These will give you an overview of not only the behavioural ecology of the species, but also of the long term projects that provided the data. Often the ideas other researchers have had about how to collect data are as useful as the actual results. Bear in mind that you can adapt ideas to suit your own study and try to learn from the experience of others.

Box 14h LONG TERM BEHAVIOURAL ECOLOGY STUDIES AT AFRICAN SITES

Listed below are some of the long term African field studies of the behavioural ecology of individual species relevant to the central African forests. For each a general and fairly easily obtainable reference is given. These references provide an overview of the project, its aims, methods and the data that has been collected. Most of these projects are still actively pursuing research to date.

For savanna elephants, *Loxodonta africana africana*
Long term research at the Amboseli Elephant Research Project in Kenya
MOSS, C. 1988. *Elephant Memories*. Fontana/Collins, London.

For forest elephants, *Loxodonta africana cyclotis*
Long term research at Dzangha Bai Forest Elephant Study in Central African Republic
TURKALO, A. & FAY, J.M. 1995. Studying Forest Elephants by Direct Observation. *Pachyderm*, **20**: 45-54.

For mountain gorillas, *Gorilla gorilla beringei*
Long term research at the Karisoke Research Centre in Rwanda
SCHALLER, G.B. 1963. *The Mountain Gorilla*. University of Chicago Press, Chicago.
FOSSEY, D. 1983. *Gorillas in the Mist*. Houghton Mifflin Co., Boston.

For western lowland gorillas, *Gorilla gorilla gorilla*
Long term research at the Station d'Etudes des Gorilles et Chimpanzés in the Reserve de la Lopé in Gabon.
TUTIN, C.E.G. 1996. Ranging and social structure of lowland gorillas in the Lopé Reserve, Gabon. In *Great Ape Societies*, edited by W. McGrew, L.F. Marchant & T. Nishida. Cambridge University Press, UK.

For chimpanzees, *Pan troglodytes*
Long term research at Gombe Stream Reserve and Mahale Mountains National Park in Tanzania and Tai Forest in Ivory Coast.
GOODALL, J. 1986. *The Chimpanzees of Gombe*. Belknap Press, Harvard.
NISHIDA, T. (Ed).1990. *The Chimpanzees of the Mahale Mountains*. University of Tokyo Press, Tokyo.
BOESCH, C. 1996. Social grouping in Taï chimpanzees. In *Great Ape Societies*, edited by W. McGrew, L.F. Marchant & T. Nishida. Cambridge University Press, UK.

For Red Colobus, *Colobus badius*
Long term research at Kibale Forest in Uganda and Tiwai Island in Sierra Leone
STRUHSAKER, T. 1975. *The Red Colobus Monkey*. University of Chicago Press, Chicago.
DAVIES, A. G. & OATES, J. 1994. *Colobine Monkeys*. Cambridge University Press, Cambridge, UK.

For buffalo, *Syncerus caffer*
Long term research at Lake Manyara in Tanzania
SINCLAIR, A.R.E. 1977. *The African Buffalo*. University of Chicago Press, Chicago.
PRINS, H.H.T. 1996. *Ecology and Behaviour of the African Buffalo*. Chapman and Hall, London.

Starting to collect behavioural ecology data of your own

Before setting out to collect behavioural data, you should ask yourself (and answer!):

- What information do I need?
- Do I need to observe the animals directly, or are there other ways of finding out what they do ?
- How will I find the animals and observe them, if that is what I decide to do ?
- Can I reliably identify the species, sexes and ages I may see?
- What data will I collect and how?

The most fundamental question to ask is "*what information do I need ?*" For instance, if you need to know when the animals breed in order to establish open and closed hunting seasons, you will be particularly interested in the timing of courtship displays, matings and births or young infants seen in the population. In this case you may be less interested in what the animal eats. However if you want to know whether males and females use the same feeding areas so that you include both in your protected area, you may be most interested in diet and in identifying male and female groups separately and subsequently in their ranging.

It is often the case that collecting data on many aspects of an animal's behaviour can be done simultaneously and that you are not obliged to collect only one type of information and exclude others. In fact, it is always a good idea to collect as much information as you can at every opportunity, as your priorities or needs may change in the future. However, if you do have specific information that you know you require immediately, you should be careful to design your observation programme so that this data is sure to be collected (see also Chapter 5 for more information on designing data collection sheets).

Having decided what you need to know, you then need to choose a method of collecting data on the subject. Decide if you need to make direct observations, or whether other methods can be used, or whether you will combine several approaches.

Will observations provide the data I need, or will other methods be better?

The methods you choose for sampling animal behaviour will depend on the information you need and the time and resources you have, the species you study and habitat you work in. Behavioural ecology data is most often collected through observation of animals, but other methods of finding out where an animal has been or what it has been doing are also available. Some of these methods are discussed in the chapters on censusing animals from the dung and nests, on animals tracks and on data from dead animals (see Chapters 10, 11 and 12). The methods in this chapter deal mainly with observational studies of behaviour, but some other methods are compared briefly at the end of the chapter.

The circumstances in which each method may be most useful are discussed in the text and summarised in Figure 14.1 above.

Direct observation versus other methods

Sometimes directly observing animals is extremely difficult. This may be a permanent problem because the habitat is dense or the animals travel long distances or are active at night, but it will often be because animals are wary of observers and will tend to flee or hide. During long term observational studies, this problem can be overcome by building hides or getting the animals used to the observer's presence, i.e. habituating them so that they no longer avoid being observed (see below). In some cases though, direct observations remain difficult There are non-observational methods of studying some aspects of behavioural ecology that can be useful in these circumstances. Some of these methods provide different data to that from direct observation and some provide essentially the same data, but are more practical if direct observation is difficult for one reason or another (see non-observational methods below).

Methods for Data Collection from Directly Observing Animals

Basic equipment for recording behavioural data

You do not need very much equipment to begin to study behaviour. For many studies you will never need more than the few things listed here. However for long-term records you will need to keep log books or computer files and find a safe, dry place to store your data archives. You may also find you need to construct hides for observation.

The essential basic equipment is:

- Field notebook (waterproof paper) and pencils;
- Data collection checksheets for specific subjects;
- Binoculars (or telescope if animals are usually seen far away);
- Watch and stopwatch;
- Maps of your study area.

Recording behaviour whenever you see animals: opportunistic sampling

You will often see animals in the course of a day when you are not concentrating on recording behaviour. You can still take valuable notes about where they are and what they do, even though the sampling wasn't planned. This is called *opportunistic* observation. Opportunistic observations are often used to study animals which are unpredictable (unhabituated and not able to be seen from hides), but it is sensible anyway to note all information you can in the course of your work as it can build up to be a valuable data set with very little effort. You can combine notes on different subjects so that a single observation contributes to several data sets, e.g. you can record the foods an animal eats, what size the group is, which animals make up the group, any breeding behaviour or aggressive behaviour and any interspecific interactions, all from the same observation, even though these data will then be analysed in different ways. For opportunistic observations it does not matter which animals you see or how long you watch them for.

There are some subjects on which you can collect some valid data from solely opportunistic data. These include *diet, group size, breeding behaviour, and rare or unusual events*. However, you will still need more standardised observations to do detailed analyses. Opportunistic sampling is usually seen as a way of recording *preliminary* data on these subjects.

A forest elephant,
Loxodonta africana cyclotis

Often many people in a team can contribute to these opportunistically collected data sets during the course of different daily routines. You may even decide that all people working in a protected area should be formally asked to contribute their opportunistic observations to long term records. Most people will find this easy to do, and it will help you to build up sample sizes in a wide range of situations. However, as opportunistic observations are unplanned, each person will have to take notes into their own fieldbook. You need to develop a centrally held record book to collate all the information. Often a cross-referenced system where each species and/or each subject (Group size data, Predation data etc) has its own log book is the best (see Chapter 5). Although copying data from personal notebooks in central records can seem a tedious task, it is essential to collaborate like this, both to maximise the information available for everyone working in the area at the present time and to safeguard long term records for future researchers.

Recording a food list

In order to record the foods that are eaten by species of animal, you can note every species of plant, animal or insect you see the animal eating. Minimal notes, which would allow you to make a "food list" for each of the species in your area and get a first impression of the diet, would be for every animal you see feeding:

- date, time of day;
- location and habitat type;
- animal species observed;
- age and sex (if possible);
- species eaten (or type of food, i.e. "insect" if species can't be determined);
- part of item eaten, i.e. "stem", "leaves", "unripe fruit seeds".

For a bushbuck observed feeding in January, your record might look like this (Table 14.2):

Table 14.2 An opportunistic observation record sheet useful for compilation of a food list.

Date	Time	Location	Habitat	Species	Age / sex	Food species	Food item
17 Jan 97	13:35	200m North of camp	grass savanna	*Tragelaphus scriptus*	Adult male	*Psidium guineensis*	new leaves

You can use these opportunistic records to compile a list of all the foods eaten to see how diverse (or specialised) the diet is. They can also tell you which foods are eaten at which times of the year, but not how much of each food is eaten. Nor can you be sure you record all foods, because things that are only available in 'hidden' places won't be recorded.

If you see animals feeding so frequently that opportunistic data collection will be too time consuming, you may want only to note each new food you see eaten. Or you may want to initiate a better study of diet that will show how much of each food is eaten, how often, or in what season and so allow you to analyse how important various foods are. In this case, you can use one (or more) of the observational sampling methods below (see ad libitum, scan sampling, focal animal watches). Conversely, if your observations are too rare and you cannot actually see the animal feeding, you can get information about its diet by searching through its dung for remains of the food it ate (see below and Chapter 10).

Collecting initial data on group sizes

Data on group sizes can be taken opportunistically into a field book or checksheet (see Chapter 5), each time you see animals. However, you will bias your estimate of group size if you count the same group too often, so it is better to have a sampling plan that will prevent this (see below). You could only include counts taken at least 2 km away from each other, or taken at the same location one year apart, to be certain that you are counting different groups. This will mean that it is quite time consuming to collect these data, but you will be sure that it is a valid representation of the groups in your area. For each sighting of a group of animals note:

- date, time of day;
- location and habitat type;
- species observed;
- number in group;
- number of each sex and age if possible;
- what the animals are doing (feeding, resting, rolling in a mudhole, etc.).

You can use a similar style checksheet to the one shown above for recording a food list.

Keeping a log of reproductive data

If you cannot recognise individual animals in a population, you can still note any matings you see and the appearance of new born infants. These observations may be made opportunistically or whilst you are taking other data on group size and composition, or on feeding and activity patterns. Noting them will help you establish when breeding seasons occur.

If you can recognise individual animals, you should keep a 'Reproductive History' log for each animal you can. In this log book you would note:

- any physical changes in the animal that suggest a breeding season (for example, changes in plumage or vocalisations in birds, swelling of the skin under the tail in female primates, or temporal gland secretions in male elephants);
- any behavioural changes, like new ranging areas, or grouping patterns;
- dates of any matings observed and with which partner;
- dates of birth ;
- how many young are born;
- what sex the offspring are;
- dates (and causes) of death for any infants that do not survive to adulthood;
- any behavioural changes that are associated with the presence of the new born young - new ranging patterns, increased aggression, changes in group membership.

Over time you will have the details of several breeding seasons noted for each individual, and will also start new log records for their offspring or even grandchildren. You will then be able to answer the questions needed to estimate the breeding potential of the population in your area.

Building up a record of rare and unusual events

As rare and unusual events cannot, by definition, be planned for, you need to record them opportunistically. You may be lucky enough to see rare events during other, planned observations, but you are still essentially recording them opportunistically, as they weren't included in your sample design. Some events are so rare, and so influential to the animals, that if you see them during a planned observation period, such as scan sampling, or focal animal watching (se below) you should still record them, even if it means interrupting your planned sample.

Notes on rare events should include:

- date and time of day, location;
- species involved and which individuals (adult males, juveniles, etc.);
- whether the animals are in groups or solitary;
- what the event is (fight over food, mating, responding to alarm calls of another species, etc.);
- which animal or species initiated any interaction;
- what each animal was doing before and does after;
- any other things that seem relevant
- if appropriate, any interpretation of why the animals behaved like this*.

*Interpretive notes are not strictly 'data' as it is your own view of a situation, rather than a measured fact. However, it is often useful to understanding patterns of behaviour to note what you understood from the event at the time.

Problems of opportunistic sampling

Although opportunistic observations are very useful, they are difficult to standardise. Your presence may influence the animal's behaviour in different ways, depending how close you are, whether you are on foot or in a vehicle, with other people or alone. You are also likely to see animals more often in some places than others, so will get a biased picture of what they do. If, for example, buffalo are usually grazing in small groups in open areas in the morning, but at midday congregate in mudholes for the afternoon. If you only make your observations opportunistically when you drive through the reserve in the morning, you could record only the small group size. You may have many observations of buffalo (a good sample size), but still not have a good idea of their behaviour. From these data

You *could* conclude that buffalo are in small groups, grazing, in the mornings.

You *could not* conclude that this is how buffalo behave all the time.

Even if you take only opportunistic observations, you should try to observe animals at several different times of the day and in different habitats and seasons before you draw conclusions about their general behaviour. If you want to make a detailed study of any aspect of animal behaviour, it is better to plan to draw up a sampling plan and observe the animals regularly, so that you know you get a balanced picture of what they do at all times in all situations. This, however, means that you have to put some effort into finding them predictably.

Systematic sampling

Planning regular observations of animals

Observing and recording an animal's behaviour is a method of describing a biological phenomenon just like describing a habitat type. In the same way that you use samples of the habitat type to get information about the whole thing (see Chapter 4), you use samples of behaviour to describe what animals do, as you cannot watch all the animals in the population all of the time. Just as samples of a physical thing, like a habitat type, must be controlled so that they are unbiased and representative of the habitat type as a whole, so your behavioural samples should be unbiased and representative of what any animal of that species, sex or age or dominance rank would do in the circumstances you are investigating. The problem with taking data from any animal you see, is that you have not controlled for these biases in the sample. You can use the data from opportunistic samples (taking notes whenever you see animals) to get 'first impressions' and to build up qualitative data sets like food lists or spot sightings in a ranging study, but you cannot answer questions about 'how much' or 'how often'.

Animal behaviour is a very complex subject and understanding why animals behave in a certain way is rarely an easy thing to do. However, unless you are careful to standardise the way you observe and record behaviour it will be impossible to interpret any patterns you may see in your data. These requirements for standardised, unbiased methods are common to all biological sampling procedures.

Standardising behavioural data collection means deciding beforehand:

1) where you will watch animals
this is often decided for you by where you can see them, but you should try to observe in all the different habitats they use;

2) when you will watch animals (dates and times of day) and establishing a routine
make sure you sample regularly through the different seasons or months of the year. Make some observations in the morning, some in the afternoon and some at night if you can;

3) how long you will watch them for (five minutes, an hour, all day..?)
generally the longer the better, but you need to decide how much time you can spend in relation to other commitments;

4) which animals you will record data from (all members of the group, just adult males, whichever you see first...?)

you need to decide which animals are most important for the questions you ask, but if you can, record data from all age and sex classes;

5) what you will call the behaviours you see

you need to have a pre-determined system for recording behaviour, so that you are sure you are consistent over time, and that you record the same behaviour as other observers. For example, everyone should call the same vocalisation the 'alarm call', rather than sometimes calling it the 'loud call', or the 'bark' or a 'cry'. Developing a checksheet with the recognised behaviours on it will go a long way to standardising descriptions of behaviour.

6) which behaviours you will record, or will you record everything?

you need to decide how much information you can record. This will depend how many animals you watch at once, and how fast they change their activity. You can best decide which behaviours you can record well, once you have watched animals for a few trial periods.

Once you have thought about these questions, you can develop a sampling plan for the data you want to collect. This should have three parts; a calendar timetable of when you will collect data, a plan of which animals you will record data from, and a method of recording data.

First devise a sampling timetable

Imagine you wish to study okapi in a forest with three habitat classes and two seasons. You have one week per month for your study, over one year and want to make sure you watch the animals in all situations, i.e. at different times of the day, in different seasons, different months of the year and in all three habitats. If you devise a sampling plan, you can check that you are recording the data you need.

Your first sampling plan might look like Table 14.3.

By following this plan you will be sure to sample each habitat each month. You will also make sure that each month, in each habitat you have a morning and afternoon sample and you have alternated the order you sample in during the week. However, you have not yet decided which animals you will watch, which behaviours you will record, how you will record data or for how long in each session.

You need to get into a position to watch animals and see how many you see, how often, and how long you can continue to watch them for, before you can decide on the most efficient way to record your observations.

Ways to find and watch animals on a regular basis

As anyone with experience of wild animals will know, they tend to try to avoid being seen. If you can see them, unless you are hidden, they can probably see you and will often run away or move behind vegetation to avoid you. Often they have smelt you and moved off before you even realise they were there, but even if they don't move away, they may still change their behaviour in response to your presence. They may move together in a group, be extra watchful or pick up youngsters which would otherwise be left alone. In this case you are seeing a special type of behaviour influenced by the animals' perception of a dangerous situation. The behaviour is not the same as what the animals would be doing if you were not there.

		Habitat 1	Habitat 2	Habitat 3
Dry season	**Jan**	2 days (Mon am, Tues pm)	2days (Wed am, Thur pm)	2days (Fri am, Sat pm)
	Feb	2days (Fri am, Sat pm)	2 days (Mon am, Tues pm)	2days (Wed am, Thur pm)
	Mar	2days (Wed am, Thur pm)	2days (Fri am, Sat pm)	2 days (Mon am, Tues pm)
Wet season	**Apr**	2 days (Mon am, Tues pm)	2days (Wed am, Thur pm)	2days (Fri am, Sat pm)
	May	2days (Fri am, Sat pm)	2 days (Mon am, Tues pm)	2days (Wed am, Thur pm)
	Jun	2days (Wed am, Thur pm)	2days (Fri am, Sat pm)	2 days (Mon am, Tues pm)
	Jul	2 days (Mon am, Tues pm)	2days (Wed am, Thur pm)	2days (Fri am, Sat pm)
	Aug	2days (Fri am, Sat pm)	2 days (Mon am, Tues pm)	2days (Wed am, Thur pm)
	Sep	2days (Wed am, Thur pm)	2days (Fri am, Sat pm)	2 days (Mon am, Tues pm)
	Oct	2 days (Mon am, Tues pm)	2days (Wed am, Thur pm)	2days (Fri am, Sat pm)
	Nov	2days (Fri am, Sat pm)	2 days (Mon am, Tues pm)	2days (Wed am, Thur pm)
	Dec	2days (Wed am, Thur pm)	2days (Fri am, Sat pm)	2 days (Mon am, Tues pm)

Table 14.3. A example of a sampling time plan

There are two solutions to this problem; hiding yourself so that the animals are unaware or you, or getting the animals used to your presence, so that they don't change their behaviour, even if you are there (*habituation*; see below). The two are not mutually exclusive and you can compromise by partially 'hiding' so that the animal is less threatened than it would be were you fully exposed, but nonetheless, it is aware of your presence. Often observations from a vehicle are a good compromise. Generally animals are less wary of a car than an observer on foot, even though they are well aware that the car is there and is not a 'normal' part of their environment. You can get observations in this way quicker than you would if you had to spend time habituating the animals to an observer on foot, but you must still keep in mind that the animals' behaviour is influenced by the car and may not be representative of what they would be doing if the car was not there.

In some situations, changing the animals' behaviour slightly in response to an observer may not matter for the data you want. This would be the case if you wanted only to document the composition of groups in terms of males, females and juveniles. As long as the groups were sufficiently far apart that they could not regroup in response to you, you would get the same record of ages and sexes in the group whether the animals were aware of you or not. You must judge for your own site and needs how important it is to observe from a hide or from a distance, or to spend time habituating animals.

Observing by stealth

At the beginning of a study, or if you only have a short period to make observations, you may first need to find out where the animals are, and which areas they use. Even in a long term study you need to know this before you can begin to plan observation hides or choose animals for habituation. Following animals on foot can be done without the animals being aware of you, but it is usually difficult to remain undetected for very long, or to get close enough to make good observations of which animals are there and what they are doing. If you try tracking animals on foot, familiarising yourself beforehand with their tracks and trail (see Chapter 11) will help you to first find them. Once you are searching, you should wear dark clothes, move quietly and stay downwind if possible. To further reduce the chance of the animal detecting you, it is always best to work alone or with the minimum number of people possible.

You are unlikely to be able to get all the data you want to understand the animal's life from observations taken during stealthy tracking. If you want to be certain of standardising the conditions of data collection and maximising the data you collect for the hours you spend in the field, you will probably need an observation hide (or several), or to embark on habituating the animals.

Observation hides

If you need to observe animals when they are not scared of you, the quickest solution to the problem is often to build some sort of shelter that hides you from the animals. The benefit of this is that when the animal is unaware of you, you can observe its natural behaviour, whereas to habituate animals to your presence can take months. The cost (apart from the monetary cost of construction) is that you have to stay in the hide and wait for the animals to come to you. This will probably limit the time you can see them and the types of behaviour you see, because the animals will always be in the same place when you observe them. You can, of course, build several different hides to cover a larger area, or use a vehicle as a 'moving hide', but you will probably still miss some periods of the animals' lives as they move away from you. You should think carefully about which aspects of an animal's life are important to you, and whether observations from a fixed point will be able to cover enough of what goes on to answer your questions before you invest the effort in constructing a hide (see also Chapter 13; Bai monitoring).

Building the hide

The first thing to think about is where to put the hide. You should choose a point that gives you the biggest field of view, that is easily accessible and that allows you to see the area the animals use well, without being too close. The second decision is how permanent a structure you need. If you want to put up a hide to monitor the nest site of a rare bird that will only use that site for a few weeks, you don't need to invest in high quality, durable materials. If you are building a hide for a long term study of a bai, or to allow tourists to watch animals all year round, you should build a good solid structure that will last, as the building work itself is disruptive and you won't want to be constantly making changes and repairs to the hide. It is often useful to investigate natural vantage points, like large trees, cliffs or rock outcrops before you start. These are often fairly easy to modify into hidden shelters and will be more quickly accepted by the animals than new constructions.

As animals are very sensitive to changes in their environment, they will probably avoid the hide itself when it is newly installed, but will get used to it and treat it as a normal part of the landscape after a while. The sort of hide you can use will depend on the species you want to observe and their ability to detect you, but generally it is best to try to make the structure blend into the environment - use wood or dark painted metal rather than reflective surfaces; don't use strong smelling preservatives on the wood; use canvas or leaf thatch to cover it, rather than plastic or corrugated iron that will be noisy in wind or rain or when you move inside; use green or other natural colours rather than bright colours or white.

Researchers monitoring bais in some central African sites have found that most animals in the bai (gorillas, elephants, colobus, sitatunga, bongo and others) will quickly accustom to a researcher fully visible on a platform. In this case the animals have become habituated to the observer, rather than remaining unaware, but the end result is that their natural behaviour is easily observable without the hide 'hiding' the observer. It is possible to observe animals in some situations from a simple platform without being fully hidden, but generally primates and antelopes have very good vision and will see you unless your hide covers you completely. Elephants or pigs rely more on smell and will be less likely to see you, even if you are only partially hidden. The ability of animals to smell you is hard to outwit. Building a hide high up will help as smell is carried up by the wind, or if you have a prevailing wind direction you can put your hide downwind of the area you want to observe. Limiting the number of observers to one or two people and forbidding urinating near the hide will help, as will reducing the 'smelly' things in the hide, like insect repellents, wood preservatives, cigarettes, food or sweaty clothes.

The hide should be able to keep equipment dry if necessary, and should be big enough for the observers to be comfortable for several hours and to arrange their equipment so that they can observe and record notes quietly, without having to move around. Often rough seats and a shelf for resting notes and binoculars on are good additions to a simple box shelter. The visibility from the hide should be maximised, without making the observers inside too easy to see if this is a problem. Camouflage can often be achieved with dark coloured mosquito netting, which has the double function of keeping the observers insect-free and able to concentrate for longer periods.

Once you have built your hide, you must be able to get into it without the animals detecting you. This may mean going in at certain times of day when the animals are not close by, perhaps before dawn, or clearing a path so that you can creep in quietly once they are there. You should think carefully about the access to a hide before you build it.

Habituating animals

Animals flee from humans because they are frightened of them. If you can persuade them that you are not threatening, they will no longer feel the need to run away from you. The process of gaining the animal's confidence, so that it will allow you to watch it, is called *habituation*. Habituated animals are completely accustomed to the presence of an observer and (ideally) will carry on their normal lives regardless of whether they are watched or not, allowing you to see and record their natural behaviour.

In order to gain the confidence of an animal, you need to meet it regularly and always interact with it in a calm and totally non-threatening way. If the animal often finds itself close to you, but is never threatened or scared by you, it will eventually stop feeling wary and therefore stop running away. As you need to always interact with the same group in order for them to remember you and get used to you, one of the first problems of habituating animals is to recognise the individuals you are trying to follow. If there are many groups in the area, you should concentrate on one at a time for habituation and be sure you recognise 'your' animals.

When you first start to follow the animals, they need to know that you are there, or they will not be learning to accept you. Do not approach as closely as you could through stalking stealthily; instead choose an open area, where the animal's can see you clearly, sit down and do not stare directly at them, but act as if you had not even noticed they were there. Then, if they have not already noticed you, make a small, natural noise, like rustling leaves, moving a branch or whistling a soft tune to yourself. You shouldn't make loud, scary noises; piercing whistles, voice calls or claps are no good as they will communicate alarm to the animals. If the animal notices you because of a noise it thinks you made by accident, and thinks you have not seen it, it may be confident enough to stay to look at you for a while, but the chances are it will then alert the rest of the group and flee from you. Hopefully it will do this more as a precaution than in panic, if you are calm and sufficiently far away.

Repeat this pattern of contacting the animals as often as you can, each time letting them see you and realise that you do not threaten them. Try not to pursue them directly when they run from you, as this will scare them. Instead circle round and be waiting in front of them as they move. Generally animals are always less scared if they approach you and find you already there, than they are if you walk towards them. Each time you meet the animals, you should carefully note the time when they see you, how close you are, if they alarm, how long before they flee and how far they go. If each time they seem to be less scared than before, i.e. less alarm calling, waiting longer before they leave, and fleeing less far away, you can continue to get closer to them each time and watch them more directly. If, however, they seem more frightened than earlier, you should return to showing yourself farther away. Animals will be naturally more frightened if they are on the limits of their territory or home range, but also will feel threatened if you are intruding near their nests, shelters or 'safe places'. Try to choose neutral grounds for the first contacts; feeding grounds, open areas or salt licks are good. If you don't know how the animals view their 'home', you may find that a group of animals which appear to be accepting you well have certain areas where they are still wary. Do not let this discourage you, but try to habituate them well in areas where they seem calm before you follow them into areas where they are obviously less confident.

Eventually, if you can regularly contact the same animals and they can see you in non-threatening circumstances, they will begin to accept you close to them, watching them. You will usually find that they will always have a 'minimum distance' they will tolerate you at, and that they are very perceptive to changes in your behaviour (different clothes, different times of visiting them, different paths you arrive on) and to new observers. The closer you stick to a routine of dress, timetable and behaviour, the quicker the animals will become confident to accept you. Throughout the habituation process you must never betray the trust of the animals. If they start to gain confidence, and then you scare them, they may become even more wary than before.

Once the animals do not appear to alter their behaviour at all because you are observing them, you can start to record the details of their natural behaviour, using some of the observational sampling methods below. You should expect to follow the animals regularly for several weeks (even months) before you get to this point, though some animals are naturally more confident than others and will habituate more quickly. Colobus monkeys are naturally calmer than guenons and buffalo calmer than bushpigs for instance.

You must be very self-disciplined and careful to habituate animals properly, but once you have a group of animals that behave naturally in front of you will be in the best position you can be to record and interpret their behaviour

Some problems of habituating animals

You may find that you cannot habituate the animals you try to follow.

This may be because

- you do not meet the same ones often enough for them to remember you and accept you.
- because you are not being careful enough never to frighten them
- because during the early contacts they never stay long enough to realise that you are not threatening them, so never start to learn that you are harmless.

This last point is particularly important when you are following animals in a dense habitat. It is important during the early days of habituation that the animal watches you for long enough to see that you are not trying to attack it. If it flees instantly each time, and you are always so close before it sees you that it is already scared, then it will never even begin to gain confidence. This is probably the biggest problem for habituation of ground dwelling animals in forests.

You may find that the animals habituate too well! This sounds not to be a problem, but in fact, if the animals get so confident that they start coming towards you, they are no longer behaving 'naturally' either. This is less of a problem than running away from you, but you must be very careful in recording behaviour to note if the animals are interacting with you (looking at you, moving toward you or even threatening you) so that you can use this in interpreting what they do.

Finally, unfortunately, you may find that well-habituated animals become a nuisance to other people, even dangerous, because they have lost all fear. They may start to raid rubbish pits or crops, or at worst attacking people. Likewise, habituated animals are more at risk from people, because they no longer flee from them, and become easy prey for hunters. Think carefully about the possible consequences of a fearless animal before you start habituating; it can lead to problems with some species of animals in areas with dense human populations.

Recording behavioural data with a standardised sampling method

Once you are in a position to be sure of seeing your animals regularly, from a hide, by stealthy tracking or because you have successfully habituated them, you can start collecting more detailed data on their behaviour. You should first organise a sampling timetable (see above) so that you are certain you are collecting data representative of all situations you are interested in. Then you need to decide what you will record during these observation periods.

Defining different behaviours

To take standardised data, you need to be sure that you know the difference between and can recognise different activities. First ask yourself whether you are seeing the same behaviour repeated or a different behaviour? For instance, if you see a bongo lying down, it could be asleep, or wallowing in mud or perhaps it may be ill, or giving birth to a calf! You must observe the animal carefully to be sure you see exactly what it is doing .

Then you need to make up rules for what you will call certain behaviours, so that your records are consistent each time you sample. For instance, as a buffalo grazes, it is also moving slowly and searching the ground for food. You may choose to call this simply 'grazing' in your notes and describe at the beginning of your data file what the term encompasses. Or, you may decide to note 'searching for food' and 'grazing' as separate behaviours. When you collect data on animals feeding, it is often useful to separate the time spent searching for food (foraging) from the time spent actually eating. This can allow you to analyse whether food is harder to find in some seasons than others.

If you don't decide beforehand how you will define what you see, you may sometimes record 'grazing' for only the time spent actually feeding and sometimes for a combination of searching and feeding. This will ruin your data collection and give you a false picture of what the animals do.

You can speed up your note taking in the field by using simplified definitions for complex behaviour patterns which are always repeated together. For instance, if a male mandrill is scent marking a tree, he will climb a few feet, sniff the bark, 'grin' and shake his head, showing his lower teeth, then rub his chest against the tree in four or five upward strokes, sniff the tree again and climb down. Obviously you can't write all this each time and also keep track of what the animal does next, so you can use a simpler term, like 'scent marks' to indicate the whole sequence just described. There are many repeated behaviour patterns where this sort of simplification can be useful, but be sure to state clearly at the beginning of your record book what you mean by each defining term used in your notes.

Sampling rules and recording rules

There are two separate decisions you must make to decide how you will record behaviour, though many researchers confuse these (see Figure 14.4). Firstly you decide 'who' or 'what' you will watch. This is the sampling method. There are four well-known sampling methods

- ad libitum sampling;
- scan sampling;
- focal animal watches;
- particular behaviour records.

Then you decide how often you will record data. This is your recording regime. There are two recording regimes possible

- continuous recording - record data all the time;
- time interval sampling - record data only at pre-determined times.

You can combine sampling methods and recording regimes in different ways to get the best picture of what the animals are doing. Below, each of the sampling methods is outlined, together with the most appropriate recording regime to use in different circumstances.

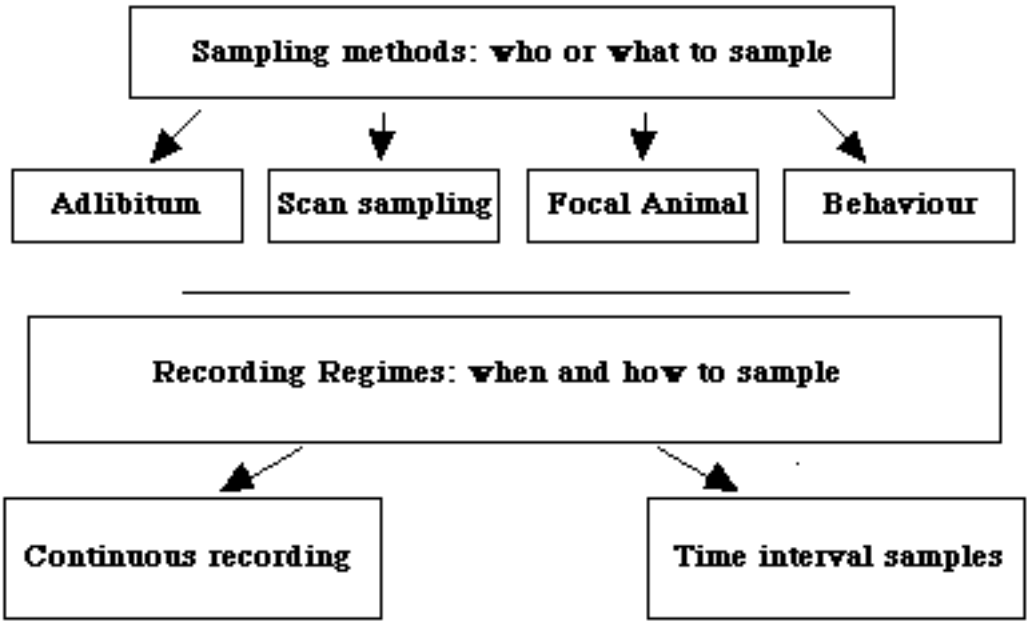


Figure 14.4. Sampling methods and recording regimes

Ad libitum sampling

When to use ad libitum sampling

Ad libitum sampling means that you watch all the animals you can and record anything that happens. This is the least standardised of the sampling methods; however it can be used to give you a good overall picture of what a group of animals is doing. It is a good method to use at the beginning of a study when you don't know what behaviours you may see. However, it is difficult to analyse the data and compare it to other sites, so you should try to use other sampling methods as well.

Ad Libitum sampling is also a useful method for recording events which happen quickly - like mating, fighting, catching prey, or visits to a fruit tree where fruit are collected and taken away. However it is not a good method for recording the duration of behaviours like feeding or grooming, because you are often distracted and miss pauses in the activity, or miss the end of the activity period. A better method for looking at how long and animal spends doing something is a focal animal watch (see below).

Designing an ad libitum sampling programme

First you need to draw up a sampling time plan to spread your observations over all the situations you want to monitor. Then you need to decide how long you will spend watching during each observation period, and which animals you will watch. Generally the longer you observe the better, but you are sure to have other constraints on your time. Choose a minimum period you can spend with the animals every time you sample and try to stick to this. On days when you have more time, you can stay longer. You always use a continuous recording regime, because you don't know what will happen, but you must decide if you will always watch groups of animals (and whether it will always be the same study group, or different ones) or whether you will alternate between groups and lone animals if both occur. The choice of animals to watch depends on the questions you are asking. For example, if you want to get an overall impression of the behaviour of group living animals compared to solitary animals, you should divide your observation periods between groups and lone animals, in the same way time is divided between habitats in the okapi study example above (see Figure 14.3).

You record descriptive notes into a fieldbook during ad libitum sampling, because it is difficult to draw up a checksheet when you don't know beforehand what you will see. However, the more systematic you are about your sampling periods, the better the data will be, so you should still try to standardise initial data collection. You can make a checklist in the back of your notebook of data to collect at the beginning of each session. Always note the date, time you start and finish sampling, the weather, how many animals are in the group and who they are, other species present, how far you are from the animals, how they react to you on arrival, and any special circumstances you feel affect the animals behaviour.

Ad libitum sampling in the field

Before you set off, you should already know how to find the animals and be in a position to record their natural behaviour (see above). You should have decided on a minimum observation period for each session, and have drawn up a sampling time plan for when you will observe.

When you first see the animals, spend a little time working out which animals are there and setting up your equipment for easy recording, then start to note all you can. Try not to wait for an interesting event before beginning (or to 'hang on' for something interesting at the end of the session). You can use descriptive language to record complicated events, but try to quantify as much as you can. For instance for a red duiker feeding under a *Mangifera* tree, it would be better to record, "eating mango fruit at a rate of 1 every 3 minutes", than just, " feeding on mangoes". The second note doesn't tell you how many fruit were eaten and so is much harder to compare in a meaningful way to other observations of duiker feeding then the first.

You could use *ad libitum* sampling to continuously record colobus behaviour during a 90 minute period of your sampling timetable.

Your notebook might look like Figure 14.5.

```
10:00 - arrive at riverside. See 10 adults and 2 juvenile black colobus present in a single
Berlinia bracteosa tree. Adult male and one female feeding on new leaves, both
juveniles playing, all others sleeping.
10:02 - one juvenile runs across the back of a sleeping adult, who does not move
10:24 - the adult female stops feeding and sleeps in same place
10:25 - adult male pauses in feeding and gives a single cough/growl, then continues to
feed, none of the animals appear to respond to the vocalisation
10:31 - juveniles return to their mothers and sleep next to them
10:40 - one juvenile leaves mother and jumps to next branch and back several times, ?
in play
10:42 - male stops feeding and moves 5m along the branch to sleep
10:43 - juvenile stops ? play jumping and returns to rest beside mother
11:20 - two adult females awake and travel into adjoining Cryptosephalum staudtii.. One
has slightly kinked tail and is perhaps recognisable from the others.
11:21 - a subadult follows the 2 females
11:30 - end of sampling period; observer leaves the site
```

Figure 14.5 *Ad libitum* sampling notes.

Analysing ad libitum samples

At the end of your sampling you will have:

a total number of minutes of observation;
records of all the different behaviours and interactions that occurred during this period;
the number of times each behaviour occurred.

Often the most useful thing gained from *ad libitum* sampling is an overall impression of the activities, the behaviours seen and the way groups and individuals interact. This is valuable in itself for your understanding of the animals, but notes which cannot be quantified into numbers are hard to compare between sites (see Chapter 4).

The simplest first analysis of *ad libitum* data is to look at the number of times each behaviour occurs over the sampling periods and calculate a frequency. Then compare between situations which may differ, like morning and afternoon samples, or dry and wet season, or males and females.

For example, you could use *ad libitum* sampling to watch otters on a large river every evening and note, amongst their other behaviours, the number of times they catch a fish or crab. Then you could compare the frequency of crab catches to the frequency of fish catches in the dry season and the wet season, or between males and females, or experienced adults and juveniles.

Scan sampling

When to use a scan sample

Scan sampling is used to record the activity of a group of animals. It is especially useful when you cannot see all members of the group, or if the animals are in a large group as it allows you to record the activity of only some animals, but then to standardise the data in order to compare between groups. Scan sampling is usually the best method to record feeding behaviour and activity patterns of groups of animals.

Designing a scan sampling method and choosing a recording regime

How to record?

During a scan sample, you try to look at several members of a group of animals and record what each is doing. You record one activity for each animal, at the moment you see it (or 10 seconds later to allow you work out what it is doing if this isn't clear immediately). It is difficult to use a continuous recording regime, because the time it takes to observe each animal and write down what it is doing prevents you from continuously observing the others, therefore scan sampling is used with a time-interval recording regime.

There is no standard time interval for scan sampling. The principle is that the more frequently you scan through the group, the closer you will get to a true picture of what they do, but you must leave yourself enough time to write down what you see. Although you scan through the group at fixed times, you cannot see and record behaviour of several individuals instantly, so a scan method allows a certain amount of time for observing the animals. For instance, your time-interval may be 15 minutes, with an observation time of 5 minutes. This means you *start* to record what the first animal is doing at the beginning of each 15 minute period, but you allow yourself 5 minutes to find and observe the others, then 10 minutes when you are not recording. You can use this time to prepare for the next scan and to record other data opportunistically into a notebook, or onto the datasheet. To record the behaviour of 5 animals, you could scan every 15 minutes quite easily, but to score 50, you may need to reduce your scan frequency to every 30 minutes and increase your observation time to 20 minutes. Try to maximise the frequency of your scans whilst leaving yourself enough time to record all the behaviour calmly so that you won't make mistakes. It is better to scan less frequently and record the data well than to scan too often and make mistakes. You will probably need a few trial periods trying different time intervals and numbers of individuals scanned before you decide on the best strategy.

During the time you are not scanning, you can record other notes. It is very useful to record the position of the group centre onto a map at the end of each scan. Data on the weather, direction the group is travelling, any events that affect the group, like loud noises or predators appearing, are useful , as are group size changes, mating or aggressive encounters.

Which animals to observe

For animals that are sometimes hidden, you may not be able to record the behaviour of all of them during each scan. In this case, you choose some subset of animals. Choose the maximum number you can always see no matter what they are doing, so that you can always complete the scan and sample each behaviour equally. For example, if you were scan sampling the behaviour of a group of talapoin, even if you could sometimes see the whole group of 15, if you could only be sure to see 6 at any one time, your scan should include 6 records, and you score the first 6 individuals you see. If you don't do this, you will have more records for things that happen very visibly, like feeding on leaf buds in a naked tree, or loud calling, than for cryptic behaviours, like eating fruits in a dense vine tangle. Your data will show a greater proportion of time spent in the obvious behaviours, even though this may not really be the case. You can accept a few scans that are incomplete in your dataset (not more than 10% of the total) as there are always some times when you cannot see even your minimum number.

You can further bias your data if your attention is drawn by the most visible or noisy animals and you miss more discreet activities. Try always to scan from left to right for the first scan, then right to left for the second, and so on, so as to minimise the risk of recording the same animal twice or being biased by recording the most obvious animals. You may decide that there are particular animals you want to look at in a known group, or particular classes of animals, like only mothers with infants, or only adult males. However, you should still try to minimise the bias towards obvious animals as if you were recording from the whole group.

You can use scan sampling to record all behaviours, or just a particular subject, like feeding behaviour, however as time is of the essence, and you know what you will be recording (unlike ad libitum sampling), it is always better to use an checksheet than a notebook for scan sampling (see Chapter 5).

Scan sampling in the field

When you set out to take your scan samples, you will have already decided the day and period of time you will sample for (sampling time plan) the time interval, the observation period (recording regime), the number of individuals you will observe in each scan and the behaviours you will record (sampling method). You will have a checksheet with you that will allow you to record the data efficiently and you will know where to observe the animals without influencing their behaviour.

You should take with you your binoculars, pencils, checksheets, notebook, maps of the area (including blank maps for recording group movements) and a watch. Go to your observation point, allow about 15 minutes to familiarise yourself with which animals are there, to let the animals settle if you disturbed them on arrival and to make yourself comfortable and set up a telescope if you need one. If you need to wait until the animals come into view, allow yourself a similar period of settling in from the moment you first see them before you collect data. Then begin your scans. Start on the hour or half hour as this will make recording scan time and calculating the beginning of the next scan easier. It also prevents you biasing your scans by waiting for an 'interesting' moment to begin recording. During the observation period scan the animals from left to right on the first scan, then right to left next scan and so on. Note the behaviour of each as soon as you see it and determine what it is doing, then move to the next animal, until you complete your scan of the minimum number of records or until the observation time is up. If you cannot see what an animal is doing 10 seconds after you see it, record a '?' and move on. At the end of the scan record the position of the centre of the group on your map. Use the time until the next scan to make any supplementary notes and to change your position if you need to be ready for the next scan.

If you scanned a group of 9 mustached guenons (known to comprise an adult male, three adult females, two subadults, 1 juvenile and 2 infants), recording all types of behaviour from the first 5 animals seen, for a 60 minute sampling period, using a 15 minute time-interval, your checksheet might look like Figure 14.6.

There is still quite a lot to write. If you are more precise about the behaviours you record, you can develop a checksheet that will allow you to make even simpler notes to record behaviour. For example, if you were only interested in feeding behaviour, you still observe all animals, but only record those that are feeding. You may have to adjust the number of records you take if the number of animals you can be sure to see feeding in a scan is less than the total number you can see. If you don't, you will record more records for foods that are easy to see, even if the animals are not really feeding on them more often . On your checksheet you only include foods you know are available, plus an 'other' category. You can obviously change the categories on the daily checksheet as food availability changes, rather than searching through a long list every time you record data.

For the same observation period, scoring only feeding records, your checksheet might look like Figure 14.7. In this case you have not managed to see 5 monkeys each time, it would be better to aim for 2 records per scan for feeding in future, so that you are sure to have equal samples every time.

Date	27 Jan 1997		**Observer**	P. Nkum	
Species	Cercopithecus cephus		**Location**	North west study zone	
Scan interval		15 minutes	**Observation period**		5 minutes
Scan time	11:00	11:15	11:30	11:45	**Comments**
Adult male	grooming female 3	feeding on kapok new leaf	resting	feeding on insects	
Adult female 1		resting			
Adult female 2	feeding on kapok new leaf		resting		
Adult female 3	groomed by Adult Male		resting	feeding on Ficus fruits	
Subadult 1		resting		travelling between trees	
Subadult 2	feeding on kapok new leaf	feeding on kapok new leaf	feeding on insects	playing with infant 2	
Juvenile		feeding on kapok new leaf			
Infant 1					
Infant 2	resting		feeding on kapok new leaf	playing with subadult 2	first leaf feeding record

Figure 14.6 Checksheet filled out during a 60-minute scan sample of a group of 9 mustached guenons

Adult male "silverback" western lowland gorilla *Gorilla gorilla gorilla*

Date	27 Jan 1997		**Observer**	P. Ntoum	
Species	Cercopithecus cephus		**Location**	North west study zone	
Behaviour sampled	Feeding				
Scan interval	15 minutes		**Observation period**	5 minutes	
Scan time	11:00	11:15	11:30	11:45	**Comments**
Trees/s			SubAd	AdM	
Kapok leaves	SubAd 2, AdF2	SubAd 2, Juv, AdM	Inf 2		1st leaf feeding record, inf 2
Okoumé fruits					
Cissus fruits				AdF 2	
Other					

Figure 14.7 Feeding records made during the 60-minute scan sample of a group of 9 mustached guenons.

Recording precise data onto this sort of checksheet is very easy; however, you must take care to include all the possible categories of food (or behaviour) on your checksheet before you start. The line 'other' allows for a few unexpected behaviours which will always occur, but unless you have designed your checksheet well, you will end up with a confusingly large number of records in the this line!

Analysing scan sample data

When you have collected your scan data, you will have:

- A total *number of scan samples* (the number of times you scanned the group, e.g. 4 for the first guenon example above);
- A *total number of records* (the number of all individuals you scored over all the scans, e.g. 20 for the example above);
- A *number of activity records* for each behaviour (the total number of times the behaviour was recorded, e.g. 6 for resting behaviour above);
- A *number of individual records* for each animal if you identified animals individually (e.g. 4 records for the adult male above. As each animal can only be scored once, this should never be more than the number of scan samples, but could be less if the individual was not seen on some scans);
- The *dates* and *times of day* that each behaviour was recorded.

*To calculate the proportion of time **any** member of the group spends in each activity*

$$\text{Divide} \quad \frac{\text{number of activity records of that behaviour}}{\text{total number of records}}$$

To calculate the proportion of time a particular individual animal spends in each activity

$$\text{Divide} \quad \frac{\text{number of activity records for that individual}}{\text{number of individual records for that animal}}$$

For these calculations to be a true reflection of the animal's behaviour, you must have a large sample sizes (see Chapter 3). A good figure to aim for is 100 scan records in which each individual appears. This means that each scan record is 'worth' one percent of the calculation, which allows you to record quite a detailed analysis of the behaviour, including some rarer behaviours, but means that no one record will be able to bias your data if it is atypical.

Remember that if you want to compare behaviour between individuals or over different seasons, or sites, you have to have 100 scans per individual, per season, or per site, then make each calculation separately before comparing the resulting figures. You can make the simple calculations above to estimate the frequency and relative importance of different behaviours in different animals, or at various times of the year. This is probably sufficient analysis to help you with management decisions.

Questions you can answer with scan sample data

When do certain activities occur?

As you record the time you start each scan, you can analyse when in the day certain behaviours are most common. If you want to do this, you should put together the records from the same time periods on different days, for example, all the morning records together and all the afternoon ones, and compare the proportions of time spent in each activity in the different periods (see box 14a).

How much time is spent in each activity? How important is the activity?

Scan sampling data tell you what proportion of the time period sampled an animal spends in each activity you record (but it cannot tell you the actual number of minutes the animal spends on each). Knowing the proportion of time an animal spends doing something can in some cases be used as an estimate of the importance of the activity. For example, you may find that the guenons spend 88% of their time eating *Pycnanthus* fruits in September. Thus you can conclude that *Pycnanthus* is the most important food for this group at this time and try to ensure that your management plan protects *Pycnanthus* trees in the monkey's habitat.

How does the time spent in the activity change during the day or during the year?

If you have spread your sampling periods throughout the day and over the seasons or months, scan sample data can also tell you when in the day or year certain behaviours are more frequent, or take up more time. You divide the scan samples into different periods of time and calculate activity proportions for each period. i.e. add all your dry season samples into one data set and all the rainy season ones into another, then you can compare the two. For example you may find that in the rainy season a monkey is eating insects in 36/100 feeding records (36%), but in the dry season 78/100 (78%) feeding records are of a monkey eating insects. This may be because the monkeys have to eat more insects in the dry season because there is not much fruit available. Be aware that you can only draw this conclusion if you have recorded 'feeding' only when the monkeys are actually eating (see above, 'Defining behaviour'). If you recorded 'feeding' when they were searching for insects as well as actually eating them, you will not know whether the increased time spent on insects is because they actually eat more, or because they spend more time searching, as insects are less abundant in the dry season.

A note on further analysis of observation data

Statistically valid comparisons of data are complex (see Chapter 4). You should refer to specialist statistical manuals before embarking on detailed analyses of behavioural data. Good texts for behavioural analyses are

- MARTIN, P. & BATESON, P. 1990. *Measuring behaviour*. Blackwell, Oxford.
- SIEGEL, S. & CASTELLAN, N. J. 1988. *Non parametric Statistics for the Behavioural Sciences* (2nd Ed). McGraw-Hill, Singapore.
- SOKAL, R. R. & ROHLF, F. J. 1995. *Biometry.* (3rd Ed) W.H. Freeman & Co., New York.

Focal Animal Watches

When to use a focal animal watch

A third sampling method is to watch a single (focal) animal and only record its behaviour. This method is good for recording the behaviour of particular individuals which might behave differently from the rest of the group, like the dominant male of a mangabey group. It is also a good method for recording rare events that scan sampling might miss. Obviously it is a good method for recording the behaviour of solitary animals. Focal animal watches can give you more precise information about the proportions of time an animal spends in particular activities than scan sampling can, but it takes longer to collect the data as the sample size for each observation period is only one.

If you want precise information about how a certain type of animal behaves, focal animal watches are best.

Designing a focal animal watching method and recording regime

You can use a focal animal sampling method with either a continuous or time-interval recording regime. As you are only watching one individual, you can usually manage to record continuously, but if you have to move a lot to follow the animal, you can use a time-interval recording regime to give yourself time to move between samples. For data on single animals, a continuous recording regime is best if you can manage it, *but* if you want to compare the data from a focal animal with that of a group which you are scan sampling, it is best to apply the same scan method and recording regime to the focal animal, so that the data you collect are directly comparable.

Focal animal watching in the field

Once you head for the field to do a focal animal watch, as for scan sampling, you should already have decided your sampling time plan, your recording regime, which animal you will watch and how you will find it.

Take with you binoculars, pencils, checksheets, notebook, maps of the area and a watch. You may add to this a telescope, compass and dictaphone if they are needed. Once you find your animal, watch it for about 15 minutes to familiarise yourself with what it is doing, to be sure it is the right animal, to let it settle if you have disturbed it on arrival and to be sure your equipment for observing and recording is set up well. Then begin to watch it. If you are using continuous recording, record *everything* it does in a notebook or onto a checksheet. Note the time the new activity starts to the nearest second if you can and describe the activity using your pre-prepared list of definitions. If the animal does several things quickly, you may not be able to record and watch at the same time. Then the best you can do is note all the activities occurring within the same minute and list them in the order the animal performed them. If a particular series of activities always occurs together, you could decide on a definition for the whole sequence to make recording easier (see above, Defining behaviour). If watching and recording together is a continual problem, you could try using a dictaphone so that you can continue to watch the animal as you record. If you say the time when you start recording, you can them play back the dictaphone against a stopwatch later to note the time when the activities changed, in the same format as your written notes would have.

If the animal is part of a group, you should periodically (say every 10 minutes) record who it is closest to; its nearest neighbour and the neighbours distance This will give you a useful index of whom it associates with most in the group and how close the association is. Often these are family relations like mother/offspring. It is also useful to note the behaviour of other animals in the group, for instance every hour. At this moment you should mark the centre of the group on a map of your study area, together with the positions of any neighbouring groups you can see or hear, or any other animals present. Knowing the details of the group's position can be very useful in interpreting behaviour and the map will allow you to calculate the day range (how far the animals travelled in the day). Day ranges can be compared between seasons to see if animals are travelling further to find food (or other resources) in some periods than others.

Your written record might look like Figure 14.8 for an hour's focal watch of a habituated adult male owl monkey (*Cercopithecus hamlyni*).

Date	18 June 1997	**Observer**	P. Nixon		
Species	*C. hamlyni*	**Individual**	#5	**Group**	#2
Location	Central valley	**Weather**	Hot, calm, sunny		
Sampling and recording		Continuous focal animal watch			

Time	Notes
10:00	find group and settle down 30m S. of them. No alarm calls or avoidance. Most of the group feeding on insects around the base of a Pentadesma microphylla tree. Group spread over 10m diameter. Male #5 on north edge of group.
10:15	begin focal watch on male #5. Feeding on ants in P. microphylla bark, eating one insect every 30 sec. approx.
10:17	moves to other side of tree, out of sight.
10:30-32	disappears and crosses to neighbouring Pycnanthus angolensis and rests sitting upright.
10:37	approaches by female #6 in his group, who begins to groom his back.
10:44	turns in groom female's hand.
10:48	female leaves, #5 stays resting in same position as 10:32.
10:50	group spread over 30m, group centre 30m south of position at 10:00. Most members now resting.
11:15	moves 10m away from Pycnanthus to south and approaches female #6 to groom her left arm.

Figure 14.8. Data recorded during a 1-hour focal watch of an adult male owl monkey.

If a focal animal goes out of sight, but you know you will recognise it when it reappears, you can continue sampling as if this was a behaviour; record 'hidden from view' and note the time as for other activities (10:17 - 10:30 in Figure 13.8).

If you cannot recognise your focal animal again, you must stop the watch and start a new sample with a new animal. There is no set time period for focal animal watches, but generally the longer the better. Watching animals for less than 15 minute focal watches is probably not very useful. You should try to stick to your sampling time plan and record for as much time as possible.

Analysing focal animal watches

At the end of your study, your data will include:

total number of minutes sampled from all animals;
total *number of minutes sampled for each known animal*, or class of animal (female, male, etc.);
total *number of minutes* of each behaviour (including time 'out of sight');
the *date* and *time of day* that each behaviour was recorded.

To calculate the proportion of time each animal spent in each activity

$$\text{Divide} \quad \frac{\text{the number of minutes in the activity}}{\text{the total number of minutes the animal was watched}}$$

To calculate the mean proportion of time spent in each activity for a class of animals

$$\text{Divide} \quad \frac{\text{the total number of minutes of that behaviour}}{\text{the number of minutes sampled from all animals in that class}}$$

Before you analyse the rest of the data, calculate the proportion of time the animal was hidden from view. If this is a large proportion of your observation time (over 50%), you have not really gained a good picture of what the animal does. You should try to change you r method of observing to enable you to see the animal for more of the observation period, or lengthen the observation period. Aim for a minimum time with the animal hidden from view; less than 25% is probably acceptable.

Questions you can answer with focal animal watch data

When do certain activities occur?

As your data are recorded against real time you can calculate when the activity occurs and the actual length of time each activity takes. If your samples are spread through the day and over different times of the year, you can also see changes in the frequency or length of time spent in different behaviours at different times. From this you can see the activity pattern of the animal. You may find that elephants spend a short time in marshes in late afternoon in the cool seasons, and a long time nearer the middle of the day in the hot season. Predicting their behaviour will help you to show them to visitors.

How much time does an animal spend in each activity?

Focal animal watches can be used for calculating the proportions of time individuals spend in certain activities. With a continuous sampling regime you can calculate absolute amounts of time in each activity. For the owl monkey above, 15 minutes out of 60 (10:15-10:30) were spent grooming (25%), 2 minutes (10:30-10:32) were spent travelling (3.3%), 28 minutes (10:32 -10:37 and 10:52-11:15) were spent resting (46.7%). Clearly these proportions are not representative of the monkey's whole life. You need many watches (a large sample size) to calculate statistically valid figures (see Chapters 4 and 16).

Do different classes of animal spend different amounts of time in a certain activity?

For focal watches, your sampling unit can be each animal or a time period, like each minute, hour, or each day. You can lump together all the minutes you have watched focal animals and calculate mean times spent in each behaviour for a certain class of animal (like dominant males) and compare the behaviour of different animals. You may find after many focal watches that male owl monkeys spend a mean of 68% of their day feeding, whereas pregnant females spend 92% of their time feeding. This could indicate that the nutritional needs of the pregnant females are higher than those of the male, and that they will be most at risk in times of food shortage.

How often do rarer behaviours occur?

If you watch a focal animal continuously, you will see all it's behaviour. However, as discussed above, it is impossible to watch the animal 24 hours a day, so you use standardised samples of behaviour to try to get a representative picture. Events that take only a short time or are infrequent, like mating or fighting, are the most likely to be misrepresented because you see them least often, so your sample size is low. A focal animal watch using continuous recording can show you how frequently an individual does these things. However, if you are particularly interested in these rarer behaviours, for instance to investigate increasing aggression in a population (see Box 14g), you may want to use a sampling method to record only specific behaviours.

Some methods for studying behavioural ecology that do not require direct observation of the animals

Dung analysis

The analysis of fecal samples to determine the composition of an animal's diet (see Chapter 10) is very useful method for getting this important information without direct observation. However it is also an example of an indirect method which provides different data to that collected during direct observation of the animal feeding.

Fecal analysis can only tell you about food items that remain identifiable in the dung, whereas direct observation can only tell you about items you can see being eaten. Many foods can be identified using both methods, but some will be better (or solely) identified by one method or the other. For example dung analysis may allow you to identify small items in the diet that it is very hard to see the animal eating or which the animal eats only rarely, like which species of ant a monkey eats from tree bark, where the remains may be fairly easily identified in the dung sample. However the converse is also true, that some dietary items which are totally digestible, like soft fruit pulp, may not be identifiable in the dung and so are only known from observations of animals feeding. Fecal analysis can sometimes tell you how much of a food item the animal ate relative to another item (for example the relative amount of sedge fibres to grass fibres in hippo dung, which would be difficult to quantify even if you were directly observing the animal grazing a mixed sward), but it cannot tell you how much time and effort the animal spent obtaining and eating each item in its diet. Direct observation of an animal feeding can be used to collect qualitative data on its diet, and also provides information about where the animal finds food and how much time it spends in foraging for and eating its food. This behavioural information about how the animal spends its time is often as important as knowing what it eats.

Radio telemetry

Radio signals emitted from a transmitter can be located by receivers some distance away (up to several kilometres, depending on the strength of the signal). Nowadays, radios and batteries are manufactured small enough to be carried by animals on collars (or even under the skin in a surgical implant), and their signal can be received by an observer with a radio receiver, far away from the animal. This technique is called radio telemetry (see box 14i). If it is difficult to follow and observe the animal because it is shy, or it ranges over long distances, or at night, or through a dense forest with low visibility, the radio allows you to replace direct observation with an indirect means of collecting information on the animal's ranging behaviour and activity patterns. Radio telemetry has been extremely useful in studies of the ranging behaviour of species in forest environments.

Camera traps

A camera 'trap' is a fixed camera, focused on a place that the animal is likely to pass, for example, a road crossing point, a baited feeding station, a salt lick or a latrine pit. The camera takes photographs automatically at set time intervals, or when the animal triggers it by stepping on a hidden pressure pad, or breaking an infra-red light beam. Camera traps are essentially automatic observations, but all they can tell you is that the animal was in that spot at a particular time noted by the camera. The pictures usually cannot tell you why the animal was there or how long it stayed or which other animals it was with. In some species you can recognise sexes and even individuals from photographs, in others you cannot; it depends on the individual variation in physical appearance. Camera traps are particularly useful for monitoring nocturnal animals and can be used for inventory surveys of the species that exist in an area. However, non-appearance in a camera trap is not evidence for the species not being present in the area. Point sightings of animals can be especially useful for documenting which species use a special area or resource, like a salt lick, and can also be used as a 'recapture' technique for capture-mark-recapture studies of animal density (see Box 14j).

Post-mortem data

The collection of data from dead animals can tell you a lot about the animal's life, even though the animal is no longer 'behaving' (see Chapter 11). Strictly, information about events in a lifetime, like the age at first breeding, the age at death, the number of offspring and when they are born are called *life history* data, but often life history data are collected during behavioural studies, because these events shape the way an animal behaves.

At death, some of the life history can be deduced from the corpse. The sex of the animal can be seen from it's anatomy, and its age often deduced from the state of its dentition, jaw, foot or horn size or from other physiological changes. Whether a female mammal has reached reproductive condition or not can be seen from ovulation scars on the ovaries, and whether she has lactated can be determined from the scarring of the nipples. If there are pelage, plumage or skin colouration changes through life or with season, these can be documented by describing the appearance at the date of death.

The immediate cause of death can also often be deduced from a carcass (Are there bullets in the body, or the claw marks of a predator? Does it show signs of disease or is it very old with worn or missing teeth? Does it show signs of starvation?). Defining the cause of death is extremely important in populations under pressure from hunting, habitat change, disease or other stress. For example, if the ages and sexes of animals killed by a new disease introduced in the population are noted, you may be able to predict which surviving animals are most at risk and target your management efforts to protect them.

Box 14i RADIO TELEMETRY

Radio transmitters can be attached to an animal by a collar or harness, or implanted under the skin. All of these operations require capture of the animal, so the first consideration in a radio telemetry study must be how you will capture the animals safely and immobilise them long enough to safely attach the radio. Generally the animal is anaesthetised, which allows a thorough health check and valuable data on size, weight, dentition etc. to be taken at the time of capture. This sort of handling of animals requires expert training and should be supervised by a qualified veterinary surgeon.

The radio is powered by a battery, also attached to the animal. The larger the battery, the longer the radio will transmit, but the heavier load the animal must carry, so a balance must be achieved. Radio transmitters for wildlife study can be made to individual requirements to maximise the time of transmission for each type of collar and weight.

Each transmitter is programmed to a certain radio frequency (usually VHF). The receiver the researcher carries can be tuned to this frequency to pick up the signal. Transmitters send a signal on a single frequency, but a receiver can usually be tuned in to several different frequencies. If several animals are radio-collared in the same area, each should transmit on a different frequency, so that the researcher can distinguish which animal is being followed. Transmitting frequencies need to be decided at the beginning of the study and programmed into the radios at manufacture.

The radio receiver can tell you quite precisely the direction from which the signal is coming, but much less precisely the distance from which it is transmitting. However, if you take a bearing from yourself to the animal (receiver to transmitter) from three different positions, you can then calculate a fairly precise location from the intersection of these three lines. This is called triangulation (see Page 78, Chapter 6, esp. Figure 6.10). If the bearing is slightly wrong (especially if the animal is distant) or if the animal moves significantly between the three 'fixes', the location will be imprecise. Tracking animals effectively using radio telemetry requires experience in interpreting the signal so that bearings are precise, a good knowledge of the study area topography and a certain amount of intuition about where to take bearings in order to get the best triangulation in a short time.

Radio telemetry studies have been widely used in the past two decades to gain information on species that are difficult to follow directly and they will continue to be invaluable aids to ecology in forested habitats. However, the equipment is expensive, considerable expertise is required to capture and handle animals and both time and expertise are required to follow the radio signals and analyse the data.

Further information on radio telemetry can be found in:

KENWARD, R.E. 1987. *Wildlife Radio-tagging: Equipment, Field Techniques and Data Analysis.* Academic Press, London.

PRIEDE, I.G. & SWIFT, S.M. 1992. *Wildlife Telemetry: Remote Monitoring and Tracking of Animals.* Ellis Harwood, Chichester, UK.

Box 14j CAPTURE - MARK -RECAPTURE CENSUSES

Capture-mark-recapture methods of animal census can be used for estimating the actual size of a population of non-territorial animals. The technique is most often used to assess populations of small animals (birds, reptiles, rodents, insects) that can easily be caught, and that live in large freely-mixing populations or colonies. Larger animals are not usually censused this way as capturing them is too problematic and sample sizes are too small.

The method is that you capture, count and mark (harmlessly) a first sample of animals, then release them to mix back into the population. At a later date you repeat the capture process and record how many individuals are caught in the second capture and what proportion of them are marked (i.e. were also captured the first time). The second 'capture' can be replaced by an observational survey if the marks are visible.

You assume that if:

1. The population is 'closed' between the samples, i.e. no animals arrive in it or leave it (including births and deaths);
2. The initial animals marked are a representative cross section of the population and not biased to either sex or any age class which may affect their likelihood of capture;
3. The marked individuals mix randomly in the population after release;
4. The capture technique, location or marking system does not make the marked animals more or less likely to be captured the second time than unmarked animals;
5. The marks are not lost between captures;

then the ratio of the number of marked animals to the total population is the same as the ratio of recaptured animals to the total number captured in the second sample. You can then calculate the population size using the Lincoln - Petersen estimate.

$$\text{Population size (N)} = \frac{mn}{r}$$

where **m** = number marked and released at the first capture, **n** = number caught at the second capture and, **r** = the number of marked animals recaptured on the second occasion.

Imagine you wanted to estimate the population of zebra mice in a savanna patch as part of a study of the prey available in a golden cat territory. On the first day of the census you caught 52 mice in live traps and marked them by dyeing their tails red with hair dye. Then a week later you caught 64 mice in the same area, 16 of which had dyed tails.

The population size in the patch would then be calculated as

$$N = \frac{52 \times 64}{16} = 208 \text{ mice.}$$

Of course, the 5 assumptions above are often not really true; there are animals which move in and out of the sampled area between captures, the marked animals may lose the marks or die because of injury during the first capture, they may be less likely to be caught the second time because of fear of the trap, and so on. The Lincoln - Petersen estimate is the simplest way of using capture-mark-recapture data to calculate population size but it does not take into account any changes in the population, loss of marks, or bias in the capturing. It is probably the most feasible method to use for simple population estimates, but you should be aware that if any of the assumptions above are likely to be completely false, then the estimate will be wrong. Some common false assumptions have predictable effects on the population estimate: if the marks come off between captures, or if the marks decrease the animal's chance of survival and it dies before the next capture, your final estimate will be too high; if some animals are more likely to be caught and re-caught than others, your final estimate will be too low.

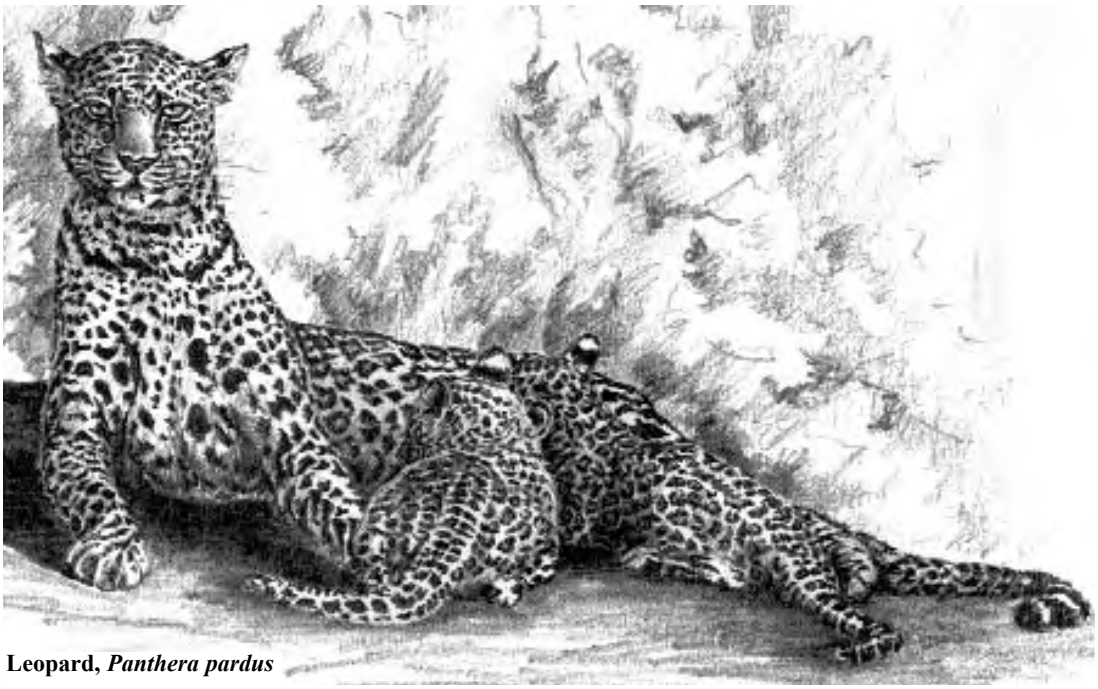
There are many refinements of the capture-mark-recapture technique which have been developed to deal with dynamic populations. Good general references are:

- BEGON, M. 1979. Investigating Animal Abundance: Capture Recapture for Biologists. Edward Arnold, London.
- SEBER, G.A.F. 1982. The Estimation of Animal Abundance and Related Parameters. Macmillan, New York.

Generally, *post-mortem* data can be very informative about the population structure and demography if the carcasses found represent a relatively large cross section of the ages and sexes. This may be the case in heavily hunted populations. However, even if carcasses are rarely found, *post-mortem* data should always be taken and stored for reference as, over time, or if circumstances change, they may become more useful than the first record seemed. If you consistently note the species, age, sex, physical characteristics, breeding status, size and date and cause of death for dead animals you find, over time you will build up a knowledge of the life history of the species in your area (see Chapter11). This sort of data can allow you to answer questions like:

- how long animals of each species live naturally;
- how often an individual in the population dies;
- whether there are times of high mortality in the population (like drought years, or times of food scarcity);
- whether there is high natural mortality of infants and whether this is linked to environmental changes, like heavy rainfall during the birth season;
- whether males die younger than females;
- how big adults can grow in the wild (especially important for species hunted for trophies like ivory or horns).

This knowledge will be directly applicable to your management strategies, even if your population is not under any pressure from changing circumstances. If you have this data from your population, you can then demonstrate any change from increased hunting pressure of other stress. For instance, you may find the carcass of a monkey that dies naturally in your area about once every 6 months and note that these are usually young individuals killed by crowned hawk eagles. If in one year you find a monkey carcass every month and these are adult males with no marks from predators, you will realise that this is an abnormally high death rate for these monkeys in comparison to your records from previous years. You may then want to find out if they have a new disease and make an effort to have samples from the monkeys tested for viruses or parasites that could put your whole population at risk (see Box 11a).



Leopard, *Panthera pardus*

CHAPTER 15
Socio-economic data and their relevance to protected area management

Bryan Curran, David Wilkie & Richard Tshombe

The earth of the twentieth century is a human-dominated landscape, and people will therefore continue to play an important role in managing natural resources. Recognition by outsiders (conservation and development NGOs, government, timber companies, sport hunters, etc.) that indigenous communities have legitimate rights and needs is an important step towards long-term conservation of natural resources. It is becoming increasingly clear that forced protection is a short term solution, particularly in developing nations that rarely have the resources to devote to biodiversity conservation. Identifying important natural assets and defining the sustainable limits of their exploitation will be a major challenge for protected area managers. It has often been claimed that the preservation of tropical forests is in the best interests of indigenous peoples that traditionally have relied on its resources for their livelihood. In reality, however, adopted conservation measures frequently constrain subsistence and economic activities, and the more intangible benefits which might accrue from long term protection (i.e. restocking overhunted areas from core protection zones) often are not readily apparent to local communities. If the legitimate concerns of these people continue to be overlooked, conservation projects will fail to achieve their objectives. Protected areas generally have been created following only biological/ecological criteria. Proper management, however, requires an understanding of the entire range of social, political, cultural, economic and legal variables that are likely to impact the project over the long term. It is imperative therefore that protected area staff establish proper rapport with local communities and encourage their participation, from design to implementation to monitoring and evaluation. Neglecting to do so may have more profound influence on project success than will the failure to collect appropriate biological data.

Gathering a socio-economic information base relevant to conservation can be accomplished with a combination of quantitative and qualitative methodologies. Although certain methods of data collection are by definition quantifiable (population census, market surveys, measures of hunting impacts), a wealth of information (e.g. local attitudes, participation of communities) can be gleaned only from more qualitative data. Experience has shown that a combination of quantitative and qualitative methods is generally most useful; the choice of appropriate methods depends upon the types of information the protected area management team requires for decision making, and on the relationship between different stakeholders.

Each project will have certain specific aims and/or research objectives. Socio-economic data are only relevant if they provide protected area managers with information pertinent to the maintenance of the natural system under their guardianship. As such, the need to appraise and monitor human activities will be somewhat different between sites, and requires that problems be identified and objectives of research clearly defined prior to data collection. Consideration must be given to the costs and logistics of proposed projects, including travel options, food, lodging, salaries, etc. The actual data collection is only one (often small) factor to consider when calculating costs; you must also accurately figure the time required for study design, preparation of data collection forms, training research assistants, pretesting, analysis and interpretation of data, and report-writing. Training needs must be assessed - how many research assistants will be required, and what level of technical competence will be necessary?

Although many of the methods presented in this chapter (and indeed throughout this manual) can be learned by protected area staff and even local assistants, there will likely be instances where outsiders (experienced researchers or university students, for instance) are better suited to collect data (longer term projects, or sensitive topics that rely on rapport with local communities which protected area staff may not enjoy). Reading this manual should never take the place of proper training *prior* to application of these methods. Protected area managers should insist on the inclusion of at least one experienced social scientist on their staff (and preferably two: one man and one woman, to facilitate working with women in those areas where it may not be culturally acceptable for men to spend time with women) who is qualified to evaluate the situation locally, determine the kinds of research which should be undertaken, and train the appropriate assistants in collection of data. These scientists may already be part of the national staff responsible for protected area management, or it may be necessary to ask advice from universities, research consulting firms, national NGOs or conservation groups in order to identify suitable individuals.

In any case, there should exist certain issues common to any protected area where local communities are present and likely to have an impact on the resource base. Entire books have been written on the methods available to assess these impacts, and they should be put to use wherever possible. In addition, all relevant background information should be consulted prior to data collection to ensure as complete a picture as possible of the project area. National and regional planning and policy reports, legal documents, historical archives, census records and so forth will provide a valuable perspective which will aid conservation planning. This chapter outlines some of the most basic approaches which can be used for gathering additional socio-economic information.

Prior to data collection, make preliminary contacts with regional and local government officials, in order to avoid misunderstandings or conflict at a later time. Clearly explain the goals and objectives of the conservation project and research programs. Local communities are often suspicious of outsiders, and tend to defer to authorities in their immediate area, even if the national government has already demonstrated its willingness to support conservation. If local official government "permission" is granted, subsequent meetings with villagers will be less troublesome. Once these authorities have been contacted, arrange preliminary and informal meetings with village chiefs or tribal councils, who can greatly facilitate village-wide reunions. The substance of village meetings will differ from site-to-site, depending upon the status of the project. For example, if the project has been around for a number of years, people may be familiar enough with its objectives that it will no longer be necessary to spend lots of time explaining why the project is there; instead, the goals and objectives of a particular research project could be discussed, and the role that local communities might play in data collection addressed. If, on the other hand, you are working in an area where a protected area is planned, it will be important to devote considerable time to discussing conservation, listening to people's concerns about natural resource management, explaining the objectives of this new project, and so forth. In either case, the importance of meeting regularly with local communities, of establishing a comfortable working relationship, cannot be overestimated. These contacts can pay enormous dividends in the long run, and make collaboration with local communities more meaningful. Keep in mind that you may need to have translators available to assist; even in those villages where many people speak a national language, there will likely always be some individuals (older people, women, illiterate folks) who do not. You want to facilitate participation and understanding by everyone, and often that only can often be done by using local dialects. Take the time yourself to learn basic phrases of greeting in local language, as people will appreciate this effort, and are often more willing to talk. After these initial meetings and sensitisation data collection can begin.

Sketch maps and village transects

Sketch maps

A village map is a sketch map of a village which roughly describes the number and locations of houses, farms, water sources, roads, paths, etc. Village maps can provide a wealth of important information, and are a useful way to put residents at ease at the very beginning of data collection. Published maps (preferably of the smallest scale available) and aerial or satellite photos should be consulted prior to village mapping exercises in order to familiarise oneself beforehand with the area. Maps can provide helpful knowledge about land-use strategies (agricultural fields, hunting territories, forest resources, etc.) demographics, problem areas (animal damage, water access) and so forth. Initially this can be done very simply, by drawing on the ground with a stick (this is often less intimidating for people, especially where paper and pens are not commonly used), and later transferred to paper. If you are using a participatory approach to mapping, it may be necessary first to sketch in some of the obvious features (roads, trails, school, dispensary, etc), but villagers should be encouraged to complete the drawing. These maps can be modified later as more information becomes available (they should help you to ask more questions of people), or compared to published maps or those done by your research team. The exact scale is generally not important, especially in those instances where the villagers themselves participate in this exercise, as their perceptions of spatial relationships are often as important as the actual map. The key is access to a good map (or series of maps) for project use which includes all houses and other important structures, water sources, roads, field locations, village limits obvious geographic features (rivers, hills, etc) and so forth (Figure 15.1).

Village transects

A village transect is a sketch made whilst walking through a village in the company of one or more of its inhabitants. Unlike sketch maps, which try to represent all features of the village in two dimensions, village transects provide a one-dimensional pass through the village which show the relative locations of areas used for different activities. This should be one of the earliest steps in working with villagers, as it tends to encourage participation, and it is generally a rather comfortable exercise. Unlike the forest transects (see Chapter 13), those in the village provide mostly qualitative data which compliments the information obtained from village maps. This type of transect will help you to understand how local communities divide and manage land in proximity to their villages, as well as socio-cultural features in the village which may prove important at a later time. For instance, if you will need to stratify the village for some future research (see sampling, below), the transect data will be very useful. Depending upon your research questions, you may want to make observations about crops (both cash and subsistence), soil types, animal (both domesticated and wild) and human activity, water sources, cultural activities, building materials, problems, needs, and so forth.

The following steps are necessary for completing village transects:

- Create village maps which provide a good overall view of the village. Remember that the transects are supposed to provide complementary data to that of the preliminary sketch maps. These maps will allow you to choose a logical starting point for the transect in order that your team will cover all zones of importance to the village (water holes, gardens, fishing spots, sources of building materials, fallow fields, abandoned villages, etc).
- Outline relevant research questions and the information that needs to be gathered to supplement the village sketch maps. If your team has several members (and this is advantageous, particularly if they have different expertise), decide who will be responsible for which observations. For example, a wildlife management specialist should note where animals are damaging crops; an agroforestry expert can record data on farming practices; your social scientist might be responsible for data on economic activities, and so forth.

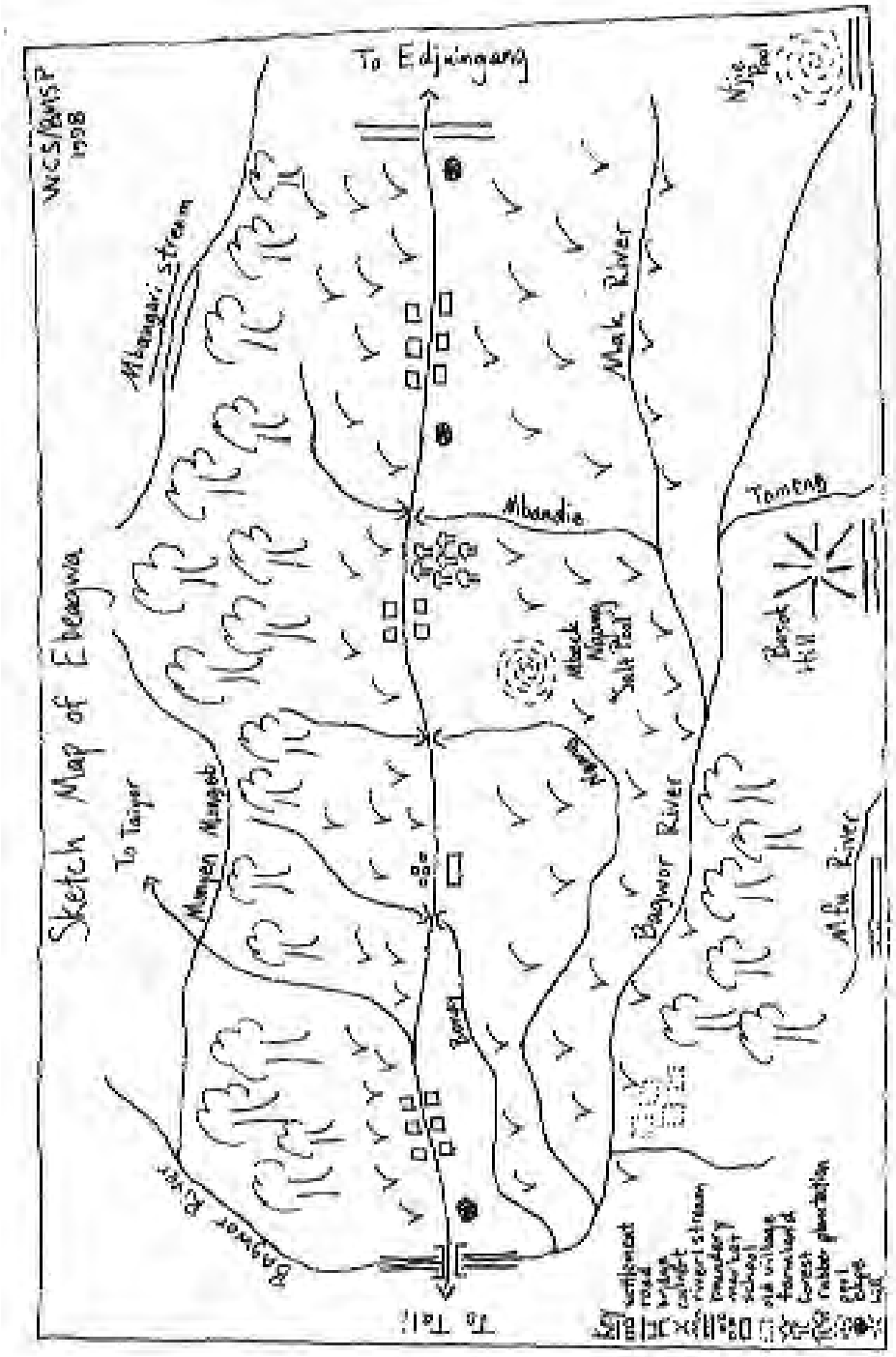


Figure 15.1 Sketch map of a village drawn by staff of the WCS Banyang Mbo project

- Identify community members (2-3 people) who are familiar with the area and willing to participate in this exercise. Explain to them the kinds of data that will be collected and the intended route, so they can help steer the research team towards appropriate areas or put them in contact with knowledgeable people.

- Walk the transect. If the village is especially large it may be more practical to drive. However, in order to minimise differences in status with villagers, it is better to plan several walking transects instead. Take care to make appropriate observations along the way. Team members should ask questions of the informants who are participating, and take time to stop along the way to informally interview villagers about pertinent issues. The types of questions you will ask will largely depend on the focus of your research, but remain flexible enough to follow lines of inquiry opened up by villager responses (see informal interviews, below). At the very minimum try to identify the most important natural, agricultural and economic zones of the village, and note characteristics of the following in each: soils, vegetation types, water, subsistence and cash crops, obvious problems (erosion, crop diseases/damage, poor child health), economic indicators (building materials, presence/absence of latrines, etc), opportunities (school, market, natural resources) and so on.

- Compile team field notes and draw the transect (Figure 15.2). Review the resulting diagram with key informants and use it to stimulate discussion in subsequent village meetings.

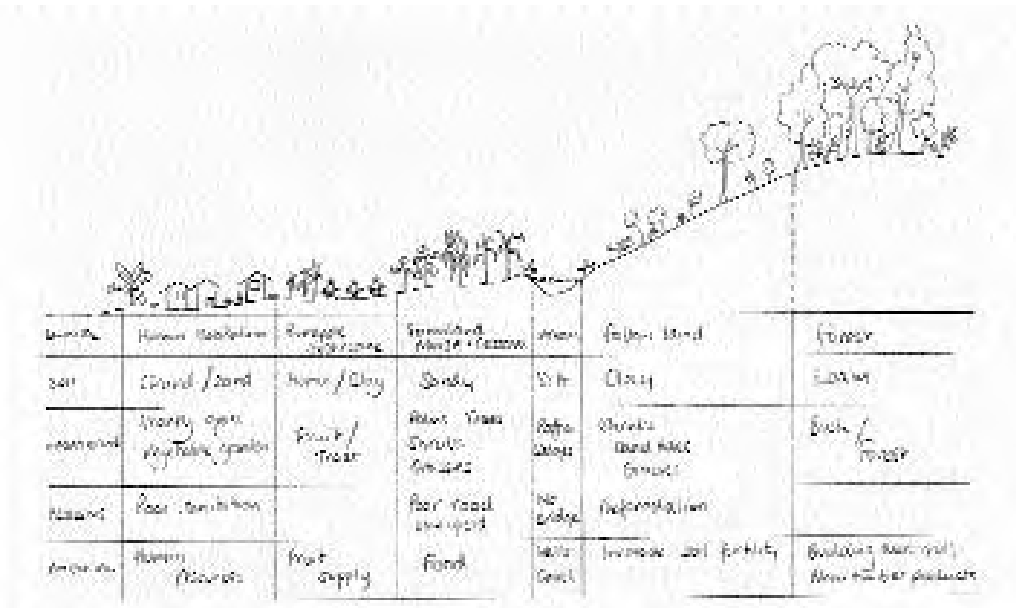


Figure 15.2 A village transect.

Population census

Most human populations in central African forests are increasing, either through natural demographic trends or immigration. Project managers must have accurate information about how many people live in or near protected areas, as well as basic socio-economic data about these communities. The first step towards acquiring this knowledge is a detailed human population census. Once these baseline data have been gathered, change can be effectively monitored. If local populations are growing, is it due to natural demographic trends, or immigration? If immigration is occurring, who is moving into the area, and why? With this information, protected area managers can make appropriate decisions about how to manage human activities so that resource use might be maintained at sustainable levels.

A population census should not simply focus on counting how many people live in any given location. The census can be used to gather a variety of village data, including (but not necessarily limited to): ethnic composition, age structure, individual tenure within villages and prior movements of people, principle subsistence and economic activities, house construction materials, presence of domesticated animals, levels of education, and so forth. It is important to remember that each conservation project will have specific research objectives which should be reflected in the census questions. Figure 15.3 is an example of a census data sheet used in southeastern Cameroon.

How to conduct a proper census of local communities

A proper census of local communities should include the following steps:

- Background search for all available census information from government records (or other sources, including missionary or health centre data). These data will be useful for evaluating trends over time, although it is important to remember that sometimes government records are inaccurate. For example, village census workers can be influenced by chiefs or local officials to inflate figures if it means that more development money will be available. Or mobile hunter-gatherers are notoriously difficult to count, and therefore are often ignored. Nonetheless, an historical perspective on population numbers can be very helpful for protected area planning.
- Preliminary contacts with regional and local government officials and traditional village authorities should occur well before the census takes place. Getting to know villagers first will facilitate data collection. It is important to put people at ease at the start of each household visit, in order to minimise distrust. For example, a common problem with census work is that people believe that the government will use the results to control or tax them. Explain carefully the goal of the census before beginning.
- Create village maps (see above) if they are not already available, making sure that each residence is included and assigned a corresponding number. In particularly large villages, it may be necessary to prepare separate maps of each quartier, for example, in order to be sure that all houses are represented.
- Define relevant research questions and design the census form. Think carefully about the information most important to your protected area, and especially about how these data will be analysed and interpreted. For instance, if there are no means of formal employment in your area, it may not be necessary to ask about occupation. Or if computers will not be available for data analysis, the census statistics collected should be as simple as possible, while still meaningful for the protected area. Be sure to include an explanatory note (Box 15a), as this often helps to identify those parts of the census forms which may be difficult for census takers to understand.

Box 15a EXPLANATORY NOTES FOR A CENSUS SHEET USED IN SOUTHEASTERN CAMEROON

Village: Name of the village where the census took place.
Observer: Name of the researcher who noted the data.
Date: Date of census - day/month/year (eg.: 12/03/1994)

: The number given to the household on the village map.

Letter / Name of person: A letter should be entered for each person resident in the household. If the project needs names note the first and family name of each person.

Sex: M for Male; F for Female.

Age: Age of the people censused. Note the age as precisely as possible. A historical events calender can be used to estimate the ages of people who have no birth certificate or other document stating their age and who do not know their date of birth. Estimate ages above 20 years in 10-year categories: 20-30, 30-40, 40-50, etc.

Relationship: Relationship to the head of the household:
W = Wife; **P** = Parent; **G** = Grandparent; **C** = Child; **GC** = Grandchild; **B** = Brother; **S** = Sister; **N** = Nephew/Niece; **U** = Uncle; **A** = Aunt; **C** = Cousin; **FL** = Father-in-Law; **ML** = Mother-in-Law; **BL** = Brother-in-Law; **BS** = Sister-in-Law; **DR** = Distant Relative; **F** = Friend; **V** = Visitor.

Ethnic Group: Name of the cultural group of the head of the household.

No. children: Total number of children (present or not, living or dead) for each person at the time of the census.

Age of children: Heads of family should give the ages of their youngest and oldest children. For example, if the youngest is 2 and the oldest 13, write 2-13. For children less than 1, write 1.

Arrival date: The arrival date is the date when the person moved to the census village. For people originally from the village note "0".

Where from?: The previous place of residence of the person before coming to this village.

Education: Education level completed, note:
I = Illiterate; **L** = Literate; **P** = Primary; **S** = Secondary;
HE = Higher Education

Occupation: Profession of each person censused (eg.: 'ménagère', farmer, carpenter, student, etc.)
NOTES: The observer should note any supplementary information which clarifies the situation of any given household. For example, the head of the household has a house for each of his three wives, status of visitors, family members who have travelled away (to where).

	Household #
	Letter / Name of person
	Sex
	Age
	Relationship
	Ethnic Group
	No. children
	Age of children
	Arrival date
	Where from?
	Education
	Occupation

Village: .....

Observer: .....

Date: .....

Figure 15.3 A census form used in southeastern Cameroon.

- Identify local research assistants who are capable of collecting accurate census data. If possible, hire from each village to facilitate data collection and reduce mistrust. Train these assistants to understand and use the village maps and census data sheets.
- When working in areas where dates of birth are unknown, it is useful to prepare a historical events calendar with the help of local communities. This calendar should include those episodes which will be known to everyone in the area, so that when censusing individuals of unknown age, their approximate dates of birth may be calculated based on their proximity to known historical events (Box 15b).
- Collect census data. Visit each residence. The house number from the village map should correspond to a number on the data sheet. The simplest way to gather these data is to ask the head of each household who normally sleeps in his/her house. This will reduce the likelihood of counting temporary visitors, and avoid any confusion about differentiating between children who have moved out of the area or died (although it is often also interesting to gather these types of data). For instance, if one is interested in family size, or mortality information, it might be important to ask about total numbers of children, regardless of where they are during the census.

Keep in mind that a complete census will be virtually impossible, as inevitably some individuals will be unavailable or unwilling to participate. Do not force anyone to cooperate, as this will only reinforce the notion that this is an official government census, and may discourage others from being honest in their responses. You are interested in as complete a demographic picture as possible, so that you will get to know people, and will be capable of monitoring future population trends that may have an impact on the protected area. In fact, with good baseline data, it may be adequate and more cost effective in the future to simply count houses in order to follow the course of demographic changes. Again, this will depend upon the kinds of information vital to management of each protected area.

Box 15b HISTORICAL EVENTS - SOUTHEASTERN CAMEROON

Moloundou taken by the French	1914
First World War	1914-1918
Opening of the Yokadouma-Moloundou road	1930-1932
Second World War	1939-1945
First car in the region	1952
Independence of Cameroon	1960
Reunification of Cameroon	1960
Troops arrive at Moloundou to fight the rebels	1964
Sous-Préfecture of Biyo'o created at Moloundou	1969
Creation of the Catholic Mission at Salapoumbe	1969
Opening of the 'College d'Enseignement General' at Moloundou	1970-1971
Unification of Cameroon	1972
Opening of air strip at Moloundou	1973
Creation of the Catholic Mission at Moloundou	1977
President Biya installed	1982
Opening of the Ngilili-Kika (SIBAF) road	1983
Opening of the Mambele-Kika (SIBAF) road	1989

A word on sampling:

The principles of choosing samples for biological research outlined in Chapter 3 are equally valid for socio-economic studies. Unlike a census, it is simply not possible (nor advisable, due to limitations on time and money) to talk to or study everyone in the population (and as we have seen, even a census is liable to miss some individuals) when using some of the methods outlined below. You must therefore select a sample from the population. The key is to select a *representative sample*; that is, a subset which will mirror the larger group in such a manner that conclusions drawn from your sample will allow you to make valid assumptions about the group as a whole. In order for this to be true, careful consideration must be given to the size of your sample, and how it is chosen.

Generally speaking, the larger a sample size, the more accurately your results will reflect the reality of a given situation in the entire population. At the same time, the study will become more expensive in terms of the time and money required. It is therefore critical that your research questions are well-defined, and that in your planning a balance is reached between the size of the sample and the cost of doing the work. The bottom line is that your sample size must first be defined in terms of the cost, and then with respect to the level of accuracy required. If you know, for instance, that you have two months to complete a certain study, and two field assistants to collect data, you should be able to calculate fairly easily how large your sample can be. For example, if your field assistants are capable of six interviews per day, and they will work for 50 days during the two months available, then you might expect that they will be able to complete a maximum of 600 interviews (2 assistants x 6 interviews/day x 50 days = 600). You must then decide if that will be sufficient for the purpose of your study, and if not, revise your research design (for example, make the questionnaire shorter, or consider other methodological approaches which will allow you to collect the same type of information in less time).

Once you know how large a sample you can reasonably afford to study, you can evaluate how *representative* it is likely to be of the target population. Again, this will depend in large measure upon the type of research you are doing. If you are pretesting certain questions, for example, or are simply trying to get a good sense of what is happening in your protected area, so that your "real" study can be better defined, you can be reasonably safe with smaller samples of 30-50 individuals. The same is true for detailed, long-term studies; fewer subjects should be sufficient. Keep in mind that you may need to understand different groups of people; that is, to *stratify* your sample. Stratification involves separating people into distinct categories. For example, you might want to consider males and females, or different age or ethnic or income groups. If you are doing a hunting study, for example, it is unlikely that you will need to be concerned with women, but you might want to evaluate differences between hunter-gatherer ("pygmy") and farmer practices. The bottom line is that it is probably better to collect less data in a carefully controlled fashion than to increase the sample size at the expense of accuracy.

Selection of the sample is just as important as its size. The larger sample sizes (over 100) should be *randomly* drawn from the population; that is, all individuals have an equal chance of being chosen. This is important, in order to *minimise bias*. There are numerous types of bias which can be introduced during data collection; the research design must overcome this problem to the extent possible. For example, if you only talked to chiefs or elites about the size of their gardens, you would almost certainly conclude that villagers had larger fields than they actually do. If you were interested in forest subsistence activities, yet only studied this issue from January to June, you would introduce a seasonal bias that might cause you to miss a particularly important resource that was collected in September. Care must be taken to avoid spatial biases also; many research projects only work with villages which are easy to reach by road, rather than those which require arriving on foot. It is equally important to avoid a temporal bias (i.e. at certain times of day, only a well-defined subset of the population can be found in the village - old or sick individuals, for example).

The purpose of any research project is to learn more about the population within your protected area so that proper and realistic management decisions can be made. Therefore your study must reduce sampling biases. This means you must get out of your car, walk in the rain, work in the hot sun, visit the poorest households, and so forth. This is the only way to make the truly informed judgments that are critical to sustainable management of protected areas.

There are two relatively easy ways to randomise the sample selection. Both depend upon the village map, with each house assigned a number. You can employ the random numbers table, as explained in Chapter 3 (See Box 3a and the accompanying table). Alternatively, the research assistants responsible for data collection can begin at one end of the village, and depending upon the targeted sample, visit every third (or second, or fifth) house. It is best to approach the targeted households in advance, and map out who the subjects will be in each village before actually beginning the work. Assuming that every adult in each household is eligible to be interviewed, these preliminary visits will allow your research team to randomly select the subject in addition to the house (simply assign each possible interviewee a number, and then use the random numbers table to select the individual). In this way, it may be possible to schedule interviews throughout the day (rather than only early morning or late afternoon when most people can be found in the village), even if it means following respondents when they go to markets or to work in fields. If the residents do not wish to participate, simply advance to the next house, and then continue on to the next pre-targeted household.

Measuring the impact of a local population on forest resources

To evaluate the effectiveness of natural resource management decisions first determine which resources are most critical to local users, as well as the levels at which these resources are being exploited. A variety of methods will be useful for gathering resource use information.

Seasonal activities calendars.

A seasonal events calender is a summary in table form of the activities undertaken by the population at different times of the year. These are a very effective early step to measure how local communities allocate their time throughout the year. Any activity which has a temporal component can be displayed in a calendar format. Not only will these calendars help you to understand what people are actually doing during different seasons, but they allow the researcher to elicit information about the importance of various resources, including cash and subsistence crops and forest products. In addition, they permit protected area staff to plan the best times of year for working with local communities. For example, in the Banyang-Mbo Sanctuary in southwestern Cameroon, plans were made to work on a community self-help initiative (clearing a path for an electric fence to keep elephants out of gardens). Although the community was clearly interested in this project, and expressed a willingness to provide the necessary labour, when construction was to begin, very few people came to work. When project staff asked about the apparent lack of interest on a project that could be very beneficial for them, it was discovered that the work had been planned during the height of the cocoa harvest season, when there was very little labour available for other activities. Calendars can also provide information about climate, availability of labour, and so forth, and can even be used to monitor how the presence of a conservation project modifies the manner in which

The following steps are important for creation of a village seasonal activities calendar:

- Begin with a background search of secondary sources (previously published social science research, climatic data, government agricultural records, etc.). Once these documents have been consulted, the protected area team can develop a preliminary regional calendar before working in the village itself. This will allow you to have a good idea of the basic activities that are likely to occur in every village, and to define the kinds of questions that will facilitate the subsequent village meeting.

- Preliminary meetings with government authorities are essential before going to the villages (see above). Traditional village leaders also should be contacted so that they will understand the purpose of the research, and encourage people to participate.

- Schedule meetings with different focus groups in villages. Farmers should be prepared to discuss the annual agricultural cycle, and another group for activities that occur in the forest. It is useful to have different calendars prepared by all stakeholders, as they will better reflect true activity patterns. For example, if you only talk to men in a village, yet most gathering of forest products is done by women, the amount of time people spend in the forest would be under-represented by the resulting calendar.

- Start the actual meetings with very general discussions of what activities occur throughout the year. Once a list of these activities has been prepared, you can begin to place them into a calendar format. Keep in mind that in some illiterate communities, the notion of months may be difficult to grasp, and it may be more productive to refer instead to seasons (e.g., What do you do in your gardens during the heavy rainy season? When do you go to the forest to collect construction material? Do you plant your cassava at the beginning or end of the first dry season? What time of year does the best hunting take place?).

- Once a fairly comprehensive list of activities has been compiled, you must decide how best to present these data. If the community or focus group is literate, you may be able to use large sheets of paper and a normal Western calendar (often 18 month calendars are more useful than 12 month, as this will allow complete planting cycles to be covered, for example), with relevant climatic data (rainfall, monthly temperature, etc.) included. Activities can then be plotted directly onto the calendar. On the other hand, if the group is illiterate, it will be necessary to develop other visual aids. For example, a "calendar" can be drawn in the dirt with a stick, and then different seasons can be represented with sticks or pebbles (more pebbles or longer sticks for more rain, for example), and symbols for each activity placed on the calendar. You can let people decide what those symbols will be from materials at hand; different leaves for different forest products, or farming implements for different crops, and so forth. Alternatively, if you have a good idea before hand from your secondary sources what the majority of activities are during the year, you can bring these "symbols" with you to facilitate the meeting.

- Once the different calendars have been prepared, a composite can be developed (see Figure 15.4). These should be continually updated as a result of additional data collection or direct observations during subsequent visits to the village.

Seasonal Calendar of Ebeagwa Men's Activities, 1998

Activities	Months											
	J	F	M	A	M	J	J	A	S	O	N	D
Clearing												
• First clearing	■	■	■	■								
• Second clearing								■	■			
• Lamba											■	■
• Palms											■	■
Burning		■	■									
Planting												
• Plantains			■	■	■	■	■	■				
• Cash crops						■	■	■				
Harvesting												
• Coffee	■									■	■	■
• Cocoa	2					1	1	1	1	2	2	2
• Plantains	■	■	■	■	■	■	■	■	■	■	■	■
• Bush mango						■	■	■				
• Njansang								■	■	■		
• Palm nuts			■	■	■	■						
Nursery	■	■	■								■	■
Pruning			■	■	■	■						
Spraying			■	■	■	■	■	■	■			
Fishing												
• Net	■	■	■	■	■	■				■	■	■
• Trap	■	■	■	■	■	■	■	■	■	■	■	■
Hunting	■	■	■	■	■	■	■	■	■	■	■	■
Trapping				■	■	■	■	■	■	■		
Weaving	■	■	■	■	■	■	■	■	■	■	■	■
Thatching												
Tapping	■	■	■	■	■	■	■	■	■	■	■	■
Peak Tapping	■	■	■	■						■	■	■
Afofo	■	■	■	■	■	■	■	■	■	■	■	■
Feasting			■	■								■

1. German
2. Amazon

Figure 15.4 Seasonal events calender

Market surveys.

One way to measure the impact of local communities on forest resources is to record which items are available for trade/sale in local and/or regional markets. Note the type and quantity of the resource, as well as it's price, where it was hunted, gathered or cultivated, and by whom (i.e., is the procurer actually selling the item, or are middle-men involved?). In addition, note the presence of goods manufactured outside of the area, particularly where forest resources are traded for the acquisition of this item (note the "exchange rate" in this instance, as a means of documenting the value of forest products in local economies).

Field size.

Villages should be sampled, and randomly-selected garden areas measured. This requires that your research assistants be familiar with the use of a compass and measuring tape, and can accurately record their observations (see Chapter 6 for details of map-making). It might be useful, depending upon your protected area's research needs, to record the following information: location of garden in relation to village; habitat type cleared for field (see Chapter 9); types of crops planted (distinguish between crops planted for subsistence and for commercial purposes); and estimates of crop yields for each type. For cash crops, research assistants should gather data on expected yields, expected point of sale and economic return. It is important also to evaluate fields in fallow, not only to gauge their importance with each succeeding year of fallow for different crops, but also to better understand how local communities use and maintain ownership of their land. Once these baseline data are available, it may be possible to monitor future forest clearing for agriculture with aerial photos or video.

Resources exploited.

A more direct assessment of resource use can be obtained through surveys in targeted communities, where the types, quantities and sources of exploited resources may be characterised. It is critical in this instance to stratify and randomly sample villages and households, such that data are collected from the full range of resource users. Once the population census has been completed, villages can be stratified by principle economic foci, for example: farmers, fishermen and hunter-gatherers may reside in distinct locations, and all should be sampled. It is particularly important that within each village a random sample of households is visited, in order to avoid bias by age, sex, social or economic status, and so forth (see note above on avoiding bias).

Once the appropriate households have been identified, employ one of two methodologies, depending upon the time available. Ideally, the observer should stay in the selected village or camp on certain randomly selected days during the sampling period (and this should not be biased by season, as different types of resources will be preferentially exploited at distinct times of year; in other words, a study of this nature should run for at least one full year in order to have a complete picture of resource use). The number of days will depend upon the resources and number of assistants available for this study, but as a "rule-of-thumb," subject households should be visited a minimum of two times each month. The assistant then records the resources (forest or otherwise, depending upon your study) which are brought into the household. If you are working in an especially small village or camp, it may be possible to choose a central location and record everything brought in by all residents. Weigh all food resources (including agricultural and forest, and perhaps even those purchased in markets) and categorise all animals into age and sex classes.

In addition, it is important to identify where and how each resource was collected (hunted, gathered, cultivated, purchased, traded for) and by whom. Two detailed examples of this kind of study are presented in the case studies on hunting impacts in southeastern Cameroon and Ituri Forest, RDC (Boxes 15c & 15d). A similar study could be effective for any number of forest resources. By modifying the methodology, for example, you could gauge the importance for local communities of fishing, or wild mango gathering, or the collection of building materials. It would be wise to consult with a trained social scientist before beginning this kind of research.

Alternatively, villages can be visited on the same randomly-selected days, and using the same data sheets, ask questions about the resources brought in the previous day. There are a number of disadvantages to this approach: some resources may not be reported, either because they've been forgotten, the respondents don't want it known, or consider something too insignificant to identify (caterpillars or snails, for example); in addition, weights must be estimated. Despite these drawbacks, this approach can still be a cost-effective method to collect baseline data about resource use among local communities.

Box 15c CASE STUDY: IMPACT OF SUBSISTENCE HUNTING IN SOUTHEASTERN CAMEROON

The objective of this study was to investigate the impact of village-level subsistence hunting on the fauna of the area, with the long-term goal of evaluating the sustainability of this activity. The following methods were employed, demonstrating the importance of collaboration between social science and biological research teams for protected area management. For more details, consult Fimbel et al (in press).

Documentation of hunting offtake

A reference list of animal names was generated in Baka and Bangando languages. Several local assistants, representing both Bangando and Baka ethnic groups, were selected and trained in the use of standardised data forms (see below) and spring balance scales. Each assistant worked with a fixed group of hunters, who were selected based on their willingness to participate in the study, and the proximity of the hunter's residence to the assistant's residence, to facilitate communication and data collection (we followed 46 Bangando and Baka hunters for 17 months, from November 1994 to March 1996). The assistant collected relevant biological data (species, weight, age, sex) on the animals killed, as well as hunting technique and location, destination of meat (local consumption or sale), etc.

Duiker inventory

The abundance of duikers in the Lobéké forest was estimated using census walks to record abundance of sign, primarily dung, along straight line transects. Because the dung pellets of medium-sized duikers (*Cephalophus callipygus, C. dorsalis, C. nigrifrons*, and *C. leucogaster*) were of a similar size and shape, it was not possible to distinguish among the dung of these four species. Thus, these duikers were combined into a common category of 'red duikers' to distinguish them from the smaller 'blue duiker' (*Cephalophus monticola*). The forest east of the main N-S road was stratified into four zones (0-10km, 10-20km, 20-30km and >30km), representing progressively decreasing levels of human intervention. Fifteen transects of 5 km length were distributed amongst the four zones, and each was sampled three times during the study period. Census walks were undertaken by a team of 3 persons: one to search for nests and primates in trees, another to search for and identify dung and signs of animals on the forest floor, and the third person to record data and coordinate team efforts. Transects were walked adhering to the general guidelines for line transect sampling described by Burnham et al. (1980) and Barnes and Jensen (1987). All animal signs were recorded, and estimated ages and perpendicular distances were included for direct sightings, nests and dung. Transect widths, and then density estimates were calculated for 'blue' and 'red' duiker dung using DISTANCE (Laake et al. 1994). Dung density was converted to animal density using the following equation:

Animal Density = dung pile density / (defecation rate/day) x (dung decay period)

Lacking data from southeastern Cameroon, values were taken from the literature (Koster and Hart 1988) for defecation rates of 4.9 and 4.4 pellet groups/day, and decay periods of 18 days and 21 days for blue and red duiker pellets, respectively.

The non-parametric Kruskall-Wallis procedure for analysis of variance was used to test for differences in the frequency of duiker sign among study zones.

DATA FORM AND INSTRUCTIONS GIVEN TO ASSISTANTS IN A STUDY OF SUBSISTENCE HUNTING IN SOUTHEASTERN CAMEROON

This study is intended to evaluate the impact of hunting on forest animals in south east Cameroon. The results of the study will enable local populations to evaluate how best to maintain their forest resources. Will future generations always be able to hunt bush meat? We do not intend to stop subsistence hunting - WCS believes that wise use of forest resources is the best way to conserve them for the future needs of local populations.

It is important, therefore, that you take great care to collect the data correctly. In this way we will be able to better our understanding of the processes of conservation and improve our collaboration with local communities.

Instructions:

- The hunter's name should remain anonymous. It is important to explain to people that we will not be able to collaborate with them unless they cooperate fully in this study. If some hunters are reluctant, do not insist that they participate.

Date	Name of Animal	Sex	Age	Weight	Hunting method	Site	Destination / Price	Comments

However, it is important to establish the number of hunters encountered in any given area

- Only choose hunters based close to your village, to avoid the need to travel long distances. If possible try to obtain a representative sample of Baka and village hunters. All animals killed should be shown to the observer who should complete the checksheet in the following way:

1) **Name of animal** - Write the name of the hunted animal in the column indicated. It is very important to write the correct local name. For example, DAWA is the generic name for MONKEYS - you must note the specific name for the type of monkey (such as NDJOGO or PUNGA).

2) **Sex** - Carefully examine the animal and note the sex. If the animal is smoked and it is impossible to be sure of the sex note "unknown" in the column.

3) **Age** - Where possible choose one of the following age classes: I = Infant; J = Juvenile; A = Adult. If possible collect the lower jaw in order to better estimate the age later.

4) **Weight** - Weigh the animal with your balance.

5) **Hunting method** - Note the weapon or method used to kill the animal. For example: snare, cross-bow, spear, dogs, shotgun, etc.

6) **Site** - Note as accurately as possible where the animal was killed. When you note towards Boumba or towards Lobéké be sure to give the distance from the village (250m or 15km).

7) **Destination / Price** - The destination is the intended use of the meat: for local consumption or for sale. If it is for sale, what was the **total** price, and was it sold locally or elsewhere.

8) **Comments** - Note the hunter's ethnic group (Bangando, Bakwele or Baka). Also, note "**R**" if the animal was already rotten, "**S**" if it was smoked, et "**P**" if it was a pregnant female. Any other information and comments are welcome.

Results.

A total of 325 blue duikers and 296 red duikers were taken over the course of 354 hunter months in the Lobéké region. Annual offtake rates for duikers were calculated using 'per hunter' kill rates in conjunction with the total number of hunters, and the size of the region. We found an average capture rate of 0.9 blue duikers/ hunter/ month (11.0 blue duikers/ hunter/ year) and 0.8 red duikers/ hunter/ month (10.0 red duikers/ hunter/ year).

Based on a combination of formal and informal village surveys, we estimated that 80% of 1,330 adult males over the age of 15 in the local population actively engage in hunting, yielding a total of 1,064 hunters.

This gives a total annual offtake of 11,725 blue duiker/ year and 10,671 red duiker/ year captured by local hunters of the region.

Finally, 98.8 % of blue duiker and 93.2 % of red duiker were reported to have been captured within 30 km of either side of the road, a capture area of 3,113 km2. These data allow us to calculate annual offtake estimates as density figures for the region within 30 km from the villages as follows:

98.8 % of 11,725 blue duiker = 11,584, divided by 3,113 km^2 = 3.72 blue duiker/ km^2

93.2 % of 10,671 red duiker = 9,945, divided by 3,113 km^2 = 3.19 red duiker/ km^2

Discussion.

We believe that hunting pressures have been mounting in the recent past, based on claims by local people that they need to go deeper into the forest to catch duikers compared with previous generations of hunters. This observation suggests a trend whereby local populations of forest antelope are being reduced due to over-exploitation. In addition, the local human population appears to be growing due to the immigration of Cameroonians and Congolese from areas where game meat has been depleted in recent years. In addition, the logging industry has brought many changes to the region, including:

1) increased access to the forest interior through the creation of logging roads;
2) the introduction of a non-indigenous labour force; and,
3) a mechanism for transporting game meat to external markets on logging trucks.

Finally, the economic crisis resulting from the collapse of coffee and cocoa markets, and the devaluation of the Central African Franc, has caused more people in the central African region to turn to the natural resource base to earn a living (Lahm 1993). The composite of these conditions has created a situation whereby pressures are mounting on a resource base that is being locally depleted, but probably at the same time renewed by immigration from adjacent source habitat such as the more remote forest interior.

We view the current high levels of hunting as alarming because the harvest data gathered are most certainly underestimates of the total rates of offtake of wildlife for the region. Our harvest figures do not account for the offtake of commercial hunters from the market centres of Moloundou, Kika, and Libongo, nor that of the to safari hunters operating in the region (believed to be minor compared with offtake by residents). Thus, harvest levels are likely greater than those observed, which does not bode well for the sustainability of forest antelope hunting, especially for the red duikers, as currently practiced in the region.

The first step in guarding against over-exploitation of heavily hunted game species is to establish a central protected area where hunting is not allowed. High faunal use habitats such as forest clearings or mineral licks should be protected from exploitation, even when they occur within community hunting zones. It is likely that ungulates frequent these habitats to add essential minerals to their diet, or to obtain high quality forage. Thus, in the interest of maintaining high levels of animal reproduction, hunting should be prohibited within a 1 km radius of such high animal-use areas. Following the creation of a protected zone, designated hunting blocks could be incorporated into a multiple-use buffer zone surrounding the central protected area. The establishment of community hunting zones would constitute a concrete example of support for indigenous land rights, which is widely recognised as one of the best options for the conservation and rational use of tropical forests outside of protected areas (Redford and Stearman 1993). The creation of adjoining hunting and protected blocks should provide a relatively risk-free opportunity for local management of the fauna, without posing a great threat to biodiversity.

The granting of a community hunting zone will likely be contingent upon acceptance of a game management plan for the zone by the government. However, the plan should be prepared locally, with technical assistance, to enhance indigenous participation in resource management. Within these zones, communities could be granted control over faunal resources (subordinate to the existing national game laws), including the option to refuse access to non-"members," or alternatively, to allow managed sport hunting that could generate revenue. In return, community members could be required to restrict their hunting to mandated areas, and to collectively desist from hunting within the protection zone. In addition, the community should be required to monitor offtake within their designated hunting sector.

Box 15d CASE STUDY ON THE SUSTAINABILITY OF HUNTING AND FARMING IN THE OKAPI WILDLIFE RESERVE, NORTHEASTERN RDC

The following case study illustrates how a coordinated program of biological and socio-economic research can yield results important to protected area management (from Wilkie et al, 1998). To examine how, at present and in the future, subsistence farming and hunting are likely to impact the extent of mature forest remaining, and the biomass of wild animals within and bordering the Okapi Wildlife Reserve (OWR), we used data on: a) the area and distribution of mature and disturbed forest; b) human population size, distribution, and growth rate; c) the rate of forest clearing for agriculture per household; d) the average fallow period employed by farmers; e) meat consumption per capita; f) the relative composition of game species commonly exploited; and, g) production levels of commonly exploited game species.

At a predicted annual growth rate of 3.18%, farmer and forager populations within the OWR will more than triple in 40 years, growing from 28,897 in 1987 to 101,082 in 2027. Overall population density will rise from 1.3 people/km2 (range 0.49-7.82) to over 4.5 people/km2 (range 1.73-27.35).

At present mature forest covers 90% of the whole region, and 85% of the landscape within 15km of settlements. Ninety-three percent of all disturbed vegetation lies within this 15km zone. Simulations suggest that after 40 years and an increase in human population of more than 300%, the area of mature forest will not have changed significantly over the entire region nor within 15km of settlements. When the data are separated by political district only three of the seven districts show a reduction in mature forest, and the average area of mature forest only declines over a 40 year period from 80% (range 16-98%) to 78% (range 16-93%). This corresponds to an annual deforestation rate of ~ 0.06%, compared to 2-3% for the Congo Basin as a whole.

In contrast to the optimistic future for mature forest conservation over the next 40 years, the outlook for animal populations is less favourable, particularly within 15 km of settlements. At present, over 1.3 million kilograms of bushmeat are consumed annually by the inhabitants of the OWR, and that is predicted to increase to over 4.7 million kilograms in 40 years. Duikers and other small antelopes account for over 80% of consumption, with over 1.1 million kilograms being exploited each year at present, rising to over 3.8 million kilograms by the year 2034. Results show that depending on the production estimates used, the sustainability of duiker exploitation in the OWR at present levels is ambiguous.

Using high antelope density estimates, only 11% of duiker production is consumed on average each year within all political districts, and only one district may at present be unsustainably exploiting duikers. Consumption in all other districts is well below the sustainable harvest rate. In stark contrast, if we use a low duiker density scenario, consumption exceeds 40% of annual production. In three districts consumption exceeds 30% of production, and in one district humans consume almost 1.5 times the number of duikers that are produced each year. With populations expected to triple in 40 years the impact of subsistence hunting on duikers will increase. Even using the high production scenario, duiker exploitation for domestic consumption alone will become unsustainable throughout the OWR within 60 years.

This simulation suggests that as long as: a) markets for commercial crops do not revitalise; b) abandoned plantations are not re-established; c) farming does not become a major subsistence activity of the majority of foragers; d) logging does not become commercially viable, and; e) outside immigration does not become a major contributor to population growth, ample fallow forest for agriculture will be available for 20-40 years even if resident populations increase substantially. Given DRC's present political gridlock, the appaling condition of the roads, and the collapse of the economy, it is likely that the majority of the above assumptions will remain true for the foreseeable future. Thus we can expect that over 85% of the OWR will remain mature forest for at least the next 40 years.

Hunting is clearly a concern for the long term conservation of duikers and small antelope throughout the OWR, and to primates in the bow-hunting districts. One can argue that the data on bushmeat consumption patterns and animal densities were collected only in a few areas of the OWR, and then generalised for the whole forest, thus obscuring any variance that may exist throughout the region. One can also question the accuracy of both consumption and production figures. Regardless, the take home message of the simulation is clear: bushmeat consumption poses either an immediate or potential threat to forest animal populations, particularly the small antelope species. Further research efforts are clearly warranted to verify the results of this study, and to provide a sound foundation for development of effective natural resource conservation policies and practices.

Iron Age pottery found in Lopé, Gabon.

Understanding attitudes/asking questions/integrating people.

The working relationship between protected area management teams and local populations is important for achieving conservation objectives. It is therefore critical that you spend time talking to people, trying to understand better their attitudes, what motivates them, who is exploiting which resources, and at what times of year, etc. The impact of your project on local communities (for example, an increase in integration into market economies as a result of access to paid jobs, or the effect of restrictions on resource use), and the level of local participation in management should also be monitored. Both quantitative and qualitative methodologies are available for gathering these data.

Village meetings / focus groups

These are qualitative in nature, but they allow local communities to discuss issues in an open setting, and often result in information that is important for management decisions. It is critical that the full demographic range of the population is represented (young, old, men, women, agriculturalists, hunter/gatherers) in these village meetings. If this is not the case, it may be necessary to hold separate meetings with each focus group in order to be sure that all opinions are being considered. For instance, women may be excluded if these meetings are perceived to be "men's work", and the tensions which often exist between horticulturalists and their hunter/gatherer neighbours might best be deflected by giving each group its own forum to express ideas. These decisions can be reached after observing who participates in initial meetings.

These village meetings, at least initially, will probably be convened and directed by project staff. It is important that local languages are used wherever possible, in order to ensure that everyone understands the discussion; older people, women and hunter/gatherers (“pygmies”) may not speak regional or national languages well enough to participate in discussion. A local research assistant can help monitor these meetings; it is critical that the assistants understand that their role is to translate, and not to interpret questions or answers according to their own beliefs. It is the role of the moderator to prepare certain questions or issues for discussion in advance, and to guide the dialogue, but it is important that the moderator not remain the focus of the meeting. The purpose of these focus groups is to foster a dialogue among the local communities that will provide useful information that can be channelled back into management decisions. Project staff must learn to monitor these meetings so as to glean the maximum relevant information without wasting large amounts of time on issues which are not relevant to the project and the conservation objectives. It is critical that written minutes be prepared for each meeting, preferably by someone other than the moderator, and distributed to relevant individuals.

Once the conservation project is operational, it may be useful for some type of "advisory committee" to be chosen by local communities, with the stipulation that the widest possible geographical/demographic range be represented. These committees (the number will depend upon the size of the protected area, logistical difficulties, the socio-political structure of local communities, and so forth) should be encouraged to meet on their own, although project staff can occasionally monitor the dynamics of these reunions. It would be important to note the types of issues discussed, the range of participation, the manner of decision-making, etc., to be sure that the views of the committee are indeed representative of the community-at-large, rather than a few select individuals with their own interests. Equally important is to monitor how these meetings translate into concrete action on the part of the community. Local participation on the part of resident communities must go beyond employing the occasional individual or attending various meetings; there must be some responses to community needs, both on the part of the project

Informal or semi-structured interviews

Depending upon how the semi-structured interview is organised, it can yield both qualitative and quantitative data. These informal interviews should be designed to stimulate relaxed, two way dialogue between protected area staff and local communities. They are typically conducted with particularly knowledgeable individuals (renowned hunters, or women's weaving groups, for instance), and are guided by a prepared list of questions, or topics, that are very open-ended (see below for differences between open- and close-ended questions). The direction the interview will take depends in large measure on the skill of the interviewer, who must put the respondent at ease and constantly reflect on the appropriate follow-up questions. The following steps will allow for successful informal interviews:

- Make sure that the interview team is trained by a well-qualified expert. Informal interview skills are more difficult than you might imagine, as the interviewer must be able to think clearly of appropriate questions while at the same time registering informant responses. They should be considerate of local customs, and at the very least be familiar with greetings in local languages. The interviewer must learn to ask non-leading questions (see below), be non-judgmental and avoid criticising the informant(s). For example, in southeastern Cameroon, one informal interview was abruptly terminated by the informant when the interviewer began calling local villagers "lazy." Compassion, patience, and forward-thinking are not skills which can easily be taught, and yet they are critical for the success of the interview. Select your team carefully, and ensure proper training!
- Prepare a list of general topics which will be discussed, and a general way the interviewer can score responses. Pretest these questions with people who will not be interviewed, in order to insure comprehension (see pretest, below).
- Identify the key informant(s). If you have spent sufficient time in the village, you will have some idea of who is knowledgeable about different topics (older people for instance, will have a much better historical perspective on resource exploitation than younger villagers. They also tend to tell the best stories, and are often the source of a wealth of information).
- Set up a time and place for the interview which is convenient for the informant(s). This may mean going out to their fields, or arriving in the village very early in the morning. The respondent(s) should be comfortable in order to get the best results, so do not insist on an inappropriate time (meals, for example) or location simply because it is more convenient for you.
- If you will use a team to conduct the interview, make sure that each person is responsible for certain key topics, in order to avoid the confusion which will result if too may people are asking questions at the same time. Again, successful interviews depend on a smooth-flowing "conversation," which can be interrupted if the informant(s) feels overwhelmed. Try not to use more than two interviewers who are well-trained, and make sure that everyone is properly introduced at the beginning. Explain the purpose of the interview, and wherever possible, make sure the informant(s) understands the level of confidentiality involved. For example, if you plan to quote exactly to government sources, and attribute quotes to specific informants, they should be aware of this prior to starting.
- Follow the general guidelines prepared in advance, but the interviewer must feel free to pursue interesting lines of questioning without straying too far from the topic. Most informal interview questions should result from informant responses. These related questions should probe deeper, and should include where appropriate "Who? What? Where? When? Why? How?" inquiries. The informant(s) should be allowed to elaborate as they see fit - the purpose of these interviews is to learn from local "experts." It is critical that the interviewer remain neutral, and not judge or be critical of the informant(s). The interviewer should take minimal notes, trying to stick to the prepared matrix. Further observations can be added immediately following the interview. Alternatively, a tape recorder can be used, although they tend to make people nervous. Do not record people's responses without permission.

- Review results of interviews each day (or discuss them among team members, if several people are involved), and adjust the interview guidelines as necessary as you proceed.
- Share your impressions with the informants. This not only enhances a two-way dialogue, but allows you to cross-check interview results.

Questionnaires

Questionnaires are a more quantitative method of collecting data on a wide range of topics (understanding of conservation objectives, attitudes towards conservation, attitudes towards project management, etc.), although there are a number of potential drawbacks involved. First of all, in order for the questionnaires to be effective, a considerable amount of time must be expended on selecting questions, translating them into local languages (almost essential), recruiting and training research assistants, testing and refining questions, and executing the final version. Data collection, analysis and interpretation can be very costly, especially if large numbers of questionnaires will be administered. Respondents must be comfortable (using local assistants will help considerably), and they must be assured of the confidentiality of their answers, particularly if controversial topics are included.

Questionnaires should be conducted only with the interviewee, one-on-one, in order to avoid influencing the answers of other potential respondents. A small gift (tobacco, cola nuts) can be given to encourage participation, although cash payments should be avoided. The following points are considered crucial for the preparation of meaningful questionnaires:

- Hold village meetings, informal interviews and focus group discussions beforehand. These methodologies are most appropriate to open-ended questions, are more relaxing for informants, and allow you to better understand relevant issues. As a result, the formal surveys will be more clearly formulated. Finally, experience has shown that administering questionnaires can be intimidating, particularly if no previous relationship of trust has been built with local communities. In this case, you can be almost certain that the responses you'll be given will be perfunctory, and often geared towards what the informant thinks you want to know. In many instances, it is probably better to forego questionnaires altogether and concentrate on the informal, confidence-building methodologies outlined above.
- Prepare well defined questions. These should be close-ended (see Box 15e), resulting from analysis of the information gathered during more informal meetings with people, and relatively easy to code for future data analysis. Take time to think about the way answers will be coded when contemplating how the questions will be worded. Always start with less controversial topics (age, name of village, etc.); potentially contentious issues (illegal subsistence activities) should be left for the end. Short questionnaires invariably work best. Experience has shown that long questionnaires are often counterproductive, as the subjects tend to tire after 30-45 minutes. It is best to prepare two short, well-defined questionnaires which can be administered at different times rather than one long one.
- Translate and pretest questions. A questionnaire can only be effective if the questions are well understood by both the research assistants and the respondents. Invariably, this means that the questions must be translated into local languages; they must be written in the local languages, as too much bias is likely to be introduced if each research assistant simply verbally translates the questions. Once the questions have been translated, they must be pre-tested on a small sample of people to insure complete comprehension of all questions (and allow the research team to think more critically about how answers will be coded). Where difficulties arise, rewording of the questions may be necessary.

- Identify, recruit and train research assistants. This is arguably the most critical element in the successful execution of formal interviews. The most well-designed questionnaire will be completely invalidated in the hands of a poorly trained or boorish assistant. Educated individuals who speak, read and write both local and national languages are essential (data are often analysed at another time and location, so answers must be written in a widely understood language such as French or English). At the same time, assistants must be patient, fair and non-judgmental, which are difficult "skills" to train. They must understand that their role is to record answers, and that although they will have to sometimes prompt the respondent, they must in no way influence the answers (see examples of leading questions, Box 15e). The assistant must become thoroughly familiar with the questionnaire; if they do not understand each question themselves, they may render certain questionnaires useless. Experience has shown that with questionnaires which cover a number of topics and include more than 40 questions, a minimum of two weeks of intensive training is essential (this is yet another reason for administering, for example, two separate, shorter questionnaires).
- As part of the training, interviewers should practice actually administering the questionnaire. In some cases, the trainer should act as the respondent, in order to test the comprehension of all questions (i.e. giving purposely misleading answers is a quick measure of how well the assistant understands). Mock interviews with local residents should also be used in order to gauge the behaviour of the assistant.
- Select the sample. Depending upon the focus of your research, you may have to choose a random, or stratified sample, or one which is both random and stratified. For more details on choosing sample size and ensuring accuracy, see Sampling (above). Contact the potential respondents and secure their permission prior to arriving to do the survey.
- Execute the survey.

Box 15e EXAMPLES OF LEADING, OPEN- AND CLOSE-ENDED QUESTIONS

Leading Questions (To be avoided always):

- The forest is important to you for bushmeat and fish, right?
- Shouldn't you put a lid over your water storage pot?
- You don't make very much money from selling bush mangoes, do you?
- Wouldn't you prefer to cultivate an improved variety of maize?

Open-ended Questions (Best for informal surveys and interviews):

- Why is the forest important to you?
- How has hunting changed for your family since the time of your grandparents?
- What differences have you noticed in the availability of non-timber forest products since the arrival of the logging company?
- What would you change about your agricultural practices if you could?

Close-ended Questions (Appropriate for formal interviews/questionnaires):

- How many years do you leave your field in fallow before recultivation?
- Between the timber company and the safari hunters, who causes the most damage to your forest?
- Do you prefer the taste of bushmeat or domesticated animals?
- What is the most important non-timber forest product for personal consumption?
- What is the most important non-timber forest product for the market?

Further reading

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A forest tree in the Lopé Reserve, Gabon, growing in the remains of an iron furnace. The furnace was dated to 700 years before present. Note the remains of clay pipes through which air was introduced into the furnace using a bellows. Humans play a key ecological role in the African rain forest today but our role in the past should never be under-estimated(see Box 9f).

CHAPTER 16
STATISTICAL TECHNIQUES

**Neil Chalmers, Phil Parker & Kevin McConway*

Chapter 4 described the principals of data analysis and described the different types of data you might be dealing with and their properties. Once you have collected information and undertaken preliminary analyses to synthesise and display your data you will often want to quantify more exactly whether your data offer a clear answer to the questions you set out to investigate. For example, if you undertake a chimpanzee survey in two strata, one logged and the other unlogged, you may expect to find more nests in the unlogged area. If the nest density is 9 nests / km^2 in the unlogged forest and 1 nest / km^2 in the logged area the answer is fairly obvious - there are fewer chimpanzees in the logged forest. However, if it is 7 nests / km^2 in the unlogged forest and 6 nests / km^2 in the logged area it is difficult to know if the difference is 'real' or simply due to variability in your samples. We use statistical techniques to decide how confident we can be about conclusions drawn from our data. In other words, statistical techniques enable us to assign a probability to the outcome of our study (see Page 41).

This chapter gives a general introduction to statistical inference (Box 16a) and then details ten statistical techniques. Before you decide which one is best suited to your particular analysis, you have to be clear about the following:

- Whether you wish to make decisions about differences between populations from which your samples come, or whether you wish to make decisions about associations between features within the population from which your sample comes.
- Whether your measurements are at the interval, ordinal of categorical level (see Page 31).
- Whether your measurements are matched or unmatched (see Box 16b).

Once you are certain about these things, consult the decision chart (Figure 16.1) which tells you which technique you might use and where to find it described. Then turn to the relevant page, to discover whether the technique is, in fact, appropriate.

The layout for each technique is the same. First, in a box, there is a summary of the situations in which you may use the test. Not all of this may be clear until you have read all of the information on the technique, but once you have done this you should read this box through again *before* carrying out the test on your own data, to check that the conditions required by the test are met.

Next the rationale of the test is discussed, in terms that are intended to appeal to your intuition, rather than to any mathematical principles.

After this there is usually a brief summary of the procedure in symbols - useful for quick reference once you become thoroughly familiar with the technique. This is followed by a worked example, which you should find particularly useful when going through the technique for the first time. Sometimes the procedure involves alternative routes, depending, for example, on whether you are working with large or small samples.

Finally there is a list of 'Do's and Don't's, which you should work through carefully. This list should help you avoid making the commonest errors found in research projects.

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Box 16a STATISTICAL INFERENCE

A major feature of biological and socio economic research is that one uses data from samples of finite, and often rather small size to make inferences about the populations from which they come. There are two kinds of inference that you will often want to make. These are: a) whether there is a difference in a particular feature between two populations, and b) how big such a difference might be. The methods used to answer these questions are called hypothesis-testing and estimation techniques, respectively.

Hypothesis testing

All **hypothesis testing** proceeds through a standard sequence of reasoning. Take, for example, the following hypothesis:

'The abundance of *Marantochloa purpurea* at sites X and Y is different because of differences in soil moisture at the two sites'.

Our chain of reasoning is as follows:

a) Assume, for the moment, that the averages of the soil moisture values from the two sites are the same.
b) Take several measurements of soil moisture at each site and see whether the values from the two sites are, on the whole, close to each other or far apart.
c) Assess the probability of getting moisture values at least as far apart as those you have obtained, if the two sites do not actually differ in overall moisture content.
d) If this probability is too low, reject the assumption made in (a) and accept that mean soil moisture values at the two sites are different.

This is a specific example, of course, but the same four-step chain of reasoning applies generally across the whole field of hypothesis testing. Put in more general terms, and refining some ideas slightly, the steps in the argument are as follows:

1) *Formulate a hypothesis that no difference exists in the specified feature, in the populations from which the two samples are taken.*

COMMENTS

i) Such a hypothesis of 'no difference' is called a **null hypothesis**. It is usually denoted by the symbol H_0. Note particularly that the null hypothesis refers to the *populations* from which the samples are taken, and not to the samples themselves.

ii) Note also that the null hypothesis refers to a *specified feature* of the populations. Most often the null hypothesis refers to **population means** of the measurements in question: that is, the null hypothesis states that the samples come from populations that do not differ in their means (for whatever feature is being measures). Another feature commonly specified in a null hypothesis is the **population median**: that is, the null hypothesis states that the samples come from populations that do not differ in their medians. A third feature that you may well wish to specify in a null hypothesis is the **population variance**: that is, the null hypothesis states that the samples come from populations that do not differ in their variances. Which of these features you should specify varies from one situation to another. In the descriptions of specific tests below we give advice on how to decide on the most appropriate feature for a given situation.

iii) In competition with the null hypothesis, as it were, you have to formulate an **alternative hypothesis**. This must be worded in such a way that if the null hypothesis is true then the alternative hypothesis is false, and vice versa. The simplest form of alternative hypothesis is the direct converse to the null hypothesis. Thus, if the null hypothesis states that no difference exists between population means, then the alternative hypothesis states that a difference *does* exist. The symbol used to denote the alternative hypothesis is H_1.

iv) The null and alternative hypotheses need not be limited just to two populations. It would be perfectly possible, for example, to compare the soil moisture values from, say, five sites, and formulate a null hypothesis that the mean soil moisture did not differ between sites.

2) *Look at the measurements obtained for the samples from the populations.*

It makes intuitive sense that if the measurements within samples are tightly clustered, and if the measurements for different samples are very different in magnitude, then we may be confident that the samples came from different populations. However, the greater the scatter of measurements *within* the samples and the smaller the difference in measurements *between* samples, the less confident we can be that such a difference exists. This statement can be re-phrased using terminology of Box 4b, namely: a null hypothesis of no difference in population means or medians is the more likely to be rejected, the bigger the difference in sample means or medians and the smaller the scatter of measurements within each sample.

3) *Work out the probability under the null hypothesis of obtaining measurements at least as far apart as the ones you have collected.*

The techniques for working out a probability under a null hypothesis can be complicated. Fortunately, the hard work has already been done by statisticians, and Tables exist from which you can read off the required probability, provided you understand certain principles. These are perhaps more readily explained with reference to an example, so we shall consider a hypothetical set of measurements on the carapace length of hatchling leatherback turtles (*Dermochelys coriacea*).

Imagine that it were possible to measure the carapace length of every single hatchling turtle along the coast of Gabon in any one year. It would then be possible to find out how many baby turtles' carapaces measured between, say, 40 and 41 mm in length, how many between 41 and 42 mm, and so on up to the very top of the range of lengths. Rather than expressing these actual numbers, you could equally well express the percentages of all the turtles counted. This has been done in Figure B16.1, which shows the percentage of all hatchling turtle carapaces between 40 and 41 mm long, and so on. The carapace lengths summarised in this Figure constitute, in the statistical sense of the word, a population. In reality it would be impossible to measure the entire population of any marine turtle; but one tiny fraction of the population of measurements might give the data listed in Table B16.1.

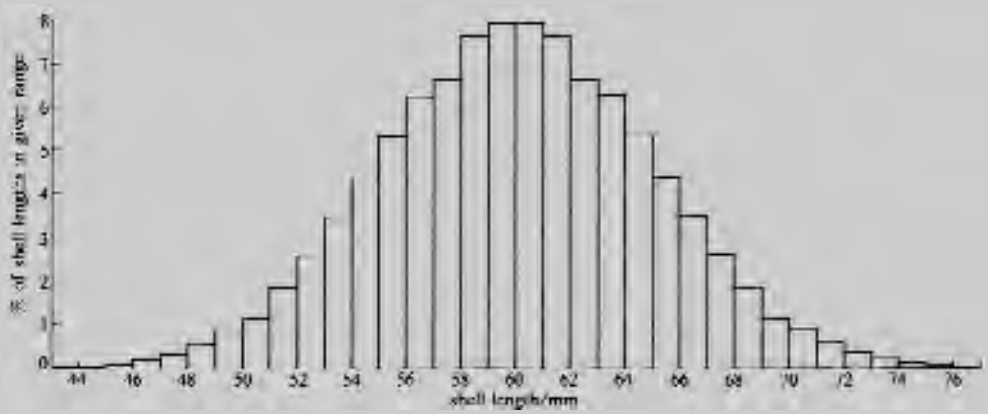


Figure B16.1 An example of the normal distribution: a hypothetical length / frequency plot for carapace length in hatchling leatherback turtles.

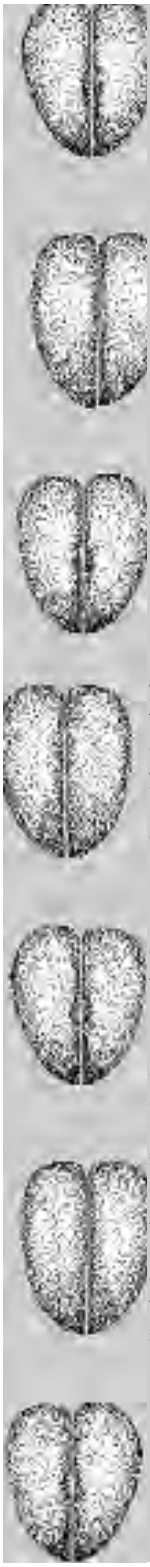


Table B16.1 Carapace lengths for 100 hatchling leatherback turtles. For this sample, mean carapace length ($\bar{x}$) = 60.45 mm, and sample standard deviation (s) = 5.04 mm.

sample measurements / mm									
60.6	66.8	59.7	71.7	60.6	71.9	57.8	61.9	63.6	69.9
67.0	57.5	57.0	58.7	61.8	55.6	60.4	62.5	53.9	56.2
60.3	55.8	61.8	58.2	55.3	54.2	60.1	59.8	50.5	55.3
59.9	67.5	57.7	56.0	63.6	55.5	58.6	59.7	59.0	72.1
63.9	61.0	65.6	60.8	50.8	55.5	57.7	61.5	64.1	71.1
59.4	53.0	61.9	54.2	62.4	55.5	54.4	63.1	57.8	67.2
62.1	62.5	54.4	57.5	56.6	55.7	61.4	67.3	56.3	64.6
58.5	62.1	71.1	55.8	71.0	62.4	61.9	68.6	63.4	55.8
56.6	53.2	60.1	62.8	63.1	65.3	64.8	60.6	67.7	54.7
52.6	58.6	59.6	57.4	64.3	56.7	54.0	56.5	68.1	62.5

Notice from Figure B16.1 that most of the carapaces are about 60 mm long, that the further from the mean value of 60 mm one goes the fewer individuals there are, and that the measurements fall symmetrically around the mean value. These characteristics are typical of certain biological measurements, and the shape of the distribution of measurements in Figure B16.1 is known as the **normal distribution**.

4) *Decide whether to reject the null hypothesis.*

If somebody brought you a hatchling leatherback turtle of unknown origin measuring, say, 62.5 mm and asked you whether you thought it could possibly have come from Gabon on the basis of carapace length, you would have little hesitation in saying that it could. This turtle is only 2.05 mm different in length from the mean of 60.45 mm, and Figure B16.1 shows that turtles differing from the population mean by this amount (or more) are relatively common around Gabon. If, however, someone brought you instead a hatchling with a carapace measuring 74.2 mm, you might be doubtful: this turtle is nearly 14 mm from the mean. Figure B16.1 shows that as few as 0.2% of turtles are more than 14mm from the mean. If, finally, you were shown a turtle whose carapace was 92.8 mm long, you would say almost certainly it did not come from Gabon, because virtually no Gabonese turtle hatchlings are as far as this from the mean.

Your decision in each of these three instances is determined by your knowledge of the probability of obtaining measurements more than various distances from the mean. You say, in effect, 'What is the probability of obtaining a turtle hatchling from the Gabonese leatherback population *at least* as far from the mean as this?' If the probability is sufficiently small, you say that the turtle is very unlikely to have come from the Gabonese population.

You may wonder how one can work out the probabilities concerned. Where, in other words, do the percentages of Figure B16.1 come from? The answer lies in the normal distribution. The normal distribution has certain key properties. In particular, approximately 33% of the measurements in the population lie more than one standard deviation from the mean, and approximately 5% more than two standard deviations from the mean. In fact, it is possible to work out what percentage of measurements lie further than any particular number of standard deviations from the mean, whether it be 1, 2, 1.5, 0.36 standard deviations or whatever. It is because we know the properties of the normal distribution exactly that we are able to calculate the probability of obtaining a given measurement that differs by more than x standard deviations from the mean, where x is any number we choose.

A similar kind of reasoning to that used above would apply if somebody brought you, not one, but several hatchling turtles and asked you if they could have come from the Gabonese population. This is equivalent to setting up a null hypothesis that there is no difference between the mean carapace length of the Gabonese population and that of the population from which the sample came.

To illustrate this reasoning, imagine that the sample size were 5, and were to come from the collection of measurements in Table B16.2. Select five measurements from this table at random (see Box 3a). calculate the mean and the median for the five measurements you have selected. Repeat the whole process for two other samples, each containing five measurements. You will thus end up with three samples of five measurements each, together with three sample means and three sample medians.

sample means / mm				
59.85	57.36	58.76	61.22	61.21
62.40	59.67	57.23	57.65	62.00
59.08	60.27	58.73	59.27	56.88
60.32	60.23	62.13	61.50	59.96
58.05	56.99	56.84	61.03	60.17
60.26	60.84	63.96	60.69	59.76
58.40	60.83	59.38	59.58	58.96
59.16	62.27	57.32	58.59	59.96
62.40	58.28	57.51	57.51	59.27
60.35	65.13	58.45	58.81	60.51

Figure B16.2 A histogram of the 50 sample means from Table B16.2

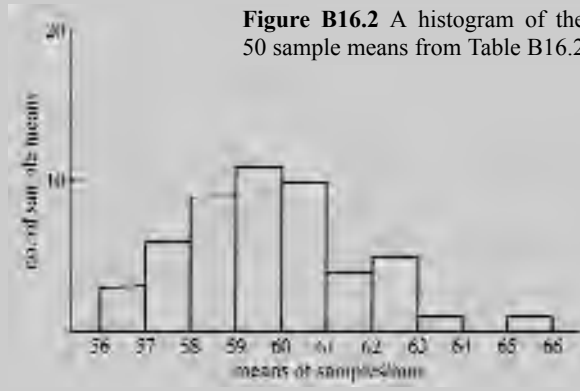


Table B16.2

The ~~means of 50 carapace-length samples,~~ each containing five measurements, taken from the population of hatchling leatherback turtles in Gabon, part of which is shown in Table B16.1. The mean of the 50 sample means ($\bar{x}$) = 59.740 mm, and the standard deviation of the 50 sample means (s) = 1.829 mm.

How do your sample means compare with each other and with the population mean?

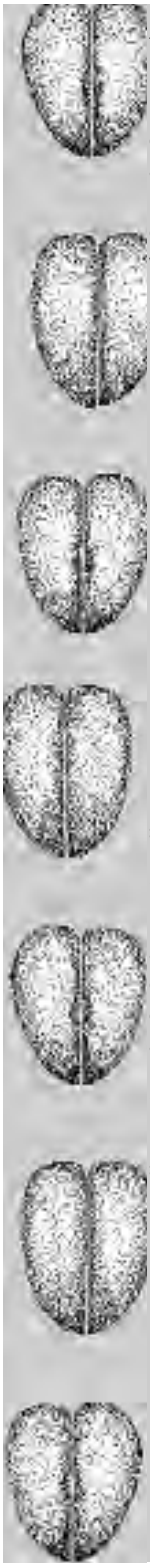
Your sample means are very likely to be close together but slightly different from one another: it is extremely unlikely that they will be exactly the same. The sample means are also likely to be close to the population mean of 60.45 mm.

The sample medians are also likely to be similar to each other but not identical. This exercise demonstrates an important feature of samples: if you take many samples from a population, and calculate the sample means or medians, they will vary from one sample to another. Just as it is possible to draw a frequency distribution of the individual measurements that make up the sample, so it is possible to draw a frequency distribution of the sample means or medians. For example, Table B16.2 shows the respective means of 50 samples (each containing five measurements) taken from the population of gabonese leatherback turtle hatchlings. Figure B16.2 is a histogram of these means. Notice three things about the histogram:

i) The means are themselves clustered around a mean value. In this instance, the mean value is 59.74 mm, which, by no coincidence, is very close to the population mean of 60.45 mm.

ii) The means appear to be normally distributed, and we ask you to take it on trust that they are, in fact, normally distributed. The significance of this is that we can therefore work out the probability that a given sample mean will be more than a certain distance from the mean of the sample means, just as we did earlier for the original measurements.

iii) The sample means are more tightly clustered around their mean than are the original measurements around *their* mean. (Compare Figure B16.2, which shows that virtually all of the sample means lie between 55 and 65 mm, with Figure B16.1 which shows that many of the original measurements lie outside these values).



In fact, it is perfectly possible to calculate a standard deviation for the sample means, to discover just how scattered they are. This turns out to be 1.829 mm. The standard deviation of the original measurements, by contrast, is 5.04 mm.

The standard deviation of a sample statistic such as the mean is very important in statistical analyses. In order to distinguish it from a standard deviation of the original measurements, it is given a special name: the **standard error.** So, the standard deviation of a collection of sample means is called the **standard error of the mean** (often abbreviated to **s.e.m.**), the standard deviation of a collection of sample medians is called the **standard error of the median**, and so on.

Suppose that the sample of five leatherback turtle carapace measurements that the person brought you had a mean length of 58.33 mm. You know the standard error is 1.829 mm, and hence this sample is less than 1 standard error from the mean value of the sample means of 59.740 mm. Because the sample means are normally distributed, you can conclude that about 67% of them lie within one standard error of the mean. Hence, it is reasonable to accept the null hypothesis that this particular sample does indeed come from the Gabonese population of leatherbacks. If, by contrast, the sample's mean carapace length had been 66.58 mm, you would have probably come to a different conclusion.

Why would the conclusion be different?

66.58 mm is more than 6.00 mm from the population mean. Figure B16.2 shows that none of the sample mean lengths are more than 6.00 mm from the mean of the sample means of 59.740: the probability of obtaining a sample with a mean length of more than 6.00 mm from the mean value for the gabonese population is very small. Therefore, you might think it reasonable to reject the null hypothesis that the sample comes from a population whose mean does not differ from that of the Gabonese population, and to accept the alternative hypothesis that it comes from a different population.

One further step in the argument is needed to make it applicable to a great deal of biological research. Suppose somebody brought you two samples of leatherback hatchlings rather than one, and asked you whether they could have come from the same population. This is equivalent to setting up a null hypothesis that the samples came from populations that do not differ in their mean carapace lengths. On the whole, you would expect that samples taken at random from the same population would tend to have mean carapace lengths close to each other rather than far apart. If, for example, you were to look at any two of the 50 sample means in Table B16.2, and were to subtract one from the other, you would expect the answer to be close to zero. Table B16.3 shows the sort of results you would get if you were to take each of the 50 mean values in Table B16.2 in turn and subtract it from another mean in the table chosen at random. Figure B16.3 presents such results as a histogram. Notice from Figure B16.3 that most of the differences are close to zero; that larger differences are progressively rarer; and that positive differences are as common as negative differences (because it is quite arbitrary which carapace length you subtract from which). It turns out that these values are normally distributed, with a mean value of zero. Once again, it is possible to calculate the standard deviation of the differences in the means, in order to give an idea of how widely scattered they are about the mean; and once again, this kind of standard deviation is called a standard error - in this instance the **standard error of the difference in means**.

Just as with the original measurements of carapace lengths and with the sample means, the knowledge that the differences in sample means are normally distributed makes it possible for us to calculate the probability of getting differences which are more than so many standard errors from the mean value. A large difference between two sample means will be unlikely if the samples come from two populations with the same mean, whereas a small difference will be much more likely.

Table B16.3 Differences between pairs of sample means taken from Table B16.2.

differences between pairs of means / mm				
-2.55	2.31	1.53	1.57	-0.79
3.32	-0.60	-1.50	0.38	5.12
-0.24	0.04	-3.40	-2.23	-3.08
2.27	3.24	5.19	0.47	-0.21
-2.21	-3.85	-7.02	0.34	0.41
1.86	-0.09	4.58	1.11	0.80
-0.76	-1.34	2.06	0.99	-1.00
-3.24	3.99	-0.19	1.08	0.69
2.05	-6.85	-0.94	-1.30	-1.24
2.99	6.37	-2.77	-2.40	0.66

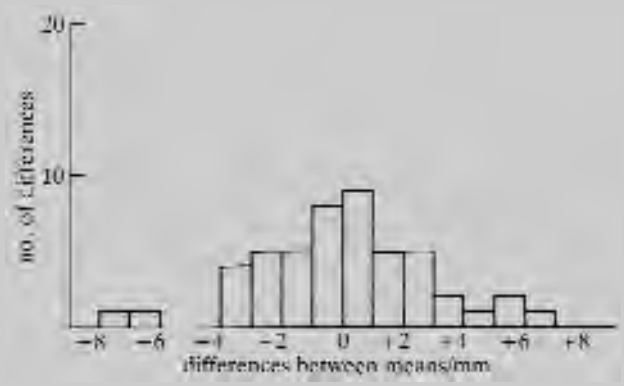


Figure B16.3 A histogram of the differences between pairs of sample means

It turns out that the standard error of the differences between sample means shown in Figure B16.3 is 2.772 mm. So, if samples are taken repeatedly from two populations of turtles with the same mean carapace length then about 67% of the differences in sample means will be less than 2.772 mm, and about 95% of the differences will be less than twice 2.772 = 5.544 mm. Hence, if two samples of leatherback carapaces were to differ in length by, say, 1,3 mm, you would accept the null hypothesis that they came from populations with the same mean shell length. On the other hand, if they were to differ in mean length by, say, 8.2 mm, you would have very strong grounds for rejecting the null hypothesis and for accepting the alternative hypothesis that they came from populations with different mean carapace lengths.

In the preceding paragraphs we have considered three processes:

i) deciding whether a single turtle could have come from a known population;
ii) deciding whether a sample of turtles could have come from a known population;
iii) deciding whether two samples could have come from the same population.

It is helpful to introduce some additional ideas that apply to all three situations. In each, the procedure is as follows:

1 Set up a null hypothesis and an alternative hypothesis.
2 Look at the measurements obtained.
3 Find out the probability of getting results at least as extreme as these if the null hypothesis were true
4 Reject the null hypothesis if this probability is too small, and accept the alternative hypothesis.

How small the probability has to be for you to reject the null hypothesis is a matter of personal judgement. It is usual in biology, however, to reject a null hypothesis only if the probability of obtaining your particular sets of measurements is less than 1 in 20. This probability is often written as a decimal, namely 0.05, or as a percentage, namely 5%. If you choose as your criterion for rejecting the null hypothesis a probability of 5%, you are said to be operating at a 5% **significance level**. Sometimes, you may find that the probability of obtaining the measurements that you have collected if the null hypothesis is true is a lot smaller than 1 in 20. It may be less than 1 in 100 or even less than 1 in 1000. If so, you can then reject the null hypothesis at the 1% or the 0.1% levels respectively.

Estimation

In the example described above, a significance test would enable you to decide whether two samples of carapace measures could have come from the same population. You could, however, have used the arguments in virtually identical fashion to answer a different question: not 'Is there a difference in the population means?' but 'Within what range does the difference in population means lie?' This amounts to **estimating** that difference. If you look at Figure B16.3, you can see that virtually all the differences in sample means lie between -7 mm and +7 mm. In fact, because the differences in means are normally distributed, with a standard error of 2.772 mm, we know that 95% of them lie within a distance of two standard errors of the mean of this normal distribution. We assumed that the difference between the means of the *populations* from which the samples come was zero, so the mean of the distribution of differences in *sample means* is zero, and therefore 95% of the differences between sample means lie between -5.544 mm and +5.544 mm (5.544 = 2 x 2.772). So far so good, but in practice you have only one pair of sample means, not the 50 in Table B16.2, and you cannot assume that the population means are equal - that is one of the things you probably want to investigate. If your pair of *sample means* differs by, say, 1.2 mm, and if the standard error of the difference in sample means is still 2.772 mm, then it is very likely that the difference between the *population means* is more than two standard errors from the difference you found, 1.2 mm. That is, a plausible range of values for the difference in population means runs from 1.2 - (2 x 2.772) mm and 1.2 + (2 x 2.772) mm, *i.e.* from -4.334 mm to + 6.744 mm. We can be 95% confident of this, since 95% of the measurements in a normal distribution lie within two standard deviations of their mean. We can therefore refer to -4.334 mm and + 6.744 mm as 95% **confidence limits** for the difference in population means. The range between upper and lower confidence limits is known as a 95% **confidence interval**.

Almost all of the statistical tests described below can be used both to test a null hypothesis and to provide confidence limits. Both features are very useful, and which of them is more applicable for you will depend upon the kind of question you are trying to answer.

Test statistics and critical values

The method for carrying out significance tests described above contains some important features that need to be explained further. Take a situation where the null hypothesis under test is that the means of the populations from which two samples come are the same. In carrying out the test, you are in effect asking 'How likely is it that I would have obtained a difference in sample means at least as big as this if the null hypothesis were true?' Putting this in more general terms, you use the measurements from the two samples to calculate a *single* number; in this instance the difference in sample means. This is what happens in all significance tests described in this manual, although the number you calculate will not always be the difference in sample means. This single number that you calculate in a significance test is called the **test statistic**.

An important feature of a test statistic is that it is always possible to calculate the probability of obtaining a particular value (of at least as large or as small as a given value) if the null hypothesis is true. Consider the carapace example above. Look at Figure B16.4 which shows the relative frequency with which you would expect to find differences between pairs of sample means if the null hypothesis (that the two samples of hatchlings came from the same population) were true. The Figure is very like Figure B16.3, but the histogram has been converted into a smooth curve, with a normal distribution, a mean of zero and a standard error of 2.772 mm, the same as for Figure B16.3. If we were to take the difference in sample means as a test statistic, then in formal terms we know its **probability distribution**. If you had decided upon a 5% significance level for your test, you would have been able to work out from the probability distribution how large a difference in sample means would be needed for the null hypothesis to be rejected. Earlier in this box we saw that this value is about 5.544 mm. Values larger than this (see Figure B16.4) allow us to reject the null hypothesis.

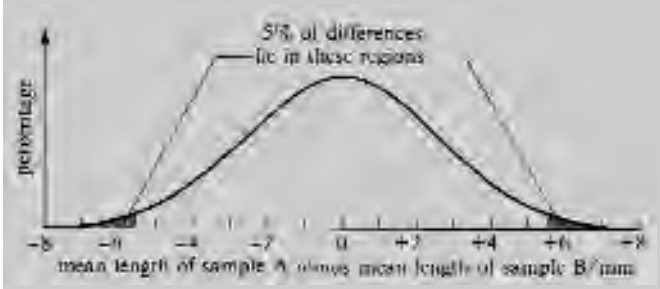


Figure B16.4 The relative frequencies at which differences between pairs of sample means would be expected, under the null hypothesis that the means of the population from which the samples were taken do not differ.

What we have done, in general terms, is to choose a **critical value** for the test statistic which, if exceeded, will cause you to reject your null hypothesis. So, in Figure B16.4, the critical value is 5.544 mm and you would reject the null hypothesis if the difference in sample means, the test statistic, exceeds this.

The following features are common to all significance tests:

1. You calculate a test statistic from your measurements.
2. You know the probability distribution of this statistic, given that the null hypothesis is true.
3. You can discover from the probability distribution the critical value which your test statistic must exceed for the null hypothesis to be rejected.

Statisticians have taken a lot of the hard work out of significance testing, in particular by preparing Tables of critical values for given test statistics. Several such Tables are provided at the end of this chapter. This means that you can calculate a test statistic from your sample measurements and then simply consult the appropriate Table for the critical value under your null hypothesis for the significance level that you require. Tables frequently come in two versions, one for '**one-tailed**' or '**one-sided**' tests, and the other for '**two-tailed**' or '**two-sided**' tests. We recommend that you use the 'two-tailed' version. Only 'two-tailed' Tables are included here.

Bay duiker,
Cephalophus dorsalis

Box 16b MATCHED AND UNMATCHED MEASUREMENTS

Many research projects involve an analysis of two samples: that is, two sets of measurements. You have to decide whether these measurements can be **matched** together in pairs or not, because some techniques described in this chapter can only be applied to matched pairs of measurements, whereas others are to be used on unmatched sets. By 'matched pairs', we mean that one measurement from one of the samples can be paired with one and only one measurement from the other sample.

For example, you may suspect that more leatherback turtles arrive to nest on any given night whilst the tide is rising, but that other factors, such as the phases of the moon also have an impact on the total number of turtles observed each night. If you walked the beach once as the tide was turning and a second time in the morning after all turtles had finished nesting (they only nest at night) and counted all fresh turtle tracks, you would have a measure of how many turtles had nested during each phase of the tide each night. Obviously the number of hours that the tide was rising and falling would differ, so you would have to calculate a number of turtles per hour of darkness for rising and falling tides each night. The total number of turtles arriving peaks when there is no moon and is greatly reduced around the full moon, and also varies through the 3-4 month breeding season. However, if your hypothesis is correct, regardless of the phase of the moon or the period in the breeding season (and hence the total number of turtles nesting each night), the average number of turtles visiting the beach whilst the tide is rising will be greater than when the tide is falling - for each night you have a matched pair of measurements, one for the rising tide and one for the falling tide.

As another example, you may suspect that the chimpanzee density inside a protected area is higher than that in an adjoining logging concession. You may decide to cut 20 randomly located 5-km line-transects in each area and estimate nest density in the protected and logged forests. In this case there will be 20 measures for each site, but there is no reason to pair any one of the measurements from the logged site with any one measurement from the protected area. In this case you will test your hypothesis on unmatched measurements.

In many research projects, you have little choice over whether the measurements are to be matched or not: the nature of the study itself dictates whether or not measurements will be paired. Sometimes, however, the choice *is* under your control. For example, you may want to assess the relative importance of two beaches for nesting turtles. You might choose to walk each beach 20 times during the 4-month breeding season and count the number of fresh tracks and nests. This would give 20 unmatched measurements as in the chimpanzee example above. However, knowing that the phases of the moon have an important effect on the number of nesting turtles, you might choose to always collect the data on the same, or successive (if it is not possible to do the two the same day) days for the two beaches. This would give 20 matched measurements with the same lunar conditions. In this way you can be sure that if there is a difference in the data for the two beaches that it reflects a 'real' preference for one of the beaches, rather than an effect of different lunar conditions, or the period in the season observations were made, in each site.

Adult leatherback turtle,
Dermochelys coriacea

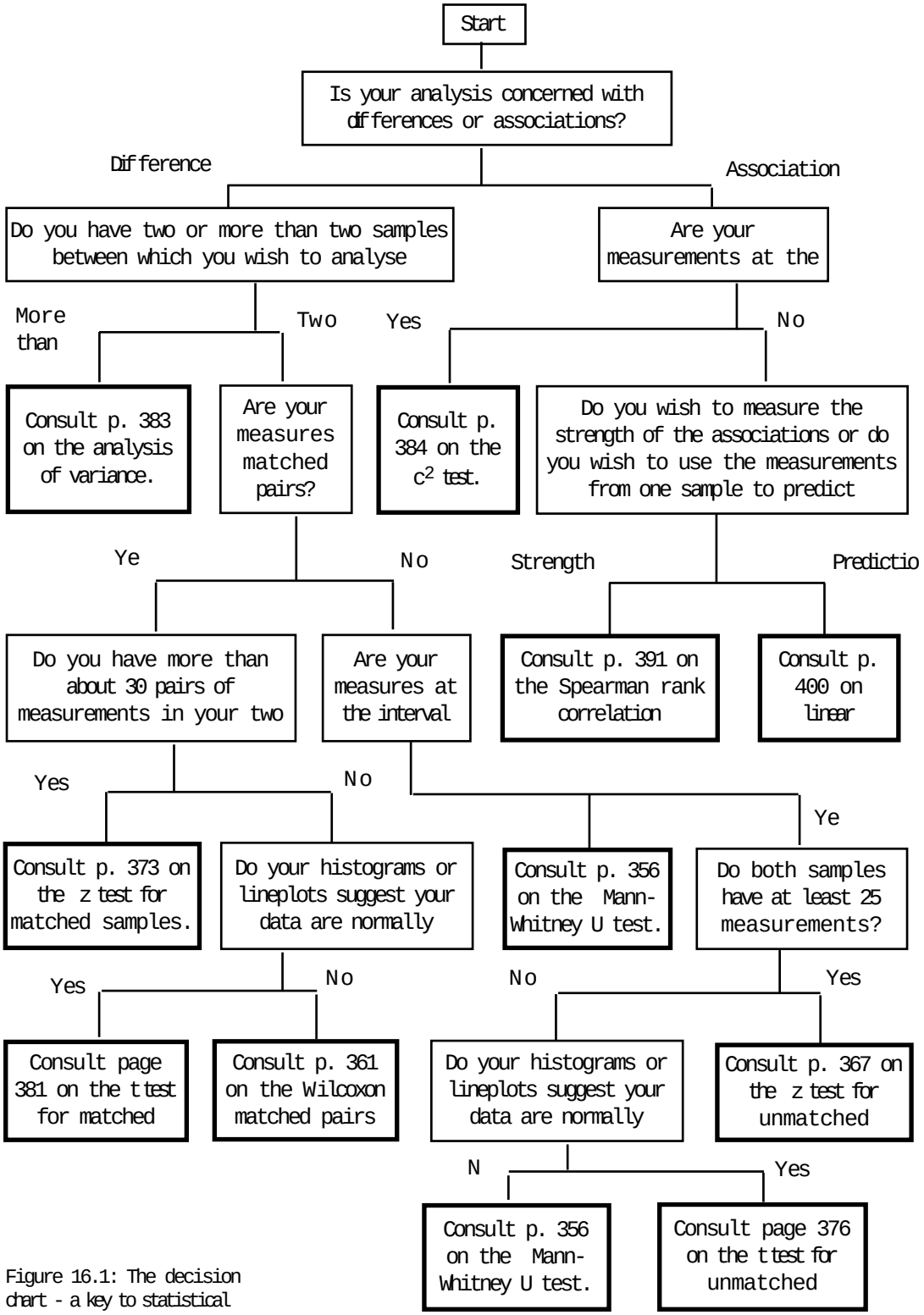


Figure 16.1: The decision chart - a key to statistical

The Mann–Whitney *U* test.

When to use:

- If you wish to test for differences in population **medians** (see Page 37).
- Provided you have two **unmatched** samples.
- Provided your data are either at the **ordinal** or **interval** level (see Page 31).
- Provided the populations from which the samples come have the **same shaped distribution** (they do *not* need to be normally distributed)
- Do not use if **one** of the samples has **only one measurement**, or if **both** samples have **under five measurements**.
- If your measurements are at the interval level, do not use when both sample sizes are **greater than 25**. Use the *z* test for unmatched samples.
- It is difficult to use the Mann-Whitney *U* test to calculate confidence intervals without the use of a computer.

Rationale of test

Suppose that you have two unmatched samples and that you wish to test the null hypothesis that they come from populations with the same medians. Sample A contains four measurements, which in increasing order of size are A_1, A_2, A_3 and A_4. The other sample, B, contains three which - again in increasing order of size - are B_1, B_2 and B_3. (It would be unwise in practice to use such small samples, but we choose small samples here to simplify the explanation.)

The exact values of these measurements do not matter, but you must be able to say which is the smallest, which the next smallest, and so on, ranking them all in order of increasing size.

If the null hypothesis (see Page 41) is true, then it is unlikely that all of the measurements in sample A will be smaller than all of those in B, or vice versa - see Figure 16.2a and b.

It is far more likely, if the null hypothesis is true, that the measurements from the two samples will overlap to some extent, as in Figure 16.2c.

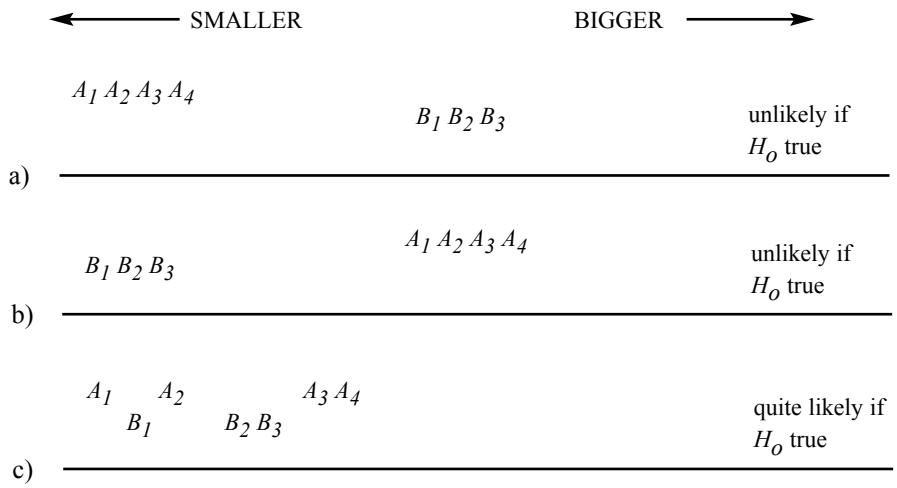


Figure 16.2 Likely and unlikely outcomes of an investigation, given the null hypothesis H_o that the two samples, A and B, come from populations with the same medians.

The Mann-Whitney U test provides a simple way of quantifying the overlap between measurements in two samples, and also tells you the probability of obtaining such an overlap if the null hypothesis is true. To measure the overlap, start with one of the samples, take each measurement from that sample in turn and count how many measurements in the other sample are smaller than it. Keep a tally as you go along. Then swap the two samples round, and go through the procedure again. For example, with the data in Figure 16.2a, you would proceed as follows. First count how many sample B measurements are smaller than each of the sample A measurements. A_1 does not have any measurements from sample B smaller than it, nor does A_2 or A_3 or A_4. So the total number of times sample B measurements are smaller than sample A measurements is zero. Now swap the two samples round and count how many times sample A measurements are smaller than sample B measurements. B_1 has four sample A measurements smaller than it, so does B_2 and so does B_3. So the total number of times sample B data were smaller than sample A data was 12.

The situation in Figure 16.2b is the mirror image of that in Figure 16.2a, and not surprisingly, if you were to go through the same procedure with these two samples, you would find that the number of times sample A data were smaller than sample B measurements was zero as well, and that the number of times sample B data were smaller than sample A data was 12.

Go through the same procedure for the data in Figure 16.2c.

Sample A:
A_1 has 0 sample B measurements smaller than it.
A_2 has 1
A_3 has 3
A_4 has 3

total 7

Sample B:
B_1 has 1 sample A measurement smaller than it.
B_2 has 2
B_3 has 2

total 5

Notice that in examples (a) and (b), where there is no overlap between the two samples, the lower of the two totals we calculated is) and the higher is 12. By contrast, in example (c) where there is considerable overlap, the lower total is 5 and the higher is 7. From this it seems that, if the null hypothesis is true, we would expect the two totals to be close together, whereas if it is false we would expect them to be far apart: a very high or a very low total might lead one to reject the null hypothesis. In fact, it is the smaller of the two that is used. The test statistic is usually denoted by the letter U. It is possible to work out the probability distribution of U when the null hypothesis is true, although we do not do this here. It turns out that the probability distribution is different for each different pair of sample sizes; for example, Figure 16.3 shows the U values obtained by taking two samples with the same median (i.e. one sample from each population). The calculated value of U will lie between 0 and 8 inclusive. If you take such samples repeatedly, the value of U will fluctuate from sample to sample. If the null hypothesis is true, there is a probability of about 0.2 (20%) of getting a value $U = 6$, a probability of just under 0.1 (10%) of getting a value $U = 3$, and a probability of 0.03 of getting a U value of 0. The probability of getting a U value of 1 is also 0.03, so the probability of getting a value of 1 or smaller is 0.03 + 0.03 = 0.06, which is greater than 0.05. Hence we can reject H_o only if U=0, assuming that the criterion for rejecting H_o is a probability of 5% or less. (Note that for Mann-Whitney U test we reject the null hypothesis if the test statistic is equal to or *smaller than* the critical value.)

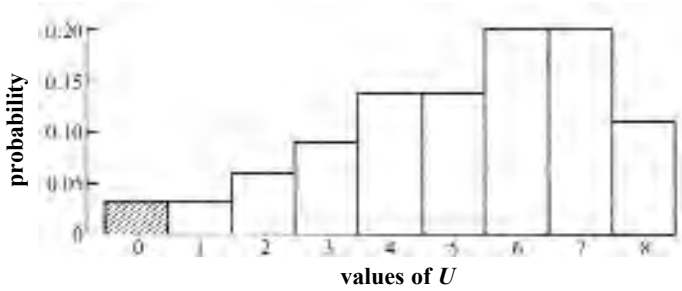


Figure 16.3 The probability distribution for the test statistic U for two samples, each with four measurements

Rather than drawing scores of probability distributions for different combinations of sample sizes, it is usual to present a Table (e.g., Table A16.1, located at the end of this chapter) of the critical values that U takes for a given significance level, for various combinations of sample size. The following example shows you how to use these Tables, and how to carry out the whole Mann-Whitney U test.

WORKED EXAMPLE

Procedure

1) Call the two samples A and B, respectively.
Rank the data in the samples in increasing order of size.

Calculation

sample A: 18 19 15 23 19
sample B: 19 21 16

← SMALLER LARGER →

sample A:	15	18	19	19	23
sample B:	16		19	21	

2) For each measurement in sample B, count how many in sample A are smaller. Write down this number. (Sometimes measurements in samples A and B will have the same value. Each time this happens, count a score of 1/2).

sample B measures	**comment**	**no. sample A measures < sample B**
16	one sample A measure - 15 - < sample B measure of 16	1
19	two sample A measures - 15 & 18 < sample B measure of 19. These contribute a score of 2; also two sample A measures of 19 have same value as sample B measure of 19 and score 1/2 giving a total of (2+1/2+1/2)	3
21	four sample A measures - 15, 18, 19 & 19 are smaller than sample B measure of 21	4
	total	8

3) When you have worked your way through all the measurements in the sample, add up the numbers you have written down to give a grand total. Call this U_A.

Therefore $U_A = 8$

4) Carry out steps 2 and 3 again, this time with the two samples swapped round.

sample A measures	comment	no. sample B measures < sample A
15	one sample B measure < 15	0
18	one measure - 16 - <18	
19	one measure - 16 - < 19 and one has the same value.	1 1/2
19	one measure - 16 - < 19 and one has the same value.	1 1/2
23	three measures - 16, 19 & 21 - are smaller than sample 23	4
	total	7

Therefore $U_B = 7$

Call the new grand total you get U_B.

5) Check that your calculations are correct in the following way. If the size of sample A is n_A and of sample be is n_B, then, if you have calculated U_A and U_B correctly, $U_A + U_B$ will equal n_A multiplied by n_B. If it does not recalculate U_A and U_B

$n_A = 5 \quad n_B = 3$
$U_A = 8 \quad U_B = 7$
$U_A + U_B = 8 + 7 = 15$
$n_A \times n_B = 5 \times 3 = 15$

Hence $U_A + U_B = n_A \times n_B$ (the calculation is correct)

6) Choose the smaller of U_A and U_B. This is the test statistic. If one of your sample sizes is greater than 20 go to step 7. Otherwise carry on reading.

Refer to Table A16.1 (located at the end of this chapter) which shows the critical values of *U* at the 5% significance levels. The critical values of *U* for the two sample sizes is found at the intersection of the appropriate row and column. If, for example, both your sample sizes are 8 then read down from the 8 in the top row and along from the 8 in the left-hand column until the two lines meet. The critical value is 13. This means that when both sample sizes are 8, the test statistic *U* has to be 13 or smaller for you to reject the null hypothesis at the 5% level. To take another example, if your sample sizes are 13 and 10, read down from the 13 in the top row and along from the 10 in the left-hand column until the two lines meet. The critical value of *U* in this case is 33. You will notice that you cannot use Table A16.1if one or both samples are very small, and indeed you are advised to ensure that both sample sizes are greater than 10 if at all possible.

Now decide whether or not to reject the null hypothesis for our example.

7) Table A16.1 (a standard Table of critical values for the test statistic *U*) can only cope with sample sizes of up to 20. If one of your samples is larger than this proceed as follows.

If n_A and n_B are the two sample sizes, and *U* is the test statistic, calculate a new test statistic called *z* as follows.

$$z = -\frac{U - (n_A\, n_B / 2)}{\sqrt{n_A\, n_B\, (n_A + n_B + 1) / 12}}$$

For example, let $U = 154$, $n_A = 18$ and $n_A = 25$. Then

$$z = -\frac{U - (18 \times 25 / 2)}{\sqrt{18 \times 25 \times (18 + 25 + 1) / 12}} - \frac{-71}{40.6202} = 1.74790$$

= 1.75 (to two decimal places).

The probability distribution of z is very well known (it is the standard normal distribution - see Page 34) and it is therefore possible to calculate critical values for z for various significance levels; three values are given below:

Critical values of z for three significance levels.

significance level:	5%	1%	0.1%
critical value for z:	1.96	2.58	3.29

You can reject the null hypothesis at a given level of significance only if the absolute value of your calculated z (ignoring the minus signs) is *greater* than or equal to the critical value. In the present example, the calculated value of z is 1.75, which is smaller than 1.96, so the null hypothesis cannot be rejected at the 5% significance level.

Do's and Don't's for Mann-Whitney U tests

- The Mann-Whitney U test is designed to be used on unmatched samples. Do not, therefore, carry out such a test on matched pairs of measurements.
- The test is based on the assumption that the measurements in the two populations (from which the samples are taken) have the same shaped distribution, like Figure 16.4a and b and unlike Figure 16.4c. Although you cannot produce histograms for your populations, you *can* do so for your samples. Your samples will probably be too small to be very informative, but if the shapes of their distributions appear to be widely different, do not use the Mann-Whitney U test.
- Since it is a test for differences in population **medians**, do not use the Mann-Whitney U test to make statements about population *means.* This is a common error; for example, one often comes across statements such as: "The *mean* of sample A was x!, and of sample B was y!. A Mann-Whitney U test showed that these differed significantly at the 5% level".
- If your measurements are at the interval level and both your sample sizes are greater than 25, do not use the Mann-Whitney U test. It is quicker and easier, in such circumstances, to use the z test.

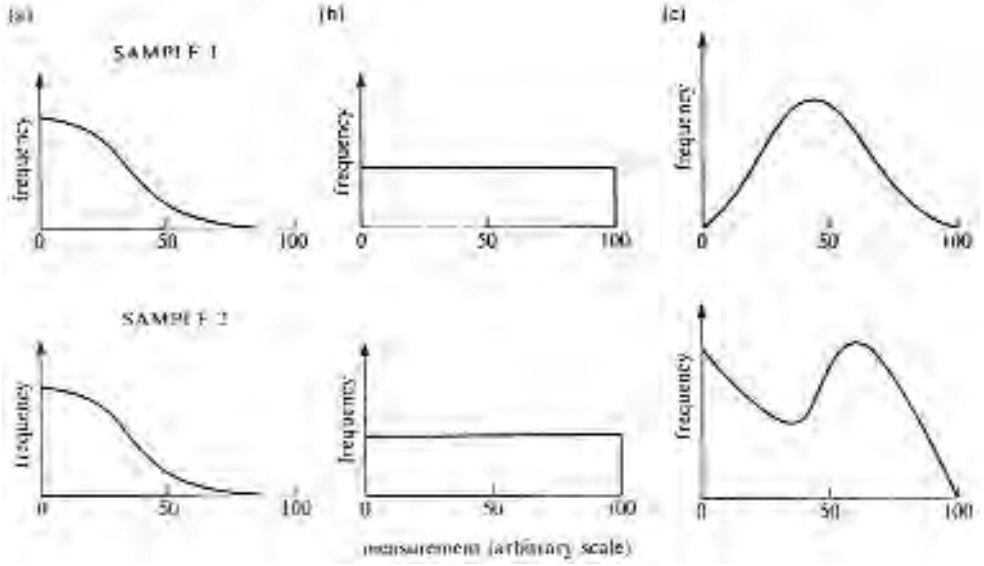


Figure 16.4 Pairs of samples from populations with distributions that are (a and b) and are not (c) the same.

The Wilcoxon matched pairs test.

When to use:

- If you wish to test for differences in population **medians**.
- Provided you have two **matched** samples.
- Provided your data are either at the **interval** level.
- Provided that the differences between matched measurements from the two samples are **symmetrically distributed**.
- Do not use if the number of non-zero differences between matched measurements is **less than 5** or **greater than 30**.
- It is difficult to use the Wilcoxon matched pairs test to calculate confidence intervals without the use of a computer.

Rationale of test

Suppose that you have two matched samples, and that you wish to test whether they come from populations with the same medians. Sample A contains four measurements: A_1, A_2, A_3 and A_4. Sample B also contains four measurements: B_1 matched with A_1, B_2 matched with A_2, B_3 matched with A_3 and B_4 matched with A_4. (Note that, although we call the measurements A_1, A_2, A_3 and A_4, we are not saying that these are ranked in order of size - that A_1 is the smallest and A_4 the largest for example. A_1 is simply the first measurement you made in sample A, and B_1 is the measurement from sample B with which A_1 is matched (see Box 16.1). For the purposes of this test, you have to know how big the measurements are; that is, the measurements need to be at the interval level. This means that it is possible to calculate how far apart the two measurements are in each matched pair: you can calculate the difference between A_1 and B_1, between A_2 and B_2, and so on.

If the null hypothesis that population medians are equal is true, it is unlikely that all of the measurements in sample A will be smaller than those in B, or vice versa; that is, it is unlikely that:

$$A_1 > B_1 \quad A_2 > B_2 \quad A_3 > B_3 \quad A_4 > B_4$$

or that

$$A_1 < B_1 \quad A_2 < B_2 \quad A_3 < B_3 \quad A_4 < B_4$$

It is more likely, if the null hypothesis is true, that some of the measurements from sample A will be larger than their matched measurements from sample A will be larger than their matched measurements from sample B, and that some will be smaller; for example:

$$A_1 > B_1 \quad A_2 < B_2 \quad A_3 > B_3 \quad A_4 < B_4$$

Consider now the *size* of the difference between each matched pair of measurements. Some of the sample A measurements may be much bigger than their matched sample B measurements, some only slightly bigger. Similarly, some of the sample B measurements may be much bigger than their matched sample A measurements, some only slightly bigger. If the nul hypothesis is true, then it is unlikely that all of the big differences will be in one direction and all of the small ones in the other; for example, the following result is unlikely:

A_1 much bigger than B_1 $\quad$ A_2 just smaller than B_2
A_3 much bigger than B_3 $\quad$ A_4 just smaller than B_4

It is more likely, if the null hypothesis is true, that there will be as many big differences and as many small differences in one direction as in the other; for example:

A_1 much bigger than B_1 $\quad$ A_3 just bigger than B_3
A_2 much smaller than B_2 $\quad$ A_4 just smaller than B_4

The idea behind the Wilcoxon matched pairs test is to sort differences only according to their various magnitudes, and to see whether any trend is apparent - whether big differences occur in one direction and small differences in the other, or whether both are more or less equally common in each direction. To do this, it is necessary to rank all of the differences in order of increasing size.

Example 1

Consider the pair of matched samples, each containing six measurements of elephant tusk weight, in columns 2 and 3 below. The smallest difference is between measurements A_6 and B_6, a difference of only 1 (see column 4) so this gets a rank of 1 in column 5. The next smallest difference is between measurements A_2 and B_2, a difference of 2, so this gets a rank of 2. The largest difference is between A_3 and B_3, a difference of 9, so this gets a rank of 6. We do not, at this stage, take into account whether the differences are positive or negative.

(1) measurement no.	(2) tusk weight sample A	(3) tusk weight sample B	(4) difference (A minus B)	(5) rank
1	8	12	-4	4
2	9	7	+2	2
3	12	3	+9	6
4	6	9	-3	3
5	14	6	+8	5
6	10	11	-1	1

The ranking procedure has told us the size of the differences between matched measurements, but has not told us in which directions they occur. To discover this, divide the ranks you have just calculated into two groups: those that are associated with positive differences, and those that are associated with negative ones. In the present example, this produces the following result:

measurement no.	rank	direction of difference
1	4	–
2	2	+
3	6	+
4	3	–
5	5	+
6	1	–

The result is perhaps clearer if positive and negative differences are set out in two separate columns:

	ranks of positive differences		ranks of negative differences
	2		4
	6		3
	5		1
total	13	total	8

If the null hypothesis is true, you would expect the sum of the ranks of the negative differences to be roughly equal to the sum of the ranks of the positive differences. You would not expect them to be wildly different. In the present example the sum of the negative differences is 8 and of the positive differences is 13, which is some way from equality, but not enormously so.

Example 2

Consider another example which is of two matched samples of seed weights (see below). Here, all of the differences are negative. As a result, the sum of the ranks of the positive differences, 0, is much smaller than that of the negative differences, 21. It is unlikely that you would get such large differences in these totals if the null hypothesis were true.

measurement no.	seed weights/mg sample C	seed weights/mg sample D	difference (C minus D)/mg	rank of difference	ranks positive differences	ranks negative differences
1	10	20	–10	5		5
2	11	12	–1	1		1
3	14	23	–9	4		4
4	13	15	–2	2		2
5	13	26	–13	6		6
6	21	24	–3	3		3
				total	0	total 21

Example 3

Try to do this example yourself:

The table below shows two matched samples of relative humidity. The null hypothesis is that the samples come from populations with equal medians. Calculate the sum of the ranks of the positive differences and of the negative differences. Are you likely to get such totals if the null hypothesis is true?

	relative humidity / %	
measurement no.	**sample E**	**sample F**
1	57	64
2	49	54
3	53	51
4	50	54
5	57	51
6	56	48

First find the differences, and rank them. The two totals are very close together, a result that is very likely if the null hypothesis is true.

	relative humidity/%					
measurement no.	**sample E**	**sample F**	**difference (E minus F)/%**	**rank of difference**	**ranks positive differences**	**ranks negative differences**
1	57	64	–7	5		5
2	49	54	–5	3		3
3	53	51	+2	1	1	
4	50	54	–4	2		2
5	57	51	+6	4	4	
6	56	48	+8	6	6	
				total	11	total 10

Below we summarise the pairs of totals that we have calculated in the last three examples, and the conclusion that can be drawn in each case about the null hypothesis. The table bears out the statement made earlier that if the null hypothesis is true, you would expect the sum of the ranks to the positive and negative differences to be roughly equal rather than to be wildly different.

	Example 1	**Example 2**	**Example 3**
sum of positive differences	13	0	11
sum of negative differences	8	21	10
likelihood of obtaining such sums if H_o true	moderate	low	high

The results suggests that either of the sums could act as a test statistic. The test statistic is usually denoted by the letter W. It is not difficult to work out from first principles the probability distribution of W given that the null hypothesis is true, but we do not do this here.

It turns out that the probability distribution of W is different for each pair of sample sizes. You can work out the critical value of W from Figure 16.5 for a 5% significance level. If two samples, each containing six measurements, come from populations with the same median, the calculated value of W will lie between 0 and 21 inclusive (since 6 + 5 + 4 + 3 + 2 + 1 = 21). If you take such samples repeatedly, the value of W will fluctuate from sample to sample. If the null hypothesis is true, there is a probability of about 0.03 (i.e. 3%) that W will be as extreme as 0 or 21. Hence if we obtain a value for W of 0 or 21, we can reject the null hypothesis at the 5% level.

Rather than drawing several probability distributions for different sample sizes, it is usual to present a Table of critical values for W for given significance levels (see Table A16.2 at the end of this chapter). To use this Table, first choose whichever of the two sums of differences (positive or negative) is the smaller for your test statistic W.

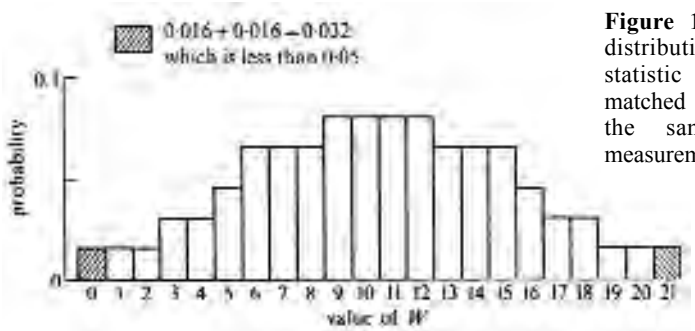


Figure 16.5 The probability distribution of W, the test statistic for the Wilcoxon matched pairs test, when both the samples contain six measurements.

WORKED EXAMPLE

Procedure

1) Arrange the measurements from the two samples in matched pairs side by side (columns 1 and 2)

2) Subtract each measurement for sample B from its paired measurement in sample A (column 3)

3) Ignoring whether the differences are positive of negative, rank the differences in increasing order of size (column 4). The smallest difference should be given rank 1, the next smallest rank 2, and so on. Differences of equal magnitude should be given an average rank. Thus the two differences of 4 share the average rank of 4.5. If two matched measurements have the same value, so that the difference between them is zero, remove them altogether from the calculations.

(1) sample A	(2) sample B	(3) difference (A minus B)	(4) rank of differences
16	15	+4	4.5
15	15	0	–
10	14	–4	4.5
9	10	–1	1
7	5	+2	2
17	14	+3	3
8	13	–5	6

4) Divide the ranks into two groups: those associated with positive differences and those associated with negative differences.

5) Add up the ranks in each of the two groups.

6) Check the accuracy of the calculations as follows. Let R^+ be the positive ranks and R^- the sum of the negative ones. Let the number of differences that are not zero be N_D.
Then, if your calculations are correct, you will find that

$$R^+ + R^- = 1/2\, N_D\, (N_D + 1)$$

If it does not, re-do your calculations.

7) Choose the smaller of R^+ and R^-, and let this be the test statistic *W*.

Refer to Table A16.2 at the end of this chapter which shows the critical values of *W* at various significance levels and N_D values. Reject the null hypothesis only if the calculated value of *W* is equal or *smaller* than the critical value given in the Table for your chosen level of significance and calculated value of N_D. (Note that the Table does not give critical values for N_D less than 5, so it is not worth carrying out the test with matched samples with fewer than five non-zero differences).

	ranks of positive differences, R^+		ranks of negative differences, R^-
	4.5		4.5
	2		1
	3		6
total	9.5	total	11.5
	$R^+ = 9.5$		$R^- = 11.5$

$N_D = 6$
(There are 7 pairs of measurements, hence 7 differences [A minus B], but one of these is zero, so $N_D = 6$).

$R^+ = 9.5$ $R^- = 11.5$
Therefore $W = 9.5$

At the 5% significance level for $N_D = 6$, the critical value of $W = 0$

W is greater than this, so do not reject H_0.

Do's and Don't's for the Wilcoxon matched pairs test

- The Wilcoxon matched pairs test can only be used on **matched** samples. Do not, therefore, carry out such a test on unmatched samples.
- The test is based on the assumption that the differences in measurements taken from the two populations are symmetrically distributed. Figure 16.6 shows some symmetrical distributions. Notice that the distribution of the differences does not have to be normal, but it does have to be symmetrical. In practice, you will usually be working with small samples (see point 3 below), and even if you were to plot frequency distributions of your differences, it would be very difficult to discover whether they were symmetrically distributed or not. Most people therefore work on the assumption that the differences *are* symmetrically distributed, unless there are good reasons for supposing the contrary.
- Do not use the test for sample sizes of more than 30. The test becomes increasingly cumbersome with large sample sizes, and there are quicker tests that can be used on large samples (see the *z* test for matched samples).

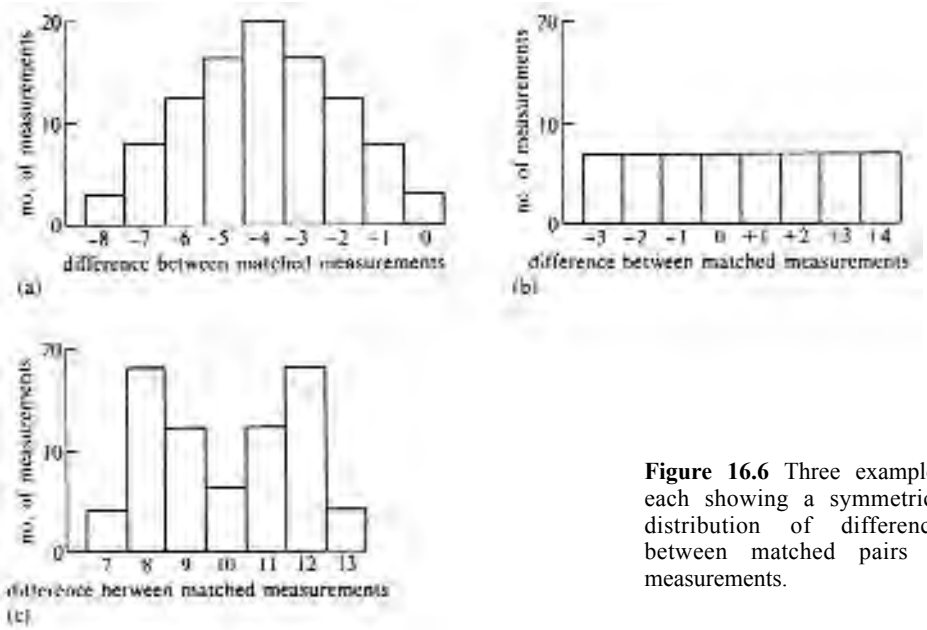


Figure 16.6 Three examples, each showing a symmetrical distribution of differences between matched pairs of measurements.

The *z* test for unmatched samples.

When to use:

- If you wish to test for differences in population **means**.
- Provided you have two **unmatched** samples.
- Provided that your measurements are at the **interval** level.
- Provided that your samples have **at least 25** measurements each.

Rationale of test

Suppose that you have two unmatched samples, each containing 25 or more measurements, and that you wish to test whether they came from populations with the same means. For the *z* test to be valid, your measurements must be at the interval level: lengths, weights, durations and other common biological measurements fall into this category.

Suppose that you have two samples of carapace lengths of hatchling leatherback turtles (*Dermochelys coriacea)*, A and B, which come respectively from populations with mean carapace lengths of μ_A and μ_B (The Greek letter μ, pronounced mu to rhyme with new, is the symbol commonly used to denote a population mean). The null hypothesis H_0 would be that the mean carapace lengths of the two populations were the same, and the alternative hypothesis H_1 would be that they were different. In symbols these hypotheses could be written as:

H_0: $\mu_A = \mu_B$

H_1: $\mu_A \neq \mu_B$

Another way of writing the null hypothesis is:

H_0: $\mu_A - \mu_B = 0$

That is, if $\mu_A = \mu_B$, then the difference in population means is zero.

If you repeatedly take pairs of samples from populations A and B and calculate the difference in sample means for each pair, then, if the null hypothesis is true, these differences will be clustered around a mean value of zero and will be normally distributed (see Box 4b).

If the differences in sample means are normally distributed, we know that about two-thirds of them will lie within one standard error of the mean, about 95% within two, and that we can calculate the percentage of differences that lie within any chosen number of standard errors from the mean. All that you have to do, therefore, is to find out how many standard errors away from the mean is the difference in sample means that you have calculated. Taking the turtle example in Box 4b, the standard error of the difference in the sample means was 2.772 mm. If the difference in means of two carapaces happened to be 2.772 mm this would be exactly one standard error from the mean difference of zero; if the difference happened to be 4.200 mm, this would be 4.200 / 2.772 = 1.515, or roughly 1.5 standard errors from the mean, and so on. Figure 16.7 illustrates this.

Figure 16.7b is especially important because it shows the shape of the normal distribution that you get when you express your difference in sample means as 'so many standard errors from the mean'. It is called the **standard normal distribution**. Figure 16.7b tells us that about 13% of differences in sample means are more than 1.5 standard errors away from the mean. Thus under the null hypothesis that our two samples of turtle carapaces come from populations with the same mean length, the probability of obtaining a difference in sample means of at least 1.5 standard errors is 13%. This is considerably greater than the value of 5% or less that we require to be able to reject the null hypothesis; so we would accept it.

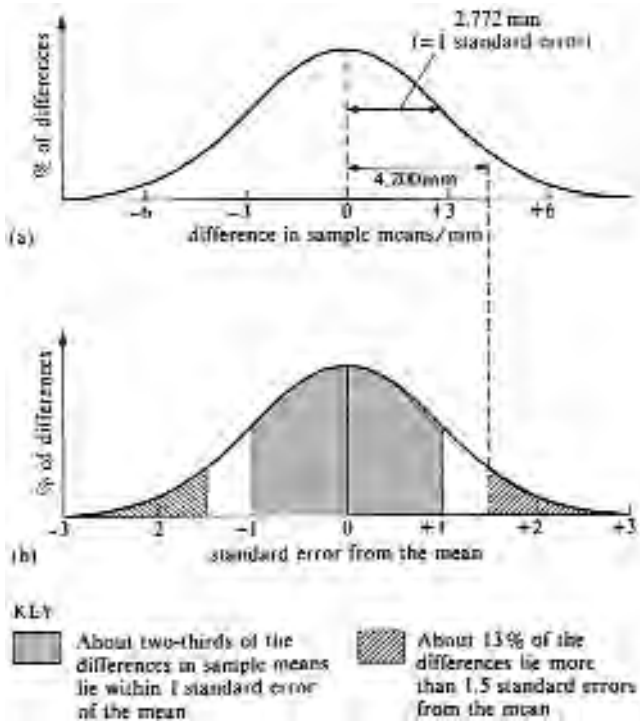


Figure 16.7 Differences in sample means (a) the difference in sample means for the seed example in Box 4b and; (b) the same differences expressed in terms of standard errors of the mean.

The preceding paragraphs present the essence of the *z* test. In it, you *convert* your difference in sample means to a new number: the number of standard errors your difference lies from the mean difference expected under the null hypothesis. This new number, the test statistic, denoted by the letter *z*, has a normal probability distribution with a mean of zero and a standard deviation of 1. There are Statistical Tables (as presented for the Mann Whitney *U* test above) that give the critical values of *z* for tests at various levels of significance. The critical values can be used to calculate confidence intervals (see the worked example above).

To put the above into symbols:

Let two samples, A and B, have means of $x!_A$ and $x!_B$ respectively. Let the standard error of the difference in the sample means be SE_D. Then the difference in the sample means is $x!_A - x!_B$, and the number of standard errors this difference lies from zero, denoted by z, is given by:

$$z = \frac{x!_A - x!_B}{SE_D}$$

One final problem remains. Given two samples A and B, say of turtle hatchlings, it is easy to calculate their respective means $x!_A$ and $x!_B$; but how do we discover the standard error of the difference in the sample means?

As Box 4b showed, it is impossible to collect all possible pairs of samples from two populations of turtles, and to work out from them the distribution of all differences in mean sample lengths. Instead, you have to *estimate* the standard error of the difference in sample means, and you do this from measurements in your two samples. If sample A has n_A measurements with a sample standard deviation of s_A, and if sample B has n_B measurements with a sample standard deviation of s_B, then it turns out that the estimated standard error for the differences in sample means SE_D is given by

$$SE_D = \sqrt{\frac{s_A^2}{n_A} + \frac{s_B^2}{n_B}}$$

But $z = \dfrac{x!_A - x!_B}{SE_D}$

Thus, by substituting for SE_D,

$$z = \frac{x!_A - x!_B}{\sqrt{\frac{s_A^2}{n_A} + \frac{s_B^2}{n_B}}}$$

from which z can be calculated

Blue duiker
Cepholophus monticola

WORKED EXAMPLE

Procedure

1) Arrange your sample measurements x_A and x_B in two columns (columns 1 and 3) and work out the sample mean and variance of each using the procedure given in Box 4c, or a suitable computer program.

Following the procedure in Box 4c:

(i) Square each measurement from each sample, to give x_A^2 and x_B^2 and enter them in columns 2 and 4.

(1)	(2)	(3)	(4)
sample A		sample B	
length	x_A^2	**length**	x_B^2
60.3	3636.09	57.2	3271.84
61.9	3831.61	56.9	3237.61
59.8	3576.04	58.1	3375.61
58.7	3445.69	55.0	3025.00
61.1	3733.21	56.3	3169.69
60.7	3684.49	54.1	2926.81
62.4	3893.76	55.5	3080.24
58.1	3375.61	56.0	3136.00
58.9	3469.21	54.2	2937.64
60.0	3600.00	55.8	3113.64
59.7	3564.09	53.9	2905.21
59.9	3588.01	55.1	3036.01
62.1	3856.41	54.9	3014.01
61.1	3733.21	55.4	3069.16
60.3	3636.09	54.3	2948.49
61.2	3745.44	53.9	2905.21
		53.2	2830.24

(ii) Add up the numbers in each column, to give • x_A, • x_A^2, • x_B and • x_B^2.

□ x_A=966.2 **□ x_B=939.8**
□x_A^2=58368.96 **□x_B^2=51982.42**

(iii) Divide the totals of the measurements by the sample size, n_A or n_B, to give the sample means (note that in order to simplify calculations we are using sample sizes of less than 25 here, whereas in practice this should not be done)

$$\frac{\bullet\ x_A}{n_A} = \frac{966.2}{16} \qquad \frac{\bullet\ x_B}{n_A} = \frac{939.8}{16}$$

$= 60.3875$ $\qquad = 55.2824$

(iv) Square the totals of the measurements

$(\bullet\ x_A)^2 = 966.2^2 = 933542$ $\qquad (\bullet\ x_B)^2 = 939.8^2 = 883224$

(v) Divide each of the figures you have just calculated by the corresponding sample size to give $(\bullet\ x_A)^2/n_A$ and $(\bullet\ x_B)^2/n_B$

$(\bullet\ x_A)^2/n_A = 933542 / 16 = 58346.4$
$(\bullet\ x_B)^2/n_B = 883224/17 = 51954.4$

(vi) Subtract the figures from step (v) from the totals of the squared measurements ($\bullet\ x_A^2$ and $\bullet\ x_B^2$)

58368.96 - 58346.4 = 22.56 $\qquad$ 51982.42 - 51954.4 = 28.02

(vii) Divide each of the figures from step (vi) by one less than the corresponding sample size

$$\frac{22.56}{16 - 1} = \frac{22.56}{15} \qquad \frac{28.02}{17 - 1} = \frac{28.02}{16}$$

$= 1.504$ $\qquad = 1.751$

(viii) These figures are the sample variances, s_A^2 and s_B^2

$s_A^2 = 1.504$ $\qquad s_B^2 = 1.751$

2) Calculate the estimated standard error of the the difference between the sample means, SE_D using the equation

$$SE_D = \sqrt{\frac{s_A^2}{n_A} + \frac{s_B^2}{n_B}}$$

$$SE_D = \sqrt{\frac{1.504}{16} + \frac{1.751}{17}}$$

$$= \sqrt{0.094 + 0.103}$$

$$= 0.443847$$

3) Calculate z, using the equation

$$z = \frac{x!_A - x!_B}{SE_D}$$

$$z = \frac{60.3875 - 55.2824}{0.443847}$$

$$= 11.5019$$

$$= 11.50 \text{ (to two decimal places)}$$

4) Choose which of the following you require:

(i) a significance test: if so, go to step 5
(ii) a confidence interval, if so use step 6

5) The value of z that you have calculated may be either positive or negative. This will depend upon which of your two samples you decide to call sample A, and which sample B. This is an arbitrary decision - if in the worked example you had reversed the labels on the two samples, your calculated value of z would have been -11.50

mean of sample A = 55.2824
mean of sample B = 60.3875
SE_D = 0.443847 (as before)

$$z = \frac{60.3875 - 55.2824}{0.443847}$$

$$z = -11.5019$$

$$z = -11.50 \text{ (to two decimal places)}$$

Take the absolute value of z, ignoring whether it is positive or negative, and compare it with the critical values for z (z_{crit}) at various levels of significance. The critical values are as follows:

significance level:	**5%**	**2%**	**1%**	**0,1%**
z_{crit}	1.96	2.33	2.58	3.29

If your calculated value of z is greater than or equal to a critical value, you can reject H_0 at the corresponding significance level.

11.50 is greater than 3.29, so we can reject H_0 at the 0.1% level of significance in favour of the alternative hypothesis.

6) The most usual confidence interval to calculate is the 95% confidence interval for the difference in population means. You may, however, choose other confidence intervals if you wish.

(i) Choose from the following list the appropriate critical value of z:

confidence level:	**95%**	**99%**	**99.9%**
z_{crit}	1.96	2.33	2.58

For this worked example, we choose a 95% confidence interval, for which the critical value of z is 1.96.

(ii) Calculate the difference in sample means.	$x!_A - x!_B$ = 60.3875 – 55.2824 = 5.1051 mm
(iii) The lower limit of the confidence interval is given by $(x!_A - x!_B) - (z_{\mathbf{crit}} \times SE_D)$	*lower limit* 5.1051 - (1.96 x 0.443847) = 5.1051 - 0.869940 = 4.23516 = 4.24 mm (to two decimal places)
(iv) The upper limit of the confidence interval is given by $(x!_A - x!_B) + (z_{\mathbf{crit}} \times SE_D)$	*lower limit* 5.1051 + (1.96 x 0.443847) = 5.1051 + 0.869940 = 5.97504 = 5.98 mm (to two decimal places)
(v) So the confidence limits are given by $(x!_A - x!_B) \pm (z_{\mathbf{crit}} \times SE_D)$	The 95% confidence interval in the worked example is 4.24 mm to 5.98 mm. Thus we estimate, with 95% confidence, that the difference between the population means lies between 4.24 mm and 5.98 mm.

Do's and Don't's for z tests for unmatched samples

- The *z* test, as described here, can only be used on **unmatched** samples. Do not, therefore, use this test on matched samples. A different version, which can be used on matched samples, is described below.
- Your measurements should be at the interval level, but they need *not* be normally distributed (this is because the means from large samples of the same population will be approximately normally distributed, even though the individual measurements from the population may not be).
- Do not use the *z* test if your samples are small. As a rough guide, both samples should be at least 25. If your measurements appear *not* to be normally distributed (and frequency distributions will help you decide this) use sample sizes well over 25.
- When deciding how large your samples should be, consider the following points:

  (i) Larger samples give better indications of the populations from which they come than do smaller ones, so from one point of view, the bigger the samples, the better. But . . . .

  (ii)There is a law of diminishing returns. For example, an increase in sample size from 25 to 35 might alter the value of the test statistic *z* considerably, whereas an increase from 125 to 135 would have less effect.

  Thus, although large samples provide a sounder basis for making inferences about populations, it may not be worth while working with very big samples. Here, practical considerations come into play. How difficult, expensive, or time consuming is it to collect your measurements? Could you better spend your time on extending your project in other directions, rather than collecting very big samples to answer just one question? These are matters for your own judgement.

The z test for matched samples.

When to use:

- If you wish to test for differences in population **means**.
- Provided you have two **matched** samples.
- Provided that your measurements are at the **interval** level.
- Provided that you have **at least 30 measurements** in each of your two samples.

Rationale of test

First read the rationale behind the z test for unmatched samples above. The z test for matched samples is a simple extension of the unmatched test. Imagine that you have two matched samples A and B of say 30 measurements each, with the measurements at the interval level, and you wish to test whether they come form populations with identical means. In symbols,

$$H_0\text{:}\ \mu_A = \mu_B$$

Suppose that you now calculate the difference between each pair of measurements, by subtracting one from the other. If the null hypothesis is true, you would expect that some differences will be positive and some negative but that on average they will be close to zero, as seems probable from the early part of the left hand table below. By contrast, if the average is far from zero, as in the early part of the right hand table, then H_0 is unlikely to be true. In fact, under the null hypothesis, you expect the mean of the differences to be zero, and, moreover, that the mean differences will be normally distributed around zero with a certain standard error (SE_D). The rationale of the z test applied to matched pairs, is, therefore, to calculate the mean of the differences, and to discover how many standard errors away from zero this mean lies. This gives the z statistic, which can be treated in the same way as for unmatched samples.

Likely outcomes for differences in paired measurements from samples A and B, if H_0 (that population means are equal) is true.

measurement no.	sample A	sample B	difference (A B)
1	A_1	B_1	D_1 (+ve)
2	A_2	B_2	D_2 (+ve)
3	A_3	B_3	D_3 (-ve)
4	A_4	B_4	D_4 (-ve)
5	A_5	B_5	D_5 (+ve)
.	.	.	.
.	.	.	.
.	.	.	.
30	A_{30}	B_{30}	D_{30} (-ve)

Unlikely outcomes for differences in paired measurements from samples A and B, if H_0 (that population means are equal) is true.

measurement no.	sample A	sample B	difference (A B)
1	A_1	B_1	D_1 (+ve)
2	A_2	B_2	D_2 (+ve)
3	A_3	B_3	D_3 (+ve)
4	A_4	B_4	D_4 (+ve)
5	A_5	B_5	D_5 (+ve)
.	.	.	.
.	.	.	.
.	.	.	.
30	A_{30}	B_{30}	D_{30} (+ve)

WORKED EXAMPLE

Procedure

1) Arrange your sample measurements in two columns (2 and 3), with the matched pairs on the same row (e.g. 14 and 16 in row 3 are a matched pair). Subtract each sample B measurement from its matched sample A measurement, writing the answer in column 4.

Calculate the sample mean and variance of the differences in column 4, using the procedure given in Box 4c.

Following the procedure in Box 4c:

(i) Square each difference in column 4, and put the results in column 5.

Calculation

(1) measurement no.	(2) sample A	(3) sample B	(4) difference (A B)	(5) difference squared
1	13	10	+3	9
2	15	12	+3	9
3	14	16	-2	4
4	20	15	+5	25
5	21	16	+5	25
6	18	20	-2	4
7	11	10	+1	1
8	11	15	-4	16
9	21	17	+4	16
10	19	10	+9	81
.	.	.	.	.
.	.	.	.	.
.	.	.	.	.
30	14	17	-3	9
		total*	45	328

*Most of sample measurements are not shown, for the sake of brevity. The totals are for *all* the sample measurements, both those that are shown and those that are not.

(ii) Add up the numbers in columns 4 and 5. This gives • (A-B) and • $(A\text{-}B)^2$

$$\bullet\ (A\text{-}B) = 45 \qquad \bullet\ (A\text{-}B)^2 = 328$$

(iii) Divide the total in column 4 by the sample size. The number you have calculated is the mean of the differences between matched pairs of measurements, *D*!.

$$\bullet\ (A\text{-}B)/n = 45/30 = 1.5$$

$$D! = 1.5$$

(iv) Square the total of column 4 to give $(\bullet\ (A\text{-}B))^2$

$$(\bullet\ (A\text{-}B))^2 = 45^2 = 2025$$

(v) Divide the figure from step (iv) by the sample size to give $(\bullet\ (A\text{-}B))^2/n$

$$(\bullet\ (A\text{-}B))^2/n = 2025 / 30 = 67.5$$

(vi) Subtract the figure from step (v) from the total of column 5 to give
$\bullet\ (A\text{-}B)^2 - (\bullet\ (A\text{-}B))^2/n$

$$\bullet\ (A\text{-}B)^2 - (\bullet\ (A\text{-}B))^2/n = 328 - 67.5 = 260.5$$

(vii) Divide the figure from step (vi) by one less than the sample size

$$\frac{\bullet\ (A\text{-}B)^2 - (\bullet\ (A\text{-}B))^2/n}{n - 1} = \frac{260.5}{29} = 8.98276$$

(viii) This is the sample variance of the differences in the paired measurements, s_D^2.

$$s_D^2 = 8.98276$$

2) Calculate the estimated standard error of the mean differences in paired measurements, SE_D using the equation

$$SE_D = \sqrt{\frac{s_D^2}{n_A}} \qquad SE_D = \sqrt{\frac{8.98276}{30}} = 0.547198$$

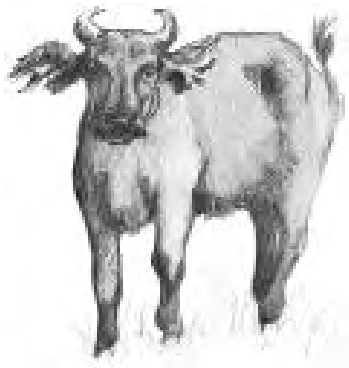
3) Calculate z using the equation

$$z = \frac{D!}{SE_D} \qquad z = \frac{1.5}{0.547198} = 2.74124 = 2.74 \text{ (to two decimal places)}$$

4) For a significance test go to step 4 of the z test for unmatched samples, and for a confidence interval go to step 6. Proceed exactly as for unmatched samples.

Do's and Don't's for z tests for matched samples

- The z test, as described here, can only be used on **matched** samples. Do not, therefore, use this test on unmatched samples.
- Do not use the z test if your samples are small. As a rough guide, both samples should be at least 30. If your measurements appear *not* to be normally distributed (and frequency distributions will help you decide this) use sample sizes well over 30.
- The other do's and don'ts are the same as for the unmatched z test (above).



The *t* test for unmatched samples.

When to use:

- If you wish to test for differences in population **means**.
- Provided you have two **unmatched** samples.
- Provided that your measurements are at the **interval** level.
- When your samples are small; that is when one or both have **fewer than 25** measurements.
- Provided that the measurements can be assumed to come from **normally** distributed populations with **equal variances**.

Rationale of test

First, make sure that you are familiar with the rationale of and the procedure for the *z* test for unmatched samples (above). You will notice that the *z* test can only be used when sample sizes are large - each containing 25 or more measurements.

If you have smaller samples than this, which often happens in biological research, then you cannot use the *z* test. Recall that the equation for calculating *z* is

$$z = \frac{\bar{x}_A - \bar{x}_B}{SE_D}$$

where the difference in sample means, $\bar{x}_A - \bar{x}_B$, is assumed to be normally distributed, and SE_D is the estimate of the standard error of the difference in means. It turns out that with small samples there are two problems. First, $\bar{x}_A - \bar{x}_B$ will not be normally distributed unless the populations from which the samples come are themselves normally distributed (remember with large samples, $\bar{x}_A - \bar{x}_B$ will be normally distributed). Second, the method of estimating SE_D [the standard error of the differences in sample means] used in the *z* test is not appropriate when small samples are used. So, with small samples the *z* test cannot be used, and the *t* test is used instead.

The *t* test differs from the *z* test in certain important respects to cope with the problems just mentioned. First, you use a different method of estimating SE_D. Second, you calculate your test statistic, *t*, using the equation

$$t = \frac{\bar{x}_A - \bar{x}_B}{SE_D}$$

and compare this, not with critical values obtained from the normal distribution, but with critical values obtained from a different probability distribution. We shall consider these two points in more detail.

(i) Calculating SE_D

First. we have to make one major assumption, which is that the two populations from which the samples come have the same variance. This is usually called the **common population variance**, and is denoted by S_C^2. Suppose that you wish to estimate SC^2 from two samples: sample A containing n_A measurements and with a sample variance of s_A^2, and sample B containing n_B measurements and with a sample variance of s_B^2. (You can calculate these sample variances using the method in Box 4c or by using appropriate computer programs).

Then the estimate of the common population variance is given by

$$S_C^2 = \frac{(n_A - 1)\ s_A^2 + (n_B - 1)\ s_B^2}{(n_A - 1) + (n_B - 1)}$$

From this, you can calculate SE_D from the equation

$$SE_D = \sqrt{\frac{S_C^2}{n_A} + \frac{S_C^2}{n_B}}$$

You then use this value of SE_D to find the value of t, using:

$$t = \frac{x!_A - x!_B}{SE_D}$$

(ii) The sampling distribution of t

Unlike the test statistic z, the sampling distribution of t varies with the sample size. More precisely, it varies with what is called the number of **degrees of freedom**. To obtain the number of degrees of freedom, you subtract 1 from each sample size, and add the two resulting numbers together. So, if the two sample sizes are n_A and n_B, the number of degrees of freedom is $(n_A - 1) + (n_B - 1)$. (Another way of writing this is $(n_A + n_B - 2)$. Thus the number of degrees of freedom is slightly smaller than the combined size of both samples. Figure 16.8 shows the sampling distribution of t for various degrees of freedom. It also shows the standard normal distribution, for comparison. You can see that if the number of degrees of freedom is small, the distribution of t is flatter and more widely spread than the standard normal distribution: a larger area of the distribution lies towards the extremes in the t distribution than in the standard normal distribution. So critical values for t for a given level of significance have to be larger than corresponding critical values of z. You will also notice that when the number of degrees of freedom is large, the distribution of t comes to closely resemble that of z. This is one reason why, with large samples, you can use the simpler z test rather than the t test.

The implication of this figure 16.8 is that when you wish to discover the critical value of t for a given significance level, you have to take into consideration the number of degrees of freedom. There are Statistical Tables (e.g. Table A16.3 at the end of this chapter) that give critical values of t for various degrees of freedom. The following example shows how to use these tables.

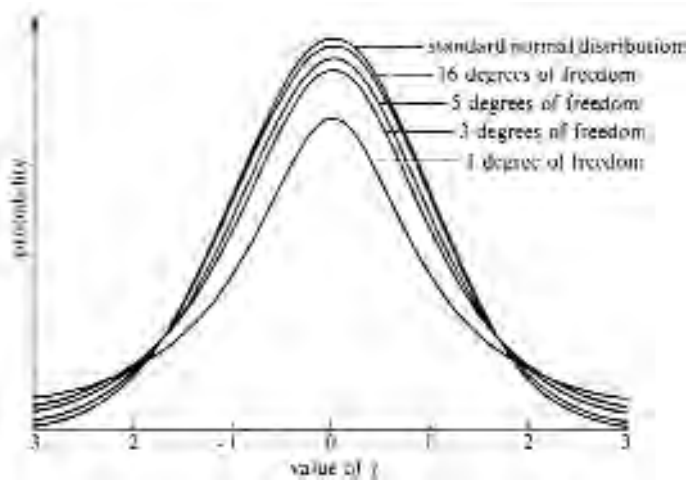


Figure 16.8 The sampling distribution of the test statistic, t, for various degrees of freedom, compared with the normal distribution.

WORKED EXAMPLE

Procedure

1) Arrange your sample measurements x_A and x_B in two columns (columns 1 and 3) and work out the sample mean and variance of each using the procedure given in Box 4c, or a suitable computer program.

Following the procedure in Box 4c:

(i) Square each measurement from each sample, to give x_A^2 and x_B^2 and enter them in columns 2 and 4.

(1)	(2)	(3)	(4)
sample A		**sample B**	
Weight / grams		**Weight / grams**	
x_A	x_A^2	x_B	x_B^2
3.0	9.0		
4.6	21.16	1.9	3.61
2.8	7.84	3.1	9.61
4.1	16.81	3.3	10.89
2.5	6.25	2.8	7.84
5.0	25.00	3.8	14.44
3.2	10.24	3.7	13.69
3.4	11.56	3.4	11.56

(ii) Add up the numbers in each column

$\bullet\ x_A = 28.6$ $\quad \bullet\ x_A^2 = 107.86$ $\qquad \bullet\ x_B = 22.0$ $\quad \bullet\ x_B^2 = 71.64$

(iii) Divide each of the totals of the measurements by the sample size.

$$\frac{\bullet\ x_A}{n_A} = \frac{28.6}{8} = 3.5750 \qquad \frac{\bullet\ x_B}{n_A} = \frac{22.0}{7} = 3.1429$$

These are the means of samples A and B, $x!_A$ and $x!_B$ respectively.

$$x!_A = 3.5750 \qquad x!_B = 3.1429$$

(iv) Square the totals of the measurements to give $(\bullet\ x_A)^2$ and $(\bullet\ x_B)^2$

$$(\bullet\ x_A)^2 = 28.6^2 = 817.96 \qquad (\bullet\ x_B)^2 = 22.0^2 = 484.0$$

(v) Divide each of the figures from step (iv) by the corresponding sample size to give $(\bullet\ x_A)^2/n_A$ and $(\bullet\ x_B)^2/n_B$

$$(\bullet\ x_A)^2/n_A = 817.96 / 8 = 102.245 \qquad (\bullet\ x_B)^2/n_B = 484/7 = 69.1429$$

(vi) Subtract the figures from step (v) from the totals of the squared measurements to give $\bullet\ x_A^2 - (\bullet\ x_A)^2/n_A$ and $\bullet\ x_B^2 - (\bullet\ x_B)^2/n_B$

$$\bullet\ x_A^2 - (\bullet\ x_A)^2/n_A = 107.86 - 102.245 = 5.615$$

$$\bullet\ x_B^2 - (\bullet\ x_B)^2/n_B = 71.64 - 69.1429 = 2.4971$$

(vii) Divide each of the figures from step (vi) by one less than the corresponding sample size

$$\frac{5.615}{8 - 1} = \frac{5.615}{7} = 0.802143 \qquad \frac{2.4971}{7 - 1} = \frac{2.4971}{6} = 0.416183$$

(viii) These figures are the sample variances, s_A^2 and s_B^2

$$s_A^2 = 0.802143 \qquad s_B^2 = 0.416183$$

2) Calculate the estimated standard error of the difference in sample means from the equation

$$S_C^2 = \frac{(n_A - 1)\, S_A^2 + (n_B - 1)\, S_B^2}{(n_A - 1) + (n_B - 1)}$$

$$S_C^2 = \frac{(7 \text{ x } 0.802143) + (6 \text{ x } 0.416183)}{(8 - 1) + (7 - 1)}$$

$$= \frac{5.615 + 2.4971}{13}$$

$$= 0.6240$$

[Notice that the two numbers added together on the top of the fraction defining S_C^2 are the two numbers calculated in step 1 (vi) during the calculation of sample variances]

3) Calculate the estimated standard error of the difference in sample means from the equation

$$SE_D = \sqrt{\frac{S_C^2 + S_C^2}{n_A + n_B}}$$

$$SE_D = \sqrt{\frac{0.6240}{8} + \frac{0.6240}{7}}$$

$$= \sqrt{0.0780 + 0.0891}$$

$$= 0.40878$$

4) Calculate t from the equation

$$t = \frac{x!_A - x!_B}{SE_D}$$

$$t = \frac{3.5750 + 3.1429}{0.40878}$$

$$= 1.05705$$

$$= 1.057 \text{ (three decimal places)}$$

5) Calculate the number of degrees of freedom,
$= (n_A - 1) + (n_B - 1)$

$= (8 - 1) + (7 - 1) = 13$

6) Choose which of the following you require
(i) a significance test: if so, go to step 7
(ii) a confidence interval, if so use step 8

7) The value of t that you have calculated may be positive or negative. This will depend upon which of your two samples you decided to call sample A, and which sample B. Take the absolute value of t (that is, ignore whether t is positive or negative).

The value of t would be either 1.057 or -1.057, depending on which sample you called A and which B.

The absolute value of t is 1.057.

Refer to table A16.3, which shows critical values of t at various significance levels.
If H_0: $\mu_A = \mu_B$
and H_1: μ_A ¤ μ_B
then reject in favour of at a given level of significance if your calculated value of t is bigger than the critical value of t for that significance level.

From Table A16.3, the critical value of t for 13 degrees of freedom at the 5% significance level is 2.160. The calculated value of t, 1.057, is smaller than this, so H_0 cannot be rejected.

8) The most usual confidence interval to calculate is the 95% confidence interval. You may, however, choose other confidence intervals if you wish.

(i) Select from Table A16.3 (at the end of this chapter) the value of t appropriate to the confidence interval of your choice and to the number of degrees of freedom.

We choose a 95% confidence level, and there are 13 degrees of freedom. Therefore,

$t = 2.160$

(ii) Calculate the difference in sample means.

$x!_A - x!_B = 3.575 - 3.1429 = 0.4321$

(iii) The lower limit of the confidence interval is given by
$(x!_A - x!_B) - (t \times SE_D)$

Lower limit
$= 0.4321 - (2.160 \times 0.40878)$
$= - 0.450865$

(iv) The upper limit of the confidence interval is given by
$(x!_A - x!_B) + (t \times SE_D)$

Upper limit
$= 0.4321 + (2.160 \times 0.40878)$
$= 1.31507$

(v) So the confidence limits are given by
$(x!_A - x!_B) \pm (t \times SE_D)$

The 95% confidence level is

- 0.45 g to +1.32 g (two decimal places)

This signifies that we can be 95% confident that the mean of the population from which the sample was drawn is no more than 1.32g greater than, and no more than 0.45g less than, the mean of the population from which sample B was drawn.

Do's and Don't's for z tests for matched samples

- The *t* test, as described here, can only be used on **unmatched** samples. An alternative version for matched samples is described below.

- Your measurements must be at the interval level and must be **normally distributed**. With small samples it is very difficult to discover whether the populations from which they come are in fact normally distributed. You should, however, draw line plots or histograms of the measurements in your two samples. If they appear to be very skewed, then do not use the *t* test: use medians and the Man -Whitney *U* test instead (see above).

  Most biological measurements involving weights, lengths, volumes and areas, etc. are not normally distributed. Only those relating to even-aged stands of plants and discrete age-classes of animals are likely to fit that distribution. Be careful also when working with measurements, such as percentages, where it may be theoretically impossible for a measurement to exceed or be smaller than a particular value. For example, a researcher might measure the percentages of mosquitoes killed by two brands of insecticide, A and B in two sets of 50 trials. Histograms of the results might be as in Figure 16.9. Most of the mosquitoes are killed by both brands, but the researcher might still wish to test whether the mean percentages were different. You clearly cannot get more than 100% of the mosquitoes killed in any one trial, so you end up with very irregularly-shaped distributions, with most of the measurements clustered just below 100% and a drop to zero above 100%. A *t* test would be inappropriate in such a case. Instead, you should test for a difference in population medians using a Mann -Whitney *U* test.

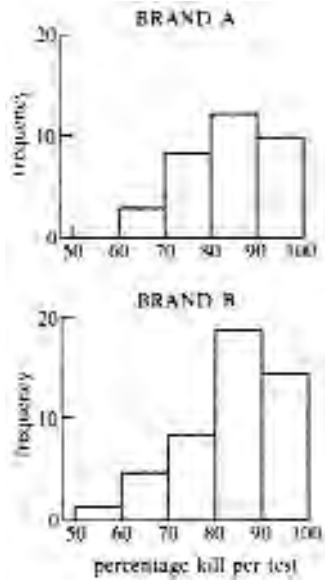
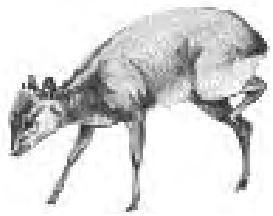


Figure 16.9 An example to illustrate a circumstance in which *t* tests cannot be used: the results from (hypothetical) experiments on two brands of insecticide.

- The variances of the populations from which your samples come should be equal. Again it is difficult to find out whether this is the case if you have small samples, because the variances of small samples even if taken from populations with equal variances can fluctuate widely. It turns out, however, that differences in the variances of the two populations do not severely affect the value of *t* provided that the two samples are roughly equal in size, so it is worth keeping your sample sizes as close to each other as possible.

- When both sample sizes are over 25 (and hence with 48 or more degrees of freedom), use the *z* test rather than the *t* test.



Blue duiker
Cepholophus monticola

The *t* test for matched samples.

When to use:

- If you wish to test for differences in population **means**.
- Provided you have two **matched** samples.
- Provided that your measurements are at the **interval** level.
- When your samples are small; that is when one or both have **fewer than 30** measurements.
- Provided that the measurements can be assumed to come from **normally** distributed populations.

Rationale of test

The rationale for this test is very similar to that for the *z* test for matched samples. Before reading any further make sure that you understand the rationale for the *z* test for matched samples, and for the *t* test for unmatched samples.

Just as the *t* test (rather than the *z* test) must be used on small unmatched samples, the same applies to small matched samples. Imagine that you have two matched samples of, say, 10 measurements each, with measurements at the interval level, and that you wish to test whether they come from populations with the same mean. In symbols

H_0: $\mu_A = \mu_B$

As in the *z* test for matched samples, you calculated the difference between each matched pair of measurements by subtracting one from the other. If the null hypothesis is true, you would expect the mean differences, $D!$, to be zero, with a standard error SE_D. For large samples, you can calculate the test statistic *z* from:

$$z = \frac{D!}{SE_D}$$

For small samples, you have to calculate the test statistic *t*, using the same equation:

$$t = \frac{D!}{SE_D}$$

Here, as in the *t* test

$$SE_D = \sqrt{\frac{S_D^2}{n}}$$

Summary

$D!$ = mean difference between pairs of sample measurements
n = number of measurements
S_D^2 = variance of differences in paired measurements

$$t = \frac{D!}{SE_D} \text{ where } SE_D = \sqrt{\frac{S_D^2}{n}}$$

Compare the calculated value of *t* with the critical value, for n - 1 degrees of freedom.

WORKED EXAMPLE

Procedure

1) Arrange your sample measurements in two columns (2 and 3), with each matched pair on the same row. Subtract each sample B measurement from its matched sample A measurement, writing the answers in column 4.

Calculate the mean *D*!, of the differences in column 4. Also, calculate the variance of the differences in column 4, s_D^2, using the procedure given in Box 4c.

(i) Square each difference in column 4, and put the results in column 5.

(ii) Add up the numbers in columns 4 and 5. This gives • (A-B) and • $(A-B)^2$

(iii) Divide the total in column 4 by the sample size. The number you have calculated is the mean of the differences between matched pairs of measurements, *D*!.

(iv) Square the total of column 4 to give $(\bullet\ (A-B))^2$

(v) Divide the figure from step (iv) by the sample size to give $(\bullet\ (A-B))^2/n$

(vi) Subtract the figure from step (v) from the total of column 5 to give $\bullet\ (A-B)^2 - (\bullet\ (A-B))^2/n$

(vii) Divide the figure from step (vi) by one less than the sample size

(viii) This is the sample variance of the differences in the paired measurements, s_D^2.

2) Calculate the estimated standard error of the mean difference in paired measurements, SE_D using the equation

$$SE_D = \sqrt{\frac{s_D^2}{n}}$$

3) Calculate *t* using the equation

$$t = \frac{D!}{SE_D}$$

4) Calculate the number of degrees of freedom.

5) Go to step 6 of the *t* test for unmatched samples and proceed exactly as for unmatched samples.

Calculation

(1) measurement no.	(2) sample A	(3) sample B	(4) difference (A-B)	(5) difference squared
1	16	12	+4	16
2	15	15	0	0
3	18	13	+5	25
4	17	18	-1	1
5	19	11	+8	64
6	18	15	+3	9
		total*	19	115

$\bullet\ (A-B) = 19 \qquad \bullet\ (A-B)^2 = 115$

$\bullet\ (A-B)/n = 19/6$
$= 3.1667$
$D! = 3.1667$

$(\bullet\ (A-B))^2 = 19^2$
$= 361$

$(\bullet\ (A-B))^2/n = 361 / 6$
$= 60.1667$

$\bullet\ (A-B)^2 - (\bullet\ (A-B))^2/n = 115 - 60.1667$
$= 54.8333$

$$\frac{\bullet\ (A-B)^2 - (\bullet\ (A-B))^2/n}{n-1} = \frac{54.8333}{5}$$
$= 10.9667$

$s_D^2 = 10.9667$

$$SE_D = \sqrt{\frac{10.9667}{6}} = 1.35195$$

$$t = \frac{3.1667}{1.35195}$$
$= 2.34230$
$= 2.34$ (to two decimal places)

n - 1 = 6 - 1 = 5

Do's and Don't's for z tests for matched samples

- The *t* test, as described here, can only be used on **matched** samples. Do not use it, therefore, on unmatched samples.

- When the number of degrees of freedom exceeds about 30, you can use the matched pairs *z* test instead, since the two tests give virtually identical results.

- Your measurements must be at the interval level and must be **normally distributed**. (Strictly, it is the population of differences between paired measurements that needs to be normally distributed). In practice, with small samples it is difficult to assess whether the population distribution is normal or not. You should, however, draw a line plot or histogram of the differences between your pairs of measurements. If their distribution appears to be very skewed, then do not use this test.

Analysis of variance

When to use:

- When testing for differences in the **means** of **more than two populations**, especially when samples from these populations have been subjected simultaneously to more than one kind of treatment.
- Provided that several assumptions about your measurements are satisfied. These are discussed in the references cited below.

Rationale of test

Each of the significance tests described in the earlier pages of this chapter is suitable only for situations where you wish to compare two samples. As many biological research projects require no more than this, these techniques are very useful. However, one is often faced with more complicated situations than simple comparisons between two samples, and more elaborate statistical techniques are needed. For example, you might wish to test whether three or more samples came from populations with t he same mean (or median): for instance whether the mean group sizes of monkeys (of a given species) were the same in samples collected from three sites that were logged at different times in the past. In addition, biologists often wish to investigate the effect of two or more factors simultaneously. For instance, you might wish to discover not only the effect of time since logging on monkey group size, but also the effects of different levels of hunting pressure and of the number of trees cut per unit area during logging (logging intensity), as well as the combined effects of these factors.

There is a very flexible and useful statistical technique called the analysis of variance (often abbreviated to ANOVA) that is suitable for such problems as these. The principle is quite simple, although the detailed way in which the technique is applied to each particular situation can be complicated. The principle is as follows. Suppose you have several samples of, say, monkey group size and that each sample (study area) has underwent logging at different times, has different hunting pressure and experienced different logging intensity. The group sizes in the study will, of course, vary: some of the variation will be caused by factors you have measured, while the rest will be caused by other factors which, from your point of view, are a nuisance in that they obscure the effects of the factors in which you are interested.

Maybe some group sizes vary because of differences in forest composition before logging in the various study sites, or because of a historical factor such as a disease that affected some sites but not others; and so on. In the analysis of variance, you work out how much variation there is in the samples taken together. The measure of the variability is the variance (see Box 4c). That is, you calculate the total variance present in your measurements. You then work out what proportion of this variance is due to each of the factors in which you are interested. What proportion is due to logging history? What to hunting pressure? What to intensity of logging? What to the combined effects of logging and hunting, or to hunting and logging intensity? And so on.

Each of these proportions forms the basis of a separate test statistic (F). Thus when you carry out an analysis of variance, you generate several test statistics: one for each factor or combination of factors in which you are interested. For each F that we calculate, we know the sampling distribution of F under the null hypothesis, which is that the group size of monkeys in areas which have experienced different logging histories, hunting pressure and logging intensity do not differ. By carrying out a single analysis of variance, therefore, we can carry out a whole series of significance tests, and so assess simultaneously the relative importance of the several factors, acting either singly or in combination, on the measurement in which we are interested.

There are many different versions of the ANOVA, each appropriate to a different situation. Given the complex nature of this test we do not describe it in detail here. Two text books which give good descriptions of this test are:

N. T. J. Bailey (1981) *Statistical methods in biology,* 2nd edn. Hodder and Stoughton.
R. E. Parker (1979) *Introductory statistics for biology*, 2nd edn. Edward Arnold.

Other tests (such as Kruskal-Wallis and Friedman tests - see Siegel & Castellan, 1988) should be used with small samples from populations which are not normally distributed.

When to use:

- When you wish to test for an association between **two or more** sets of measurements, each at the **categorical** level.
- Provided that each measurement is **independent** of the others.
- Provided that the expected **frequencies**, under the null hypothesis of no association, are **larger than five**.

The c^2 test for association

Rationale of test

Many of the features of animal and plant life that interest biologists and protected area managers are measured at the categorical level. For example, before starting a conservation education programme in a protected area you might undertake a questionnaire on the attitudes of school children to wildlife - and then repeat it a year after the programme has been initiated. Suppose that you asked the question "Do you think elephants are good or bad?" before and after initiating the conservation education programme. Say you questioned 60 children initially and

You can arrange these numbers in a Table called a **Contingency Table**, as follows.

Are elephants good or bad?		**good**	**bad**	**total**
Before education programme		20	40	60
One year later		70	20	90
	total:	90	60	150

Just looking at these numbers suggests that there was a change in attitude as a result of the education programme. Can we test this statistically? The c 2 test - pronounced kai (to rhyme with sky) squared, where c is the Greek letter chi - can be used for this purpose. First you must set up a null hypothesis H_0 which proposes that there is *no* change in attitude as a result of the education programme. The alternative hypothesis H_1 is that there is a change (hence that the education programme has had an impact on childrens' attitudes to elephants).

In the present example, a total of 150 children were questioned. The next step is to work out how many children would have been expected to reply 'good' or 'bad' *if the null hypothesis were true.* We describe below how to calculate there 'expected figures', as they are called, but ask you to accept for the moment that these are numbers given in parentheses in the Table below. If you compare each expected figure with the actual figure next to it (usually referred to as the 'observed figure'), you will see that they are quite a long way apart.

Are elephants good or bad?		**good**	**bad**	**total**
Before education programme		20 (36)	40 (24)	60
One year later		70 (54)	20 (36)	90
	total:	90	60	150

Imagine, now, that the numbers of children replying 'good' and 'bad' in a school that had not been part of your education programme (but where questionnaires were filled out at the same one-year interval to act as a 'control' sample) turned out to be as in the Table below.

Are elephants good or bad?		**good**	**bad**	**total**
Before education programme		38 (40)	42 (40)	80
One year later		47 (45)	43 (45)	90
	total:	85	85	150

Casual inspection of these results suggests that there was little change in attitudes over the one-year period. We have again calculated how many children would have been expected to reply 'good' or 'bad' *if the null hypothesis were true.* Here these expected figures are much closer to the observed figures.

Comparing the data for children exposed to the education programme with those for the control sample, it seems that a large discrepancy between observed and expected figures is unlikely if the null hypothesis is true, whereas a small discrepancy is much more likely. We need, therefore, a method of saying just how big this discrepancy is. You might suggest that it would be a good idea to subtract each expected value from its observed value (or vice versa), and add up all the differences of the result. Unfortunately, the result is always zero.

To overcome the problem that some differences are positive and others negative, we square them. Hence our control sample above gives:

	observed minus expected $(O - E)$	**(observed minus expected)^2** $(O - E)^2$
	-2	4
	+2	4
	-2	4
	+2	4
total:	0	16

One further refinement is needed. A difference of 100 between an observed number (O) and an expected number (E) would seem to be much more extreme when the observed and expected values are, say, 105 and 5 than when they are 1100 and 1000; so you need to put each difference into perspective, as it were. This is achieved by dividing each (observed minus expected)^2 value by the expected value. In symbols,

$(O - E)^2 / E$

(observed minus expected)^2 $(O - E)^2$	**(observed minus expected)^2 / expected** $(O - E)^2 / E$
4	4 / 40 = 0.1
4	4 / 40 = 0.1
4	4 / 45 = 0.0888889
4	4 / 45 = 0.0888889
	total: 0.3777778

If you add each of the $(O - E)^2 / E$ values together, you end up with a single number that indicates the overall discrepancy between the observed and expected figures: the bigger this number, the bigger the discrepancy. This number is called c^2. Thus:

$$c^2 = \bullet \quad \frac{(O - E)^2}{E}$$

c^2 is a test statistic whose sampling distribution under the null hypothesis is known. Although the examples here involve Contingency Tables with only two rows and columns, larger Tables can be treated in exactly the same way. The sampling distribution in fact differs with the numbers of rows and columns in the Contingency Table. To find the correct sampling distribution you calculate the **degrees of freedom**, using the rule:

degrees of freedom = (number of rows in the Contingency Table - 1) multiplied by (number of columns in the Contingency Table minus 1)

In the present example,

degrees of freedom = (2 - 1) x (2 - 1) = 1

There are Statistical Tables that give critical values of c^2 for different degrees of freedom and for various significance levels (e.g. Table A16.4 at the end of this chapter). You reject the null hypothesis in favour of the alternative hypothesis if your test statistic is greater than the critical value for the appropriate number of degrees of freedom at your chosen significance level.

There remains the problem of how to calculate the expected values (*E*). Refer back to the data for the children who participated in the education programme (above). Notice that of the 150 children questioned, 90 said that elephants were 'good' and 60 'bad'. If the null hypothesis is true, we should expect the answers 'good' and 'bad' in the ratio of 90:60 in the survey before and after the conservation education programme. Now 60 children were questioned before the education programme began, so we have to find out how many of these would have been expected to say 'good', to give a 90:60 ratio. The answer is 90 / 150 of 60 would have been expected to reply 'good' and 60/150 of 60 would have been expected to reply 'bad'; that is, 36 would have replied 'good' and 24 'bad' - as given in the parentheses in the first row of the Table. Similarly, 90 children were questioned one year after the conservation education programme was initiated, and again we have to find out how many of these would have been expected to say 'good', to give a 90:60 ratio. The answer is 90 / 150 of 90 would have been expected to reply 'good' and 60/150 of 90 would have been expected to reply 'bad'; that is, 54 would have replied 'good' and 36 'bad' - as given in the parentheses in the second row of the Table. To calculate expected values for observations in Contingency Tables with more than one row, therefore, the procedure is:

1) Identify the total for the column in which the first observed value lies.
2) Divide this by the grand total (i.e. the number at the foot of the right-hand column in the tables above.
3) Multiply the result by the total for the row in which the observed value lies. This gives the expected value for this position in the Table. (*Note*: you will often find that the expected value is *not* a whole number. This is a consequence of the arithmetical procedure that you have to perform and is not a cause for alarm).
4) Repeat, to obtain the full set of expected values.
5) Check your answers: expected values along a row should add up to the row total, and expected values down a column should add up to the column total.

This procedure can be used on any size of Contingency Table

Summary

O = the observed number of items (in any one position in a Contingency Table)

E = the expected number for that position, under the null hypothesis (where E is calculated as ' column total divided by grand total multiplied by row total')

$c^2 = \bullet \frac{(O - E)^2}{E}$, where you sum $\frac{(O - E)^2}{E}$ over all positions in the Contingency Table

If there are *a* columns and *b* rows in a Contingency Table, then there are (*a* - 1) x (*b* - 1) degrees of freedom and the appropriate significance level, as given in Table A16.4 and *reject* the null hypothesis if

$c^2 >$ critical value.

WORKED EXAMPLE

Procedure

1) A common use of the c^2 test in ecology is to discover whether two (or more) species are distributed independently of one another, or whether, conversely, there is an **association** between the distribution of one and the other.

Calculation

In this example, we are to investigate whether two plant species, A and B, grow independently of one another or not.

Assume that we have sampled in a forest using plots of a given area. Each plot can be classified according to the presence or absence of one or both species.

a) both A and B present;
b) A present, B absent;
c) A absent, B present;
d) both A and B absent.

These four categories are arranged in a Contingency Table.

Set up a Contingency Table in which you clearly identify each of the mutually exclusive categories into which you are going to classify your results.

		species A	
		present	**absent**
species B:	present	(a)	(b)
	absent	(c)	(d)

2) Count how many instances of each category you observe in your sample.

Suppose that 225 plots were taken overall in the forest, and that the results were:

		species A		
		present	**absent**	**row total**
species B	present	92	58	150
	absent	41	34	75
		133	92	225

3) Calculate row and column totals for these observed frequencies (*O*).

sum of row totals: 150 + 75 = 225
sum of column totals: 133 + 92 = 225

4) Check that the sum of the column totals equals the sum of the row totals. If not re-do your calculations.

5) Calculate the expected frequencies (*E*) for each position in the Table under the null hypothesis of no association, using the rule (column total divided by grand total) multiplied by (row total).

	species A		
species B	**present**	**absent**	**row total**
present	$\frac{133}{225}$ x 150 =88.6667	$\frac{92}{225}$ x 150 =61.3333	150
present	$\frac{133}{225}$ x 150 =88.6667	$\frac{92}{225}$ x 150 =61.3333	150
total:	133.0000	92.0000	225

6) Check that the expected frequencies add up to the correct (observed) row and column totals, as in step 3. If not, re-do your calculations.

1st row: 88.6667 + 61.3333 = 150
2nd row: 44.3333 + 30.6667 = 75
1st column 88.6667 + 44.3333 = 133
2nd column 61.3333 + 30.6667 = 92

7) For each position, calculate

$$\frac{(O - E)^2}{E}$$

a) $\frac{(92 - 88.6667)^2}{88.6667}$ = 0.125311

a) $\frac{(58 - 61.3333)^2}{61.3333}$ = 0.181156

a) $\frac{(41 - 44.3333)^2}{44.3333}$ = 0.250622

a) $\frac{(34 - 30.6667)^2}{30.6667}$ = 0.362311

total: 0.9194000

8) Add together all of the figures that you calculated in step 7. This is your test statistic, c^2, where

$$c^2 = \bullet \quad \frac{(O - E)^2}{E}$$

c^2 = 0.919400

= 0.919 (to three decimal places)

9) Calculate the degrees of freedom using the rule '(number of columns minus one) multiplied by (number of rows minus one)', i.e.

$$(a - 1)(b - 1)$$

degrees of freedom = (2 - 1) x (2 - 1)
= 1

10) Compare your test statistic with the critical value given in Table A16.4 (at the end of this chapter) for the appropriate degrees of freedom and significance level. If your test statistic is larger than the critical value, reject the null hypothesis and accept the alternative hypothesis.

From Table A16.4, the critical value of c^2 for 1 degree of freedom and a 5% significance level is 3.841. The calculated value is smaller than this. So we **cannot** reject the null hypothesis, and we accept that there is **no association** between the distributions of species A and B.

Do's and don't's for c^2 tests of association

- Make sure that you work with sufficiently large numbers. A generally accepted rule of thumb is that none of the expected values under the null hypothesis should be less than five. If you have a large Contingency Table with several rows and columns, and it turns out that some of your expected values *are* smaller than five, you may be able to overcome this problem by combining some of your rows or columns; that is, by lumping together some of your categories. You should only do this if you think it makes biological or socio-economic sense to do so.
- Make sure each item that you classify is independent of every other one. For example, in the worked example on plant associations, we assumed that the category into which you put any one plot was independent of the category into which you put any of the others. If your plots were, in fact, sub-divisions of one long, continuous botanical transect, this is not the case because adjacent 'plots' are likely to be located in similar vegetation and are more likely to be assigned in the same category. The important point is to ensure that any one item that you sample is not in a position to influence the measurements you take from any other items in the sample.

- Make sure that you only use the c^2 test for associations when you are dealing with categorical variables. That is, you must be able to say to yourself for each item in your samples 'Into which category do I put this?', and having done that, you add one to the frequency of items in the category that you have identified. The calculations must then be carried out using the frequencies themselves. A common error is to calculate a table of percentages from the frequencies, and then to apply the test to the percentages.

- It is possible to use other versions of the c^2 test in certain situations which do not lead to a contingency table in the way described. But there is a situation you should be very cautious about. Suppose, for example, you were to collect insects of a given species from one quadrat in each of three sites, in an attempt to see if there is a difference in abundance of the species between sites. Your results might be as follows:

	Site		
	1	**2**	**3**
no. of insects	37	100	15

The data are in a table very like a contingency table, except that is has only one row. It would be possible to use a version of the c^2 test to investigate the null hypothesis that the abundance of the species is the same at each site (though the details are not the same as for the c^2 test for association). With the data given above, the null hypothesis would be rejected. However, this does not actually tell you anything very useful. It might have occurred because there really is a difference in the abundance of this insect between the three sites. But it might instead have occurred because the insects within each site do not fly around independently of each other, but instead tend to cluster together. With many species of insect, such clustering is very likely. Maybe there is no overall difference in abundance between the sites, but by chance the quadrat in site 2 happened to include a cluster of insects. The problem arises in the *design* of the study rather than in the statistical analysis of the data. If you really wished to discover whether there was a difference in abundance of this insect between site 1 and site 2, for instance, you would have to take several quadrats from each area, count the number of insects in each quadrat, and hence obtain several measurements from each area. You could then treat these counts as samples of measurements at an interval level, and carry out an appropriate significance test, such as a Mann-Whitney U test or a t test.

Often, if an ecological study leads to data in the form of a table of counts with only one row, there will be similar problems in interpreting the results, because the design of the study is inadequate. For this reason we do not describe how to analyse such data here. If your study is going to provide data of this sort, we advise you to discuss the design of the study with a statistical expert before you go ahead with data collection.

The Spearman rank correlation coefficient

When to use:

- When you wish to discover whether there is an **association** between two sets of measurements, that are at the **ordinal** or **interval** level.
- When you have samples containing **from 7 to 30 pairs** of measurements.
- When these measurements are reasonably **scattered**, and do not have a U-shaped or inverted U-shaped relationship with one another.

Rationale of test

It often happens in biological research that you collect two sets of measurements from a sample (i.e. a pair of measurements from each member of the sample) and that you wish to discover how strongly these two sets of measurements are associated with one another. For example, Box 13n gives examples such as whether the number of elephant dung piles seen on transects and recce surveys show a consistent relationship. There are a number of statistical techniques, known as **correlation** techniques, whose function is to assess the strength of such an association. Each of these techniques ends up by calculating a single number, called a **correlation coefficient**, which indicates the strength of the association. The coefficient lies within the range of +1 to -1.

A coefficient of +1 indicates a perfect positive correlation between the two sets of measurements. For the Spearman rank correlation coefficient, described here, this means the following: if you were to plot a scatter diagram of your two sets of measurements, they would either be in a straight line, rising from low values for both sets of measurements on the left of the diagram to high values for both on the right, or, an *increase* in the value of one set of measurements would always be accompanied by an *increase* in value in the other. Figure 16.10a shows three examples of perfect positive correlations. Notice that it does not matter how steeply the line rises or how curved it is: the correlation is perfect so long as both sets of measurements increase together.

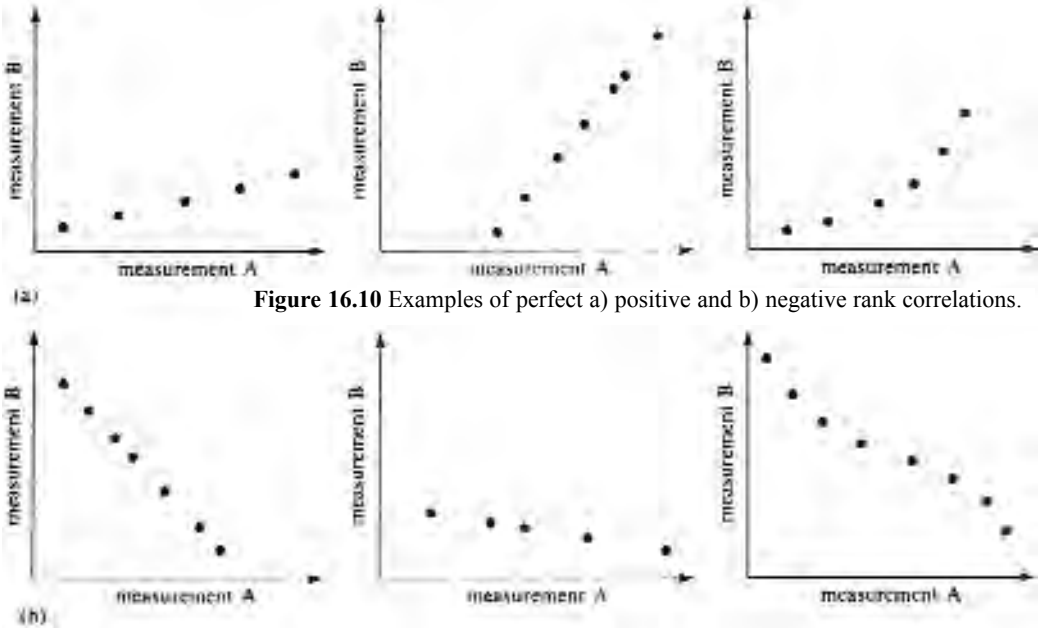


Figure 16.10 Examples of perfect a) positive and b) negative rank correlations.

A Spearman rank correlation coefficient of -1 indicates a perfect negative correlation. The higher one of the measurements is, the lower is the other measurement of the pair. Again, if you plot a scatter diagram, all the points either lie on a straight line - but this time with the line running from top left to bottom right - or show that an *increase* in one set of measurements is always accompanied by a *decrease* in the other. Again, it is not how steeply the line slopes that is important, but the fact that one set of measurements increases while the other decreases (Figure 16.10b).

It is very rare, in biological research, to find perfect correlations. Therefore correlation coefficients are seldom as strongly positive as +1 or as strongly negative as -1. There are several reasons for this:

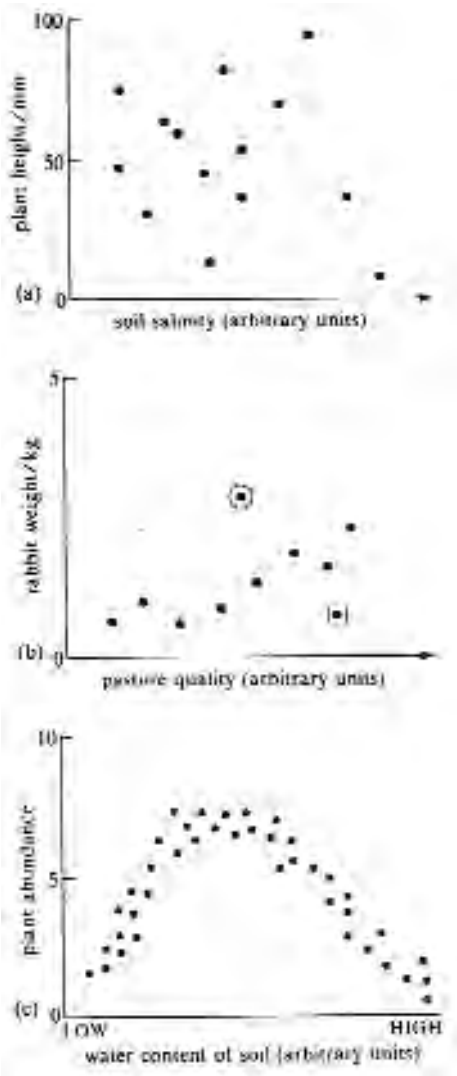


Figure 16.11 Three hypothetical examples illustrate correlation problems: a) absence of correlation; b) a positive correlation is partially obscured by random variation; a relationship in the shape of an inverted U.

1) There may be no association between two sets of measurements, so that the correlation coefficient is zero. Figure 16.11a shows such a situation. Here, both soil salinity and the height of a species of plant found growing in the soil vary considerably, but the variation in the two sets of measurements is independent; hence the points in the scatter diagram are dotted around without any clear pattern.

2) There may be an association, but it is obscured by the random variation that is commonly found in biological measurements. Figure 16.11b shows a situation where there is a positive correlation between rabbit body weight and the quality of pasture on which the rabbits feed, but that the points are scattered about a straight line rather than lying directly on it. Possibly the rabbit indicated by the circled point, which lies above the line, was unusually heavy because it was pregnant, whereas the rabbit indicated by the point enclosed by a square was unusually light because of disease. The correlation coefficient in this example turns out to be +0.65.

3) There may be an association, that is not a simple one. If you were to plot a scattergram of plant abundance against water content of the soil, you might get a picture similar to Figure 16.11c. The plant in question does not flourish in very dry or very wet soil, and its numbers are highest at intermediate levels of water content. There *is* a relationship between plant numbers and soil water content here, but the relationship is neither a simple straight line nor a simple curve which steadily rises or falls. Rather, it is a curve of a shape often called an 'inverted U'. Commonly-used correlation techniques will not detect a relationship if the curve is either U-shaped or an inverted U, because the coefficients produced turn out close to zero.

4) There may be an association, but all your measurements are so highly clustered together that a correlation analysis will not reveal the association. For example, there might be a strong association between gorilla nest counts on transects and recces which would be perfectly clear if you had sampled in strata with a wide enough range of densities. However, if sampling is concentrated in just one stratum, the association might be difficult to detect (see Figure 16.12).

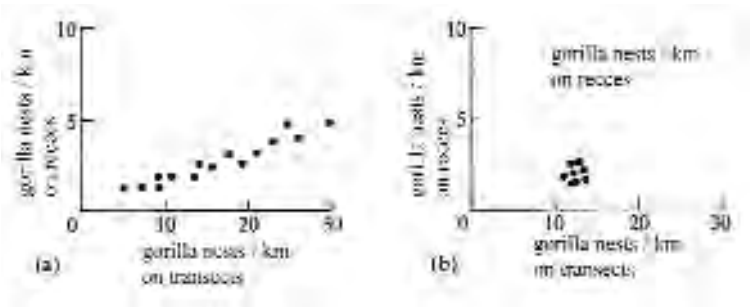


Figure 16.12 The association between nest counts on transects and recces is much clearer when the measurements are taken over a wide range (a) than a narrow range (b) of density.

To discover which of the situations 1-4 applies, it is important to draw a scattergram of your results in order to see what they look like. Only if they are not in the shape of a U or an inverted U should you go ahead and calculate a correlation coefficient.

In the Spearman rank correlation coefficient, described here, you arrange your two sets of measurements in two columns, side by side (Columns 2 & 4 below). Work out the ranks of the measurements in each of these columns, entering the results in columns 3 & 5 respectively. Suppose, for example, that you wished to discover the extent to which the maximum lengths and weights of *Irvingia gabonensis* seeds are correlated; the two sets of measurements might appear as follows:

(1) seed no.	(2) length / mm	(3) rank of length	(4) weight / g	(5) rank of weight
1	40.5	3	39.3	2
2	43.2	4	42.0	4
3	38.3	1	41.6	3
4	45.3	5	43.8	5
5	39.7	2	38.9	1

Looking first at lengths, seed number 3 has the smallest length, so is ranked number 1, seed number 5 has the second smallest, and is ranked number 2, and so on. Ranking the seed weights, seed number 5 is the lightest, and so is ranked number 1; seed number 1 is the second lightest; and so on. Next, you calculate the difference in ranks for each pair of measurements, by subtracting each rank of weight from the corresponding rank of length. The results are listed in Example 1 on Page 386.

If there were a perfect positive correlation between the two measurements, then the seed which ranked 1 for length would rank 1 for weight, the seed ranked 2 for length would rank 2 for weight, and so on. The difference in ranks would, as a result, all be zero, as in example 2.

If, by contrast, there was a perfect negative correlation, then the seed that ranked 1 for length would rank 5 for weight, the seed ranked 2 for length would rank 4 for weight, and so on, as in example 3. The differences in ranks would be large: in fact they would be as large as it was possible to achieve for that particular sample size.

Example 1 Ranks of the lengths and weights of five *Irvingia gabonensis* seeds.

seed no.	rank of length	rank of weight	difference
1	3	2	1
2	4	4	0
3	1	3	-2
4	5	5	0
5	2	1	1
		total	0

Example 2 Positive correlation of ranks.

seed no.	rank of length	rank of weight	difference
1	2	2	0
2	5	5	0
3	1	1	0
4	4	4	0
5	3	3	0
		total	0

Example 3 Negative correlation of ranks.

seed no.	rank of length	rank of weight	difference
1	3	3	0
2	1	5	-4
3	4	2	2
4	5	1	4
5	2	4	-2
		total	0

Notice that although in the last example the differences in ranks are bigger than in both of the previous two examples, the summed differences are all zero. This is no accident: if they did not add up to zero you would have ranked them incorrectly. Nonetheless, because the **magnitude** of the differences varies with the degree of association between the two sets of measurements, this suggests that if we could concentrate on the magnitude, eliminating the minus signs in some way, we would have a measure of association. This is achieved by squaring the differences:

	Example 1		Example 2		Example 3	
seed no.	difference	difference squared	difference	difference squared	difference	difference squared
1	1	1	0	0	0	0
2	0	0	0	0	-4	16
3	-2	4	0	0	2	4
4	0	0	0	0	4	16
5	1	1	0	0	-2	4
total	0	6	0	0	0	40

The totals of the squared differences now reflect (in some way) the degree of association between the two sets of measurements. Where there is a perfect positive correlation the total is zero, where there is a weak positive correlation the total is 6, and where there is a strong negative correlation, the total is 40.

For brevity, we can denote the sum of the squared differences by the symbol $\bullet\ D^2$. $\bullet\ D^2$ is not itself a correlation coefficient: in the three examples here it varies from 0-40, rather than from +1 to -1.

Suppose we ask the question 'How big is $\bullet\ D^2$ in relation to the maximum value ($\bullet\ D^2_{max}$) it could take?' In example 1, $\bullet\ D^2$ is 6, and we know that the maximum possible value of $\bullet\ D^2$ is 40, so $\bullet\ D^2$ is 6/40 = 0.15 times the maximum value, $\bullet\ D^2_{max}$. In Example 2, $\bullet\ D^2$ is 0, which is 0/40 = 0 times the maximum value. In Example 3, $\bullet\ D^2$ is 40, which is 40/40 = 1 times the maximum possible value. This is set out below:

	Example		
	1	**2**	**3**
$\bullet\ D^2$	6	0	40
$\bullet\ D^2_{max}$	40	40	40
$\dfrac{\bullet\ D^2}{\bullet\ D^2_{max}}$	$\dfrac{6}{40} = 0.15$	$\dfrac{0}{0} = 0$	$\dfrac{40}{40} = 1$

This ratio $\bullet\ D^2\ /\ \bullet\ D^2_{max}$, looks more promising as a correlation coefficient: at least the numbers are in the range of 0 to 1, instead of 0 to 40. But we are not quite there yet, because for a perfect positive correlation the value of $\bullet\ D^2\ /\ \bullet\ D^2_{max}$ is 0, rather than the 1 we require.

To turn $\bullet\ D^2\ /\ \bullet\ D^2_{max}$ into a number with the required properties, you multiply it by two and subtract the result from 1:

$$\text{correlation coefficient} = 1 - 2(\bullet\ D^2\ /\ \bullet\ D^2_{max})$$

Hence our calculated values become:

	Example		
	1	**2**	**3**
$\dfrac{\bullet\ D^2}{\bullet\ D^2_{max}}$	6	0	40
$1 - \dfrac{2\bullet\ D^2}{\bullet\ D^2_{max}}$	1-(2 x 0.15) = 0.7	1 - (2 x 0) = 1	1 - (2 x 1) = -1

It can be seen that this manipulation produces numbers that range from +1 where there is a perfect positive correlation to -1 where there is a perfect negative correlation. There is one further step to simplify calculations of $\bullet\ D^2_{max}$.

It would be tedious to work out the maximum possible value for $\bullet D^2$ for every sample size by calculating this value when the two sets of measurements are perfectly negatively correlated (as in Example 3). Fortunately it is possible to show mathematically that:

$$\bullet D^2_{max} = n(n^2 - 1) / 3$$

where n is the sample size. (If you do not believe this, try it out for yourself on a few sample sizes). Substituting for $\bullet D^2_{max}$ in the equation:

$$\text{correlation coefficient} = 1 - \frac{2 \bullet D^2}{\bullet D^2_{max}}$$

this gives what is known as the Spearman rank correlation coefficient, r_s:

$$r_s = 1 - \frac{6 \bullet D^2}{n(n^2 - 1)}$$

Usually in biological research, one is not so much interested in the precise value of r_s as in whether (or not) r_s is close to zero. If r_s is close to zero, you accept the null hypothesis that there is no correlation between the two features under investigation. If, by contrast, r_s is sufficiently far from zero, then you reject the null hypothesis, and accept the alternative hypothesis that there is a correlation between the two features - positive or negative according to whether r_s is positive or negative. The coefficient r_s can therefore be used as a test statistic in a significance test. The sampling distribution of r_s under a null hypothesis of no correlation is known, and Table A16.5 at the end of this chapter shows the critical values for r_s for different sample sizes and different levels of significance. You reject the null hypothesis in favour of the alternative hypothesis at the appropriate level of significance if the calculated value of r_s is greater than or equal to the critical value given in the Table.

To illustrate the use of the Table, suppose that your sample size is 18 and that your calculated value of r_s is 0.522.

Reading along the row for sample sizes of 18 until you come to the column for a 5% significance level, the critical value for r_s turns out to be 0.475. For a 2% level of significance the critical value is 0.564. The calculated value of r_s lies between these two values, and so one can reject the null hypothesis in favour of the alternative hypothesis at the 5% level but not at the 2% level of significance.

Summary

D = the difference in rank between a pair of measurements

$\bullet D^2$ = the sum of the squares of all such differences

n = the sample size: that is, the number of pairs of measurements

r_s = Spearman's rank correlation coefficient

$$= 1 - \frac{6 \bullet D^2}{n(n^2 - 1)}$$

WORKED EXAMPLE

Procedure

1) Arrange your two sets of measurements side by side in two columns, so that the two measurements from a given pair lie on the same row.

Calculation

seed no.	length / mm	weight / g
1	20.5	19.3
2	23.2	22.0
3	18.3	21.6
4	25.3	23.8
5	19.7	18.9
6	25.3	24.7
7	15.6	18.2
8	20.8	19.5

2) Draw a scatter diagram of one set of measurements against the other (Fig. 16.13)

If the points appear to lie on a U-shaped or inverted U-shaped curve (Fig. 16.11c) or if they are all squashed together (Fig. 16.12b) do not proceed with the correlation test: simply describe the apparent relationship between the two sets of measurements verbally. Otherwise proceed to step 3.

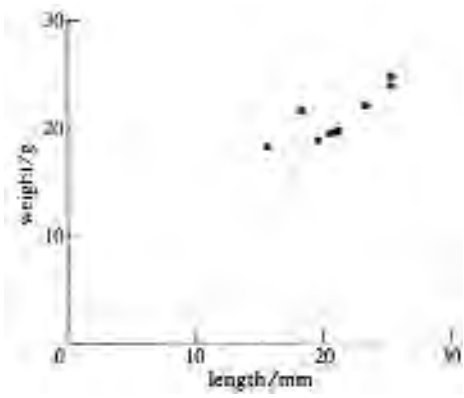


Figure 16.13. The relationship between weight and length of *Coula edulis* seeds from the table above.

3) Rank each measurement in column 2. Give the smallest measurement rank 1, the next smallest rank 2, and so on, and enter them in column 3. If some of the measurements have the same value give them the average of their ranks (for example, 4 and 6 are both 25.3 mm long and would occupy ranks 7 and 8 if they were not quite identical, so give them both the average of these ranks of 7.5).

Do the same for the measurements in column 4, entering the ranks in column 5.

(1) seed no.	(2) length / mm	(3) rank of length	(4) weight / g	(5) rank of weight
1	20.5	4	19.3	3
2	23.2	6	22.0	6
3	18.3	2	21.6	5
4	25.3	7.5	23.8	7
5	19.7	3	18.9	2
6	25.3	7.5	24.7	8
7	15.6	1	18.2	1
8	20.8	5	19.5	4

4) Subtract each rank in column 5 from the corresponding rank in column 3.

(1) seed no.	(3) rank of length	(5) rank of weight	difference in ranks (length minus weight)
1	4	3	1
2	6	6	0
3	2	5	-3
4	7.5	7	0.5
5	3	2	1
6	7.5	8	-0.5
7	1	1	0
8	5	4	1
		total	0

5) Add up the differences; if the total is not zero, check your calculations, since you have made a mistake.

6) Square each of the differences.

seed no.	difference in ranks	difference squared
1	1	1
2	0	0
3	-3	9
4	0.5	0.25
5	1	1
6	-0.5	0.25
7	0	0
8	1	1
	total (• D^2)	12.5

7) Add up the squared differences. The total is • D^2.

8) Work out the sample size, n (the number of pairs of measurements).

$n = 8$

9) Work out $n(n^2 - 1)$

$8(8^2 - 1) = 8 \times 63 = 504$

10) Multiply • D^2 by 6 and divide by $n(n^2 - 1)$.

$$\frac{12.5 \times 6}{504} = 0.14881$$

11) Subtract the result from 1 to find r_s;

$$r_s = 1 - \frac{6 \bullet D^2}{n(n^2 - 1)}$$

$$\begin{aligned} r_s &= 1 - 0.14881 \\ &= 0.85119 \\ &= 0.851 \text{ (to three decimal places)} \end{aligned}$$

12) Compare r_s with the critical values in Table A16.5 at the end of this chapter for a test with a sample size of n.

Critical value of r_s with $n = 8$ is 0.833 at the 2% significance level and 0.881 at the 1% significance level.

The calculated value of r_s lies between these two values, so we reject the null hypothesis of no correlation between weight and maximum length at the 2% (but not at the 1%) level of significance.

Do's and don't's for the Spearman rank correlation coefficient

- Avoid working with sample sizes of less than 7 or 8, since random variation is more likely to obscure a genuine association with very small samples. With large samples the procedure of working out the ranks becomes very tedious, and so it is best to avoid samples of more than 30.
- Do not use the Spearman rank correlation coefficient if (i) the relationship between the two sets of variables appears to be U-shaped, or (ii) if the amount of variation in one or both sets of measurements is very small.
- Be careful how you interpret the results of your analysis. If you show a correlation exists, this does not explain *why* it exists. For instance, in our example of *Coula edulis* seed weights and lengths, the measurements show a significant positive correlation. Say a similar correlation had been found between body weight and wing span for moths. There could be three potential explanations for this:
  i) big bodies are responsible for the growth of big wings;
  ii) big wings are responsible for the growth of big bodies;
  iii) some other factor promotes both big wings and big bodies, and body weights and wing spans do not have any direct effect on one another.

  But correlation does not imply causation. There may be no biological explanation for the correlation in terms of cause and effect.

  Use of a correlation cannot tell us which of these three alternatives is correct, and you would have to carry out further investigations to find the answer. Remember, therefore, that there are in general four possible explanations for any correlation that you reveal.

Explanation 1:	A causes B
Explanation 2:	B causes A
Explanation 3:	A and B independently depend upon C
Explanation 4:	Coincidence

Male bushbuck
Tragelaphus scriptus

Linear regression

When to use:

- When you have two sets of measurements at the **interval** level, which are arranged in **pairs**.
- When you wish to use the measurements in **one set** to **predict** values in **the other**.
- Provided that any underlying relationship between the two sets of variables is **linear**, that both sets of measurements are reasonably **scattered**, and that the **residuals** (see below) do not vary with the size of the measurements belonging to the set from which the predictions are to be made.

Rationale of test

Before reading further, make sure you understand the previous section on correlation.

Linear regression is similar to correlation, in that both deal with two sets of measurements, and both are concerned with the association between them. There is, however, an important difference. When you calculate a correlation coefficient, you are interested in discovering *how strong* the association is, and this strength is indicated by the size of the correlation coefficient. In regression analysis, by contrast, you are interested in *predicting* the values of measurements in one of your sets, from values of measurements in the other set. In the previous section on correlation, the worked example on *Coula edulis* seed weight and length showed that the Spearman rank correlation coefficient r_S was 0.851, indicating a strong association between the two features. Linear regression applied to the same measurements would help us to answer the question 'If I have a seed length x mm, what is the weight likely to be?' or the converse, 'If I have a seed of weight y g, what is the length likely to be?'

To take a different example, you might want to know how temperature affects plant growth. You might conduct experiments on three different species, growing several individuals of each plant under a variety of different temperature regimes for a given length of time. Suppose, for the moment, that you obtained the unlikely result that in each case there was a perfect positive correlation between temperature and plant growth, so that when you plotted the increases in plant height against temperature for each species, you obtained the results shown in Figure 16.14. In each case, the correlation is perfect and the line joining the points is straight. However, the lines differ one from another. In species 1, there is no growth at all if the temperature is 0ºC, and a higher temperature only slightly increases the growth rate. In species 2, again there is no growth if the temperature is zero, but a higher temperature has a much larger effect on growth. In species 3, by contrast, there is some growth even at OºC, but the increase in growth is only slight when the temperature is higher.

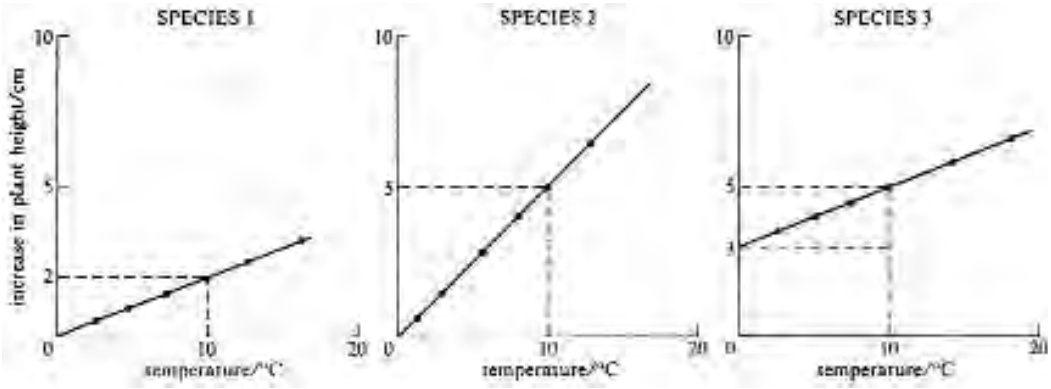


Figure 16.14 Three different relationships between temperature and plant growth

If you knew the temperature at which a plant of species 1 was to be grown, you could predict exactly how much it would grow during the course of the experiment, and similarly with a plant from species 2 or species 3. You would simply find the point on the temperature scale for the appropriate species, move vertically upwards from that point until you came to the sloping line, and then read off the point on the vertical scale that you had reached. Note, however, that you have to use a different graph for each species: there is a perfect correlation in each instance, but the nature of the straight line relationship differs in each.

Referring again to Figure 16.14 you can see that the two features in which the sloping lines differ are:

i) their slope - for one species it is steep, for the other two it is shallow;

ii) the point at which they cross the vertical axis - for two species it is at zero, for one it is at 3 cm.

When there is a straight line relationship between two sets of measurements, you can define exactly where the line goes provided you know the slope, and the point at which the line crosses the vertical axis. To measure the slope, you ask the question 'By how many units does the measurement plotted along the vertical axis change for every unit that the measurement plotted along the horizontal axis changes?' If, for example, plant growth increases by 2 cm for every degree Celsius rise in temperature, the slope is 2; if the increase in plant growth is 3 cm per degree, the slope is 3; and so on.

The rule for predicting a measurement on the vertical axis from a measurement on the horizontal axis is therefore as follows: take your horizontal axis measurement, multiply it by the slope, and add to the result the value on the vertical axis at which the sloping line crosses that axis. In symbols:

Let x = horizontal axis measurement (e.g. temperature);
y = vertical axis measurement (e.g. increase in plant height);
m = slope;
c = point at which sloping line crosses vertical axis.

Then, $y = mx + c$

Figure 16.14 shows the slopes and values of c for the three plant species growing at different temperatures, and illustrates how you can use the equation $y = mx + c$ to predict plant growth from temperature.

There is no reason, in principle, why you should wish to use this prediction in only one direction. You might, for some reason, wish to use your plants as a thermometer, and predict temperature from plant growth. To do so, you would simply plot plant lengths along the horizontal axis, temperatures along the vertical, and proceed exactly as above.

Perfect straight line relationships are very rare in biological research. More usually, points plotted on a graph are scattered to a greater or lesser extent about a line, the scatter being the result of random variation.

When we carry out linear regression, we assume that there really is a straight line relationship between the two features under investigation (which is why the technique is called *linear* regression), and we estimate the position of the best-fit line through the scattered points (see Figure 16.15). But how do we decide what *is* the best line? Intuitively, it seems that a good line will run close to all of the points, whereas a bad one will be a long way from them. Judged on this criterion, line A in Figure 16.15 is much better than line B, and C is marginally better than B.

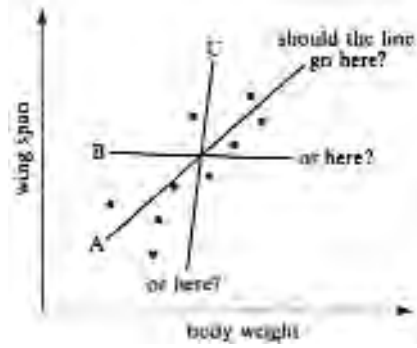


Figure 16.15 The problem of where best to draw the straight line representing the relationship underlying the two sets of measurements.

Clearly there needs to be an objective way of measuring how close a given line runs to all the points in a scattergram. The method used is as follows. Imagine that a scattergram of, say, plant height against temperature looks like Figure 16.16, and that you draw a straight line through the scattered points. For a given temperature, x, the line predicts that the corresponding plant height will be y. Suppose however, that a plant that has been grown at temperature x is in reality much taller than predicted - perhaps it has a height of y'. The vertical distance $y' - y$ between the point and the line, shown by the double-headed arrow in Figure 16.16, tells us how big is the difference between the observed and the predicted heights of the plant. Put differently, the vertical distance tells us how large is the *error* in the prediction made by the line.

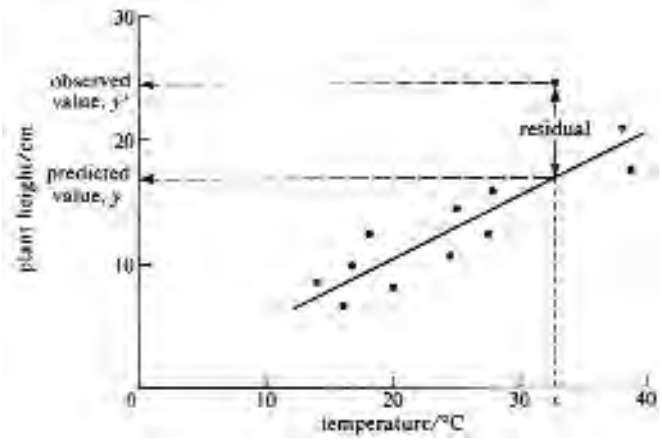


Figure 16.16 A scattergram of plant height against the temperature at which each individual was grown. The dots depict data points: the diagonal presents a summary of the general relationship between plant height and temperature.

The technical term for this error is the **residual**. If you look at all of the points in Figure 16.16, you will see that some lie above the line and some below: in some the residuals are positive and in others negative. To measure how good a fit the line achieves, it is necessary to take all the residuals into account. If you were simply to add them all up, the positive residuals would tend to cancel out the negative ones; so to overcome this problem each residual is squared. (When you multiply a negative number by itself, the result is a positive number, and hence squaring the residuals produces a positive set of numbers). You can then add together all the squared residuals, and this total gives a measure of how well the line fits the points in the scattergram: the smaller the total, the better the fit. The line with the best fit of all is the one that gives the smallest total value for the squared residuals.

There is a technique for finding this straight line, often referred to as the 'least squares' method of regression analysis. It allows you to calculate the slope (m) of the 'best fit' line and the point ($x = 0$, $y = c$) at which it crosses the vertical axis, from the data in the scattergram. That is, the technique enables you to identify the line that gives the smallest sum of squared residuals. The calculation can be time-consuming, and you are advised to use a calculator or computer program if possible. We shall not describe the rationale behind the technique in this chapter, but simply summarise how to calculate m and c as follows:

Summary

Let the sample plotted along the horizontal axis be called sample X. Let it have a sample mean of x!, and let any one measurement in the sample be called x.

Let the sample plotted along the vertical axis be called sample Y. Let it have a sample mean of y!, and let any one measurement in the sample be called y. The measurements in the samples must be arranged in their pairs, just as when calculating a correlation coefficient.

The slope of the requires straight line is given by

$$m = \frac{\bullet\ (x - x!)\ (y - y!)}{\bullet\ (x - x!)^2}$$

The point at which this line crosses the vertical axis is given by

$$c = y! - mx!$$

Having calculated m and c, you can draw the straight line with slope m which crosses the vertical axis at point c. You can also predict any particular value of sample Y from any particular value of sample X, by using either the line you have drawn or the equation $y = mx +$

WORKED EXAMPLE

Procedure

1) Arrange your two sets of measurements side by side in two columns, so that each pair of measurements lies on the same row. The measurements *from* which you wish to make predictions should go in the second column, the measurements *about* which you wish to make predictions should go in the fourth. (Leave columns 3 and 5 empty for the present).

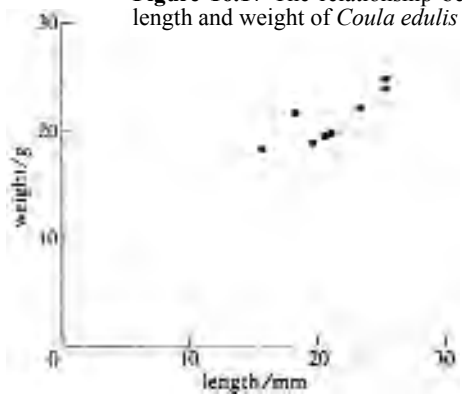
Calculation

We shall use the same *Coula edulis* seed measurements as for the correlation analysis above.

(1) seed no.	(2) length x / mm	(3)	(4) weight y / g	(5)
1	20.5		19.3	
2	23.2		22.0	
3	18.3		21.6	
4	25.3		23.8	
5	19.7		18.9	
6	25.3		24.7	
7	15.6		18.2	
8	20.8		19.5	
total	168.7		168.0	

2) Draw a scatter diagram of one set of measurements against the other (Fig. 16.17) The measurements *from* which you wish to make predictions should go along the horizontal axis; the measurements *about* which you wish to make predictions should go along the vertical axis. If the points appear to lie on a curve rather than a straight line, or if they all appear to be squashed together, do not proceed with the regression analysis but use another method. (See Do's and Don'ts for more details.)

Figure 16.17 The relationship between length and weight of *Coula edulis* seeds.



3) Calculate the mean for columns 2 and 4, then subtract each measurement in these columns from their means, entering the answers in columns 3 and 5 respectively.

mean length, $x! = 168.7 / 8$ = 21.0875 mm

mean weight $y! = 168.0 / 8$ = 21.0 g

Total columns 3 and 5. They should both be zero (or very close to it, since rounding errors may result in slight deviations from zero). If they are not, re-do your calculations.

(1) seed no.	(2) length x / mm	(3) x □x!	(4) weight y / g	(5) y □y!
1	20.5	-0.5875	19.3	-1.7
2	23.2	2.1125	22.0	1.0
3	18.3	-2.7875	21.6	0.6
4	25.3	4.2125	23.8	2.8
5	19.7	-1.3875	18.9	-2.1
6	25.3	4.2125	24.7	3.7
7	15.6	-5.4875	18.2	-2.8
8	20.8	-0.2875	19.5	-1.5
total	168.7	0.0000	168.0	0.0

4) Multiply each value in column 3 by the corresponding value in column 5. Enter the results in column 6, then total them.

	(3) (x □x!)	(5) (y □y!)	(6) (x □x!) □(y □y!)
	-0.5875	-1.7	0.99875
	2.1125	1.0	2.11250
	-2.7875	0.6	-1.67250
	4.2125	2.8	11.79500
	-1.3875	-2.1	2.91375
	4.2125	3.7	15.58630
	-5.4875	-2.8	15.36500
	-0.2875	-1.5	0.43125
total	0.0000	0.0	47.53000

This gives • $(x - x!)\,(y - y!)$

5) Square each value in column 3. Then add the results together and enter them at the foot of column 7.

(3) x □x!	(7) (x □x!)^2
-0.5875	0.34516
2.1125	4.46266
-2.7875	7.77016
4.2125	17.74560
-1.3875	1.92516
4.2125	17.74560
-5.4875	30.11270
-0.2875	0.082656
total	80.188750

This gives • $(x - x!)^2$

• $(x - x!)^2 = 80.18875$

6) Divide the number you calculated in step 4 by the number you calculated in step 5. This gives the slope m:

$$m = \frac{\bullet\,(x - x!)\,(y - y!)}{\bullet\,(x - x!)^2} \qquad m = \frac{47.53}{80.18875} = 0.592727$$

7) Multiply the slope m by the sample mean length x! which you calculated in step 3.

m x x! = 0.592725 x 21.0875
= 12.4991

8) Subtract this figure from y!, the sample mean weight that you calculated in step 3. This gives the value of c, the point at which the sloping line crosses the vertical axis:

$c = y! - mx!$

$c = y! - mx!$
= 21.0 - 12.4991
= 8.5009

9) Write down the equation relating y to x:

$y = mx + c$

$y = 0.59x + 8.5$ (to two significant figures)

10) Draw a straight line through your scattergram with slope m and crossing the vertical axis at c, by the following method:

i) Mark the value of c on the vertical axis. This gives you your first point on the line, the point $x = 0$, $y = c$. (Figure 16.18)

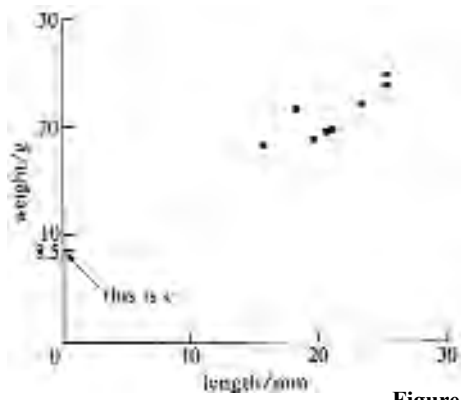


Figure 16.18

ii) Work out the value of y corresponding to a convenient large value of x near the right-hand end of the horizontal scale, using the equation

$y = mx + c$

Choose, say, $x = 30$. Then

$y = 0.59 \times 30 + 8.5$
= 17.70 + 8.5
= 26.2

iii) Enter this second point on the scatter diagram.

iv) Work out another value of y from a value of x somewhere in the middle of the horizontal scale. Enter this third point on the scatter diagram.

v) Join the three points you have entered with a straight line (Figure 16.19). (If the three points do not lie on a straight line, check your calculations.) This is called the *regression line of seed weight on seed length.*

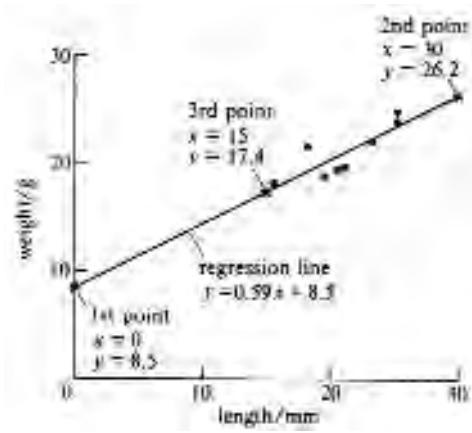


Figure 16.19

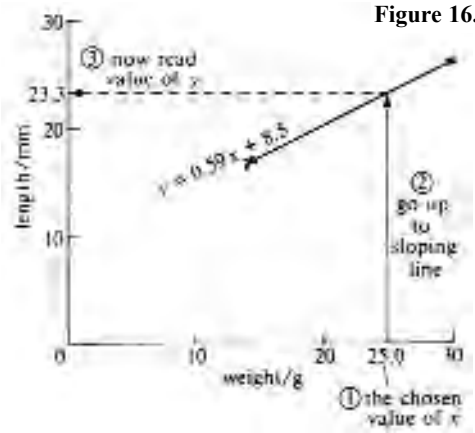
11) Predict the value of y corresponding to any value of x. There are two methods:

i) use the regression line to predict y

What is the value of y predicted from $x = 25.0$?

i)

Figure 16.20



From the regression line, if $x = 25.0$, then $y = 23.3$

ii) or use the equation $y = mx + c$ to predict y.

ii) Using the equation, if $x = 25.0$

$$\begin{aligned} y &= 0.59 \times 25.0 + 8.5 \\ &= 14.75 + 8.5 \\ &= 23.25 \end{aligned}$$

Do's and Don't's with Linear Regression

- Always plot a scattergram of your measurements before you carry out a regression analysis. The regression technique described here assumes that there *is* a straight line relationship between the two sets of measurements, so check from the scattergram that the relationship, if it exists, is at least not curved. (If you have small samples, it may not be possible to be sure of this.) The technique also makes certain assumptions about the distances that the points lie from the regression line (the 'residuals'). One of these assumptions is that the size of the residuals does not vary with the size of the measurements along the horizontal axis (see Figure 16.21a). You should check this visually from your scattergram.

  If the residuals at one part of the horizontal scale are clearly different in size from those at other parts of the scale, then do not use regression analysis (Figure 16.21b).

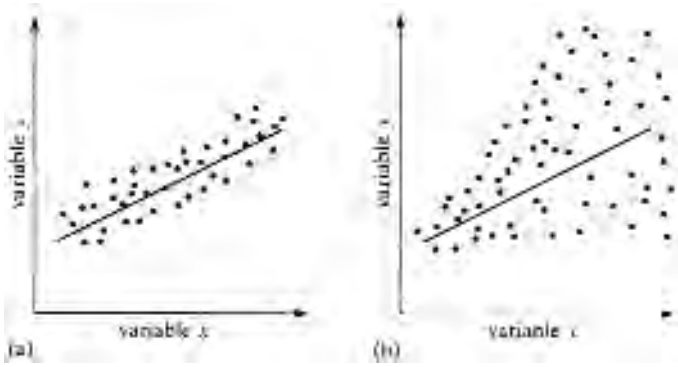


Figure 16.21

- Be very cautious about using your straight line to predict values of x which are a long way from the measurements you have made. For instance, you might think of using the straight line in Figure 16.19 to predict the weight of a seed 5 mm long ($x = 5$). But there is no evidence from the measurements in the table that a straight line accurately describes the relationship between length and weight for seeds this small. All the seeds on which the straight line is based are between 15 and 26 mm long. The straight line should not, therefore, be used for seeds much less than 15 mm long.
- There are many elaborations of the basic technique of linear regression. For example you can, if you wish, calculate confidence limits for the values of m (the slope) and c (the value of y when the sloping line crosses the vertical axis). You can, if you wish, do significance tests on whether, for example, your sample could have come from a population with a slope of a particular value, and so on. Consult a statistician or an advanced textbook if you wish to pursue any of these avenues.
- Do not carry out regression analysis unless your measurements are at the interval level.

A final note

This chapter, although relatively long, describes only a limited number of statistical tests and even for some of those described we have not been able to give all necessary details. We thought it vital to include this chapter as an introduction to statistical tests and because few people in Central Africa have access to up to date text books. However, we strongly recommend that you purchase one or more books which treat this key subject in greater detail. The following are some texts which are recommended:

R. E. Parker (1979) *Introductory statistics for biology*, 2nd edn. Edward Arnold.
N. T. J. Bailey (1981) *Statistical methods in biology,* 2nd edn. Hodder and Stoughton.
S. Siegel & N. J. Castellan (1988) *Nonparametric statistics for the behavioural sciences*. McGraw-Hill
R. C. Campbell (1989) *Statistics for biologists*, 3rd edn. Cambridge University Press.
J. Fowler & L. Cohen (1990) *Practical statistics for field biology*. John Wiley & Sons.
R. R. Sokal & F. J. Rohlf. (1995) *BIOMETRY - the principals and practice of statistics in biological research*. 3rd edn. W. H. Freeman & Co.
S. A. Glantz (1996) *Primer of Biostatistics*, 4th edn. McGraw-Hill (also available in French: *Introduction aux biostatistiques. McGraw-Hill [1998])
*J. Bouyer (1996) *Méthodes statistiques Médecine - Biologie*. Estem, Editions Inserm.
*B. Falissard (1998) *Comprendre et utiliser les statistiques dans les sciences de la vie*. 2nd edn. Masson, Paris.

* French language

Female sitatunga
Tragelaphus spekei

Table A16.1 **The Mann–Whitney U test**

Critical values of U at the 5% level. Reject your null hypothesis at the 5% level if your value of U is less than or equal to the tabulated value.

n_1/n_2	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15	16	17	18	19	20
1	—	—	—	—	—	—	—	—	—	—	—	—	—	—	—	—	—	—	—	—
2	—	—	—	—	—	—	—	0	0	0	0	1	1	1	1	1	2	2	2	2
3	—	—	—	—	0	1	1	2	2	3	3	4	4	5	5	6	6	7	7	8
4	—	—	—	0	1	2	3	4	4	5	6	7	8	9	10	11	11	12	13	13
5	—	—	0	1	2	3	5	6	7	8	9	11	12	13	14	15	17	18	19	20
6	—	—	1	2	3	5	6	8	10	11	13	14	16	17	19	21	22	24	25	27
7	—	—	1	3	5	6	8	10	12	14	16	18	20	22	24	26	28	30	32	34
8	—	0	2	4	6	8	10	13	15	17	19	22	24	26	29	31	34	36	38	41
9	—	0	2	4	7	10	12	15	17	20	23	26	28	31	34	37	39	42	45	48
10	—	0	3	5	8	11	14	17	20	23	26	29	33	36	39	42	45	48	52	55
11	—	0	3	6	9	13	16	19	23	26	30	33	37	40	44	47	51	55	58	62
12	—	1	4	7	11	14	18	22	26	29	33	37	41	45	49	53	57	61	65	69
13	—	1	4	8	12	16	20	24	28	33	37	41	45	50	54	59	63	67	72	76
14	—	1	5	9	13	17	22	26	31	36	40	45	50	55	59	64	67	74	78	83
15	—	1	5	10	14	19	24	29	34	39	44	49	54	59	64	70	75	80	85	90
16	—	1	6	11	15	21	26	31	37	42	47	53	59	64	70	75	81	86	92	98
17	—	2	6	11	17	22	28	34	39	45	51	57	63	67	75	81	87	93	99	105
18	—	2	7	12	18	24	30	36	42	48	55	61	67	74	80	86	93	99	106	112
19	—	2	7	13	19	25	32	38	45	52	58	65	72	78	85	92	99	106	113	119
20	—	2	8	13	20	27	34	41	48	55	62	69	76	83	90	98	105	112	119	127

Dashes indicate no decision possible at the stated level of significance.

Table A16.2 **The Wilcoxon matched pairs test**

Critical values of W at various significance levels. Reject the null hypothesis if your value of W is less than the tabulated value at the chosen significance level, for the number of non-zero differences N_D

	significance level			
N_D	10%	5%	2%	1%
5	0	—	—	—
6	2	0		
7	3	2	0	—
8	5	3	1	0
9	8	5	3	1
10	10	8	5	3
11	13	10	7	5
12	17	13	9	7
13	21	17	12	9
14	25	21	15	12
15	30	25	19	15
16	35	29	23	19
17	41	34	27	23
18	47	40	32	27
19	53	46	37	32
20	60	52	43	37
21	67	58	49	42
22	75	65	55	48
23	83	73	62	54
24	91	81	69	61
25	100	89	76	68
26	110	98	84	75
27	119	107	92	83
28	130	116	101	91
29	140	126	110	100
30	151	137	120	109

Male sitatunga
Tragelaphus spekei

Table A16.3 **The *t* test for matched and unmatched samples**

Critical values of *t* at various significance levels. Reject the null hypothesis if your value of *t* is larger than the tabulated value at the chosen significance level, for the calculated number of degrees of freedom.

degrees of freedom	significance level					
	20%	10%	5%	2%	1%	0·1%
1	3·078	6·314	12·706	31·821	63·657	636·619
2	1·886	2·920	4·303	6·965	9·925	31·598
3	1·638	2·353	3·182	4·541	5·841	12·941
4	1·533	2·132	2·776	3·747	4·604	8·610
5	1·476	2·015	2·571	3·365	4·032	6·859
6	1·440	1·943	2·447	3·143	3·707	5·959
7	1·415	1·895	2·365	2·998	3·499	5·405
8	1·397	1·860	2·306	2·896	3·355	5·041
9	1·383	1·833	2·262	2·821	3·250	4·781
10	1·372	1·812	2·228	2·764	3·169	4·587
11	1·363	1·796	2·201	2·718	3·106	4·437
12	1·356	1·782	2·179	2·681	3·055	4·318
13	1·350	1·771	2·160	2·650	3·012	4·221
14	1·345	1·761	2·145	2·624	2·977	4·140
15	1·341	1·753	2·131	2·602	2·947	4·073
16	1·337	1·746	2·120	2·583	2·921	4·015
17	1·333	1·740	2·110	2·567	2·898	3·965
18	1·330	1·734	2·101	2·552	2·878	3·922
19	1·328	1·729	2·093	2·539	2·861	3·883
20	1·325	1·725	2·086	2·528	2·845	3·850
21	1·323	1·721	2·080	2·518	2·831	3·819
22	1·321	1·717	2·074	2·508	2·819	3·792
23	1·319	1·714	2·069	2·500	2·807	3·767
24	1·318	1·711	2·064	2·492	2·797	3·745
25	1·316	1·708	2·060	2·485	2·787	3·725
26	1·315	1·706	2·056	2·479	2·779	3·707
27	1·314	1·703	2·052	2·473	2·771	3·690
28	1·313	1·701	2·048	2·467	2·763	3·674
29	1·311	1·699	2·043	2·462	2·756	3·659
30	1·310	1·697	2·042	2·457	2·750	3·646
40	1·303	1·684	2·021	2·423	2·704	3·551
60	1·296	1·671	2·000	2·390	2·660	3·460
120	1·289	1·658	1·980	2·158	2·617	3·373
∞	1·282	1·645	1·960	2·326	2·576	3·291

Table A16.4 **The c^2 test of association**

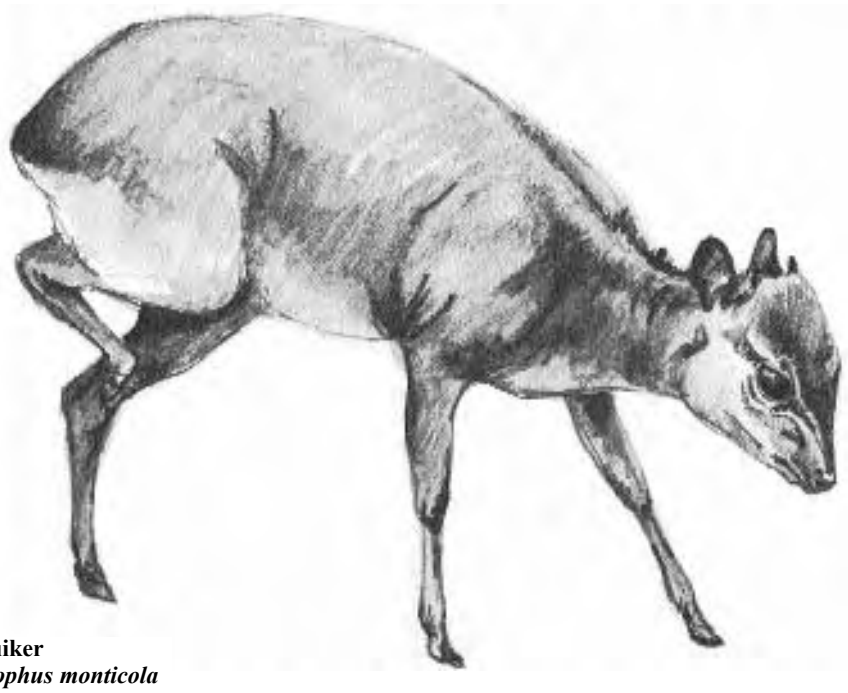
Critical values of c^2 at various significance levels. Reject the null hypothesis if your value of c^2 is bigger than the tabulated value at the chosen significance level, for the calculated number of degrees of freedom.

degrees of freedom	significance level:												
	99%	98%	95%	90%	80%	70%	50%	30%	20%	10%	5%	2%	1%
1	0·000157	0·000628	0·00393	0·0158	0·0642	0·148	0·455	1·074	1·642	2·706	3·841	5·412	6·635
2	0·0201	0·0404	0·103	0·211	0·446	0·713	1·386	2·408	3·219	4·605	5·991	7·824	9·210
3	0·115	0·185	0·352	0·584	1·005	1·424	2·366	3·665	4·642	6·251	7·815	9·837	11·341
4	0·297	0·429	0·711	1·064	1·649	2·195	3·357	4·878	5·989	7·779	9·488	11·668	13·277
5	0·554	0·752	1·145	1·610	2·343	3·000	4·351	6·064	7·289	9·236	11·070	13·388	15·086
6	0·872	1·134	1·635	2·204	3·070	3·828	5·348	7·231	8·558	10·645	12·592	15·033	16·812
7	1·239	1·564	2·167	2·833	3·822	4·671	6·346	8·383	9·803	12·017	14·067	16·622	18·475
8	1·646	2·032	2·733	3·490	4·594	5·527	7·344	9·524	11·030	13·362	15·507	18·168	20·090
9	2·088	2·532	3·325	4·168	5·380	6·393	8·343	10·656	12·242	14·684	16·919	19·679	21·666
10	2·558	3·059	3·940	4·865	6·179	7·267	9·342	11·781	13·442	15·987	18·307	21·161	23·209
11	3·053	3·609	4·575	5·578	6·989	8·148	10·341	12·899	14·631	17·275	19·675	22·618	24·725
12	3·571	4·178	5·226	6·304	7·807	9·034	11·340	14·011	15·812	18·549	21·026	24·054	26·217
13	4·107	4·765	5·892	7·042	8·634	9·926	12·340	15·119	16·985	19·812	22·362	25·472	27·688
14	4·660	5·368	6·571	7·790	9·467	10·821	13·339	16·222	18·151	21·064	23·685	26·873	29·141
15	5·229	5·985	7·261	8·547	10·307	11·721	14·339	17·322	19·311	22·307	24·996	28·259	30·578
16	5·812	6·614	7·962	9·312	11·152	12·624	15·338	18·418	20·465	23·542	26·296	29·633	32·000
17	6·408	7·255	8·672	10·085	12·002	13·531	16·338	19·511	21·615	24·769	27·587	30·995	33·409
18	7·015	7·906	9·390	10·865	12·857	14·440	17·338	20·601	22·760	25·989	28·869	32·346	34·805
19	7·633	8·567	10·117	11·651	13·716	15·352	18·338	21·689	23·900	27·204	30·144	33·687	36·191
20	8·260	9·237	10·851	12·443	14·578	16·266	19·337	22·775	25·038	28·412	31·410	35·020	37·566
21	8·897	9·915	11·594	13·240	15·445	17·182	20·337	23·858	26·171	29·615	32·671	36·343	38·932
22	9·542	10·600	12·338	14·041	16·314	18·101	21·337	24·939	27·301	30·813	33·924	37·659	40·289
23	10·196	11·293	13·091	14·848	17·187	19·021	22·337	26·018	28·429	32·007	35·172	38·968	41·638
24	10·856	11·992	13·848	15·659	18·062	19·943	23·337	27·096	29·553	33·196	36·415	40·270	42·980
25	11·524	12·697	14·611	16·473	18·940	20·867	24·337	28·172	30·675	34·382	37·652	41·566	44·314
26	12·198	13·409	15·379	17·292	19·820	21·792	25·336	29·246	31·795	35·563	38·885	42·856	45·642
27	12·879	14·125	16·151	18·114	20·703	22·719	26·336	30·319	32·912	36·741	40·113	44·140	46·963
28	13·565	14·847	16·928	18·939	21·588	23·647	27·336	31·391	34·027	37·916	41·337	45·419	48·278
29	14·256	15·574	17·708	19·768	22·475	24·577	28·336	32·461	35·139	39·087	42·557	46·693	49·588
30	14·953	16·306	18·493	20·599	23·364	25·508	29·336	33·530	36·250	40·256	43·773	47·962	50·892

Table A16.5 **The Spearman rank correlation coefficient, r_s**

Critical values of r_s at various significance levels. Reject the null hypothesis if your value of r_s is greater than or equal to the tabulated value at the chosen significance level, for your number of pairs, *n*.

no. of pairs, *n*	significance level: 10%	5%	2%	1%
5	0·900	1·000	1·000	—
6	0·829	0·886	0·943	1·000
7	0·714	0·786	0·893	0·929
8	0·643	0·738	0·833	0·881
9	0·600	0·683	0·783	0·833
10	0·564	0·648	0·746	0·794
12	0·506	0·591	0·712	0·777
14	0·456	0·544	0·645	0·715
16	0·425	0·506	0·601	0·665
18	0·399	0·475	0·564	0·625
20	0·377	0·450	0·534	0·591
22	0·359	0·428	0·508	0·562
24	0·343	0·409	0·485	0·537
26	0·329	0·392	0·465	0·515
28	0·317	0·377	0·448	0·496
30	0·306	0·364	0·432	0·478



Blue duiker
Cepholophus monticola

CHAPTER 17
Presenting and conserving your findings

Ann Edwards & Lee White

No research is complete until the results have been analysed and made available in the form of reports and publications. If you fail in this last step of the process your research is of little value. Furthermore, information, if well presented, can be a powerful tool that you can use to your advantage. Information can serve to educate decision makers, inspire the uninformed, and direct the intentions of all willing readers towards support of a cause. Specifically, when information is presented well it can highlight the importance of your protected area as a refuge for endangered species or habitat types, or as a site of unique cultural, spiritual, educational or recreational value. Well presented information can educate and inspire people locally, nationally and internationally, or graphically highlight threats to an ecosystem or reserve, which in turn will help to increase political and financial support to the protected area.

Information should be Presented in Different Forms for Different Audiences

Information can be presented in many different forms, including technical reports and scientific publications, short brochures, lists or "fact sheets", lectures or talks, permanent displays, and guided tours. The way in which information is presented should depend on the intended audience. For example, technical documents should be written for park directors, scientific advisors, and other people who are accustomed to critically reading detailed material. On the other hand, brochures, lists and other non-technical materials should be written for tourists, park visitors and other people who want to learn quickly about the most exciting, interesting and unique things in the area.

If the information is presented so it is interesting and easily understood by a particular group of people, then it will make the forest and its needs more real and accessible. This in turn will lead to increased support for the area.

A suggested goal for each protected area is to have information available in an appropriate form for each category of people who may take an interest in the area.

Guidelines for Presenting Information

Whilst the written word can be very powerful when well used, if poorly presented or analysed, or if your data are inadequate, any report or paper you produce is likely to have the opposite effect. Never rush into publication of data which is minimal, and always bear in mind that your reputation is at stake when your put you name to a document.

In order to be persuasive, information must be presented with care. Documents should be carefully edited to eliminate information that is not relevant. The total length of the document and the order in which the information is presented should be carefully considered. Graphics and illustrations should be used wherever appropriate, because they make the document more appealing to the reader.

Technical and non-technical documents have different purposes, and therefore should be written with different things in mind. Technical documents are used to present detailed information. They should contain enough information to answer the reader's detailed questions. Non-technical documents are used to introduce the reader to a subject, and therefore do not need to explain any one point in great detail. Extra care should be taken with non-technical documents to make them immediately interesting and enjoyable to read.

When you begin to compile a report or other document think about what you would do when first handed such a report. If you are unsure, try giving reports to colleagues and watching their immediate response. In general they will probably do something like the following sequence:

- first they will look at the cover and read the title and the name of the author(s);
- then they will hold the spine of the report and flick over the pages from back to front to get an overall impression of the layout and content - if they see many interesting looking figures their interest will be increased. They may pause to look at any photographs or maps;
- next they will turn to the table of contents to get a better idea of the information included in the report;
- they may then read the summary or abstract to get an overall idea about the ideas and conclusions within;
- they will often dwell on the acknowledgments, to see if they or anybody they know is thanked for helping with the study;
- finally, they may glance through the references cited at the end of your report, to see if you have cited key papers or reports which deal with your subject (or those they or their friends have written), and to assess the breadth of your background knowledge, which will be reflected by the completeness of your bibliography.

If their overall impression is good, they may then think about reading the report in more detail. Their actions provide you with a clear set of guidelines for preparing reports which will be read.

Technical Reports

- Choose the words of the title carefully. The title should describe what is contained in the document. From the title alone, the reader should be able to determine the kinds of information that will be in the report. Present the title in bold letters on the front cover of your report, along with your and your colleagues names and affiliations. If possible, put a photo or drawing on the front cover to catch the eye of any potential reader.
- Include an abstract or summary at the beginning of a long document. Not everyone who is given the document will have time to read the entire report. An abstract or summary will allow these people to be exposed to the essential elements of the document and possibly to be influenced by its contents. An interesting abstract also may entice a reader to read more of the document than he or she would otherwise.
- Organise the information carefully. Put the appropriate kinds of information in the appropriate sections of the report. This will make it easier for the reader to locate the kinds of information he or she is seeking.
- Use chapters, sections, headings and sub-headings, when appropriate. It is easier to find specific information within a large document when the document is broken into smaller sections. Include a "Table of Contents", listing each section next to the page number where it can be found.
- Use enough words in titles, headings and subheadings so that the reader has a clear understanding of what is in that section. This will be particularly helpful for readers who are skimming a document, but who do not have time to read all of it.
- Use appendices to present lengthy but relevant material, such as the "raw data" collected in the forest, (which may be of use in the future for monitoring or comparison purposes but which hold little interest for most readers), examples of data sheets or questionnaires, very long lists of species seen or people interviewed, etc.

- Use graphics (such as graphs, maps, figures, sketches, photographs, etc.), whenever possible. In order to present an idea graphically, it must be simple and easy to understand even before it is graphed. Therefore, graphs can be an effective way of presenting an idea or result in a simple, easy to understand manner. Furthermore, the reader's attention will always be drawn to pictures and graphs. Such visual aids make reading a report easier and more entertaining. In particular, use graphs to illustrate research results, such as trends in species abundance. Use maps to illustrate where the research took place, for example, the trails walked, the locations of plots, etc. Use maps to show the distributions of species, the distribution of habitat disturbance, etc.
- Emphasise the most useful information. This will make the report more interesting to read. Furthermore, the report will be used, and referred to, more often.
- Include enough background material to support your conclusions. In a technical report, all conclusions are open to examination. In order to make your conclusions convincing, you must provide the reader with enough information so that he or she can assess your conclusions.

Non-technical Materials

- Report what is interesting. If it is interesting to the writer it probably will be interesting to the reader. What is unusual? What is spectacular? What is exceptionally large, or long, or fast, or high? For example, does the area contain an unusually high number of species of birds, mammals, monkeys, carnivores, frogs, bats, etc.? Is it the last refuge for any endangered species? Are there some species that do unusual things? What are the usual group sizes, social structures, litter sizes, eating habits, movement patterns, etc. of the large mammals or birds? How do local people use the plants and animals of the forest?
- Keep it short and simple, and make it easy to understand. The entire document, display, talk, etc. should be kept short. More importantly, each idea or fact should be presented simply, with different ideas clearly separated from each other. If the audience does not understand what you are trying to say then the brochure, talk, etc. is a wasted effort.
- Use figures and illustrations whenever possible. People are attracted to pictures. Using them will encourage people to read or listen more in order to learn more about the picture.

Research reports

Research reports are used to report the results of a research project. In addition, they are used to report why the project was undertaken in the first place, the methods used to collect and analyse the information, and the real significance of the results. Research reports are written in order to record fully and permanently all the details of the research process.

Often research reports are not read from start to finish. Instead, a reader may look at only one or two sections. For example, the reader may be interested only in the numerical results, or only in a description of the study animals, or only in the initial purpose of the study or the final interpretation of the results. Because technical research reports contain so much information, the order in which the information is presented is very important.

The categories listed below represent the standard format for a scientific report or publication. When a report is written using these standard categories, it will be easier for the reader to locate specific information within the report.

Research reports do not always have to follow the standard format outlined below. However, whenever the results of a study are presented, the basic information found in those categories should be included; specifically, the purpose of the study, the study species and study site, the methods used, the results (including the mean and variance of all estimates), and an interpretation of the results.

Introduction section

State the specific objective of the study. Include the species or taxonomic group studied (See Box 17a), and the exact area where the study took place. For example, you can say: "The purpose of this study was to estimate the densities of primate groups throughout Tai National Park"; or, "The purpose of this study was to assess the composition and structure of the trees of Falali Forest Reserve, east of the Rangu River".

Include any background information that is relevant to the purpose of the study. This information can help justify why the study was undertaken. For example, you can say, "Evidence suggests that with the increase in human settlement around the park over the last 10 years, the rate of primate hunting by people has increased (Owens, 1990) [see Box 17b for how to correctly cite other peoples' work]. Currently, there exist no reliable estimates of primate densities within the park. Reliable baseline information is needed if the affect of continued hunting on primate densities is to be assessed."

Study area section

The purpose of describing the study site is to give enough information so the reader can evaluate the methods, results and discussion in relation to environmental factors found in the area.

A map of the study area is always useful and often vital. It should show where the park or area of interest is located within the province or country, and can include major rivers, mountains, roads, villages, etc.

The amount of detail used to describe the study area depends on how familiar the intended reader is likely to be with the site (for example, an internal park document may require less detail than a report destined for publication), and on the availability of other reports that already describe the study area. If the study area is described well in another document, and that document is widely available, then it is acceptable to refer to that document for a description of the study area. For example, "the study area is described in detail in Olanga, 1989" (but bear in mind that a reader who does not have access to the report in which the detailed description is given will be extremely frustrated).

The following types of information can be included in a description of the study area:

- geographic coordinates of the study area;
- location of study area relative to regional habitat types and topographic features (for example, the park is located less than 40 km from the ocean but its vegetation is not influenced by the marine environment);
- local topography (hilly, flat, etc.), distinct features (rivers, mountains, etc.), range of elevations within the study area, a summary of local geology and soil characters;
- habitat types found within the study area, possibly including a list of common tree species;
- average annual rainfall, and average maximum and minimum temperatures in or near the study area;
- background about local ethnic groups and their histories.

Study species or human population section

If the paper or report describes some aspect of a particular species, this section contains relevant information about the species. This information generally comes from existing reports or observations by other people, and not from the study itself. Such information can include details about the diet, habitat preferences, or social structure of the species, estimates of abundance in other forests, whether the species is listed as threatened or endangered, or any other information that may be relevant and important to tell the reader. It is not always appropriate that the report contains a section describing the study species. For example, if the report is about the socio-economics of an area this section would be replaced by one documenting local human populations and their history and traditions; or if it described a general botanical survey it might document the habitat types present.

Box 17a SCIENTIFIC NAMES

A common fault in reports is for scientific names to be cited incorrectly . There is an international convention for writing scientific names (also known as Latin names) which should be closely followed:

Each scientific name in fact consists of two names for example, *Homo sapiens*. The first is that of the genus, which may be common to several or even hundreds of closely related species. After the generic name there is a specific name. Several species in different genera may have this specific name, which sometimes describes a character of the species (e.g., *grandifolia,* meaning 'large leaf' is a common specific name for plants) or is the name of the first person to collect or describe the species, for example, *Engomegoma gordonii* is a new genus of tree in the Olacacaea family named for Gordon McPherson, the botanist who first collected it in Gabon.

Each combination of generic and specific name is unique to only one species. However, when there are two or more recognised forms of a species, a third name can be added. This is the name of the subspecies. For example the African elephant is called *Loxodonta africana* and is divided into two sub-species, the savanna elephant *Loxodonta africana africana* and the forest elephant *Loxodonta africana cyclotis* (this last name is Latin for «round ears»).

When writing a scientific name in a report it should be distinguished from the main text by using a different style. The two standard means of presentation are to either underline the generic and specific names (Loxodonta africana) or to put them in italics (*Loxodonta africana*). If the name features in a title which is in italics the scientific name is presented in plain text. The generic name always begins with a capital letter and the specific (and sub-specific) name always begins with a small letter. It is sometimes interesting to include the name of the family to which a species belongs, particularly for plants. The family name is written in normal text but with a capital letter, for example *Aucoumea klaineana* (Burseraceae). Another convention which is sometimes followed is to add the 'authority' (the name of the expert who named the species), which begins with a capital, being a proper name, but which is not italicised or underlined: *Aucoumea klaineana* Pierre (Burseraceae).

Sometimes the generic name is known but the species has not been fully determined. This may be because the collection was not well preserved; it lacked certain features necessary for identification, such as flowers; because the taxonomy of the group in question is poorly documented; or even because it is probably an undescribed species. If this is the case, write «sp.» in normal text after the generic name (e.g., *Beilschmiedia* sp.). If several different but incompletely identified species occur you can write *Beilschmiedia* sp. 1, *Beilschmiedia* sp. 2, *Beilschmiedia* sp. 3 etc. Alternatively if you know there are several species but are not sure you can distinguish between them all, or for the sake of simplicity you wish to lump them in a report, you could write *Beilschmiedia* spp. (where «spp.» signifies that there are two or more species in this genus).

One confusing and annoying thing about scientific names, particularly botanical ones, is that they sometimes change. There are several reasons why scientific names change. Sometimes species are mistakenly classified into the wrong genus. When this is discovered it has to be corrected. Sometimes two or more taxonomists name a single species with different names because they were not aware that it had been previously described. In this case the earliest name is the correct one, even if a more recent one is in general use. Occasionally a name which has already been used for something else is chosen inadvertently by a ill-informed taxonomist. Examples you might have heard about or encounter in literature include *Milicia excelsa,* the new name for the well known tree *Chlorophora excelsa* (Iroko), and *Papio sphinx,* a previously accepted name for *Mandrillus sphinx,* the mandrill.

Box 17b: CITATIONS

When you write a report or publication you should refer to research previously undertaken by yourself or other authors. It is standard practice to 'cite' results or ideas gleaned from other peoples' work by referring to their publication(s). For example, you might want to refer to a previous study by 'Tom Struhsaker' who had found similar densities of red colobus monkeys in 1975 to those you have calculated. You would say «Struhsaker (1975) reported similar densities in the Kibale forest, Uganda» or « similar densities have previously been reported from Kibale in Uganda (Struhsaker, 1975)». If you want to cite two studies at the same time you should either present them in alphabetic order (Oates, 1977; Struhsaker, 1975) or chronological order (Struhsaker, 1975; Oates, 1977). By citing references you acknowledge work undertaken by previous researchers, and allow your readers to delve further into a topic they find interesting (by referring to the references you cite).

Each reference cited in your report should then be grouped in alphabetic order in a 'References' section. By doing this you provide your reader with all the information necessary to enable them to find the references you have referred to. You should choose a standard format for citation of other peoples' publications in your report and use this consistently (each scientific journal has a standardised format for references, described in the 'instructions for authors' section. Inconveniently, this differs from journal to journal). For example,

Struhsaker, T. T. (1975) *The red colobus monkey*. Chicago University Press, Chicago. 311pp.

Struhsaker, T. T. (1976) A further decline in numbers of Amboseli vervet monkeys. *Biotropica* **8**: 211-214.

The formatting of the text contains coded information. In the first citation, as is standard, the title of the book has been italicised (you can also underline) and the number of pages is given at the end. In the second the journal name is italicised, the journal volume is emboldened and the page numbers of the first and last pages of the article are noted.

References are frequently badly cited in reference lists. Authors' names are spelled incorrectly, the year in which articles were published is mis-quoted or the page numbers given are wrong. If you make these silly mistakes anybody wanting to look up an article you have cited will be unable to find it. Furthermore, they may wonder if your data were collected in the same sloppy fashion, and the validity of your data may be called into question.

For researchers based in Africa it is often difficult to obtain key or up to date references because few libraries have sufficient funds to subscribe to journals or buy new books. This is obviously a great handicap. When you read a paper which reports the published work of another researcher which is of direct relevance to your own study, it is tempting to simply add that reference to your own citations. Many people do this, but it is not a good habit to get into since you cannot appreciate the full implications of a study unless you have actually read and evaluated what the methodology and results were. If you cannot obtain a publication but feel it is important to acknowledge it, the best way is to write « (Struhsaker, 1975 cited in Oates, 1977)». You can often obtain papers direct from the author (s) if you write to them requesting a «reprint» - try choosing attractive stamps, or an interesting post card, to accompany your request, as this may sway the decision of an author who has received many such requests. Be aware that published results and conclusions are sometimes over-simplified or even mis-quoted by careless researchers. If you inadvertently repeat these mistakes by not studying the original article, you are equally at fault.

Methods section

The methods section has two purposes. First, it should allow the reader to assess how appropriate the methods were, given the conditions under which you were working (i.e., were the chosen methods appropriate and can they be expected to have produced reliable, representative, accurate data?). Second, it should allow the reader to repeat the methods. This is an important function. When good methods are described in detail, it makes them available for other people to use. Furthermore, if similar methods are used in more than one study, then the results become more comparable between studies (one justification for this manual).

The methods should be described in enough detail to let someone else duplicate your study. They should be presented in a clear, logical order which leads the reader through all the steps you went through in chronological order. Explain new techniques in detail, such as a description of the trap you designed and built to capture small mammals. Be specific about how you collected your data, for example, "monkeys that were heard but not seen were not counted on transects", or, "dead trees were not included in estimates of tree density".

If some procedure has been described in detail elsewhere, it is acceptable to refer to that document. For example, "The line transect method (Burnham et al., 1980; Whitesides et al., 1987) was used to estimate the density of primate groups". If Burnham et al., 1980 is likely to be unavailable to your readers, then describe the procedure in detail in your report.

If plots or transects are used, the following information should be included in the methods section:

- *Dates and times* - List the range of dates and times (if applicable), in which data were collected. For example, "Botanical plots were enumerated between August and December, 1992", or "Transects were walked monthly between June 1994 and May 1996, at the beginning of each month, between 06:00 and 13:00".
- *Location of plots or transects* - Describe the locations of the plots or transects within the study area (if possible provide geographic coordinates). Specifically, relate their locations to those of any rivers, mountains, villages, trails, logged areas, secondary forest, etc. (generally this is best done using a map).
- *Size of plots or length of transects* - Describe in detail the exact sizes of the plots and lengths of the transects used. If there are any exceptions, describe them. For example, "seven transects measured 4.0 km, and one transect (which was bounded at each end by a major river) measured 3.6 km".
- *Number of plots or transects* - List the number of plots or transects used, and explain why that number was chosen. For plots, state what percentage of the area (e.g., the reserve) was sampled. Calculate this percentage by dividing the total area of the plots by the total sampling area, and multiply that number by 100.
- *Field staff* - if the author(s) of the report is not the only person who collected the data, then the number and qualifications of the field assistants should be described (and their names should be cited in the acknowledgments section).

Results Section

Results are a summary of the data. They represent the "facts" established during the course of your research. They should be presented in the same order as the methods section as far as possible and sensible. The results section should contain the results together with a description of any patterns or trends detected in the results. For example, do not say simply, "Elephant densities in different parts of the park are listed in Table 1". Instead, explain that "Table 1 shows that elephant densities increased with increasing distance from villages, but were unaffected by habitat type".

Presenting and conserving your findings

The "raw data" (the actual numbers collected in the forest) usually are not included in the results section. However, whenever possible, lists of the raw data should be included in an appendix. If the raw data are included in the report, then as methods of analysis change and improve, the data will be available for analysis using the new methods. This allows for more accurate comparisons between studies. If the raw data are not included in the report, then make them available to the reader by explaining where the data are filed or stored.

If the raw data are not included in the report, then it is important that estimates of variability of the data, such as confidence limits, be calculated. Always report the sample size, the mean and the standard deviation.

The report will be more useful if the data are summarised and reported in a variety of ways. This allows the reader more opportunities to interpret and use the results. For example, the results from counting monkey groups on transects can be summarised in each of the following ways:

- Total number of groups detected;
- Total number of groups seen;
- Total number of groups heard;
- Mean number (and standard deviation) of groups seen per kilometre walked (an index of abundance);
- Range of groups seen per transect walked;
- Mean number (and standard deviation) of groups seen per kilometre squared (density);
- Density of groups seen in each habitat type.

Don't hesitate to search for meaning within the data. Look for relationships, for example, between the distributions of duikers and monkeys, or between the decay rate of dung and the amount of rainfall. As long as they are supported by your data, results of any sort can be included in the results section.

Interpretation and discussion section

In this section the results are interpreted, critically evaluated, and compared to the results of other research projects. What interesting things do you now know that you didn't know before this study? How do the results compare to studies conducted in other areas, or at other times, or with different species? What are the management or conservation implications of these findings?

All conclusions should be based on the results of the study. Present all arguments for and any which contradict your conclusions. This demonstrates that your ideas have been well considered. Draw positive conclusions from the results whenever possible.

Highlight for the reader what you have found to be of the greatest significance. What is most interesting to you will hopefully be the most interesting to the reader. You have spent lots of time thinking about what the results mean. The reader may spend only a few minutes reading your entire report. You should help him or her understand the most interesting conclusions of the study.

Most importantly, the discussion should answer, or say why it does not answer, the question(s) posed in the introduction. The end of the report should always state if any advance has been made by the study on the situation or state of knowledge before the study began.

Recommendations

What actions do you consider to be of priority in the light of your findings? This is the section where you should illustrate the practical implications of your research and suggest courses of action which need to be followed or new lines of research which are considered to be a priority. For example, you might recommend that increased anti-poaching efforts are needed in a particular section of a reserve, where gorillas are being killed by poachers; or that hunting by traditional methods should be allowed in the buffer zone of the National Park; or that further studies of dung decay in relation to rainfall and temperature need to be undertaken across the African forest zone in order to establish a firm relationship which can be applied to density estimations of duikers.

Conclusions (or Summary)

You should provide a list of conclusions at the end of your report or paper. For example:

1) Dung counts can be used reliably to assess elephant densities in rain forest areas.

2) Elephant densities were negatively correlated to the distance to the nearest road.

3) Future elephant surveys should be stratified according to the distance to the nearest road or village.

4) In order to improve elephant density estimates from dung counts further studies on decay rates and defecation frequencies are needed.

A "Summary" at the beginning of a report, which also provides details of what was done and where, may be included in preference or in addition to the "Conclusions" section at the end.

Figures and Tables

Graphs should be used instead of tables if they are used to illustrate meaning in the data, for example a trend, or a relationship between two factors.

Tables should be used instead of graphs if the readers might want to use the data for their own calculations.

Figures can be in the form of graphs, diagrams or photographs. The meaning of each figure should be described in the text. Each figure should have its own number, title and explanatory legend. The axis of graphs should always be fully numbered and labelled. All maps should show scale and direction. Both figures and tables must show the units of measurement (for example, duikers/km^2).

When graphing or mapping data it is important to distinguish between data points of zero (i.e., an attempt was made to count animals, nests, etc., but none were seen) and no data collected.

Draft versions

The first version of a technical report or paper is rarely if ever perfect. In fact, unless you have a lot of experience of writing such documents the first draft may bear little resemblance to your final version. There is nothing better than sitting down to write a report to clarify your ideas. If your subject is a complex one you might need to re-write several times before you have put it into a format with which you are happy. Avoid scientific jargon whenever possible - you should aim to present your work in a simple, easily understood manner which any interested person will be able to understand. Keep sentences short.

If you submit a paper to an international journal for publication the editor of the journal will send your paper out for 'review'. Most journals send papers they receive to two or three independent specialists for evaluation. These people assess the interest and originality of the paper, whether the methods used were appropriate for your study, the validity of the analyses and extrapolations, the relevance of the conclusions, and the completeness of the bibliography. They generally make suggestions for improvements and point out any problems. If the paper is of sufficient quality it may be accepted unconditionally or subject to your making certain modifications, or you might be invited to resubmit a much revised version for further review.

Even if you do not intend to send your report to a journal for publication, you should be aware that your own reputation is at stake when you put your name on a written report. Whoever reads the report will evaluate it. If it is well written, interesting and useful the reader will think well of you. If it is not then your reputation will suffer. You should therefore check through your work meticulously. Once you are happy that it is well written and gives a clear representation of your findings and recommendations, or if you begin to feel you have spent so much time on something that you can no longer be objective about it, you should ask a number of your colleagues to read and comment on your 'draft' version.

You should not be offended if they make extensive comments (particularly if they are constructive). In fact, very few people are good at reading papers critically and making constructive suggestions about how they can be improved. Most reviewers simply point out a few spelling mistakes and ask you to rephrase a few sentences. If you receive a 'first draft' version back from somebody with very little 'red ink' on it, it usually means they have not read it properly. Try to get somebody else to look at it.

Many people think that by writing "**draft**" in big letters, to indicate that the final version of the report has not been prepared, that they are denying any responsibility for poor work. This is not the case. Of course, you can give incomplete versions to close colleagues to stimulate useful discussion as you write a report or paper, but avoid circulating the report to other people until you are happy that it is a good reflection of what you want to say. At its best the written word is a permanent record of well planned and executed research which will be of value to managers and researchers for many years to come. At worst it is a waste of time and paper.

Research summaries

Research reports can take a very long time to produce. Unfortunately, sometimes, they are not produced at all, or copies are not sent to the protected area staff, so the results of the research remain unavailable (see Chapter 1). In contrast, research summaries can be produced quickly, even before the research project has been completed. Furthermore, research summaries allow the protected area staff to remain informed about the purpose and the progress of all research projects conducted within the protected area.

A research summary is a short statement which includes some or all of the following:

- The name and address of the principal researcher (both locally, and in his or her home city);
- The purpose of the study;
- The (proposed) dates of the study;
- Descriptions of the study species and the study site;
- A description of the methods;
- General summaries of the results, to date; and
- The significance of the research, especially in relation to management and protection.

Protected area managers should require research summaries of all proposed, on-going or newly-completed research projects within the protected area. This requirement is particularly important for foreign researchers who may leave the country without adequately informing the protected area management about the results or the significance of their research. By requiring written accounts of research projects, protected area managers can remain informed about the purpose, the progress and the anticipated outcomes of each project.

Each visiting researcher should be required to write two research summaries. The first should outline the proposed research, and should be submitted before any field work begins. The second should be submitted at the completion of all fieldwork, but before the researcher leaves the area to return to his or her own city. Be sure that the researcher also leaves a contact address before their departure.

This second report should be more extensive and should include a description of the work that was actually done, including the names and addresses of assistants, and the exact locations of the study sites. The researcher should be encouraged to report any preliminary results, and in particular, to address issues that are relevant to the management and protection of the protected area. However, the researcher should not be asked to make unsupported conclusions.

In addition to writing a summary of their research, each researcher should be asked to leave lists of plants or animals that he or she identified, and to leave voucher specimens if available (see Chapters 8 & 11).

Brochures, fact sheets, displays and presentations

Brochures, fact sheets and displays are all formats for presenting non-technical information quickly and cheaply to a large number of people. Their purpose is to orient a visitor to the features of the protected area, and to inform and inspire them about the many interesting things that can be found there.

Brochures

A brochure is designed to give a visitor his or her first introduction to an area. It should contain enough information so that the visitor understands the significance of the area and why it is protected. Furthermore, it should make them feel comfortable visiting the area by outlining what facilities are available for tourists, such as walking trails, guided tours, overnight facilities, etc. It should also contain a simple map of the area.

Fact Sheets

A "fact sheet" is a one or two page description of a narrow topic, or set of related topics. Fact sheets are intended to educate visitors about specific topics of interest or importance. Since they are written for a non-technical audience, fact sheets should present each "fact" in simple language, and without too much detail.

For example, one fact sheet could be about birds. Interesting "facts" that could be reported include:

- the total number of bird species in the park;
- the number of birds that migrate seasonally;
- the fact that some birds eat mostly fruit, while others eat mostly seeds, insects, or meat;
- the fact that female hornbills of many species are sealed within the nest from the time the eggs are laid to the time the young birds fledge and they depend on the male to feed them;
- the fact that some birds lay their eggs in the nests of other bird species in order to get those birds to feed and raise their young, etc.

Another fact sheet could inform visitors about what activities are legal or illegal within the park.

Species Lists

Lists of mammals, birds, under-storey plants, trees, etc. can be printed on single sheets of paper and made available to interested visitors. Often visitors from other countries come to tropical forests specifically to see (or study) rare plants or animals. A species list can help orient these visitors and encourage them to explore the forest.

Displays

A display is a large, colourful presentation of pictures and writing, usually found on a centrally located bulletin board. Displays provide another means of orienting and educating visitors. Displays are special because they offer the opportunity to post large, colourful, detailed maps, or striking photographs of elusive forest animals. Visitors are immediately drawn to displays, and they tend to read what is written there, if it is kept brief.

Presentations

Presenting your work in public is an important aspect of research. For your results to be appreciated people have to know about them. Many people are notoriously lazy when it comes to reading publications and reports, but will listen attentively when you make a formal or informal presentation. Never pass up an opportunity to present and discuss your research, since, like writing, discussion and spoken presentations often help clarify certain aspects of your research in your own mind. If you cannot present your research clearly to an audience it is unlikely that you have thought it through sufficiently.

When asked to give formal presentations you should plan them carefully and think through how you can best get your ideas and results across to your audience. There are several easy ways to improve your talks:

- *Wear appropriate clothing*. Formal presentations may call for a jacket and tie. Less formal events impose fewer constraints but dress well. Avoid scruffy clothing, brush your hair and teeth, make sure your hands are clean. You will appear more professional and make a better impression on your audience, who will generally act accordingly.
- *Make use of as many visual aids as possible*. If a slide projector is available present some good quality photographs of your study area. Be sure to check that slides are well focussed. Prepare overhead projection sheets summarising the aims of your study, your methods, the principal results, your conclusions and your recommendations. Be sure that the text is large enough for all of your audience to read them without straining their eyes. If you do not have access to such sophisticated aids prepare figures and tables on paper which can be held up or passed around, or use a blackboard.
- *Make eye contact with your audience*. If you need to consult notes make them as brief as possible. A clear summary of key points on small pieces of paper or card is ideal. Avoid reading out long sections from a script if you possibly can. Present the talk to the audience, making frequent eye contact around the room. Be sure to talk clearly and to project your voice to the back of the room or meeting.
- *Be enthusiastic*. It is astonishing how few researchers are unable to present their life's work with enthusiasm. To be a conservationist you need to be a communicator, since many of the problems you will have to deal with involve people. Try to break the ice with a joke or an amusing story about an event which occurred during your research. Do not speak in a monotone.
- *Involve the audience*. In any informal setting do not hesitate to ask questions of the audience, and encourage them to ask questions while you are presenting, in order to help you to clarify any difficult points. It is important to find out before your presentation what levels of education and experience your prospective audience will have, in order to avoid using language they will find too technical, or to avoid giving them the impression they are being talked down to.
- *Repeat key points*. Key ideas and recommendations can by repeated in order to emphasise them and ensure that the take away message is clear. Summarise key points at the end of your talk. People tend not to take in more than three key points, so organise your presentation around three main 'take home concepts' which you want the audience to remember.
- *Try to enjoy yourself*. We do not all enjoy being the centre of attention, but seize the opportunity to put your ideas across and have a good time - its your chance to get up on a stage and act a part in your very own play. Make your talk into a one-person show and try to entertain as well as to educate your audience.

A suggested list of materials that should be made available at each protected area

In order for each protected area to present itself well to a variety of audiences; in order to make the forest accessible to both tourists and visiting scientists; and in order to have adequate information on which to base management decisions, protected areas should have information available in each of the formats listed below.

An introductory brochure

A one or two page brochure can be used to give a general introduction to the area. The brochure can briefly include the following types of information:

- a description of the protected area (size, location, habitat types, elevation, climate, names of local ethnic groups, size of local villages and towns, etc.);
- an explanation of why the protected area was created;
- descriptions of special features within the protected area (for example, endangered species, large rivers or mountains, etc.);
- a list of activities that are legal or illegal within the protected area; and
- facilities and activities that are available for visitors, the dates and times that they are available, and the costs.

Maps

Maps are essential for orienting and educating visitors. One large, colour map should be put on display at some central location. Smaller, single page, black and white maps can be made available for visitors to take with them as they travel through the forest. Maps for visitors, or visiting researchers, can show some or all of the following features: walking trails, established transects, roads, bridges, villages, tourist facilities, research camps, rivers, mountains, habitat types, and points of special interest such as waterfalls or viewpoints.

Species lists

Species lists can be used by both tourists and visiting scientists. Separate lists should be written for mammals, birds, amphibians and reptiles, fish, insects, trees, herbaceous vegetation, etc. The list for birds can indicate which species are present only in certain seasons.

Fact sheets

Fact sheets are used to educate visitors on specific topics of interest. Topics can include, the story of forest succession, the inter-dependence between fruit trees and fruit eating animals, how a strangler fig grows from a seedling to a mature tree, the importance of the forest to local people, etc. They can be produced as a series, so that visitors can select those that interest them.

An introductory talk or lecture

It is advisable to have a talk prepared (with slides or overhead projections, if possible) describing the protected area and how it functions. This talk can be used locally to introduce visitors to the area. Or, if a speaker is invited to talk at some formal function, he or she can use the talk as an opportunity to interest people in the beauty, the potential and the needs of the protected area.

A list of reports and publications about research conducted in the protected area

In order to make information about the protected area more accessible, a reference list should be made of all the reports and publications that cover research conducted in the protected area.

The list should contain the following information about each report or publication:

- the name(s) of the author(s);
- the title of the report;
- the year of the report;
- where the report can be found;
- what language the report was written in (if this is likely to be an issue); and
- ideally a few words or sentences describing the report in more detail.

A separate list can include all the non-technical, educational materials, such as fact sheets, that have been written about the protected area.

Summary of each research project conducted within the protected area

Research within the protected area is useless unless the results of that research are accessible to protected area managers and researchers. Therefore, each research project within the protected area should be required to submit periodic, technical summaries of their work (see above), in addition to a final research report.

Summary of what is currently known for each important species or habitat type

Although it requires extra work, compiling a summary of what is currently known about species or habitats of special importance can be very helpful for both management and public education, and can save time in the long run. Once the information is summarised, it can be incorporated easily into talks, displays, fact-sheets, and reports. More importantly, this information can be used to manage those species or habitats more effectively and to establish priorities for future research. A summary of what is known will also highlight what is not yet known and can help determine future research priorities.

A single page, with the following headings, can be used to summarise what is known about each species or habitat:

- Species name, or Habitat description;
- Global distribution. This category will highlight the relative importance of the protected area for the long-term survival of the species or habitat;
- Distribution in and around the protected area. The distribution of the species or habitat in and around the protected area can be drawn on a map and attached to this summary. Comments can include:
    - The historical or past distribution of the species or habitat;
    - Recent changes in distribution;
    - Seasonal movements of an animal species;
    - Habitat preferences or needs; and
    - The distributions of discreet populations of the species within the protected area.
- Estimates of global abundance. Include comments on trends in global abundance, and whether the species is listed as threatened or endangered by the Convention on International Trade in Endangered Species (CITES);
- Estimates of local abundance. In addition to listing the most recent abundance estimate, list all past estimates next to the years the estimates were made. Comment on increases or decreases in abundance over time. List the method(s) used to estimate abundance;
- Threats to survival. Summarise what is known about the threats to the survival of the species or habitat (for example, rates and locations of hunting, logging, clearing, etc.). Include both known and assumed threats. Comment on the distribution and intensity of these impacts (for example, include the rate of extraction, the price, who is involved, etc.);

- Actions taken to protect species or habitat. Summarise the management actions that have been taken (or are planned) to minimise the above threats;
- Diet. Summarise what is known about the diet of the animal, and possibly the availability of its foods;
- Reproduction. Summarise what is known about annual reproductive rates, mating or birthing. Include the ratio between males and females, and the ratio between infants, juveniles, and adults. Also, comment on the seasons when mating or births occur, and related behaviours; and
- Unusual behaviour, other comments. Comment on any unusual or interesting behaviours, etc. Include those facts that would be surprising or interesting to visitors.

Organising, storing and retrieving information

If information is not accessible, then it is as if it doesn't exist. With all the time, money and effort that goes into collecting information in the forest, it is a waste to lose that information in the office. Therefore, care should be taken to store information in an organised manner so it can be retrieved reliably and efficiently, and so that any new information can fit well into an existing storage system.

Even after data have been summarised, or even published, it should be possible to return to the original data sheets or field notes. This might be necessary in order to address specific questions that may have arisen since the data were summarised or analysed.

A lot of time may pass between the time the data are collected and the time they are analysed. Furthermore, many different people may be involved in collecting and summarising these data. Therefore, in order not to lose any information, it becomes important to standardise and organise the way information is labelled and stored. Suggestions for doing these are outlined below.

Labels

Every field book, data sheet, report, etc. should be labelled. A label allows someone to tell at a glance what information the field book, data sheet, etc., contains. This becomes very important once things are filed or stored. Labels should contain four key pieces of information.

- The *date* the information was collected.
- The *location* where the information was collected.
- The *subject* of field notes or data sheets (for example, "Large mammal survey, Transect 3").
- The *name(s) of the observer(s)*.

Other information also can be included on a label. However, labels should be kept simple so they remain easy to read at a glance.

The location of the label, or key information, should be the same on every data sheet or field book. This makes it easier to find the information quickly. For example, key information can be recorded consistently in the upper right hand corner (or the upper left hand corner, etc.).

Guidelines for organising a filing or storage system

A storage or filing system can consist of a room of filing cabinets, a shelf of loose-leaf binders, or a drawer full of boxes. Whatever form information is stored in, it should be organised so that existing information can be found easily, and so new information can be added easily to the existing system.

There are many ways to do this, depending on the quantity of information and the facilities available to house the information. However, all organised filing systems should follow a few general guidelines:

- *Only trained people should file information.*

Only those people who have been trained about the organisation of the filing system should be allowed to place new material into the filing system. This will help ensure that information does not get lost through misfiling. For example, instead of filing their own completed data sheets, field assistants can put them in a specially marked box, and a trained file manager can place them in the appropriate files.

- *Clearly label documents, file folders, file sections, file drawers, etc.*

Clearly label as many different parts of the storage system as possible. For example, when using filing cabinets, label each file drawer, each subject divider within each drawer, and each file folder. As stated above, each document also should be clearly labelled.

File folders in filing cabinets are a good way to store data sheets, and other unbound papers. However, if filing cabinets are unavailable, or in short supply, unbound pages can be stored in clearly marked (and sectioned) loose-leaf binders.

- *Never store something in an unmarked file or box.*

If labelled information is stored in an unmarked place, it may be lost. Don't store written materials in boxes unless everything in the box is listed on the outside of the box.

- *File information alphabetically and chronologically.*

Information should be filed according to some rule. Usually, information is filed alphabetically (by subject), and then chronologically (by time) within each subject. For example, you could file data about animals in the following order: Duikers (1988-1992), Elephants (1991-1992), Gorillas (1990), Monkeys (1988-1992).

- *Keep master lists.*

Make an updated "master list" of all the major categories of your filing system. For example, you could list the labels of each file drawer, and each subject divider within each file drawer. Post this master list in a central location. This will not only make it easier to find information, it will also force you to organise the overall structure of your filing system.

If time permits, make master lists of categories of available information. For example, you could list all the books in the reference library, or all the published research reports, or all the large maps, or all the available educational hand-outs, etc.

Protecting your data

Copy all original data sheets

Data analysis can always be repeated, but original data, once lost, cannot be reproduced. Therefore, it is important to make copies of all data sheets, field books and field notes of any kind, in order to guard against their loss or destruction. The original and the copy then should be stored in two separate places.

Copies can be made as data are collected by placing a sheet of carbon paper between a data sheet (or a page in a field book) and a blank sheet of paper.

Summary sheets

Summarising data on to "summary sheets" is a form of data analysis, but it also provides some insurance against the loss of original data. Furthermore, summary sheets provide an important way of organising and storing information for easy retrieval. Therefore, in addition to filing the original data sheets, you should summarise the data soon after collecting it, and file the summary sheets in a separately labelled file.

The data can be summarised in many different ways, depending on how often, and in what places, data were collected. Typical summaries include:

- data summarised by time (daily, weekly, monthly or yearly);
- data summarised by location (by transect, by section of the park, etc.); and
- data summarised by the distance walked (every 500m, every 10km, etc.).

Log books

As discussed in Chapters 5 and 14, detailed notes on animal behaviour, reproduction, diet, etc., or sightings of rare animals such as leopards or okapis, can be organised and protected from loss by keeping separate "log books" for each species or behaviour of interest. A log book is a notebook with blank or lined pages in which observations are described in detail. Subjects of log books could include:

- "large carnivores: sign, sightings and kills";
- "elephant sightings: group size and composition, known individuals and foods";
- "gorilla or mandrill sightings: group size and composition, known individuals and foods";
- "buffalo sightings: group size and composition, known individuals and foods";
- "reproductive behaviour (and timing) of monkeys: observed copulations and ventral infants";
- "hornbill nest sites";
- "predation by crowned hawk-eagles"
- "medicinal plants";
- "local names for animals"; etc.

Log books are used to compile these rare sightings and encounters so that they are available in one, well protected location. In order to further protect this rare and valuable information, copies should be made of log books.

Because it may be difficult to find the original field notes, log books should contain detailed accounts of each observation or encounter, and not just a summary. If possible, the original observer should write in the log book in order to include as much detail as possible, including a reference to the location of the original information (for example, which page in which field book).

Making information available

In many places one encounters the unfortunate attitude best described as "knowledge is power", which greatly handicaps exchange of information between researchers and protected area managers, and within both of these groups. This goes against the scientific ideal of improving knowledge by exchanging results and ideas, and against any management goals. If you are in possession of an interesting document, be it a paper or report, try to make it available to and discuss it with your colleagues. Bear in mind that not all of the information presented in reports and papers is collected using appropriate methods, and several interpretations are sometimes possible for the same data set. The best way to assess the merits of data and ideas presented in papers and reports is to hold informal discussion groups. If there are researchers working in your protected area encourage them to make regular presentations of their work and arrange informal seminars for discussion of key research and management topics. Be sure that all papers, reports, books and other written materials are available to all of your colleagues in a central library. If your protected area does not have a library you should encourage your colleagues and superiors to begin one.

Presenting and conserving your findings

Such sharing of information is key to any management exercise and is fundamental to this methods manual. The various authors of this manual have shared some of their knowledge and experience with you. Their hope is that they will assist and stimulate further research which they consider to be important for the wise management of the African tropical rain forest ecosystem.

If you see a copy of this manual in a colleague's home, at head office, or on a researcher's shelf, and feel it would be useful for you and your colleagues, it is available free of charge from the Wildlife Conservation Society. The only 'price' we put on it is that you send us your comments in order to help us revise and improve any future versions. We hope it will encourage protected area managers and researchers alike to get to know the African rain forest and to work together towards its conservation and sensible management.

Barteria fistulosa

Recommended reading

Many key references are mentioned in the various chapters in this manual. Below we give a (far from exhaustive) list of some useful texts which will help you to further develop your technical ideas and refine and target your research methods, or which provide practical advice about undertaking fieldwork in remote African forests:

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