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THE UNITED REPUBLIC OF TANZANIA
MINISTRY OF HEALTH

NATIONAL INFECTION PREVENTION AND CONTROL GUIDELINES FOR HEALTHCARE SERVICES IN TANZANIA

Tanzanian Ministry of Health, P.O. Box 9083, Dar es Salaam.
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THE UNITED REPUBLIC OF TANZANIA
MINISTRY OF HEALTH

NATIONAL INFECTION PREVENTION AND CONTROL GUIDELINES FOR HEALTHCARE SERVICES IN TANZANIA

USAID FROM THE AMERICAN PEOPLE

JHPIEGO

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FOREWORD

The Ministry of Health of Tanzania is firmly committed to ensuring safe, quality healthcare services to the people of our nation and to providing protection from outbreaks of infectious diseases. The infection prevention and control guidelines contained in this document are a reflection of this commitment.

Infection prevention is a critical component of quality health services, yet it has received insufficient attention. Nosocomial infections, or infections acquired in healthcare facilities, may be transmitted in different ways, either from a patient, a relative or a staff member. They may also be transmitted through the air or contaminated water, food, drugs, medical equipment and objects in the environment such as furniture or dishes. In addition, the elevated rates of prevalence of highly infectious and potentially life threatening diseases in Tanzania, such as HIV/AIDS, cholera, tuberculosis and bloody diarrheal diseases, also demand that special attention be placed on safe and effective infection prevention practices.

The purpose of this manual is to provide all healthcare providers with basic infection prevention and control guidelines and safety precautions applicable in their day-to-day activities.

The Ministry of Health is dedicated to strengthening and supporting these practices, and will ensure proper implementation through increased budgetary allocations to meet the requirements for improved infection prevention and control. Likewise, the combined efforts of every healthcare worker will ultimately result in improved quality of care for all patients and health personnel.

M. J. Mwaffisi
PERMANENT SECRETARY
MINISTRY OF HEALTH

ACKNOWLEDGEMENTS

This document is the product of extensive and wide consultation among organizations and individuals with vested interest in providing quality services especially in the area of infection prevention (IP) and control for healthcare services in Tanzania. The Ministry of Health wishes to extend sincere gratitude to all those who have contributed materially, physically and technically towards the development of these important guidelines.

Special thanks go to USAID for financial support, and to the World Health Organization, the Commonwealth Regional Health Community Secretariat (CRHCS) and the East Central and Southern African College of Nursing (ECSACON) for the IP materials adapted from *Infection Prevention Control, Policies and Guidelines* (prepared by Dr. Una Redd), November 2001. We would also like to thank JHPIEGO for technical assistance, facilitation and for permission to adapt IP materials used in this activity, (especially Tietjen, L., D. Bossemeyer and N. McIntosh. 2003. *Infection Prevention - Guidelines for Healthcare Facilities with Limited Resources*. JHPIEGO Baltimore, Maryland. © JHPIEGO).

Finally we would like to acknowledge and congratulate the experts who have devoted their time, energy and knowledge to the development of these guidelines. Their names are given in Appendix II. These experts and medical colleagues were drawn from various departments of the Ministry of Health, national and referral hospitals, Kairuki Memorial University, and private hospitals in Tanzania.

Finally we would like to extend our cordial gratitude in advance to all those who on being exposed to these guidelines will feel it is their duty to offer to us their constructive criticism and comments aimed at improving the document.

Dr. G. L. Upunda
CHIEF MEDICAL OFFICER

ACRONYMS AND ABBREVIATIONS

AIDS	-	Acquired Immunodeficiency Syndrome
CDC	-	Centers for Disease Control
CSSD	-	Central Sterilization and Supplies Department
HBV	-	Hepatitis B Virus
HCV	-	Hepatitis C Virus
HCF	-	Healthcare Facility
HCW	-	Healthcare Waste
HCWM	-	Healthcare Waste Management
HIV	-	Human Immunodeficiency Virus
HLD	-	High-Level Disinfection
ICU	-	Intensive Care Unit
IPC	-	Infection Prevention Control
IUD	-	Intrauterine Device
MoH	-	Ministry of Health, Tanzania
PPE	-	Personal Protective Equipment
PVI	-	Povidine Iodine
SARS	-	Severe Acute Respiratory Syndrome
TB	-	Tuberculosis
UP	-	Universal Precautions
WHO/AFRO	-	World Health Organization /African Regional Office

HOW TO USE THIS MANUAL

Infection prevention and control is multidisciplinary and requires compliance by all categories and levels of healthcare providers. Infection prevention deals primarily with preventing the spread of infectious diseases through the air, blood or body fluids, and contact, including fecal-oral and food-borne. Such compliance is obligatory to prevent and control nosocomial and other infections in healthcare facilities and settings as well as in the community.

These guidelines have been developed by the MOH of Tanzania to aid health workers to understand and use evidence-based infection prevention practices. A training manual for all levels will accompany these guidelines at a later date.

The expected users of this guideline include:

a. Policy makers, health managers and administrators
b. Healthcare providers and trainers
c. Programme officers
d. Government and private health facilities and training institutions
e. Regional Health Management Teams and Council Health Management Teams
f. People working at the community level to promote quality of healthcare, e.g., Facility Health Management Committees
g. Individuals, groups, and international organizations engaged in healthcare service provision

These guidelines are made up of five parts with 26 units as follows:

i. Part I: Background
ii. Part II: Fundaments of Infection Prevention and Control
iii. Part III: Processes in Infection Prevention and Control
iv. Part IV: Preventing Infection in Special Settings
v. Part V: Infection Prevention and Control Management

BACKGROUND

1.0 INTRODUCTION

Transmission of infection continues to be a major problem in Tanzania with the burden of infectious disease very high, as reflected in the WHO IP needs assessment report of the year 2000. As of that year:

- over 2 million people in Tanzania were HIV-infected
- an estimated cumulative total of 722,490 people were suffering from AIDS
- there were 54,442 cases of tuberculosis and 1300 cases of cholera
- bloody diarrhea cases numbered 26,450

The data mostly reflect reported cases and not the actual situation of the burden of infectious disease, which may be much higher. Infectious diseases, if not controlled, may lead to high rates of nosocomial infections (those acquired in healthcare facilities). Infection prevention is therefore one of the prerequisites for ensuring safe healthcare service delivery, as well as protecting the population from outbreaks of infectious diseases.

2.0 SITUATION ANALYSIS

A situation analysis of infection prevention practices in Tanzanian health facilities revealed that IP practices are poor for the following reasons.

Lack of guidelines and standards for certain procedures
A number of procedures have no formal guidelines and standards on infection prevention.

Inadequate knowledge and skills among healthcare service providers
Many healthcare providers have not had any updates of information related to infection prevention despite the fact that over the past two decades many changes have occurred due to the emergence of infections such as HIV/AIDS.

Deficiency of equipment and materials
There are inadequate amounts of personal protective equipment (PPE) and supplies such gloves, goggles, plastic aprons and boots. The lack of PPE increases the risk of occupational infections among healthcare workers and clients. In recent years there has been a progressive decline in provision of equipment and materials in healthcare facilities for prevention of infection.

Inadequate supportive supervision
There is a shortage of qualified supportive supervisory staff; a lack of supportive supervision has been identified at all levels of healthcare service delivery.

Lack of renovation and maintenance of infrastructure
Systems such as electrical, water and drainage are often not fully functional, and facility conditions are often overcrowded. These problems are due to a lack of awareness, inadequate qualified human resources, financial constraints and the lack of involving frontline health workers in planning.

3.0 RATIONALE

The reasons for developing and implementing infection prevention and control (IPC) guidelines include the following.

- The HIV/AIDS epidemic has increased the risk for transmission of infections of various kinds; every person should be considered as potentially infected with HIV.
- The availability of new scientific information simplifies provision of safe and effective prevention measures.
- An individual's right to good health requires a safe healthcare environment for both providers and clients.
- There is increased awareness of how risky it is to work in healthcare facilities.
- Healthcare facilities are prone to infection transmission due to:
  - routine invasive procedures
  - exposure; infected or contaminated service providers may transmit infectious agents among clients or one another
  - patients who are susceptible to infection because of compromised immunity
  - patients who often have infections that can be easily transmitted to others
  - services that are sometimes provided in congested physical settings

3.1 IMPORTANCE OF INFECTION PREVENTION AND CONTROL

Infection prevention and control is important for patients, clients, healthcare service providers and communities.

3.1.1 For Patients /Clients

- Health facility-acquired (nosocomial) infections are difficult and costly to deal with because they:
  - increase the length of hospitalization
  - require treatment with expensive, antimicrobial agents
  - increase use of other interventions (laboratory, surgery, etc.)
  - increase drug resistance

3.1.2 For Healthcare Workers

- Risk of infection from airborne, waterborne and blood-borne pathogens such as HBV, HCV, and HIV is high.

3.1.3 For Communities

- Health facility-acquired infections may contribute to preventable morbidity and mortality in Tanzania
- Anti-microbial resistance acquired in a hospital can spread to families and the community when colonized patients are discharged
- Hazardous hospital wastes may carry microorganisms and their disposal can pose serious risks for communities and the environment
- Caretakers of infected persons at the household level need to observe basic IPC for household members' safety and for patient protection
- Protects the community from infectious agents.

4.0 GOAL AND OBJECTIVES OF INFECTION PREVENTION AND CONTROL GUIDELINES

4.1 OVERALL GOAL

The overall goal of IPC is to achieve safe, effective healthcare practices at all health care facilities. The aim of these guidelines for infection prevention and control is to provide a comprehensive reference for all healthcare service providers in healthcare settings in Tanzania.

4.2 OBJECTIVES

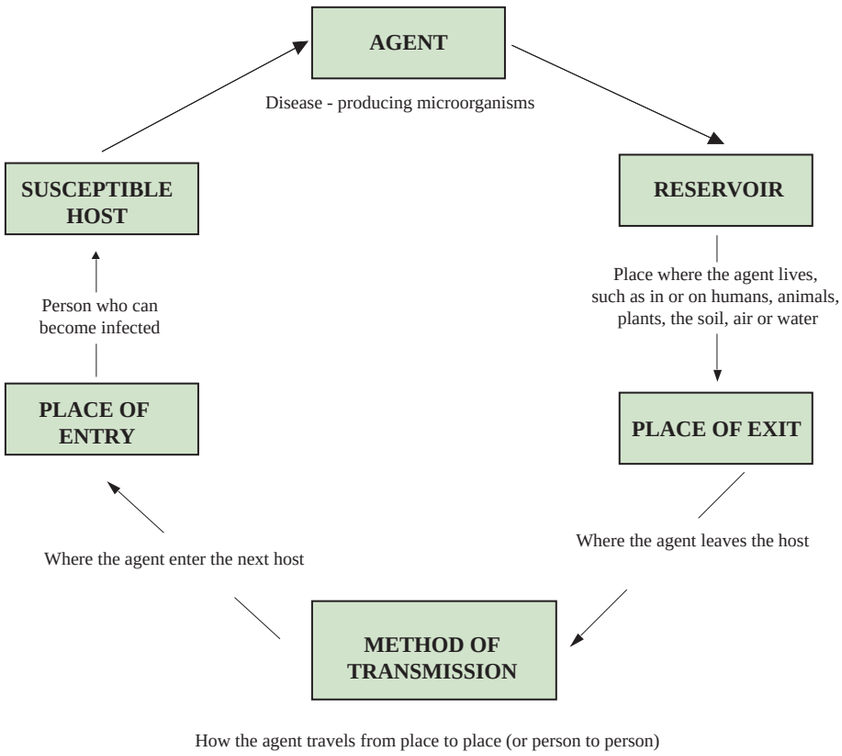
1. To protect patients /clients from nosocomial infections
2. To protect health care workers from occupational infections
3. To protect communities from infectious diseases
4. To prevent the environment from pollution

FUNDAMENTALS OF INFECTION PREVENTION AND CONTROL

5.0 THE INFECTIOUS DISEASE TRANSMISSION CYCLE

For bacteria, viruses and other infectious agents to successfully survive and spread, certain factors or conditions must exist. The essential factors in the transmission of disease-producing microorganisms from person to person are illustrated in Figure 1.

Figure 1. The Infectious Disease Transmission Cycle



Source: Adapted by JHPIEGO from APIC 1983; WPRO/WHO 1990.

Agent: The source of the infectious agent may be patients or clients, health workers, or visitors. It may include persons with active disease, those in the incubation period of the disease or those who are colonized by the infectious agent, but have no apparent disease (carriers). Other sources of infectious microorganisms can be the patient's own endogenous flora (autogeneous infection), which may be difficult to control, and inanimate environmental objects that have become contaminated, including equipment and medications.

Reservoir: The reservoir is the second element in the spread of infection. The reservoir is the source, human or otherwise, or habitat of the infectious agent, where it can survive for long periods if undisturbed.

Transmission Microorganisms are transmitted in healthcare facilities through several routes, and the same microorganisms may be transmitted by more than one route. There are five (5) modes of transmission (see also Section 5.1 below):

- Contact
- Droplet
- Airborne
- Common vehicle
- Vector borne

Susceptible Host Persons lacking effective resistance to a particular microorganism are susceptible to those microorganisms. Resistance to pathogenic microorganisms varies greatly. Some persons may be immune or able to resist colonization by an infectious agent; others exposed to the same agent may establish a commensal relationship with the infecting microorganism and become asymptomatic carriers, and still others may develop a clinical disease.

Host features that may render patients more susceptible to infection are: age; underlying diseases such as diabetes; certain treatments with antimicrobials, corticosteroids or other immunosuppressive agents; irradiation; and breaks in the first line of defense mechanism caused by such factors as surgical operations, anesthesia, and indwelling catheters.

5.1 NOSOCOMIAL INFECTIONS AND THEIR TRANSMISSION

Definition

Nosocomial infections are infections that are acquired in the health facilities. They are also called healthcare facilities-acquired infections. These infections may be transmitted from either patient to patient, patient to health worker or can be acquired from environmental factors such as air or contaminated water, food, drugs, furniture, medical equipment and other objects.

The most important transmission routes within the healthcare facilities are **contact**, **droplet and airborne. Common vehicle and vector borne** are additional routes of transmission.

5.1.1 Contact Transmission

This is the most important and most frequent mode of transmission of nosocomial infection and is divided into two sub-groups: direct-contact transmission and indirect-contact transmission. Direct-contact transmission involves a direct body surface-to-body surface contact and physical transfer of microorganisms between an infected or colonized person and a susceptible host. Indirect-contact transmission involves contact of a susceptible host with a contaminated intermediate object, usually inanimates, such as contaminated instruments, needles or dressings, or contaminated hands and gloves.

5.1.2 Droplet Transmission

Droplets are generated from the source person primarily during coughing, sneezing and talking or during the performance of certain procedures such as resuscitation, suctioning and bronchoscopy. Transmission occurs when droplets containing microorganisms generated by the infected person are propelled a short distance through the air and deposited on the host's conjunctivae, nasal mucosa, or mouth. For transmission to occur, the source and the susceptible host need to be within approximately one meter (3 feet) of one another.

5.1.3 Airborne Transmission

Airborne transmission occurs by dissemination of either airborne droplet nuclei (small-particle residue) of evaporated droplets containing microorganisms that remain suspended in the air for long periods of time, or of dust particles containing the infectious agent. Microorganisms carried in this manner can be dispersed widely by air currents and may be inhaled by a susceptible host within the same room or over a long distance from the source patient, depending on environmental factors. Microorganisms transmitted by airborne transmission include:

- Mycobacterium tuberculosis
- Rubella
- Varicella viruses

Such transmission may result in an explosive outbreak.

5.1.4 Common Vehicle Transmission

Common vehicle transmission applies to microorganisms transmitted by contaminated items such as:

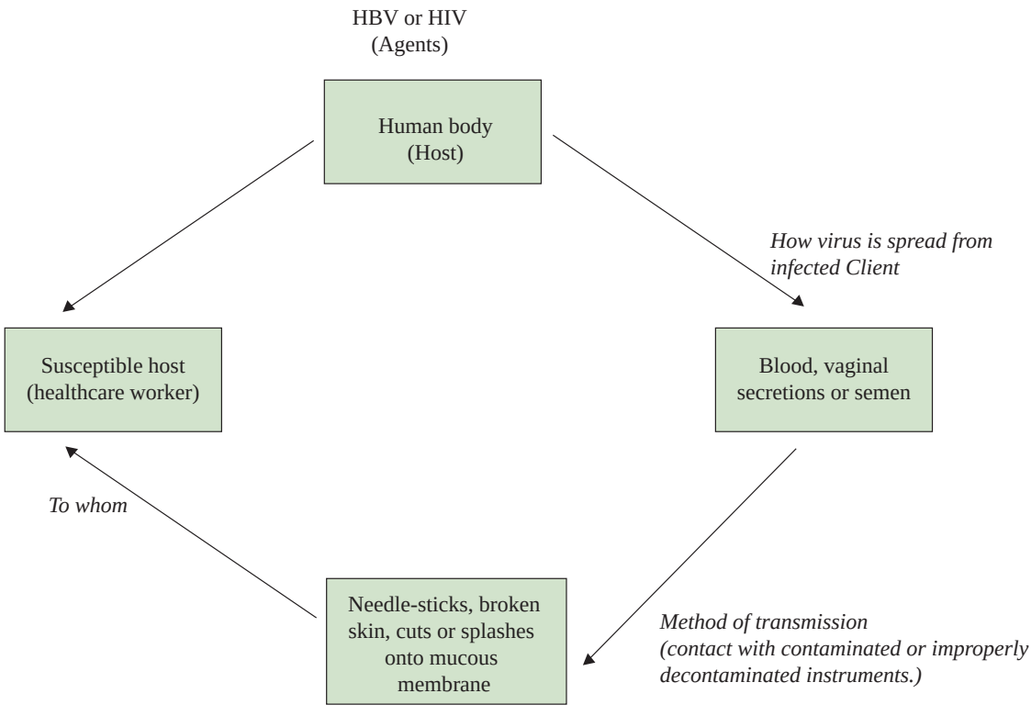
- Foods – e.g., salmonella
- Water – e.g., shigellosis
- Injections/intravenous solutions – e.g., HIV, Hepatitis B
- Blood – e.g., Hepatitis B, C, HIV
- Equipment and devices – e.g., HIV, Hepatitis B

5.1.5 Vector-Borne Transmission

Vector-borne transmission refers to transmission by animal, including insect, vectors. Vector-borne transmission occurs when vectors such as mosquitoes, flies, rats and other vermin transmit microorganisms.

Figure 2 depicts the steps in the transmission of the hepatitis B (HBV) and human immunodeficiency (HIV) viruses from colonized patients or clients (e.g., a family planning client or a pregnant woman attending an antenatal clinic) to healthcare workers. Spread of these viruses from person to person can occur when a staff member (physician, nurse or housekeeping personnel) is exposed to the blood or body fluids of an infected person (e.g., needle-stick injury).

Figure 2. Transmission of HBV and HIV from Patients to Healthcare Workers



Source: *Tietjen, L., D. Bossemeyer and N. McIntosh. (2003) Infection Prevention - Guidelines for Healthcare Facilities with Limited Resources. JHPIEGO Corporation. Baltimore, Maryland.*

6.0 HEALTHCARE WORKER SAFETY AND STANDARD PRECAUTIONS

6.1 HEALTHCARE WORKER SAFETY

6.1.1 Background

Health care workers are exposed to blood and other body fluids in the course of their work. Consequently, they are at risk of infection with blood borne viruses including Human Immunodeficiency Virus (HIV), Hepatitis B virus (HBV) and Hepatitis C Virus (HCV). The risk of infection for health workers depends on the prevalence of disease in the patient population and the nature and frequency of exposure.

6.1.2 Occupational Exposure

Occupational exposure to blood can result from percutaneous injury (needle-stick or other sharp injury), mucocutaneous injury (splash of blood or other body fluids into the eyes, nose or mouth) or blood contact with non-intact skin. The most common form of occupational

exposure to blood and the most likely to result in infection is needle-stick injury. The most common causes of needle-stick injury are two-handed recapping and unsafe collection and disposal of sharps. Healthcare workers in such areas as operating, delivery, and emergency rooms and laboratories have a higher risk of exposure. Cleaners, healthcare waste collectors and others whose duties involve handling blood-contaminated items are also at risk.

6.1.3 Strategies to Protect Healthcare Workers

Most exposure to blood and body fluids in health care settings are preventable.
Strategies to protect health workers include:

- implementation of Standard Precautions (see below)
- immunization of all health workers, especially those working in health care settings, against Hepatitis B
- provision of personal protection
- the management of exposures
- elimination of unnecessary sharps and injections

Successful implementation of these strategies requires an effective quality improvement or infection prevention committee with support from the health setting management team.

6.2 STANDARD PRECAUTIONS

6.2.1 Background

With the emergence of the HIV/AIDS epidemic in the 1980's, Universal Precautions (UP) was developed by CDC to protect healthcare workers from becoming infected with HIV and other blood-borne infections (e.g., HCV). However, Universal Precautions did not address the risks to patients, risks from other potentially infected body fluids and did not recognize that most HIV- infected people do not have symptoms (CDC 1985).

In 1987 a new system of health worker and patient precautions was introduced as an alternative to the diagnosis-driven Universal Precautions system called Body Substance Isolation (BSI) (Lynch et al., 1987). Body Substance Isolation (BSI) includes:

- protecting patients and health personnel from all moist and potentially infected body substances (secretions and excretions), not just blood

- protecting susceptible patients and staff against infectious diseases that are transmitted by airborne or droplet routes through immunization, such as measles, mumps, chickenpox and rubella, as well as blood borne pathogens hepatitis B and C and HIV

- revised instructions to persons wishing to enter a patient's room or care for patients with infections transmitted by the airborne route (Lynch et al., 1990) the use of gloves.

Disadvantages of BSI include the added cost of protective barrier equipment, particularly gloves; overuse of gloves to protect staff at the expense of patients (Patterson et al., 1991); and the lack of standards/protocols for use with patients in isolation. **New guidelines** issued by CDC in 1996 involve a two-level approach:

- Standard Precautions that apply to all clients and patients attending health care facilities and Transmission-Based Precautions that apply only to hospitalized patients (Garner and HICPAC, 1996). (See also Section 17 of this manual on Preventing Nosocomial Infection.)

6.2.2 Definition of Standard Precautions

These are simple set of effective practice guidelines (creating a physical, mechanical and chemical barrier) to protect healthcare workers and patients from infection with a range of pathogens including blood-borne pathogens. The practices are used when caring for all patients regardless of diagnosis.

6.2.3 Reasons for Setting Standard Precautions

Standard Precautions are designed:

- for the care of all persons, patients, clients and staff, regardless of whether or not they are infected
- for handling of blood and all other body fluids, secretions and excretions (except sweat), non-intact skin and mucous membranes
- to reduce the risk of transmitting microorganisms from known or unknown sources of infection (e.g., patients, contaminated objects, used needles and syringes, etc.) within the healthcare system

Standard Precautions has become the primary strategy for preventing nosocomial infections in healthcare settings.

6.2.4 Components of Standard Precautions

The key components of Standard Precautions include:

- Consider every person (Patient or staff) as potentially infectious and susceptible to infection
- **hand hygiene** including handwashing, hand antisepsis, antiseptic handscrub and surgical handscrub
- **personal protective equipment (PPE)** including gloves, masks, goggles, caps, gowns, boots and aprons.
- **appropriate handling** of sharps, patient resuscitation and patient care equipment, linen, patient placement and patient environmental cleaning
- Safe disposal of infectious waste materials to protect those who handle them and prevent injury or spread to the community.
- Process instruments by decontamination, cleaning and then either sterilization or HLD using recommended procedure.

6.2.5 Interventions of Standard Precautions

In practice, implementation of Standard Precautions includes the following interventions:

- handwashing before and after any direct contact with patients
- Do not recap needles
- safe collection and disposal of needles (hypodermic and suture) and sharps (scapel blades, lancets, razors, scissors), with required puncture-proof and liquid-proof safety boxes in each patient care area
- wearing gloves for contact with body fluids, non-intact skin and mucous membranes
- wearing a mask, eye protection and gown (and sometimes a plastic apron if blood or other body fluids might splash
- covering all cuts and abrasions with waterproof dressing
- promptly and carefully cleaning up spills of blood and other body fluids
- using a safe system for healthcare waste management and disposal.

Table 1: Strategies to Protect Health Workers

Hepatitis B Immunization	Personal Protection	Post-exposure Management
Routine immunization of health workers against infection with HBV is an effective way to protect them. HBV is the most infectious blood-borne virus and the most prevalent. Long-term sequelae of HBV infection include liver cirrhosis and hepato cellular carcinoma. HB vaccine is effective, cost-effective relatively inexpensive (less than US$ 0.5 a dose) and widely available. • Immunize all health workers • Pre-vaccination serological testing is not necessary • Use a 0, 1 and 6 months schedule of three injections • If possible, measure antibody levels between two to six months after the last dose • Do not administer boosters routinely as protection is life long.	Personal protective equipment includes gloves, goggles or glasses, masks, gowns and plastic aprons. • Use needle-stick prevention devices (i.e., devices where the sharp is sheathed or retracted after use) • Ensure adequate supplies of personal protective equipment in all areas • Involve staff in the selection of personal protective equipment as equipment that is of poor quality or uncomfortable to wear will not be used • Train staff in the correct use of personal protective equipment • Use influential senior staff as role models to promote the use of personal protective equipment • Monitor compliance and inappropriate use. Inappropriate use of gloves wastes resources. Compliance with eye protection often requires additional efforts • Dispose of used personal protective equipment safely.	The risk of infection following a needle-stick injury with a needle from an infected source patient is 0.3% for HIV, 3% for hepatitis C and 6-30% for hepatitis B. An effective response to occupational exposure to blood or other body fluids involves: • clear policy guidelines and procedures posted in visible places • confidentiality of exposed and source person • management of exposures training of healthcare personnel • rapid access to: - clinical care - post-exposure prophylaxis (PEP) - testing of source patients/exposed persons • injury prevention assessment **Elements of Post-exposure Management** • Wound management • Exposure reporting • Assessment of infection risk - type and severity of exposure - blood-borne infection status of source person • Appropriate treatment, follow-up, and counseling. See also Section 11.4.6: PEP guidelines.

7.0 HAND HYGIENE

Hand hygiene includes care of hands, nails, skin, and the use of lotions and surgical scrub. Failure to perform appropriate hand hygiene is considered to be a leading cause of no socomial (hospital-acquired) infections and the spread of multiresistant micro-organisms, and has been recognized as a significant contributor to outbreaks of disease (Boyce and Pittet 2002).

Hand hygiene can be accomplished by:

- Handwashing with or without antiseptic agent, see 7.1 below
- Surgical hand scrub
- Antiseptic hand rub using a waterless, alcohol-based antiseptic agent

7.1 TYPES OF HAND HYGIENE

7.1.1 Routine Handwashing

Definition

Handwashing is a process of mechanically removing soil, debris and organisms from the skin using plain soap and water.

Four elements are essential for effective handwashing:

- Soap
- Running water
- Friction
- Drying

When Handwashing Should Be Done

- Before and after eating, after using the toilet and when soiled
- Immediately on arrival at work and before leaving work
- Before and after each patient contact
- Before and after gloves are removed
- Before putting on gloves for performing clinical and invasive procedures (e.g., insertion of an IUD)
- Before preparing, handling, serving or eating food, and before feeding a patient
- Before medication preparation
- Whenever there is a chance of contamination

Handwashing should also be done after the following:

- Touching blood, body fluids, secretions, excretions, and exudates from wounds
- Contact with items known or considered likely to be contaminated with blood, body fluids, secretions, or excretions (e.g., bedpans, urinals, wound dressings) whether or not gloves are worn

- Attending to children's toilet needs
- After personal body functions such as using the toilet, wiping or blowing one's nose

Handwashing should be done between all procedures done on the same patient where soiling of hands is likely, to avoid cross-contamination of body sites.

Patients and family members should be educated in proper handwashing.

How To Wash Hands

- Turn on tap.
- Wet hands thoroughly under running water to at least 4 inches above the wrist.
- Soap hands adequately.
- Handwashing should be done by vigorously rubbing together all surfaces of lathered hands.
- Rub hands vigorously back and front, in between fingers up to and including the wrist, followed by thorough rinsing under running water. This should be for 10 – 15 seconds.
- Dry hands from tip of fingers to wrist with paper towel. If paper towels are not available, shake off excess water and allow hands to air-dry.
- Use the same paper towel to turn off tap if tap not elbow controlled.

Important Notes

- Immediate re-contamination of the hands by touching sink fixtures may be avoided by using paper towel to turn off taps.
- When running tap water is not available, use a bucket with a tap that can be turned on to wet hands, off to lather hands and turned on again for rinsing. Design of the taps/sinks and the right purchase of the taps, e.g., elbow, is desirable.
- If a bucket with a tap is not available, a bucket/basin and pitcher can be used to create a running stream of water. A helper can pour water from the pitcher over the hands being washed.
- Handwashing should not be repeated in the same container of water.
- Hands should be dried with paper towels/sterile towels per procedure.

7.1.2 Handwashing with Antiseptic and Running Water or with Alcohol Hand Rub

removes transient microorganisms, dirt and kills or inhibits the growth of resident microorganisms It also may reduce the risk of infections in high-risk situations such as:

- when there is heavy microbial contamination before performing invasive procedures, (e.g., the placement and care of intravascular devices, indwelling urinary catheters)
- before contact with patients who have immune defects, damage to the integumentary system (e.g., burns, wounds) and pericutaneous implanted devices
- before and after direct contact with patients who have antimicrobial-resistant organisms

7.1.3 Alcohol Hand Rub is only one kind of antiseptic hand rub. It

- kills or inhibits the growth of most transient and resident micro-organisms, but does not remove micro-organisms or dirt
- can be used when handwashing with soap and running water is not possible, as long as hands are not visibly soiled with dirt, blood, or other organic material

The use of an antiseptic hand rub is more effective in killing transient and resident flora than handwashing with antimicrobial agents or plain soap and water; it is quick and convenient to perform, and gives a greater initial reduction in hand flora (Girou et al., 2002). Antiseptic hand rubs also contain a small amount of an emollient such as glycerin, propylene glycol or sorbitol that protects and softens skin.

The steps for performing antiseptic hand rub include the following.

Step 1: Apply enough antiseptic hand rub to cover the entire surface of hands and fingers (about a teaspoonful).

Step 2: Rub the solution vigorously into hands, especially between fingers and under nails, until dry.

To be effective, an adequate amount of hand rub solution should be used. For example, by increasing the amount of hand rub from 1 mL to 5 mL per application (about 1 teaspoonful), the effectiveness is increased significantly (Larson, 1988).

Since antiseptic hand rubs do not remove soil or organic matter, if hands are visibly soiled or contaminated with blood or body fluids, handwashing with soap and water should be done first.

Antiseptic agents: Liquid soap with or without antimicrobial agent.

Antiseptic agents recommended are: Povidone-iodine 7.5% surgical scrub or Chlorhexidine 5% surgical scrub (undiluted).

Alcohol-Based Solution for Hand Rub

A non-irritating antiseptic hand rub can be made by adding glycerin[a], propylene glycol or sorbitol to alcohol (2mL in 100mL of 60-90% ethyl or isopropyl alcohol solution) (Larson, 1990; Pierce, 1990). Use 5mL (about one teaspoonful) for each application and continue rubbing the solution over the hands until they are dry (15-30 seconds).

[a] Glycerin is often sold in cosmetic departments because it is used as a hand softener.

7.1.4 Surgical Hand Scrub

Definition: Scrubbing of hands with soap, water, antiseptic and friction.
Note: The use of a brush is not recommended.

When is it done? Before beginning surgical procedures.

Purpose of surgical hand scrub

- To prevent wound contamination by microorganisms from hands and arms of surgeons and assistants
- To prevent the growth of microorganisms (scrubbing with antiseptic before beginning surgical procedures)

Steps of the surgical hand scrub procedure

Step 1: Remove hand/arm-worn jewellery, e.g., rings, watches, bracelets.

Step 2: Wet hands and arms up to the elbow under clean running water, always holding hands with fingers up in a vertical position.

Step 3: Clean nails with a nail cleaner.

Step 4: Apply soap generously.

Step 5: Using a circular motion to avoid abrasions, begin at the fingertips of one hand and lather and wash between the fingers, continuing from fingertips to elbow; continue washing for 3-5 minutes.

Step 6: Wash surfaces between fingers, sides of hands, tips of fingers, palms and dorsum of hands up to the elbow of one arm.

Step 7: Repeat procedure for the second hand and arm.

Step 8: Rinse each arm separately, fingertips first, holding hands above the level of the elbow.

Step 9: Dry hands in fingers-up, vertical position with a sterile towel; wipe from the fingertips to the elbow.

Step 10: Apply 5 ml (about one teaspoonful) of a waterless, alcohol-based hand rub to hands, fingers and forearms and rub until dry; repeat application and rubbing 2 more times for a total of at least 2 minutes, using a total of about 15 ml (3 teaspoonfuls) of hand rub.

7.2 SKIN CARE

It is important to note the following.

- Frequent handwashing and gloving can irritate skin.
- Handwashing cannot reduce the bacterial counts of personnel with dermatitis.
- Staff responsible for processing instruments and who have open sores or cuts on their hands or forearms should not clean instruments until the lesions are healed unless covered with waterproof dressings.
- Healthcare providers with dermatitis carry high numbers of microorganisms and may be at increased risk of exposure to blood-borne pathogens. Intact skin is a major defense against infection.
- Lotion can ease the dryness resulting from frequent handwashing. It can also help prevent dermatitis from frequent glove use.

7.3 IMPROVING HAND HYGIENE PRACTICE

In Tanzania conducive/enabling environments should be created and maintained to improve health workers' hand hygiene practices. These include:

- provision of adequate water supply, soap, disinfectants and antiseptics
- provision of facilities for running water
- creating awareness of all health professionals on the importance of improving hand hygiene practices

Although it is difficult to change behaviour in this area, there are certain steps that can increase the chances of success. These steps include the following.

- Widely disseminate current guidelines for hand hygiene practices, the evidence supporting their effectiveness in preventing disease and the need for health workers to adhere to the guidelines.
- Involve hospital administrators in promoting and enforcing the guidelines by convincing them of the cost benefits of handwashing and other hand hygiene practices.
- Use successful educational techniques including role modeling (especially by supervisors), mentoring, monitoring, and positive feedback.
- Use participatory performance improvement approaches targeted to all healthcare staff, not just physicians and nurses, to promote compliance.
- Consider the needs of staff for convenient and effective options for hand hygiene that make compliance easier.

7.4 OTHER ISSUES AND CONSIDERATIONS RELATED TO HAND HYGIENE

Hand hygiene, coupled with the use of protective gloves, is a key component in minimizing of the spread of diseases and maintaining an infection-free environment (Garner and Facero, 1986). In addition, understanding when sterile or clean gloves are required and, equally important, when they are not, can reduce costs while maintaining safety for both patients and staff.

Healthcare workers wear gloves for three reasons:

- to reduce the risk of staff acquiring bacterial infections from patients
- to prevent staff from transmitting their skin flora to patients
- to reduce contamination of the hands of staff by microorganisms that can be transmitted from one patient to another (cross-contamination).

For types of gloves, see Section 8.2.1 below.

7.4.1 Hand Lotions and Hand Creams

Several studies have shown that regular use (at least twice per day) of such products can help prevent and treat contact dermatitis (McCormick et al., 2000). In addition, moisturizers can prevent drying and damage to the skin and loss of skin fats. There is also biological evidence that emollients, such as glycerol and sorbitol, with or without antiseptics, may decrease cross-contamination because they reduce shedding of bacteria from skin for up to four hours.

7.4.2 Lesions and Skin Breaks

Cuticles, hands and forearms should be free of lesions (dermatitis or eczema) and skin breaks (cuts, abrasions and cracking). Cuts and abrasions should be covered with waterproof dressings. If covering them is not possible, surgical staff with skin lesions should not operate until the lesions are healed.

7.4.3 Fingernails

Research has shown that the area around the base of nails (subungual space) contains the highest microbial count on the hand (McGinley, Larson and Leydon, 1988). In addition, several recent studies have shown that long nails may serve as a reservoir for gram-negative bacilli (P. aeruginosa), yeast and other pathogens (Hedderwick et al., 2000). Moreover, long nails, either natural or artificial, tend to puncture gloves more easily (Olsen et al., 1993). As a result, it is recommended that nails be kept moderately short – not extend more than 3 mm (or 1/8 inch) beyond the fingertip.

7.4.4 Artificial Nails

Artificial nails (nail wraps, nail tips, acrylic lengtheners, etc.) worn by healthcare workers can contribute to nosocomial infections (Hedderwick et al., 2000). In addition, because there is evidence that artificial nails may serve as a reservoir for pathogenic gram-negative bacilli, their use by health workers should be restricted, especially by surgical team members, and those who:

- work in specialty areas such as neonatal ICUs
- care for patients highly susceptible to infection
- manage patients who have infections with resistant organisms (Moolenaar et al., 2000)

7.4.5 Nail Polish

Although there is no restriction on wearing nail polish, it is suggested that surgical team members and those staff working in specialty areas may wear freshly applied, clear nail polish. Chipped nail polish supports the growth of larger numbers of organisms on finger nails compared to freshly polished or natural nails. Also, dark-colored nail polish may prevent dirt and debris under fingernails from being seen and removed (Baumgardner et al., 1993).

7.4.6 Jewelry

Although several studies shown that skin under rings is more heavily colonized than comparable areas of skin on fingers without rings (Jacobson et al., 1985), at the present time it is not known whether wearing rings results in greater transmission of pathogens. It is suggested that surgical team members not wear rings because it may be more difficult for them to put on surgical gloves without tearing them.

Table 2: Soaps and Antiseptic/Antimicrobial Agents for Handwashing

Products	Indications	Special Considerations
Plain soap, bar soap, liquid soap granules	• For routine care of patients • For washing hands soiled with dirt, blood or other organic material	• May contain very low concentrations of antimicrobial agents to prevent microbial contamination growth in the product • Bar soap should be on racks that allow water to drain; small bars that can be changed frequently are safest
Waterless antiseptic agents: • Alcohol rinses • Alcohol foams • Alcohol wipes • Alcohol novelettes • Germicidal hand rinse (Hibistat)	• Demonstrated alternative to conventional agents • For use where handwashing facilities are inadequate, impractical or inaccessible (e.g., ambulances, home care, mass immunization) • For situations in which the water supply is interrupted (e.g., planned disruptions, natural disasters)	• Not effective if hands are soiled with dirt or heavily contaminated with blood or other organic material • Follow manufacturer's recommendations for use • Efficacy affected by concentration of alcohol in product • Lotions should be readily available to protect skin integrity
Antiseptic/Anti-microbial agents: • Chlorhexidine gluconate scrub strengths: 2% aqueous foam or 4% liquid preparation, 0.5% tincture • Povidone-iodine scrub strengths: 10%, 7.5%, 2%, 0.5%	• May be chosen for hand scrubs prior to performance of invasive procedures (e.g., placing intravascular lines or devices) • When caring for severely immunocompromised patients • Based on risk of transmission (e.g., specific micro-organisms) • Critical care areas • Intensive care nurseries • Operating theatre hand scrub • When caring for individuals with antimicrobial resistant organisms	• Antiseptic agents may be chosen if it is felt important to reduce the number of resident flora or when the level of microbial contamination is high • For use in high risk areas such as ICUs, neonatal units, operating theatres, labor and delivery rooms, isolation areas, laboratory and dialysis units, for invasive procedures • Antiseptic agents should be chosen when persistent antimicrobial activity on the hand is desired; they are usually available in liquid formulations; antiseptic agents differ in activity and characteristics

Table 3: Proposed Strategies to Improve Handwashing Techniques and Compliance

Obstacle	Strategy
Lack of knowledge	• Education with supportive literature, videotaped instructions, handwashing demonstrations; frequent involvement of personnel in education and feedback on infection rates
Lack of motivation	• Direct observation and feedback on a regular basis, role models; involvement of staff in studies; application of new technologies • Programmes on hand hygiene for patients and families
Unavailability of handwashing facilities	• Handwashing facilities conveniently located throughout the healthcare facility • Available running water • Handwashing facilities in or adjacent to rooms where healthcare procedures are performed • Accessible, adequate supplies of soap and disposable towels • Waterless antiseptic agents readily available in wall-mounted dispensers, or in small containers for mobile care such as home care and for emergency responders
Non-acceptance of handwashing products	• Availability of handwashing products that have a high level of acceptability to staff, with appropriateness, cost, supply, etc. being taken into consideration
Dermatitis	• Lotions to prevent skin dryness • Lotion supplies in small non-refillable containers • Compatibility between lotion and antiseptic products and effect on glove integrity • Lotions approved by the Infection Prevention and Control Committee

8.0 PERSONAL PROTECTIVE EQUIPMENT - BACKGROUND

Protective barriers, now commonly referred to as personal protective equipment (PPE), have been used for many years to protect patients from microorganisms present on staff working in healthcare settings. More recently, with the emergence of AIDS, HCV, SARS and the resurgence of tuberculosis in many countries, use of PPE now has become important for protecting staff as well. As a consequence, hospital administrators, supervisors and healthcare workers need to be aware not only of the benefits and limitations of specific PPE, but also of the actual role PPE play in preventing infection so that they can use them effectively and efficiently.

8.1 TYPES OF PERSONAL PROTECTIVE EQUIPMENT

Personal protective equipment includes: gloves, masks/respirators, eyewear (face shields, goggles or glasses), caps, gowns, aprons and other items.

8.1.2 Gloves

Gloves protect hands from infectious materials and protect patients from microorganisms on staff members' hands. They are the most important physical barriers for preventing the spread of infection. There are **three types of gloves:**

- Surgical gloves (sterile/single use)
- Examination gloves (non-sterile)
- Heavy duty/utility/household gloves

When to use gloves and the types of various procedures

- Gloves should be worn when contact with body and blood fluids is anticipated.
- Gloves should be worn as additional measures, not as a substitute for handwashing.
- Gloves are not required for routine care activities in which contact is limited to a patient's skin.
- Examination gloves shall be worn:
  - for examination and non-surgical procedures
  - for contact with blood, body fluids, secretions and excretions, mucous membranes, draining wounds or non-intact skin (open skin lesions or oxidative rash)
  - for handling items visibly soiled with blood, body fluids, secretions or excretions
  - when the healthcare worker has non-intact skin on his/her hands
  - when inserting an intravenous line
- Surgical gloves shall be worn for surgical and invasive procedures
- Utility gloves are used for decontamination of large equipment, cleaning of floors, walls, healthcare facility furniture such as beds, etc.
- Gloves shall be changed between care activities and procedures with the same patient after contact with materials that may contain high concentrations of microorganisms.
- Gloves shall be removed before moving to another patient or after completion of specific task.

- Hands shall be washed and dried immediately after removing gloves.
- With the exception of utility gloves other gloves shall not be washed, decontaminated and reused.
- Gloves shall not be worn while walking in corridors and traveling in elevators, unless in special circumstances, e.g., transporting laboratory specimens.

8.1.3 Double Gloving

The transmission of HBV and HCV from surgeon to patient and vice versa has occurred in the absence of breaks in technique and with apparently intact gloves (Davis, 2001a). Even the best quality, new latex rubber surgical gloves may leak up to 4% of the time. Moreover, latex gloves, especially when exposed to fat in wounds, gradually become weaker and lose their integrity.

Although double gloving is of little benefit in preventing blood exposure if needle sticks or other injuries occur, it may decrease the risk of blood-hand contact. For example, one recent study showed that surgeons wearing single gloves had a blood-hand contact rate of 14% while surgeons wearing double gloves had only a rate 5% (Tokars et al., 1995; Tokars et al., 1992).

Based on this study, the following are reasonable guidelines for when to **double glove:**

- when the procedure involves coming into contact with a large amount of blood or other body fluids (e.g., vaginal deliveries and cesarean sections)
- for orthopedic procedures in which sharp bone fragments, wire sutures and other sharps are likely to be encountered

8.1.4 Elbow Length Gloves for Obstetrical Procedures

Blood contact with the skin and mucous membranes of providers occurs in 25% of vaginal deliveries and 35% of cesarean sections (Davis, 2001b). Where the hand and forearm need to be inserted into the vagina (manual removal of a retained placenta) or deep into the uterus to deliver the infant's head (cesarean section), elbow-length, so-called "gauntlet" gloves, help protect the provider from significant blood and amniotic fluid contamination. If gauntlet gloves are not available, an inexpensive, effective alternative can be made easily using the following steps.

Step 1: Perform surgical hand scrub, including the forearms up to the elbows, as detailed in section 7.1.4 above, using an alcohol-based antiseptic agent.

Step 2: Put intact Sterile Surgical gloves on both hands.

Step 3: Cut the four fingers completely off each sterile surgical glove just below where all the fingers join the glove.

Step 4: Put fingerless, sterile gloves on both hands and pull up onto the forearm(s).

Step 5: Put intact sterile surgical gloves on both hands so that the distal end of the fingerless glove is completely covered. In this way the fingerless glove will completely cover the forearm and the intact glove will cover the hand, overlapping the fingerless glove at mid-arm.
(Tietjen, L., D. Bossemeyer and N. McIntosh, 2003)

8.1.5 Standard Operating Guidelines for Gowns

1. The unnecessary use of gowns is **not** recommended.
2. Gowns shall be used for protective isolation.
3. Gowns shall **not** be worn outside the area for which they are intended.
4. Long gowns shall be worn to protect uncovered skin and to prevent soiling of clothing during procedures and patient care activities likely to generate splashes or sprays of blood, body fluids, secretions, or excretions. Plastic aprons are recommended where splashes are likely to occur. Clinical coats and scrub suits should remain in the working place; taking them home increases the risk of infection to the home environment.

8.1.6 Surgical Masks

Masks are worn in an attempt to contain moisture droplets expelled as health workers or surgical staff speak, cough or sneeze, as well as to prevent accidental splashes of blood or other contaminated body fluids from entering the health worker's nose or mouth. Unless the masks are made of fluid-resistant materials, they are not effective in preventing either very well.

There are three types of masks:

1. The tieback mask, which has four ties to fasten the mask around the mouth and nose. The side of the mask with the flexible metal tab is worn away from the face with the metal tab placed above the bridge of the nose to help secure the mask and minimize air escaping from the sides (venting).
2. The ear-loop mask is similar to the tieback mask except that it has two elastic bands used for fastening.
3. Surgical masks have attached faced shields to help provide a protective barrier against splashes and spatters of blood or other infectious material. These masks are fluid resistant, lightweight, and adequate for most procedures and isolation precautions.

Standard Operating Guidelines for Masks

1. Masks should be large enough to cover the nose, lower face, jaw and facial hair.
2. Masks shall be worn where appropriate to protect the mucous membranes of the nose and mouth of the service provider during procedures and patient care.

Note: *A surgical mask becomes ineffective as a barrier if the integrity is damaged or if it becomes wet (i.e., from perspiration, or if splashed with blood or other potentially infectious material). If this occurs, remove mask and replace with another.*

8.1.7 Caps

Caps are used to keep the hair and scalp covered so that flakes of skin and hair are not shed into the wound during surgery. Caps should be large enough to cover all hair. While caps provide some protection to the patient, their primary purpose is to protect the wearer from blood and body-fluid splashes and sprays.

8.1.8 Protective Eye Wear

By covering the eyes, protective eyewear protects staff from accidental splashes of blood or body fluid.

Four Types of Eye Wear

1. Plastic glasses with solid side shields
2. Goggles
3. Masks with clear visors
4. Chin-length face shields

Standard Operating Guidelines for Eye Wear

1. Protective eye wear shall be worn where appropriate to protect the mucous membranes of the eyes during procedures and patient care activities likely to generate splashes or sprays of blood, body fluids, secretions, and excretions.
2. Use protective eye wear that is appropriate for the particular procedure.
3. Holding goggles with one hand, lift the bottom strap from the back of the head to the front.
4. If gloved hands are used for these procedures, the gloves should not be contaminated with blood or other potentially infectious material.

Generally, if protective eye wear, mask, gown and gloves are worn, the order for the removal should be:

- Protective eye wear
- Mask
- Gown
- Gloves

Note:

1. *Single-use protective barriers should be discarded into the appropriate receptacle(s).*
2. *Re-usable protective barriers should be decontaminated, cleaned, and disinfected according to the appropriate guidelines.*
3. *Wash hands and dry after removing protective barriers.*

8.1.9 Boots (Footwear)

Footwear is worn to protect feet from injury by sharps or heavy items, blood and fluids.

Standard Operating Guidelines for Footwear

1. Rubber or leather boots are recommended because they protect better; they should be kept clean and free of contamination from blood or other fluid spills.
2. Shoe covers are unnecessary if clean, closed-toe, sturdy shoes are available for use only in surgical areas.

8.1.10 Apron

The apron is made of rubber or plastic to provide a waterproof barrier along the front of the health worker's body.

Standard Operating Guidelines for Aprons

1. An apron should be worn when cleaning or during a procedure in which blood or body spills are anticipated.

Table 4: How Personal Protective Equipment Blocks the Spread of Microorganisms

WHERE MICROOGANISMS ARE FOUND	HOW MICROORGANISMS ARE SPREAD	BARRIERS TO STOP THE SPREAD OF MICROORGANISMS	WHO THE BARRIER PROTECTS
Healthcare staff			
hair and scalp	shedding skin or hair	cap	patient
nose and mouth	coughing, talking and sneezing	mask	patient
body and skin	shedding skin or hair	scrub suit, cover gown	patient
hands	touching	gloves, handwashing or waterless antiseptic hand rub	patient
Patient's mucous membranes and non- intact skin	touching	gloves	patient and staff
Patient's blood and body fluids	splashing or spraying	gloves, eyewear, mask, drapes, apron	staff
Patient's unprepared skin	touching (contact)	instrument processing	patient
		utility gloves	staff
	accidental exposure with contaminated needles and scalpel blades	protective footwear, decontamination and disposal; use a safe or neutral zone during surgery	staff
	infectious waste	utility gloves, plastic bags and disposal	staff and community
	touching	skin preparation, drapes, gloves.	patient
Clinic or hospital environment	touching	gloves, handwashing dressings	staff and their family staff and community

Source: Tietjen, L., D. Bossemeyer and N. McIntosh (2003). *Infection Prevention, Guidelines for Healthcare Facilities with Limited Resources*. JHPIEGO Corp., Baltimore, Maryland, page 5-4.

9.0 ANTISEPTICS AND DISINFECTANTS

Definitions

- **Antiseptic or antimicrobial agent** *(terms used interchangeably)*
  Chemicals that are applied to the skin or other living tissues to inhibit or kill microorganisms (both transient and resident) thereby reducing the total bacterial count.

- **Antisepsis**
  Process of reducing the number of microorganisms on the skin, mucous membranes or other body tissues by applying an antimicrobial antiseptic agent.

Note: *Antiseptics should not be used on inanimate objects such as instruments and surfaces; they do not have same killing power as disinfectants. Although antiseptics are some times used as disinfectants (e.g., Savlon or Dettol) for processing instruments and other inanimate object; they are not designed for this use. They do not have the same killing power as chemical disinfectants (e.g., glutaraldehydes, hypochlorite and per oxides) and should not be used for this purpose (Rutala, 1996).*

9.1 ANTISEPTIC AGENTS

- Liquid soap
- Antiseptics, which include 0.5% chlorhexidine with or without glycerol, and povidone-iodine. These reduce both transient and resident flora on the hands.
- They also reduce the risk of infections in high-risk situations, such as:
  - heavy microbial contamination
  - before performing invasive procedures, (e.g., the placement and care of intravascular devices and indwelling urinary catheters)
  - before contact with patients who have immune defects, damage to the integumentary system (e.g., burns, wounds) and pericutaneous implanted devices
  - before and after direct contact with patients who have antimicrobial resistant organisms.

9.2 TYPES AND SELECTION OF ANTISEPTICS

Many chemicals qualify as safe antiseptics and are designed to remove as many microorganisms as possible without damaging or irritating the skin or mucous membranes. Some antiseptic solutions have a residual effect (their killing action continues for a period of time). Hence they are recommended in daily use.

9.3 WHEN TO USE ANTISEPTICS

Use antiseptics

- before a clinical procedure involving skin, cervical, or vaginal preparation

- for surgical scrub
- for handwashing in high-risk situations, e.g., before, during and after performing invasive procedures, touching a newborn or an immunosupressed patient

9.4 SURGICAL ANTISEPSIS

Postoperative wound infections (incisional and deep) remains to be a leading cause of nosocomial infections in developing countries. The vast majority of postoperative incisional or superficial wound infections are caused by microorganisms (usually bacteria or sometimes fungi) normally found on a patient's skin or from mucous membranes adjacent to the site.

Note: *Surgical wound infectins are less often caused by organisms from the nose, mouth, respiratory tract, hands of surgeons and assistants and organisms from the operating room*

Preoperative surgical antisepsis consists of three processes:

- Hand hygiene
- Gloving
- Applying an antiseptic agent to the surgical site

Whether a postoperative wound infection occurs depends on several risk factors, the most important being:

- the number of microorganisms entering the wound
- the type and virulence (ability to cause the disease) of the bacteria
- the patient's immunity
- external factors: preoperative hospital stay days or duration of the surgical technique or procedure and surgical environment

Table 5: The Most Frequently used Antiseptics, their Microbiologic activities, potential use and effectiveness

GROUP	ACTIVITY AGAINST BACTERIA								POTENTIAL USES		
	Gram Positive	Most Gram Positive	TB	Viruses	Fungi	Endespores	Relative Speed of Action	Affected by Organic Matter	Surgical Scrub	Skin Preparation	Comments
Alcohols (60-90% ethyl propyl)	Excellent	Excellent	Excellent	Excellent	Excellent	None	Fast	Moderate	Yes	Yes	Not for use on mucous membrane Not good for physical cleaning of skin, no persistent activity
Chlorhexidine (2-4%) (Hibitane, Bibiscrub)	Excellent	Good	Fair	Excellent	Fair	None	Intermediate	Slight	Yes	Yes	Has good persistent effect Toxicity to ears and eyes
Iodine preparations	Excellent	Excellent	Excellent	Excellent	Good	Fair	Intermediate	Marked	No	Yes	Not for use on mucous membranes Can burn skin so remove after several minutes
Iodophors (7.5 - 10% Betadine)	Excellent	Excellent	Fair	Good	Good	None	Intermediate	Moderate	Yes	Yes	Can be used on mucous membranes
Para chlorometaxylenol (PCMX) (0.5 -4%)	Good	Excellent	Fair	Good	Fair	unknown	Slow	Minimal	No	Yes	Penetrates the skin and should not be used on newborns
Triclosan (0.2 - 2%)	Excellent	Good	Fair	Excellent	None	unknow	Intermediate	Minimal	Yes	No	Acceptability on hands varies

Source: Adapted by Tietjen, L., D Bossemeyer and N. McIntosh (2003). *Infection Prevention, Guidelines for Healthcare Facilities with Limited resources*. JHPIEGO Corp., Baltimore, Maryland, page 6 -3. *from:* Boyce and Pitter 2002; Olmsted 1996.

9.5 USE OF ANTISEPTICS

9.5.1 Hand Hygiene

Apply an antiseptic before and after invasive procedures and when touching an immunosup pressed patient.

Note: *Antimicrobial soaps or detergents are no more effective than plain soap and clean water in reducing the risk of infection.*

9.5.2 Skin Preparation Prior to Surgical Procedures

Although skin cannot be sterilized, applying an antiseptic solution minimizes the number of microorganisms around the surgical wound that may contaminate and cause infection.

Step 1: **Do not shave hair** around the operative site. Shaving increases the risk of infection 5-10 fold because the tiny nicks in the skin provide an ideal setting for microorganisms to grow and multiply (Nichols, 1991; Seropian and Reynolds, 1971). If hair must be cut, trim the hair close to the skin surface with scissors immediately before surgery.

Step 2: Ask the patient about **allergic reactions** (e.g., to iodine preparations) before selecting an antiseptic solution.

Step 3: If the skin or external genital area is visibly soiled, gently wash it with soap and clean water and dry the area before applying the antiseptic.

Step 4: Using dry, high-level disinfected forceps and new cotton or gauze squares and antiseptic, thoroughly cleanse the skin. Work from the operative site outward for several centimeters. (A circular motion from the center out helps to prevent recontamination of the operative site with local skin bacteria.)

Step 5: Allow the antiseptic enough time to be effective before beginning the procedure. For example, when an iodophor is used, allow 2 minutes or wait until the skin is visibly dry before proceeding, because the active agent (free iodine) is only released slowly.

9.5.3 Instructions For Cervical Or Vaginal Preparation

For **cervical** and **vaginal antisepsis**, prior to inserting a uterine elevator for minilaparotomy or doing an endometrial biopsy, select an aqueous (water-based) antiseptic, such as an iodophor (povidone-iodine) or 2-4% chlorhexidine gluconate (e.g., Savlon if properly prepared). Do not use alcohols or alcohol-containing preparations.

Step 1: Ask the patient about **allergic reactions** (e.g., to iodine preparations) before selecting an antiseptic

Step 2: If the external genital area is visibly soiled, gently wash it with soap and clean water and dry the area before applying the antiseptic.

Step 3: After inserting the speculum, apply antiseptic solution liberally to the cervix and vagina (two times).

Step 4: If an iodophor is used, allow time (2 minutes) before proceeding.

9.6 STORING AND DISPENSING ANTISEPTICS

Contaminated antiseptics can cause subsequent infection when used for handwashing or preparing a client's skin. The following can prevent contamination of antiseptic solutions.

- Unless supplied commercially in small quantities, pour the antiseptic into a small, re-usable container for daily use. This prevents evaporation and contamination. Make sure the correct name of the solution is on the container each time you refill it. **Do not store gauze or cotton wool in antiseptics because this promotes contamination.**
- Establish a routine schedule for preparing new solutions and cleaning reusable containers. (Solution is at increased risk of becoming contaminated after 1 week of storage). **"Do not "top off" antiseptic dispensers.**
- Wash re-usable containers thoroughly with soap and clean water, rinse with boiled water if available and drip dry before refilling.
- Label re-usable containers with the date each time they are washed, dried and refilled. Concentrated antiseptic solutions should be stored in a cool, dark area. Never store them in direct sunlight or in excessive heat (e.g., upper shelves in a tin-roofed building).

9.7 DISINFECTANTS

Definition

Disinfectants are chemicals that kill or inhibit all microorganisms except bacteria endospores on inanimate objects.

9.7.1 There are three types of disinfectants.

High-level disinfectants

- These are substances that kill all bacteria, viruses, fungi, and mycobacterium tuberculosis. Some high-level disinfectants are also chemical sterilants and, given sufficient time, will destroy bacterial endospores.
- Examples of disinfectants:
  - glutaraldehydes 2% (cidex)
  - sporicidin 2%
  - chlorhexidine 4%, centrimide 5%
  - hydrogen peroxide 6%
  - chlorine 0.5%

Intermediate-level disinfectants

- Kill bacteria and most viruses
- Examples are: alcohols:
  - isopropyl 60-70%
  - ethanol 70-90%
  - methylated spirit 60-90%
  - iodines and iodophor 10% solutions
  - povidone - iodine 2.5%
  - formaldehyde 8%

Note: *Recommended for use on blood and other potentially infectious materials. Small, non-lipid viruses, (e.g, enteroviruses) may be resistant. Used for some non-critical items or devices, or on environmental surfaces.*

Low-level disinfectants

- Kill some bacteria and some viruses and fungi, but do not kill tuberculosis-causing microorganisms and bacterial endospores.
- Examples are:
  - Hydrogen peroxide 3%
  - Phenolics 1-2%
  - Dettol
  - Lysol 5%
  - Carbolic acid 5%

Note: *Should be used only to decontaminate the environment (surfaces, floors, furniture, walls). They must not be used for processing instruments and other items.*

9.7.2 Factors Affecting Disinfection

- Nature of the items to be disinfected
- Number of microorganisms present (a higher number of microorganisms requires more time for disinfection)
- Resistance of microorganisms
  - some microorganisms are more resistant to disinfection then others, e.g., bacterial spores, mycobacteria, hydrophilic viruses, fungi, vegetative bacteria, lipid viruses in that order
  - organisms flourishing in healthcare facility environments (pseudomonas aeruginosa, antibiotic resistant microorganisms) have inherent resistance to certain disinfectants
- Types and concentration of disinfectant used
- Presence of organic materials
  - presence of organic soiling matter (blood, blood products, body fluids, and faeces containing significant amounts of proteins, and proteins) inactivate or slow the action of disinfectants

- Duration of exposure and temperature:
  - the longer the duration of exposure, the higher the degree of disinfection achieved
  - higher temperatures increase the killing power of most disinfectants whereas lower temperatures slow the killing power
- Rough surfaces (having crevices, lumen, hinges) need a longer time for disinfection

9.7.3 Choosing a Disinfection Method

Disinfectants chosen should be:

- Bactericidal not bacteriostatic
- Active against a wide range of microorganisms
- Not readily inactivated by organic matter (i.e. stable when in contact)
- Rapid activity.
- Non-toxic
- Non-corrosive
- Non-damaging to equipment/substances treated
- Cost-effective and available

9.7.4 Guide to Use of Disinfectants (instructions)

- Follow the manufacture's instructions AND ensure that the correct (optimum) dilution is used.
- Check expiry date of the solution. The date should be clearly marked on the container.
- Disinfectant containers must be thoroughly cleaned or sterilized before refill between uses. NEVER TOP UP!!
- Disinfectants must not be used to sterilize instruments or equipment (unless specified in the disinfectant policy, e.g., endoscopes).
- Disinfectants should be supplied, preferably ready for use from the pharmacy (new stocks to be supplied on receipt of empty containers). Do not discard empty containers or use them to store other solutions. Chemicals can be harmful when used in the wrong situations.
- Open containers of disinfectant should not be tolerated in any healthcare environment. There is a serious risk of contamination with multiple antibiotic-resistant bacteria such as Pseudomonas spp and spores.
- When disinfectants are indicated for use on surfaces, WIPE. (Do not wash, bathe or flood-wash).
- Always thoroughly decontaminate, then clean articles before disinfection, i.e., remove any substance such as dirt and biological materials.
- The healthcare facility pharmacy should ensure that:
  - the containers are thoroughly cleansed, washed and dried
  - the containers are clearly labeled with the type of contents, the in-use dilution and the expiry date
  - none of the disinfectants are exposed to inactivating substances, i.e., cork, rubber caps or incompatible detergents

Note: *Disinfectants should be diluted by knowledgeable personnel in manageable quantities, e.g., 5 litres or less. This will reduce waste. Partially filled containers must not be left on the wards (prevent hoarding).*

Table 6: Preparing Dilute Chlorine Solutions from Liquid Bleach (Sodium Hypochlorite Solution)

Using Liquid Bleach

Chlorine in liquid bleach comes in different concentrations. Any concentration can be used to make a dilute chlorine solution by applying the following formula:

$$\text{Total parts (TP) water} = \left(\frac{\%\ \text{concentrate}}{\%\ \text{dilute}}\right) - 1$$

Example1:

To make a 0.5% chlorine solution from 3.5% bleach

$$\left(\frac{3.5\%}{0.5\%}\right) - 1 = 6.$$

Take 1 part concentrated solution, add 6 parts of water to make a 0.5% chlorine solution

$$\textit{Total Parts (TP) water} = \left(\frac{\%\ \text{Concentrate}}{\%\ \text{Dilute}}\right) - 1$$

Example 2: Make a dilute solution (0.1%) from 5% concentrated solution

- Calculate TP (H2O) = $\left(\frac{5.0\%}{0.1\%}\right) - 1 = 50 - 1 = 49$
- Take 1 part concentrated solution and add to 49 parts boiled (filtered if necessary) water.

Note: In countries where French products are available, the amount of active chlorine is usually expressed in degrees chlorum. One-degree chlorum is equivalent to 0.3% active chlorine.

Source: *AVSC International (1999). Infection Prevention Curriculum, Teachers Manual. New York, pg. 267.*

Table 7: Formula for Making Chlorine Solution from Dry Powder

- Check concentration (% concentrate) of the powder you are using
- Determine grams of bleach powder needed

  $$\text{Grams/Litre} = \left(\frac{\%\ \text{Dilute}}{\%\ \text{concentrate}}\right) \times 1000$$
- Mix measured amount of bleach powder with 1 liter of water.

Example: To make a 0.5% chlorine solution from calcium hypochlorite powder containing 35% active chlorine:

$$\frac{0.5\%}{35\%} \times 1000 = 0.0143 \times 1000 = 14.3$$

Therefore, you must dissolve 14.3 grams of calcium hypochlorite powder in one litre of water to get a 0.5% chlorine solution.

Note: When bleach powder is used: the resulting chlorine solution is likely to be cloudy (milky)

Source: *AVSC International (1999). Infection Prevention Curriculum, Teachers Manual.* New York, pg.267.

Table 8: Preparing Dilute Chlorine Solutions from Dry Powders

Examples of dry chlorine powders in the market and their strengths

Available Chlorine Required	Required Concentrations	
	0.5%	0.1%?
Calcium hypochlorite (70% available chlorine)	7.1 g/L[a]	1.4 g/L
Calcium hypochlorite (35% available chlorine)	14.2 g/L	2.8 g/L
NaDCC[c] (60% available chlorine)	8.3 g/L	1.5 g/L
Chloramine **tablets** (1 g of available chlorine per tablet)	20 g/L (20 tablets/liter)[d]	4 g/L (4 tablets/liter) [d]
NaDCC-based **tablets** (1.5 g of available chlorine per tablet)	4 tablets/liter	1 tablet/liter

a For dry powders, read x grams per liter (example: Calcium hypochlorite – 7.1 grams mixed with 1 liter water).
b Use boiled water when preparing a 0.1% chlorine solution for HLD because tap water contains microscopic organic matter that inactivates chlorine.
c Sodium dichloroisocyanurate
d Chloramine releases chlorine at a slower rate than does hypochlorite. Before using the solution be sure the tablet is completely dissolved
Adapted from World Health Organization, 1989.

10.0 WASTE MANAGEMENT AT HEALTH FACILITY LEVEL

Safe management of healthcare waste (HCW) is a key issue to control and reduce nosocomial infections inside a hospital and to ensure that the environment outside is well protected. Wastes from hospitals and healthcare facilities may be contaminated (potentially infectious) or non-contaminated. Approximately 85% of the general wastes produced by hospitals, clinics and other health facilities is non-contaminated waste and poses no infectious risk to the person who handles it. Some wastes from healthcare facilities, however, are hazardous (intrinsic potential properties or ability of any agent, equipment, material or process that can cause harm). If not disposed of properly, contaminated waste may carry microorganisms that can infect people who come in contact with it, as well as the community at large.

Definitions

Healthcare Waste (HCW) includes all the wastes, hazardous or not, produced by health institutions, private or public, generated during medical activities.

Waste management includes all activities, administrative and operational (including transportation activities), involved in the handling, treatment, conditioning, storage and disposal of waste. Healthcare waste management (HCWM) should be part of the overall management system of a healthcare facility and reflect the quality of its services.

The absence of management measures to prevent exposure to hazardous healthcare waste results in a maximum health risk to the general public, in- and out-patients as well as to the healthcare provider. Therefore, the recommendations made in this section should be applied to all healthcare facilities of Tanzania.

10.1 PUBLIC HEALTH RISKS ASSOCIATED WITH HEALTHCARE WASTE

Occupational risks

All individuals exposed to hazardous HCW are potentially at risk of being injured or infected. During handling of wastes, healthcare providers as well as sanitary laborers can be injured if the waste has not been packed safely. Sharps are considered as one of the most dangerous wastes. Many injuries occur because syringes, needles or other sharps have not been collected in safety boxes or because these have been overfilled. On dumpsites, scavengers during their recycling activities may also come in contact with infectious waste if it has not been properly treated or disposed of.

Impact of infectious waste and sharps

Sharps are considered as one of the most hazardous healthcare wastes in a hospital. For serious virus infections such as HIV/AIDS and hepatitis B and C, healthcare workers, particularly nurses, are at greatest risk of infection through injuries from contaminated sharps (largely hypodermic needles).

Other hospital workers and waste-management operators outside healthcare establishments are also at significant risk, as are individuals who scavenge on waste disposal sites.

Source: Safe management of waste from healthcare activities, WHO, 1999

Risks to the population

The general public can be infected by HCW either directly or indirectly through several routes of contamination. Dumping HCW in open areas is a practice that can have major adverse effects on the population.

- The "recycling" practices that have been reported, particularly, the re-use of syringes is certainly the most serious problem in our country. The WHO estimates that some 10 million infections of hepatitis B, C and HIV occur yearly from the re-use of discarded syringes/ needles (WHO, 1999).

- **Indirect risks via the environment**

  The dumping of HCW in uncontrolled areas can have a direct environmental effect by contaminating soils and underground water. During incineration, if no proper filtering is done, air can also be polluted causing illnesses in nearby populations. This has to be taken into consideration when choosing a treatment or a disposal method by carrying out a rapid environmental impact assessment. If waste is not properly collected, stored and transported, domestic animals, birds and other scavengers can carry the contaminated materials and spread infection.

10.2 HEALTHCARE WASTE MANAGEMENT PROCEDURES

The implementation of safe HCWM procedures aims at *containing infections* and reducing *public health* risks both within and outside the HCF. The procedures should always contain the following measures:

- *minimization* of the quantity of HCW generated by the HCF
- *segregation* and *identification* of hazardous HCW from non-risk HCW
- *adequate packaging* and safe *storage* of the different HCW
- *proper treatment and disposal* of hazardous and non-risk HCW (intentional burial, deposit, discharge dumping, placing or release of any waste material into the air, or on land or water. Disposal is undertaken without the intention of retrieval)

10.2.1 Classification, Segregation and Disposal of Healthcare Waste

Proper classification and segregation of waste must follow standardized procedures. It fosters the reduction of risk to healthcare workers, and enhances cost control for hazardous waste disposal by decreasing treatment costs. It must be:

- *simple* to implement for the healthcare providers
- *safe,* by guaranteeing the absence of infectious HCW in the facility waste flow
- *stable* and uniformly applied in all HCFs
- *regularly monitored* to ensure that the procedures are respected

Segregation of waste materials

- The segregation of waste consists of separating the different waste streams based on the type, treatment and disposal practices. Proper segregation should identify waste according to source and type of disposal or disinfection. **Receptacles specifically suited for each type of waste should be available and used as intended.**

- Segregation takes place *at the source:* at the ward bedside, theatre, medical analysis laboratory, or any other room or ward in the health facility where the waste is generated. Segregation of waste shall be applied uniformly throughout the country.

Color coding

The colour-coding system aims at ensuring an immediate and non-equivocal identification of the hazards associated with the type of HCW that is handled or treated. In that respect, the colour coding system shall remain simple and be applied uniformly throughout the country. All healthcare facilities shall apply the following colour coding system:

COLOUR	TYPE OF WASTE
YELLOW	**Safety box with sharp, needles and syringes** Needles, blades, broken glass, lancets, scissors, broken ampoules, slides and slide covers, Bin with papers, pharmaceutical packaging, infusion bag, plastic bottle, big broken glass
RED	**wet infectious materials:** Blood, body tissues (amputations), body fluids (discharges), specimens (stool, sputum, placenta, wet dressings, catheters, blood bags)
BLUE	**Non-Infectious Materials:** Food remains

10.2.2 Minimizing HCW and Recycling by Healthcare Facilities (HCFs)

The implementation of recycling procedures in HCFs to minimize the quantity of HCW generated is highly sensitive. Recycling procedures complicate the overall segregation scheme by increasing the segregation criteria and multiplying the number of waste streams in the HCF. Nevertheless, considering the specific recycling practices in Tanzania, there is a need to implement an environmentally friendly process of HCW disposal with the financial resources of each HCF. A simple and safe recycling program should be implemented whenever it is possible. In these situations the instructions hereafter should be followed.

Recycling of non-contaminated plastic items

All *non-contaminated* plastic items (e.g., bags of i.v. fluids) should be collected, packed in separate boxes and delivered to or picked up by local contractors capable of recycling them with environmentally friendly techniques.

Recycling of glassware inside health laboratories

All *non-contaminated and non-broken* glassware (flasks of injectable vials and ampoules) can be collected separately. They should be put in a solution of 10% sodium hypochlorite for 30 minutes, carefully washed, rinsed and dried before being reused.

The disinfected glassware should be reused only for specific medical analysis (blood, urine) carried out inside the health laboratory. After having been used once, they shall be considered as contaminated and infectious.

Broken glassware shall always follow the stream of sharp waste disposal while non-broken glass flasks shall be reused only after disinfection in a solution of 10% sodium hypochlorite for 12 hours, carefully washed with a brush and soap, rinsed and dried. It is recommended that the glassware be autoclaved after washing at 121°C for at least 30 minutes to ensure a complete disinfection. During the disinfection process, hands shall always be protected with gloves.

Re-use of specific equipment

In general, to encourage reuse, each hospital shall collect separately, wash and sterilize surgical equipment and other items that are designed for re-use and can tolerate the sterilization process, either thermally or chemically in accordance with approved procedures. Pressurized gas cylinders shall be returned to suppliers for refilling and reuse.

Pharmacy department stores in each HCF shall be vigorously managed to avoid waste, and stock positions should be recorded on a regular basis.

10.2.3 General Healthcare Waste Management (HCWM) Procedures

Given hereafter are general instructions that should be respected in a HCWM plan to ensure a safe manipulation of all HCW generated in healthcare facilities.

Waste collection and on-site transportation

- All HCW or disposal of medical equipment shall be discarded at the point of use by the person who used the item to be disposed of. In case any such used equipment or supplies are found or are handled over to any person, that person should discard it.
- All the specific procedures of HCW management, which include segregation, packaging and labeling, shall be explained to the healthcare providers and displayed in each department on charts located on the walls near the HCW containers.
- When handling waste, sanitary staff and sweepers shall wear protective clothing at all times including facemasks, industrial aprons, boots, and heavy-duty gloves.
- The *waste collection trolley* should be easy to load, unload and clean. The trolley shall not be used for any other purpose. It shall be cleaned regularly, and especially before any maintenance work is performed on it.
- Yellow bags of hazardous HCW and black bags of non-risk HCW shall be collected on separate trolleys that shall be painted or marked with the corresponding colours and washed regularly.
- The collection route shall be the most direct one from the collection point to the central storage facility. The collected waste shall not be left, even temporarily, anywhere other than at the designated central storage facility.

Waste storage

- In each room where HCW is generated, an adequate place shall be dedicated for storing HCW bag-holders, bins or containers. It should be easily accessible for the sanitary staff. Instructions shall be displayed.
- In all HCFs, separate central storage facilities shall be provided for hazardous HCW, except radioactive waste that shall be stored specifically. It shall be stated clearly that the facility stores hazardous yellow-bagged HCW only. No waste shall be stored for more than two days before being treated or disposed of.
- The designated central storage facility shall be located within the hospital premises close to the treatment unit but away from food storage or food preparation areas. It should be large enough to contain all the hazardous HCW produced by the hospital

during one week, with spare capacity to cope with any maintenance or breakdown of the treatment unit.

- The designated central storage facility shall be totally enclosed and secured from unauthorized access. It shall be inaccessible to animals, insects and birds. It shall be easy to clean and disinfect, with an impermeable hard-standing base, good water supply, drainage and ventilation.

Off-site transportation

- In a case when off-site transportation is required to treat hazardous HCW at a central treatment facility, the relevant local government authority shall approve the off-site transportation plan before any transit occurs.
- Red bags shall be collected at least every second day. The transportation shall be properly documented, and all vehicles shall carry a consignment note from the point of collection to the central treatment facility.
- Vehicles used for the carriage of yellow bags shall not be used for any other purpose. They shall be free of sharp edges, easy to load and unload by hand, easy to clean/ disinfect, and fully enclosed to prevent any spillage in the hospital premises or on the road during transportation.
- All vehicles shall be cleaned and disinfected after use. They shall carry adequate supply of plastic bags, protective clothing, cleaning tools and disinfectants to clean and disinfect in case of any spillage.
- All vehicles staff handling yellow and red bags shall wear protective clothing. They shall be properly trained in the handling, loading and unloading, transportation and disposal of the yellow and red bags. They shall be fully aware of emergency procedures for dealing with accidents and spillage.

Waste treatment and disposal

Among all the current existing technologies for treating and disposing of HCW, the most appropriate technology shall be applied.

- The most reliable, affordable and sustainable technology will be used in accordance with the technical, human and financial resources of each HCF.
- HCFs will use the technology that minimizes the immediate public health risks associated with HCWM with the lowest impact on the environment.
- Incineration may be considered as the technology that ensures the most reliable disposal of hazardous HCW. When incineration is used, ash and residues of incineration shall be safely removed and properly buried in an appropriate pit. Nevertheless, in densely populated areas, large quantities of hazardous HCW shall not be incinerated at temperatures lower than 1,200^{o}c

Accidents and spillage

All health facility staff members shall be properly trained and prepared for **emergency response**, including procedures for treatment of injuries, cleanup of the contaminated area and prompt reporting of all incidents or accidents. The following actions shall be taken:

- **evacuation** of the contaminated area if required
- **decontamination or disinfection, rinsing and wiping dry** with absorbent cloth by personnel wearing adequate protective clothing
- decontamination or disinfection of the protective clothing if necessary
- cuts with sharps or needle-stick injuries shall be immediately disinfected.

Accidents shall be reported to the Infection Prevention Control Focal person. All cases shall be registered by the Management Team of the HCF and annually reported to the district health authorities. It is highly recommended that blood tests be performed after such an injury to ensure that the injured staff member has not been contaminated by any pathogen.

10.3 SPECIFIC PROCEDURES ASSOCIATED WITH HCW CATEGORIES

In healthcare facility settings, specific categories will require specific handling procedures. These procedures are outlined below.

10.3.1 Non-infectious Healthcare Waste

Non-infectious waste shall be placed in blue containers. Non-contaminated items that are designated for recycling shall be packed in specific blue containers marked "Non-contaminated plastic, to be recycled" or "Non-contaminated glassware, to be recycled". All non-risk HCW not designated for recycling shall be collected with municipal waste.

10.3.2 Infectious Waste

Infectious waste is all biomedical and healthcare waste known or clinically assessed to have the potential of transmitting infectious agents to humans or animals.

It shall be placed in yellow polyethylene bags marked "Danger Hazardous Medical Waste". Bags shall be sealed with appropriate adhesive tape, removed and replaced immediately when they are no more than three-quarters full. If not available, yellow bins or containers shall be used but systematically disinfected in a solution of 10% sodium hypochlorite or Lysol once emptied.

Infectious waste shall be incinerated (controlled burning of solid, liquid or gaseous combustible (burnable) wastes to produce gases and residues containing little or burnable material) in double-chamber incinerators. In densely populated areas, preferably an incinerator reaching 1'200°C shall be used. In other areas, decentralized, low-cost incinerators shall be used.

Health centers and dispensaries may incinerate in a simple pit hole. Sanitary land disposal (disposing of solid waste on land in a manner that can protect the environment, e.g., by spreading the waste in thin layers, compacting it to the smallest practical volume and then covering it with soil at the end of each working day) is an alternative solution when underground water is not at risk for contamination.

10.3.3 Sharps

Sharps include all objects and materials that pose a potential risk of injury and infection due to their puncture or cutting properties (e.g., syringes with needles, blades, broken glass).

- Sharps shall be placed in specific cardboard or plastic safety boxes resistant to punctures and leakage, designed so that items can be dropped in using one hand, and no item can be removed. The safety box shall be colored yellow, marked "Danger Contaminated Sharps" and with the biohazard symbol indicated on the outside of the box. It shall be closed when three-quarters full and then placed in a yellow plastic bag or containers with the other hazardous HCW.
- In particular, *all disposable syringes and needles* shall be discarded immediately following use. The needle shall not be recapped or removed from the syringe; the whole combination shall be inserted into the safety box directly after use.
- Under no circumstances are used syringes or needles, or safety boxes, to be disposed of in normal garbage or dumped randomly without prior treatment.
- Sharps are destroyed together with the hazardous HCW. The method of choice for destruction of full safety boxes is incineration, preferably in an appropriate double-chamber incinerator.
- If such an incinerator is unavailable, alternative methods may be used such as the use of sharps pits that are well protected. Under exceptional circumstances, full safety boxes may be incinerated in small numbers by open burning.
- The residues of incineration shall be safely deeply buried at sufficient depth greater than 1 meter.

10.3.4 Pathological Waste

Pathological waste includes that of all organs (including placentas), tissues as well as blood and body fluids. In operation theatres, all anatomical waste and placentas shall be collected separately. When a centralized incinerator is available they shall be incinerated. When a low-cost incinerator is used, anatomical waste and a large number of placentas can be difficult to incinerate and drastically reduces the performance of the incinerator.

If incineration cannot be performed, anatomical waste and placentas shall be dropped into a concrete lined pit or buried at a sufficient depth (greater than 1 m) inside the HCF compound in a location totally enclosed and secured from unauthorized access, at least at 100 m away from any underground water well. If transportation and disposal cannot be immediately ensured, anatomical waste should be stored in the mortuary.

10.3.5 Hazardous Pharmaceutical Waste and Cytotoxic Waste

Hazardous pharmaceutical waste and cytotoxic waste comprises expired pharmaceuticals or pharmaceuticals that are unusable for other reasons (e.g., call-back campaign). They can be hazardous (cytotoxic) or non-hazardous pharmaceutical waste.

Hazardous pharmaceutical waste shall be repacked in specific cardboard boxes marked "Danger Hazardous Pharmaceutical and Cytotoxic Waste!" It shall be returned to the medical stores department that shall ensure its disposal at central level.

Waste shall be incinerated at pyrolytic incinerator at 1'200°C at a minimum. Hazardous pharmaceutical waste and **cytotoxic waste containing heavy metals shall not be incinerated.**

10.3.6 Highly Infectious Waste

Highly infectious waste includes all viable biological and pathological agents artificially cultivated in significant elevated numbers. Cultures and stocks, dishes and devices used to transfer, inoculate and mix cultures of infectious agents belong to this category of waste. They are generated mainly in hospital health laboratories.

Highly infectious waste from the medical analysis laboratory of the HCF, shall be properly handled by autoclaving, and properly sealed. It shall be autoclaved at a temperature of 121°C at 1 bar for at least 20 minutes at source, i.e., in the medical analysis laboratory itself. Disinfected waste shall be collected and treated with the hazardous HCW.

If a distinct autoclave is not available at the medical analysis laboratory to ensure a thermal treatment, highly infectious waste shall be disinfected in a solution of sodium hypochlorite in concentrated form and left overnight. It shall then be discarded in a specific yellow bag properly sealed and itself discarded with the hazardous HCW.

If none of the above treatment options can be ensured, highly infectious waste should at least be packed in a specific yellow bag that shall be sealed and directly discarded with the hazardous HCW. This measure shall remain exceptional.

Highly infectious waste from isolation wards or permanent treatment centers (cholera) shall always be incinerated on-site.

10.3.7 Radioactive Waste and Special Hazardous Waste

Radioactive waste includes liquids, gas and solids contaminated with radionuclides whose ionizing radiations have genotoxic effects, and include X- and g-rays as well as a- and b-particles.

Special hazardous waste includes gaseous, liquid and solid chemicals, waste with a high content of heavy metals such as batteries, pressurized containers, out-of-order thermometers, blood-pressure gauges, photographic fixing and developing solutions in X-ray departments, and halogenated or non-halogenated solvents (effluents).

- All radioactive waste shall be stored to allow decay to background level. It shall be placed in a large container or drum and labelled with the radiation symbol showing the radionuclide's activity on a given date, the period of storage required, and marked 'Caution! Radioactive Waste'.
- Containers or tanks with *radioactive* waste that has not decayed to background level shall be stored in a specific marked area, preferably in a lead-shielded storage room or alternatively in a room with concrete walls 25 cm thick.
- Non-infectious radioactive waste that has decayed to background level shall follow the non-risk HCW stream while infectious radioactive waste that has decayed to background level shall follow the clinical HCW stream (class 2). Liquid radioactive

waste shall be discharged into the sewerage system or into a septic tank only after it has decayed to background level in adequate tanks.

10.3.8 Large Quantities of Chemical Waste

Large quantities of chemicals shall be returned to the supplier. Nevertheless currently appropriate controlled and standardized methodology does not exist in Tanzania to dispose of chemicals: sometimes on-site disposal may be necessary. The disposal methodology shall depend on the type of chemicals. In such circumstances, non-corrosive and non-flammable chemicals may be encapsulated and separated to avoid unwanted chemical reactions after neutralization.

10.3.9 Waste with a High Content of Heavy Metals

Waste with a high content of *heavy metals* should normally be treated in specific recovering industries. Alternatively, as for chemical waste, it may be encapsulated (filling a sharps container that is three quarters full with cement or clay; after hardening, this can be disposed of safely in a land fill). Waste with a high content of mercury or cadmium shall never be incinerated because of the risk of atmospheric pollution with toxic vapors.

10.3.10 Solid Waste

No solid waste shall be discarded into the sewer system (including conduits, pipes and pumping stations), and all liquid infectious waste shall be discharged into the sewer system only after being properly treated.

10.3.11 Wastewater

Wastewater from HCFs may contain various, potentially hazardous components such as microbiological pathogens, hazardous chemicals, pharmaceuticals and radioactive isotopes. The proper treatment of wastewater from HCFs is very expensive and currently cannot be foreseen in every HCF of Tanzania. Nevertheless basic steps can reduce the public health risk associated with liquid waste and wastewater.

- **Effluents** of all medical analysis laboratories shall always be neutralized in a buffer tank before being drained off into the sewer.
- **Radioactive effluents** of isolation wards shall be discharged into the sewer or into a septic tank only after it has decayed to background level in adequate retention tanks.

11.0 SAFE PRACTICES IN THE OPERATING ROOM

"The operating room is clearly one of the most hazardous environments in the healthcare delivery system. Surgery is an invasive procedure by its nature. Instruments that are designed to penetrate patients' tissues can easily injure the provider. Blood is everywhere and speed is essential. Emergencies can occur at any time and interrupt routines. Preventing injuries and exposures [to infectious agents] under these circumstances is indeed challenging!" (Julie Louise Gerberding in Tietjen, L., D. Bossemeyer and N. McIntosh. 2003, p. 7-1)

In this era of HIV, HBV and HCV there is a need to create awareness of the risks of exposure to blood and body fluids and to increase awareness of modern surgical infection practices. Just as patients must be protected from wound contamination and infections, so providers must be protected from intraoperative injuries and exposure to patients' blood and body fluids. Therefore, surgical aseptic techniques are designed to create such an environment, by controlling the four main sources of infectious organisms: the patient, surgical staff, equipment and the operating room environment.

11.1 DEFINITIONS

Asepsis and **aseptic technique** are combination of efforts made to prevent entry of microorganisms into any area of the body where they are likely to cause infection.

The goal of asepsis is to **reduce to a safe level** or **eliminate** the number of microorganisms on both animate (living) surfaces (skin and tissue) and inanimate objects (surgical instruments and other items).

Surgical asepsis is the preparation and maintenance of a reduced (safe) level of microorganisms during an operation by controlling four main sources of infectious organisms: the patient, personnel, equipment and the environment.

11.2 SPECIFIC TECHNIQUES TO ESTABLISH AND MAINTAIN SURGICAL ASEPSIS

- **Patient considerations:**
  skin cleaning
  pre-operative skin antisepsis
  wound and incision management
- **Operating staff considerations:**
  hand hygiene
  handwashing
  hand rub (with waterless, alcohol-based antiseptic agents)
  hand scrubbing
  proper wearing and removal of gloves and gowns
- **Equipment and room preparation considerations:**
  traffic flow activity patterns
  housekeeping practices
  decontamination, and cleaning
  HLD and sterilization of instruments, gloves and other items
- **Environmental considerations:**
  maintaining an aseptic operating field and using safer operating practices and techniques

11.3 INJURIES IN THE SURGICAL ENVIRONMENT

Instruments Which Cause Injuries

The vast majority of sharps injuries in hospitals occur in the operating room. Many other items can also cause sharps injuries and glove tears resulting in exposure to blood. Some of

the most important are:

- scalpel and suture needles
- hypodermic needles
- wire sutures
- laparoscopy and surgical drain trocars
- orthopedic drill bits, screws, pins, wires and saws
- needle point cautery tips
- skin hooks and towel clips
- sharp-pointed scissors and sharp-tipped mosquito forceps
- dissecting forceps
- sharp-toothed tenaculi

Almost all of these injuries can be easily avoided and with little expense.

11.3.1 Preventing Injuries

- Use a small Mayo forceps (not fingers) when holding the scalpel blade, when putting it on or taking it off when loading the suture needle. (Alternatively, use disposable scalpels with a permanent blade that cannot be removed).
- Always use tissue forceps, not fingers, to hold tissue when using a scalpel or suturing. Use a "hands-free" technique to pass or transfer sharps (scalpel, needles and sharp-tipped scissors) by establishing a Safe or Neutral Zone in the operative field.
- Always remove sharps from the field immediately after use.
- Make sure that sharps containers are replaced when they are only three-quarters full and place containers as close to where sharps are being used, as conveniently possible (i.e., within arm's reach).

The "Hands-Free" Technique for Passing Surgical Instruments

A safer method of passing sharp instruments (scalpels, suture needles and sharp scissors) during surgery, called the **"hands-free" technique**, has recently been recommended. (Bessinger, 1988; Fox, 1992). Using the hands-free technique, the assistant or scrub nurse places a sterile or high-level disinfected kidney basin, or other suitable small container, on the operative field between her/himself and the surgeon. The container is designated as the safe or neutral zone in which sharps are placed before and immediately after use. For example, the assistant or scrub nurse alerts the surgeon that a sharp instrument has been placed in or on the safe zone, with the handle pointing toward the surgeon, by saying "scalpel" or "sharp" while placing it there. The surgeon then picks up the instrument and returns it to the container after use, this time with the handle pointing away from her/him.

11.3.2 Designing Safer Operations

Using the least dangerous instrument or device that will effectively accomplish the task, while at the same time minimizing risks to the patient and surgical team, should be a goal of any operation. Simple things, such as a brief pre-op discussion of how sharps will be handled by the surgeon, assistant or scrub nurse, can be very helpful. Other examples of instruments or devices that protect the surgical team without sacrificing patient safety or staff performance are shown in the table below.

Table 9: Reducing the Risk of Exposure

FUNCTION	SAFER	LESS SAFE[1]
Skin incision	Cautery	Disposable scalpel
Cutting	Scissors, blunt tip or cautery probe	Scissors, sharp tip
Haemostasis	Blunt suture needles staples or cautery	Sharp suture needles
Sponging with gauze while using a scalpel	Surgeon does sponging; assistant only retracts	Assistant sponges but only by request
Retraction	Blunt retractor	Sharp retractor
harps transfer	Neutral zone	Hand-to-hand (communication)
Surgical gloves	Double gloving	Single pair of gloves or double gloving with reprocessed gloves
Closing peritoneum (small 2-3 cm incision)	Do not close	Purse-string closure using tissue forceps to grasp needle)
[1] Should be avoided if at all possible		

Source: *Tietjen, L., D. Bossemeyer and N. McIntosh. (2003) Infection Prevention - Guidelines for Healthcare Facilities with Limited Resources*. JHPIEGO Corporation. Baltimore, Maryland.2003, p. 7-6.

11.3.3 Blunt Needles for Suturing

The range of "bluntness" in commercially available blunt-tipped needles varies from minimal (no extra effort needed to use them) to very blunt (does not penetrate tissue such as fascia and requires conscious effort). Minimally blunt needles can be used for closure of all layers from fascia to skin. Intermediate blunt needles require some additional effort to close fascia, but are safer to use. Very blunt needles are seldom used except when operating deep in the pelvis where the needle absolutely must be retrieved with fingers.

11.4 POST-EXPOSURE PROPHYLAXIS GUIDELINE (HIV)

The risk of HIV transmission to medical personnel has been recognized since 1984. Correct estimation of the likelihood of transmission following occupational exposure is limited by relative infrequency with which HIV transmission to healthcare workers is reported. The estimated risk of HIV transmission following a single need prick exposure is about 0.3%.

11.4.1 Risk Factors for Occupational HIV Transmission

The likelihood of HIV infection following exposure is affected by the presence of certain factors:

- type of contact intact skin or broken skin
- quality of blood
- disease status of source patient
- increased risk with terminal illness and acute (or recent) infection
- host defenses
- post exposure prophylaxis

11.4.2 Post-exposure Site Management

- Wounds and puncture sites should be washed with soap and water.
- Exposed mucous membranes should be flushed with water.
- Post-exposure evaluation should be done (type of body fluid involved).
- Type of exposure should be determined (percutaneous, mucosal, intact skin, etc).
- Severity of exposure should be assessed – quantity of blood, duration of contact.

11.4.3 Definitions

Low risk exposure:

- Exposure to a small volume of blood contaminated with fluid from a symptomatic HIV patient with low viral titer
- Following an injury with a solid needle
- Any superficial injury or mucocutaneous exposure

High-risk exposure:

- Exposure to a large volume of blood or potentially infections fluids
- Exposure to flood or fluid contaminated with blood from a patient with high viral titre
- Injury with a hollow needle
- Deep and extensive injuries
- Confirmed drug resistance in source patients

11.4.4 Source Patient Evaluation

- Clinical status assessment
- Screening for HIV status of the patient after consent
- Treatment should not wait for test result and should be commenced within 24 hours post exposure

11.4.5 Baseline and Follow-up Testing

- Baseline testing of HIV antibody should be done to establish serostatus of the healthcare worker at time of exposure
- Repeat testing should be done at 6 and 12 weeks and 6 month post exposure regardless of the use of PEP
- Pregnancy test should be done for all female workers in reproductive age if their pregnancy status is unknown.

11.4.6 Counseling the Healthcare Worker

Healthcare workers should be counseled about their personal risk and recommendation made to start PEP. Discuss the following.

- Efficacy of drugs
- Side effects of drugs or toxicity
- Possible resistance to drugs
- Obtain history of the health worker (this will influence choice of drugs):
  - possibility of pregnancy, or any other pre-existing medical conditions
- Recommend a specific regimen and discuss the rationale for choosing that regimen; emphasize adherence
- Exposed healthcare workers should be encouraged to seek medical advice with any development as acute illness, or drug reaction
- Should be encouraged to adopt safer sex practices, not to donate blood, or discontinue breast-feeding
- No need to modify his/her work
- HBV vaccination should be considered if not already vaccinated

11.4.7 Prophylaxis Regimens

Post-exposure prophylaxis regimen

Two or three drugs regimens are recommended in order to reduce chances of drug resistance.

Basic prophylaxis

- For occupation HIV exposure for which there is recognized risk of transmission.
- Zidovudine 600mg daily in divided doses for 28 days
- Lamivudine150mg twice daily for 28 days

Extended prophylaxis

Considered when exposure poses increased risk for transmission e.g., large volume of blood, or high virus titer in blood.

- Drugs: Basic regimen plus indinavir 800mg tid or nelfinavir 720mg tid.

Risk Category	ARV Prophylaxis	Duration
Low risk	Zidovudine (ZDV) 300mg bd. Lamivudine (3T C) 150mg bd. (Combiviour or Duovir 1bd.	28 days
High risk	Zidovudine (ZDV) 300mg bd. Lamivudine 3TC 150mg Combivir or Duovir 1bd plus Indinavir 800mg tbs	28 days

11.5 HEPATITIS B POST-EXPOSURE GUIDELINES

Several studies have demonstrated that in susceptible persons (i.e., negative hepatitis B surface antigen [HbsAG] test and no history of receiving immune serum globulin), giving hepatitis B immune globulin (HBIG) is better than conventional immune serum globulin (ISG), or by inference doing nothing, in preventing acute hepatitis B and sero-conversion.

The suggested steps for managing an injury are as follows.

Step 1: Treat the exposure site appropriately (e.g., an open wound or cut).

Step 2: Give tetanus immunization or booster if indicated (e.g., > 10 years since immunization).

Step 3: Assess the risk of HBV exposure and determine the immune status of the patient (i.e., history of jaundice, hepatitis or previous immunization with hepatitis B vaccine). If status is unknown, continue assessment.

Step 4: Collect a specimen from the source person (the carrier or person suspected of being infected) if possible and from the patient for HBsAGg testing. If testing is not possible, base the HBV status of the infected person on clinical history and clinical findings.

Step 5: Give HBIG (5mL IM) as soon as possible and within 7 days of exposure, and also give the first dose of hepatitis B vaccine, which should be repeated at 1 and 6 months. If active immunization with hepatitis B vaccine is not possible, a second dose of HBIG should be given 1 month later (Chin 2000).

11.6 HEPATITIS C POST-EXPOSURE GUIDELINES

There is no post-exposure vaccine or drug prophylaxis for hepatitis C (immune globulin is ineffective). Prevention of exposure, therefore, is the only effective strategy for prevention of HCV.

The CDC (1998) has recommended the following guidelines that institutions should consider for follow up of health workers exposed to HCV-positive blood or other body fluids:

- baseline testing of the source patient (if available and a consent form is signed) for anti-HCV antibody (if the test is available)
- baseline and 6-month follow-up testing of exposed health worker for anti-HCV anti body and liver function screen
- if available, treatment of early HCV infection with pegylated interferon alfa before significant liver damage has occurred

PART III

PROCESSES IN INFECTION PREVENTION AND CONTROL

12.0 TRAFFIC FLOW AND ACTIVITY PATTERNS

Regulating the flow of visitors, patients and staff plays a central role in preventing disease transmission in healthcare facilities. The number of microorganisms in designated areas tends to be related to the number of people present and their activities. Microbial contamination is expected and found to be high in areas such as waiting rooms and places where soiled surgical instruments and other equipment are initially processed. Contamination can be minimized by reducing the number of people permitted into an area and by defining the activities that take place there.

An important objective of infection prevention is to minimize the level of microbial contamination in areas where patient care and instrument processing take place.

Such areas include:

- **Procedure areas** where patients are examined and procedures are carried out
- **Surgical units** where major and minor operations are performed including preoperative and recovery rooms
- **Work areas** where instruments are processed; these include dirty and clean areas where soiled instruments, equipment and other items are first cleaned and are either high-level disinfected or sterilized and then stored

12.1 SPACE AND EQUIPMENT REQUIREMENTS

Healthcare facilities vary in the types of services they provide. Regardless of the size of a facility, however, the specific space and equipment requirements to perform a particular procedure generally do not vary.

The space, equipment and need for well-defined traffic flow and activity patterns become progressively more complex as the type of surgical procedure changes from general surgery and obstetrics to open heart surgery. As a guide, the space requirements for the types of surgery typically performed at the district hospitals are roughly the same as for a busy surgical center or polyclinc. These include the following.

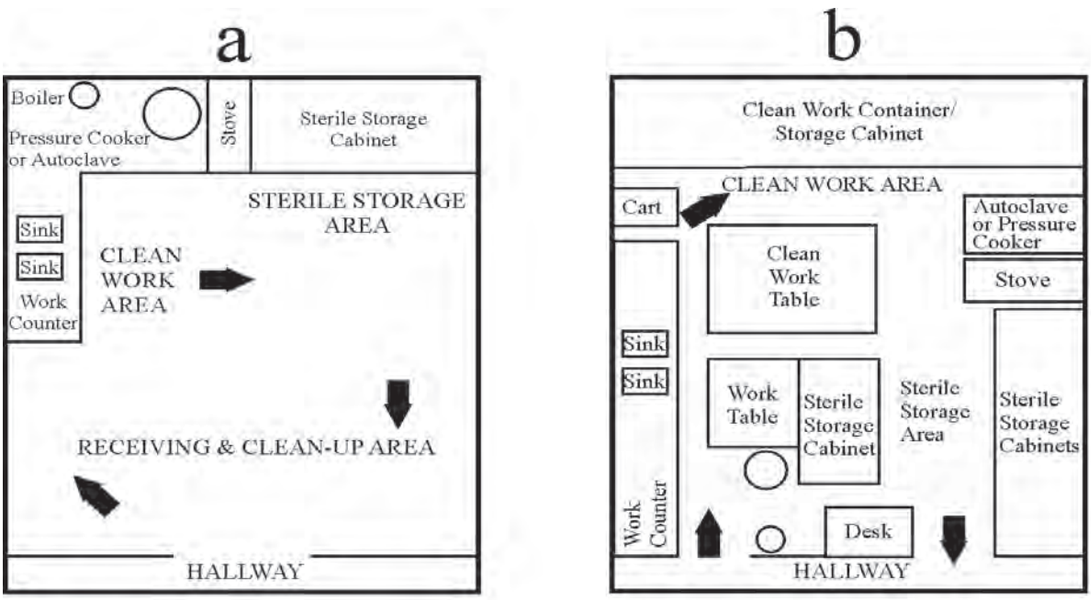
- Changing room and scrub area for clinic staff
- Preoperative area where clients are examined and evaluated prior to surgery
- Operating room
- Recovery area for patient observation after surgery (may be combined with the preoperative area)
- Processing area for cleaning and sterilizing or high-level disinfecting instruments and other items
- Space for storing sterile packs and/or high-level disinfected containers of instruments and other items

The recommended infection prevention practices for **minimizing microbial contamination** of specific areas in healthcare facilities are briefly described below.

12.1.1 Procedure Areas

- Limit traffic to authorized staff and patients at all times.
- Permit **only** the patient and staff performing and assisting with procedures in the procedure room (family members should be limited with obstetrical procedures).
- Patients can wear their own clean clothing.
- Staff should wear attire and personal protective equipment (PPE) according to procedures performed.
- Have covered containers filled with a 0.5% chlorine solution for immediate decontamination of instruments and other items once they are no longer needed.
- Have a leak-proof, covered waste container for disposal of contaminated waste items (cotton, gauze, dressing) at point of use.
- Have a puncture-resistant container for safe disposal of sharps.
- Have storage space in procedure rooms for clean, high-level disinfected and sterile supplies. (Storage shelves should be enclosed to minimize dust and debris collecting on stored items.)

Floor plans for the process of instrument cleaning, high-level disinfecting and sterilizing areas in a clinic and larger facility



Design for small clinic with minimal space available and low volume of services

Design for larger facility (district hospital) where service volume is greater

Source: SEARO/WHO, 1988.

12.1.2 Surgical Unit

The surgical unit is often divided into four designated areas, which are defined by the activities performed in each as follows.

Unrestricted area: This area is the entrance from the main corridor and is isolated from other areas of the surgical unit. This is the point through which staff, patients and materials enter the surgical unit.

Transition zone: This area consists primarily of dressing rooms and lockers. It is where staff put on surgical attire that allows them to move from unrestricted to semi-restricted or restricted areas in the surgical unit. Only authorized staff should enter this area.

Semi-restricted area: This is the peripheral support area of the surgical unit and includes preoperative and recovery rooms, storage space for sterile and high-level disinfected items, and corridors leading to the restricted area. In this area the following should be observed.

- Limit traffic to authorized staff and patients at all times.
- Have a work area for processing clean instruments.
- Have storage space for clean and sterile or high-level disinfected supplies with enclosed shelves to minimize dust and debris collecting on stored items.
- Have doors limiting access to the restricted area of surgical unit.
- Staff members who work in this area should wear surgical attire and caps.
- Staff should wear clean, closed shoes that will protect their feet from fluid and dropped items.

Restricted area: This area consists of the operating room(s) and scrub sink areas. The following should be observed in this area.

- Limit traffic to authorized staff and patients at all times.
- Keep the door closed at all times, except during movement of staff, patients, supplies and equipment.
- Scrubbed staff must wear full surgical attire and cover head and facial hair with a cap and mask.
- Staff should wear clean, closed shoes that will protect their feet from fluids and dropped items.
- Masks are required when sterile supplies are open and scrubbed staff members are operating.
- Patients entering the surgical unit should wear clean gowns or be covered with clean linen, and have their hair covered.
- Patients do not need to wear masks during transport (unless they require airborne precautions).

12.1.3 Guidelines for Working in Operating Room(s)

Enclose the operating room to minimize dust and eliminate flies; central air conditioning is preferred. (If windows are the only ventilation, provide tight-fitting screens.)

- The operating room should be located away from areas of the hospital or healthcare facility that are heavily traveled by staff and patients.

Before Surgical Procedures

- Place a clean, covered container filled with 0.5% chlorine solution or another locally available and approved disinfectant for immediate decontamination of instruments and other items once they are no longer needed.
- Place a plastic bag or leak-proof, covered waste container for contaminated waste items (cotton gauze, old dressings).
- Place a puncture-resistant container for the safe disposal of sharps (e.g., suture needles, hypodermic needles and syringes, and disposable scalpel blades) at the point of use but without contaminating the sterile field.
- Place a leak-proof, covered waste container for soiled linen away from sterile items.
- Organize tables, mayo and ring stands side by side in an area away from the traffic patterns and at least 45 cm (18 inches) from walls, cabinets and other non-sterile surfaces.
- Place a clean sheet, a lift sheet and arm-board covers on the operating room bed.
- Check and set up suction, oxygen and anesthesia equipment.
- Place supplies and packages that are ready to open on the tables, not on the floor.
- The mayo stand and other non-sterile surfaces that are to be used during the procedure should be covered with a sterile towel or cloth.

During Surgical Procedures

- Limit the number of staff entering the operating room to only those necessary to perform the procedure and to patients. Make the surgical team self-sufficient so that outside help is not required.
- Keep the doors closed at all times, except during movement of staff, patients, supplies and equipment.
- Keep the number of people and their movements to a minimum; the number of microorganisms increases with activity.
- Keep talking to a minimum in the sterile field.
- Scrubbed staff should wear full surgical attire, including:
  - sterile surgical gowns on top of scrub suit
  - a clean surgical cap that covers the head
  - clean, closed shoes (or boots that can be wiped clean) that protect the feet from fluids or dropped items
  - surgical gloves, protective eyewear and a mask covering the mouth, nose and any facial hair
  - scrubbed staff should keep their arms and hands within the operative field at all times and touch only sterile items or areas
- Non-scrubbed staff should wear surgical attire, including:
  - clean, scrub suit
  - clean surgical cap that covers the head
  - clean, closed shoes that protect the feet from fluids or dropped items, and a mask covering the mouth, nose and any facial hair
  - non-scrubbed staff should stay at the periphery of the operating room keeping their distance from sterile areas; they should not lean or reach over the operative field

- Clean accidental spills or contaminated debris in areas outside the surgical field with 0.5% chlorine solution as promptly as possible. (A non-scrubbed staff member wearing utility gloves should do this).

After Surgical Procedures

Non-scrubbed staff wearing utility gloves should do the following.

- Collect all waste and remove it from the room in closed leak-proof containers.
- Close and remove puncture-resistant containers when they are three quarters full.
- Remove covered containers with 0.5% chlorine solution with instruments and surgical gloves from the room.
- Remove soiled linen in closed leak-proof containers.
- Remove waste, soiled linen, soiled instruments and equipment, and supplies that have been opened but not used, in a leak-proof, covered waste container. (Make sure that these items do not re-enter the restricted area.)

13.0 CENTRAL STERILIZATION SUPPLY DEPARTMENT (CSSD)

The CSSD is the area where instruments and equipment are processed, and where staff should be specially trained in handling, processing and storing instruments, equipment and other clean, sterile or high-level disinfected items. The CSSD is considered a semi-restricted area, so all the recommendations for traffic patterns and proper attire described above should be followed. A CSSD consists of four areas:

1. **the "dirty" receiving/cleanup area**
2. **the "clean" work area**
3. **the cleaning equipment storage area**
4. **the sterile or high-level disinfected storage area**

The function and equipment requirements for the four areas of a typical CSSD are summarized below.

1. **"Dirty" Receiving/Cleanup Area:** In this area soiled items are received, disassembled and washed, rinsed and dried. The "dirty" receiving/cleanup area should have:

   - a receiving counter
   - two sinks if possible (one for cleaning and one for rinsing) with a clean water supply
   - a clean equipment counter for drying

2. **"Clean" Work Area:** In the clean work area, cleaned items are:

   - inspected for flaws or damage
   - packaged (if indicated), and either sterilized or high-level disinfected
   - sent for storage as packaged or air dried and placed in a sterile or high-level disinfected container

The clean work area should have:

- a large work table
- shelves for holding clean and packaged items
- a high-pressure steam sterilizer, a dry-heat oven, a steamer or a boiler

3. **Cleaning Equipment Storage Area:** Store clean equipment in this area on shelves (preferably enclosed) for storing clean equipment; have an office desk for record keeping. CSSD staff should enter the CSSD through this area.

4. **Sterile or High-Level Disinfected Storage Area:** Store sterilized packs and covered sterile or high-level disinfected containers in this area. This area should be separated from the central sterile supply area.

- Limit access to the storage area and/or store items in closed cabinets or shelves. (Enclosed shelves or cabinets are preferred as they protect packs and containers from dust and debris. Open shelves are acceptable if the area has limited access, and housekeeping and ventilation practices are controlled).
- Keep the storage area clean, dry, dust-free and lint-free by following a regular housekeeping schedule.
- Packs and containers with sterile or high-level disinfected items should be stored 20 to 25 cm (8 to 10 inches) off the floor, 45 to 50 cm (18 to 20 inches) from the ceiling and 15 to 20 cm (6 to 8 inches) from an outside wall.
- Do not use cardboard boxes for storage (Cardboard boxes shed dust and debris and may harbor insects).
- Date and rotate the supplies (first in, first out). This process serves as a reminder that the package is susceptible to contamination and conserves storage space, but it does not guarantee sterility.
- Packs will remain sterile as long as the integrity of the package is maintained.
- Sterile or high-level disinfected containers remain so until they are opened.
- Dispense sterile and high-level disinfected articles from this area.

13.1 SHELF LIFE

The shelf life of a packaged sterile item is event-related and not time-related. An event can compromise the integrity and effectiveness of the package. Events that can compromise or destroy package sterility include multiple handling, loss of package integrity, moisture penetration and airborne contamination. Sterility is lost when the package has tears in the wrapper, has become wet, has been dropped on the floor, has dust on it or is not sealed.

Factors that can destroy sterility or compromise the efficiency of the packaging material to act as a bacterial barrier are:

- dust
- moisture
- holes, breaks, rupture of seals
- opening the package

13.2 HANDLING AND TRANSPORTING INSTRUCTIONS AND OTHER ITEMS

- Keep clean and high-level disinfected or sterile instruments and other items separate from soiled equipment and waste items. Do not transport or store these items together.
- Transport high-level disinfected and sterile instruments and other items to the procedure or operating room in a closed cart or container with a cover to prevent contamination.
- Remove supplies from all shipping cartons and boxes before bringing such supplies into the procedure room, the operating room or the clean work area of the CSSD. (Shipping boxes shed dust and harbor insects that may contaminate these areas).
- Transport soiled supplies and instruments to the receiving/cleanup area of the CSSD in leak-proof, covered waste containers.
- Transport contaminated waste to the disposal site in leak-proof, covered waste containers.

14.0 PROCESSING INSTRUMENTS

The basic infection prevention processes recommended to reduce disease transmission from soiled instruments and other reusable items are **decontamination, cleaning** and either **sterilization** or **high-level disinfection (HLD).** Regardless of the type of operative procedure, the steps in processing surgical instruments and other items are the same.

14.1 DEFINITIONS

Decontamination. A process that makes inanimate objects safer to be handled by staff before cleaning (i.e., inactivates HBV, HBC and HIV and reduces, but does not eliminate, the number of other contaminating microorganisms.

Cleaning. A process that physically removes all visible dirt, soil, blood or other body fluids from inanimate objects as well as removing sufficient numbers of microorganisms to reduce risks for those who touch the skin or handle the object. It consists of thoroughly washing with soap or detergent and water, rinsing with clean water and drying.

High-level Disinfection (HLD). A process that eliminates all microorganisms except some bacterial endospores from inanimate objects by boiling, steaming or the use of chemical disinfectants.

Sterilization. A process that eliminates microorganisms (bacteria, viruses, fungi, parasites, bacterial endospores) from non living objects by high-pressure steam (autoclave), dry heat (oven), chemicals or radiation.

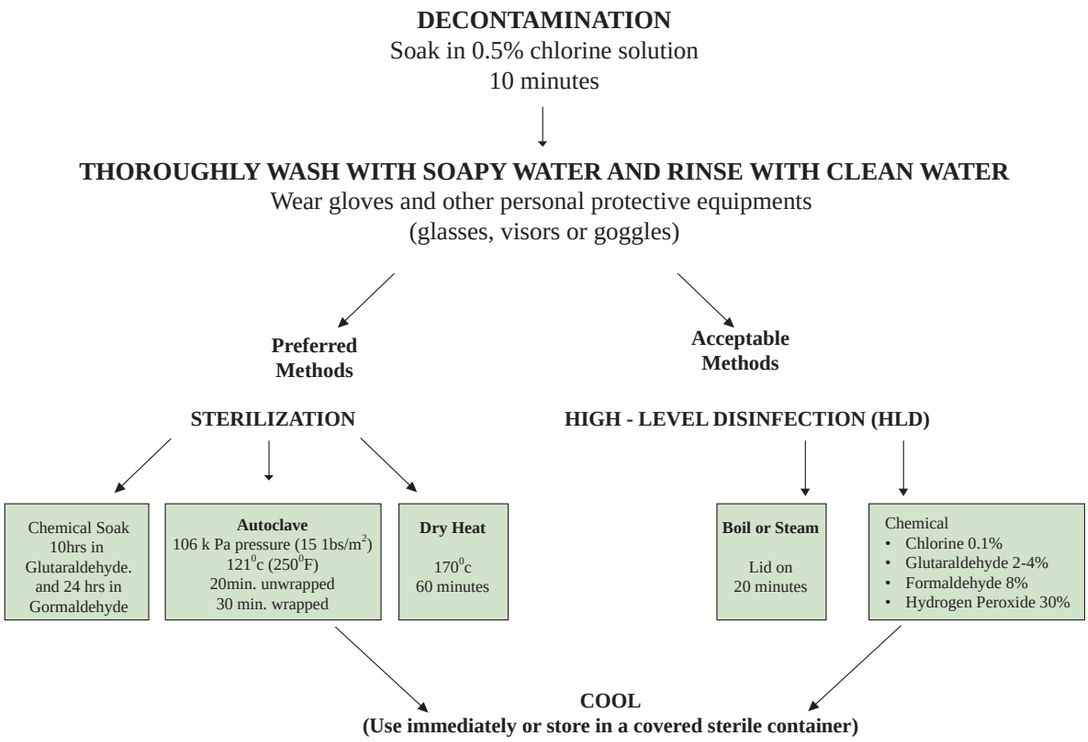
14.2 PROCESSING ITEMS

Each item, whether a soiled metal or plastic instrument, requires special handling/processing in order to:

- minimize the risk of accidental injury or blood or body fluid exposure to housekeeping staff

- provide a high quality end product (i.e. sterile or high-level disinfected instruments and other items)

PROCESSING INSTRUMENTS, AND OTHER ITEMS



Source: *Tietjen,* L, D. Bossemeyer and N. McIntosh. 2003. *Infection Prevention - Guidelines for Healthcare Facilities with Limited Resources*. JHPIEGO Corporation. Baltimore, Maryland. p. 9-2.

- Specific guidelines for processing instruments, equipment and other items used to provide healthcare services are summarized in Table 10a-d below.

Table 10 a-d: Guidelines for Processing Instruments and Other Items

Process	Decontamination is the first step in handling used items; it reduces risk of HBV, HCV and HIV viruses.	Cleaning removes all visible blood, body fluids and dirt.	Sterilization destroys all microorganisms, including all endospores.	High-Level Disinfection destroys all viruses, bacteria, parasites, fungi and some endospores.
INSTRUMENTS OR OTHER ITEMS	**DECONTAMINATION**	**CLEANING**	**STERILIZATION[A]**	**OR HIGH-LEVEL DISINFECTIONB**
Airways (plastic)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse and wash immediately.	Wash with soap and water. Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.
Ambu bags and CPR face masks	Wipe exposed surfaces with gauze pad soaked in 60B90% alcohol or 0.5% chlorine; rinse immediately.	Wash with soap and water. Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.
Aprons (heavy plastic or rubber)	Wipe with 0.5% chlorine solution Rinse with clean water. Between each procedure or each time they are taken off.	Wash with liquid soap and water. Rinse with clean water, air or towel dry at the end of the day or when visibly soiled.	Not necessary	Not necessary
Bed pans, urinals or emesis basins	Not necessary	Using a brush, wash with disinfectant solution (soap and 0.5% chlorine). Rinse with clean water.	Not necessary	Not necessary.
Blood pressure cuff	If contaminated with blood or body fluids, wipe with gauze pad or cloth soaked with 0.5% chlorine solution.	If soiled, wash with soap and water. Rinse with clean water, air or towel dry.	Not necessary	Not necessary
Diaphragms or fitting rings (used for sizing with clients)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.	Wash with soap and water. Rinse with clean water. Air or towel dry.	Not necessary but can be autoclaved at 121°C (250°F) 106 kPa (15 lbs/in2) for 20 minutes (unwrapped).	• Steam or boil for 20 minutes. • Chemically high-level disinfect by soaking in 8% formaldehyde, or a 2–4% glutaraldehyde for 20 minutes. Rinse well in water that has been **boiled.**
Exam or operating room tables or other large surface areas (carts and stretchers)	**Wipe off with 0.5% chlorine solution.**	Wash with soap and water if organic material remains after decontamination.	Not necessary	Not necessary

INSTRUMENTS OR OTHER ITEMS	DECONTAMINATION	CLEANING	STERILIZATIONA	OR HIGH-LEVEL DISINFECTION[b]
Footwear (rubber shoes or boots)	Wipe with 0.5% chlorine solution. Rinse with clean water. At the end of the day or when visibly soiled.	Wash with liquid soap and water. Rinse with clean water, air or towel dry at the end of the day or when visibly soiled.	Not necessary.	Not necessary.
IUDs and inserters (never reuse)	Not appropriate.	Not appropriate.	Not recommended. Most IUDs and inserters come in sterile packages. Discard if package seal is broken.	Not recommended
Laparoscopes	Wipe exposed surfaces with gauze pad soaked in 60-90% alcohol; rinse immediately.	Disassemble, then using a brush wash with soap and water. Rinse with clean water, towel dry.	Sterilize daily using chemical sterilization. Soak in: • glutaraldehyde (usually 2%) for 10 hours, or • 8% formaldehyde for 24 hours. Rinse with **sterile** water or water which has been boiled for 20 minutes three times.	Between cases, soak for 20 minutes in: • a glutaraldehyde (usually 2–4%), or • 8% formaldehyde, or • 0.1% chlorine solution with boiled and filtered (if necessary) water. Rinse three times with water that has been **boiled** for 20 minutes.
PPE (caps, masks, covergowns)[d]	Not necessary. (Laundry staff should wear plastic aprons, gloves and protective foot and eyewear when handling soiled linen.)	Wash with soap and hot water. Rinse with clean water, air or machine dry. Wrap for reuse.	Not necessary.	Not necessary.
Stethoscopes	Wipe with gauze pad soaked in 60–90% alcohol.	If soiled, wash with soap and water. Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.
Storage containers for instruments (metal	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.[c]	Wash with soap and water. Rinse with clean water, air or towel dry.	Dry heat for 1 hour after reaching 170°C (340°F), or Autoclave at 121°C (250°F) and 106 kPa (15 lbs/in^2) for 20 minutes (30 minutes if wrapped).	Boil container and lid for 20 minutes. If container is too large: • Fill container with 0.5% chlorine solution and soak for 20 minutes. • Rinse with water that has been **boiled** for 20 minutes and air dry before use.

INSTRUMENTS OR OTHER ITEMS	DECONTAMINATION	CLEANING	STERILIZATION[A]	OR HIGH-LEVEL DISINFECTION[b]
Suction bulbs (rubber)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse and wash immediately.	Wash with soap and water. Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.
Suction cannulae (plastic) for manual vacuum aspiration (MVA)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.	Pass soapy water through cannulae three times, removing all particles.	Not recommended. (Heat from autoclaving or dry- heat ovens will damage cannulae.)	Steam or boil for 20 minutes.
Surgical gowns, linen drapes and wrappers[d]	Not necessary. (Laundry staff should wear plastic aprons, gloves and protective foot and eyewear, when handling soiled linen.)	Wash with soap and hot water. Rinse with clean water, air or machine dry.	Autoclave at 120°C/250°F and 106 kPa (15 lbs/in2) for 30 minutes.	Not practical.
Surgical instruments metal)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.[c]	Using a brush, wash with soap and water. Rinse with clean water. If to be sterilized, air or towel dry and wrap in packs or individually.	**Preferable:** • Dry heat for 1 hour after reaching 170°C (340°F)e, or • Autoclave at 121°C (250°F) and 106 kPa (15 lbs/in2) for 20 minutes (30 minutes if wrapped). **For sharp instruments:** Dry heat for 2 hours after reaching 160°C (320°F).[e]	**Acceptable:** • Steam or boil for 20 minutes. • Chemically high-level disinfect by soaking for 20 minutes. Rinse well with boiled water and air dry before use or storage.
Thermometers (glass)	Not necessary.	Wipe with disinfectant solution (soap and 0.5% chlorine). Rinse with clean water, air or towel dry.	Not necessary.	Not necessary.

INSTRUMENTS OR OTHER ITEMS	DECONTAMINATION	CLEANING	STERILIZATION[A] DISINFECTION[B]	OR HIGH-LEVEL
Transfer forceps (cheattle) and container (metal)	Soak in 0.5% chlorine solution for 10 minutes prior to cleaning. Rinse or wash immediately.c (Reprocess per shift or when contaminated.)	Using a brush, wash with soap and water. Rinse with clean water. If to be sterilized, air or towel dry.	**Preferable:** • Dry heat for 1 hour after reaching 170°C (340°F), or • Autoclave at 121°C (250°F) and 106 kPa (15 lbs/in2) for 20 minutes (30 minutes if wrapped).	**Acceptable:** • Steam or boil for 20 minutes. Chemically high-level disinfect by soaking for 20 minutes. Rinse well with boiled water and air dry before use.
Ventilator tubing or circuits	Not necessary.	Using a brush, wash with soap and water. Rinse with clean water and air dry.	Not possible using an autoclave or dry heat oven.	Acceptable • Steam or boil for 20 minutes. • Air dry before use.

[a] If unwrapped, use immediately; if wrapped, reprocess if package becomes damaged or contaminated.
[b] If sterilization (dry-heat or autoclave) is not available, these items can be high-level disinfected either by boiling, steaming or soaking in a chemical disinfectant.
[c] Avoid prolonged exposure (> 20 minutes) to chlorine solution (> 0.5%) to minimize corrosion (rusting) of instruments and deterioration of rubber or cloth products.
[d] Paper or plastic gowns, caps or masks. Place in a plastic bag or leak-proof, covered waste container for disposal.
[e] Instruments with cutting edges or needles should not be sterilized at temperatures above 160C to avoid dulling.

Source: Tietjen, L., D. Bossemeyer and N. McIntosh. (2003). *Infection Prevention Guidelines for Healthcare Facilities with Limited Resources*. JHPIEGO Corporation. Baltimore, Maryland, pp. 9-2 – 9-7.

14.3 DECONTAMINATION AND CLEANING

Definition

Decontamination is the first step in processing soiled instruments, gloves and other items. It is the first step before cleaning. Decontamination is placing used items in 0.5% chlorine solution for 10 minutes. This step rapidly inactivates HBV, HCV and HIV and makes the items safer to handle by cleaning personnel. (AORN 1990; ASHCSP 1986). Decontamination and cleaning are two highly effective infection prevention measures that can minimize the risk of transmission of these viruses to healthcare workers.

14.3.1 Decontamination Tips

Use a plastic container for decontamination of instruments to help prevent:

- dulling of sharps (e.g., scissors) due to contact with metal containers
- rusting of instruments due to a chemical reaction (electrolysis) that can occur between two different metals (i.e., the instrument and container) when placed in water

Do not soak metal instruments that are electroplated (i.e., not 100% stainless steel) even in plain water for more that an hour because rusting will occur. After **decontamination,** instruments should be rinsed immediately with cool water to remove visible organic material before being thoroughly cleaned.

Large surfaces, such as pelvic examination or operating tables that may have come in contact with blood and body fluids should be decontaminated. Wiping with a suitable disinfectant such as 0.5% chlorine solution before reuse or when visibly contaminated is an easy, inexpensive way to decontaminate these large surfaces.

14.3.2 Cleaning

Cleaning is the removal of organic materials and debris from used items by washing with soap and water and friction.

- Cleaning that follows decontamination can remove up to 90% of microorganisms (bacteria, viruses, fungi and parasites) and is the best way to reduce the number of endospores, which cause tetanus and gangrene.
- Cleaning should be done under water, using liquid soap and friction to remove all organic material from instruments.
- After cleaning, rinse items in clean water until no detergent remains.
- Air-dry items whenever possible.
- Use heavy-duty gloves for cleaning instruments.
- Wash hands after removing gloves.

Care of all instruments

- Those instruments with moving parts should be lubricated after drying.
- Avoid oils that may protect bacteria during autoclaving.
- Water-soluble lubricant is recommended (Karl Zsort or Olympus instrument oil).

- Never use steel wool or abrasive powders on stainless steel instruments.
- Never label surgical instruments with masking tape.
- Staining/spotting of instruments can be caused by moisture or water.
- When instruments do stain in spite of all good care taken they can be cleaned by using a commercially available rust and stain remover.

New instruments

- All new instruments are supplied without lubrication. It is recommended that all be carefully washed and dried and any moving part lubricated.
- Whenever cleaning, regardless of method, keep ratchets unlocked and box joints open.
- When instruments are no longer new, avoid as far as possible contact between stainless steel instruments and any of the following substances: barium chloride, aluminium chloride, bromide and iodine containing compounds.

Manual cleaning of soiled instruments and equipment

- When an operation is in progress do not drop instruments into a holding solution of disinfectant. If the instruments are not cleaned first, disinfectants such as glutaraldehyde or alcohol act as fixatives of any organic material present, making it difficult to remove.
- Instruments should not be soaked in saline, as they will become pitted.
- Dilute detergent properly as per supplier's directions.
- Completely dismantle all items and leave instruments open.
- Use warm water, detergent and a hard brush to completely remove the blood, tissue, food and other residue, paying special attention to small teeth of instruments and joints.
- Finally rinse with clean water to remove traces of detergent.
- Dry properly. Failure to remove water from trapped areas will cause corrosion.
- Consider the item contaminated when packaging is torn, damaged, wet, dropped on the floor and when the expiry date has passed.

14.3.3 High-Level Disinfection (HLD)

HLD is the process that eliminates all microorganisms (including bacteria, viruses, fungi and parasites), but does **not** reliably kill all bacterial endospores, which cause diseases such as tetanus and gas gangrene. HLD is suitable for instruments and items that come in contact with broken skin or intact mucous membranes.

Note: *Sterilization kills all microorganisms, including bacterial endospores; it is preferable to HLD for instruments and other items that will come in contact with the bloodstream or tissues under the skin. If sterilization is not available, HLD is the only acceptable alternative.*

HLD can be performed by:

- Boiling
- Soaking in chemicals
- Steaming

HLD by boiling

Step 1: Decontaminate and clean all items to be boiled.
Open all hinged items and disassemble those with sliding or multiple parts.
Completely submerge all items in the water in the pot or boiler.
Place any bowls and containers upright, not upside-down, and fill with water.

Step 2: Cover the pot or close the lid on the boiler and bring the water to a gentle, rolling boil.

Step 3: When the water comes to a rolling boil, start timing for 20 minutes. Use a timer to make sure to record the time that boiling begins. From this point on, do not add or remove any water and do not add any items to the post or boiler.

Step 4: Lower the heat to keep the water at a gentle, rolling boil.

Note: *If the water boils too vigorously, it will evaporate, and the items may become damaged if they bounce around the container and hit the sidewalls and other items being boiled. Lower heat also saves fuel or electricity.*

Step 5: After 20 minutes, remove the items using dry, HLD pickups (lifters, cheatle forceps). Place the items on an HLD tray or in an HLD container away from insects and dust.

Note: *An HLD tray or container can be prepared by boiling it for 20 minutes or by filling it with 0.5% chlorine solution and letting it soak for 20 minutes, then draining the solution and rinsing thoroughly with sterile water.*

Step 6: Allow to air-dry before use or storage

Step 7: Use items immediately or keep them in a covered, sterile or HLD container for up to one week.

Note: *Never leave boiled items in water that has stopped boiling; they can become contaminated as the water cools down.*

Tips for HLD by Boiling

- Items must be completely covered with water. Open all hinged instruments and disassemble items with sliding or multiple parts.
- Always boil for 20 minutes. Start timing when the water reaches a rolling boil. If you forget to start timing the procedure, start timing at the point at which you realize this.
- Do **not** add **anything** to or remove **anything** from the boiler once boiling begins.

14.3.4 Steaming

After instruments and other items have been decontaminated and thoroughly cleaned, they are ready for HLD by steaming.

Step 1: Place instruments, plastic MVA cannulas and other items in one of the steamer pans with holes in the bottom. To make removal from the pan easier, do not overfill the pan.

Step 2: Repeat this process until up to three steamer pans have been filled. Stack the filled steamer pans on top of a bottom pan containing water for boiling. A second empty pan without holes should be placed on the counter next to the heat source.

Step 3: Place a lid on the top pan and bring the water to a full rolling boil. (When water only simmers, very little steam is formed and the temperature may not get high enough to kill microorganisms).

Step 4: When steam begins to come out between the pans and the lid, start the timer or note the time on a clock and record the time in the HLD log.

Step 5: Steam items for 20 minutes.

Step 6: Remove the top steamer pan and put the lid on the pan that was below it (the pan now on top). Gently shake excess water from the pan just removed.

Step 7: Put the pan just removed onto the empty pan. Repeat until all pans are restacked on this empty pan and the top pan is covered with the lid. (This step allows the items to cool and dry without becoming contaminated).

Step 8: Allow items to air dry in the steamer pans (1 to 2 hours) before using.

Step 9: Using a high-level disinfected forceps, transfer the dry items to a dry, high-level disinfected container with a tight-fitting cover. Instruments and other items can also be stored in the stacked and covered steamer pans as long as a bottom pan (no holes) is used.

Note: *Both boiling and steaming share some advantages and disadvantages over chemical high-level disinfection, which is the only other method of HLD.*

Advantages

- Inexpensive procedure
- Easily taught to healthcare workers
- Require no special chemicals or dilutions and leave no chemical residue
- Heat sources (boilers or rice cooers) are commonly available

Disadvantages

- Length of processing time must be carefully measured (i.e., start timing only after steam begins to escape or water has reached a rolling boil)

- Objects cannot be packaged prior to HLD; therefore, there is a greater chance of contamination if items are to be stored
- Requires a fuel source that may be unreliable

14.3.5 HLD by Chemicals

Step 1: Decontaminate, clean, and thoroughly dry all instruments and other items to be processed. Water from wet items will dilute the chemical solution, thereby reducing its effectiveness.

Step 2: **When using glutaraldehyde solution:** Prepare the solution according to the manufacturer's instructions. Ideally, an indicator strip should be used each time the solution is used to determine if the solution is still effective. After preparing the solution, place in a clean container with a lid. Mark the container with the date the solution was prepared and the date it expires.
When using a chlorine solution: Prepare the 0.1% chlorine solution, or 0.5% if using unboiled water, as described. Fresh solution should be made each day, or more often if the solution becomes cloudy. Put the solution in a clean container with a lid.

Note: *Use chlorine solution with boiled water and not tap water*

Step 3: Open all hinged items and disassemble those with sliding or multiple parts. The solution must contact all surfaces in order for HLD to be achieved. Completely submerge all items in the solution. All parts of the items should be under the surface of the solution. Place any bowls and containers upright, not upside-down, and fill with the solution.

Step 4: Cover the container, and allow the items to soak for 20 minutes. Do not add or remove any instruments or other items once timing has begun.

Step 5: Remove the items from the solution using dry, HLD pickups (lifters, cheatle forceps).

Step 6: Rinse thoroughly with sterile or boiled water to remove the residue that chemicals leave on items. This residue is toxic to skin and tissue.

Step 7: Place the items on an HLD tray or in an HLD container and allow to air dry before use or storage. Use items immediately or keep in a covered, dry HLD container and use within one week.

Note: *An HLD tray or container can be prepared by boiling it for 20 minutes or by filling it with a 0.5% chlorine solution and letting it soak for 20 minutes, then draining the chlorine solution and rinsing thoroughly with boiled water.*

Tips for Chemical HLD

- Items must be completely covered with solution.
- Open all hinged instruments and disassemble items with sliding or multiple parts.
- Soak for 20 minutes. If you forget to start timing, start at the point you remember.
- Do not add or remove anything once timing begins.
- Rinse items thoroughly with boiled water.
- Antiseptics should **never** be used for HLD.

14.3.6 Sterilization

Sterilization protects patients by eliminating **all** micro-organisms (bacteria, viruses, fungi, and parasites), including bacterial endospores, from instruments and other items. Sterilization is recommended for instruments and other items that will come in contact with the bloodstream or tissues under the skin, as well as on draped and some surgical attire.

Sterilization can be performed using:

- dry heat (oven)
- high pressure steam (autoclaving)
- soaking in chemicals (cold sterilization)

Heat (autoclaving/steam and dry heat) is the most effective method of sterilization and reliable if monitored carefully. It is also cheaper than chemical methods. It should be considered first for all medical equipment that can withstand heat. **Chemicals** are the alternative where heat cannot be used, e.g., ethylene oxide and glutaraldehyde.

Sterilization by Dry Heat

Time/Temperature	1 hour at 170 degrees C (340 degrees F)
	2 hours at 160 degrees C (320 degrees F)
	$2^1/_2$ hours at 150 degrees C (300 degrees F)
	3 hours at 140 degrees C (285 degrees F)

Sterilization by Steam

Time	20 minutes (or 30 minutes if wrapped)
Temperature	121 degrees C (250 degrees F)
Pressure	106 KPA (15 lbs/sq inch)

Sterilization by Chemicals

Chemical sterilization method is used for instruments and other items that are heat-sensitive or when heat sterilization is not available.

Step 1: Decontaminate, clean, and thoroughly dry all instruments and other items to be sterilized. Water from wet instruments and other items dilutes the chemical solution, thereby reducing its effectiveness.

Step 2: Prepare the glutaraldehyde or other chemical solution by following the manufacturer's instructions or use a solution that was prepared previously, as long as it is clear (not cloudy) and has not expired. After preparing the solution, put it in a clean container with a lid. Always mark the container with the date the solution was prepared and the date it expires.

Step 3: Open all hinged instruments and other items and disassemble those with sliding or multiple parts, the solution must contact all surfaces in order for sterilization to be achieved. Completely submerge all instruments and other items in the solution. Place any bowls and containers upright, not upside-down, and fill with the solution

Step 4: Follow the manufacturer's instructions regarding the time necessary for sterilization to be achieved. In general, if the solution contains glutaraldehyde, cover the container, and allow the instruments and other items to soak for 10 hours. Do not add or remove any instruments or other items once time has begun.

Step 5: Remove the instruments and other items from the solution using large, sterile pickups (lifters, cheatle forceps).

Step 6: Rinse thoroughly with sterile water to remove the residue that chemicals leave on instruments and other items; this residue toxic to skin and tissues.

Step 7: Storage: Place the instruments and other items on a sterile tray or in a sterile container and allow air-drying before use. Use the instruments and other items immediately or keep in a covered, dry, sterile container and use within one week.

Step 8: After processing, items should be used immediately or stored in such a way that they do not become contaminated. Proper storage is as important as proper processing.

15.0 PROCESSING LINEN

In processing linen, staff performing these tasks should be appropriately trained and regularly supervised. To reduce the risk of contamination, staff at each healthcare facility should determine the best way to handle, process and store linens. Without this, accidents will happen and staff will be at increased risk of exposure to infectious materials and acquiring work-related infections (Economics Report 1994).

Definitions

Linens. Cloth items used by housekeeping staff patients/clients (bedding, towels, cleaning cloths, gowns, caps, masks, scrub suits, surgical gowns, drapes and wrappers).

Soaps or detergents (terms used interchangeably). Cleaning products such as bar soap, liquid soap and powder. These substances lower surface tension, thereby helping remove dirt, debris and transient microorganisms from hands.

Soiled or contaminated linen. Linen from multiple sources within the hospital or clinic that has been collected and brought to the laundry for processing. All items, regardless of whether or not they are visibly dirty or have been used in a surgical procedure, must be washed and dried.

Sorting. Process of inspecting and removing foreign, and, in some cases dangerous, objects (e.g., sharps or broken glass), from soiled linen before washing. This step is extremely important because soiled linen from the operating room or clinic occasionally contains sharps (e.g., scalpels, sharp-tipped scissors, hypodermic and suture needles and towel clips).

***Note**: Staff responsible for washing soiled items should wear utility gloves, protective eye wear plastic or rubber aprons and protective foot wear.*

Table 11. Key Steps in Processing Linen

- Housekeeping and laundry personnel should wear gloves and other personal protective equipment as indicated when collecting, handling, transporting, sorting and washing soiled linen.
- When collecting and transporting soiled linen, handle it as little as possible and with minimum contact to avoid accident, injury and spreading of microorganisms.
- Consider all cloth items (e.g., surgical drapes, gowns, wrappers) used during a procedure as infectious. Even if there is no visible contamination, the item must be laundered.
- Carry soiled linen in covered containers or plastic bags to prevent spills and splashes, and confine the soiled linen to designated areas (interim storage areas) until transported to the laundry.
- Carefully sort all linen in the laundry area before washing. Do not presort or wash linen at the point of use.

15.1 COLLECTING AND TRANSPORTING SOILED LINEN

Soiled linen should be collected after invasive medical or surgical procedures or when changing linen in patient rooms.

- Collect used linen in cloth or plastic bags or containers with lids. If linen is heavily contaminated with blood or body fluids, carefully roll the contaminated area into the center of the linen and place in a leak-proof bag or container with a lid.
- Cloth bags are adequate for the majority of patient care linen. They require the same processing as their contents.
- Handle soiled linen as little as possible and do not shake it. This helps prevent spreading microorganisms to the environment, personnel and other patients.
- It is not necessary to double-bag or use additional precautions for used linen from patients in isolation.

- Do not sort and wash soiled linens in patient-care areas (CDC 1988; OSHA 1991).
- Collect and remove soiled linen after each procedure, and daily or as needed from patients' rooms.
- Transport collected soiled linen in closed leak-proof bags, containers with lids or covered carts to the processing area daily or more often as needed.
- Transport soiled linen and clean linen separately. If there are separate carts or containers available for soiled and clean linen, they should be labeled accordingly. If not, thoroughly clean the containers or carts used to transport soiled linen before using them to transport clean linen.

15.2 SORTING SOILED LINEN

The processing area for soiled linen must be separate from other areas such as those used for folding and storing clean linen, patient care and food preparation. In addition there should be adequate ventilation and physical barriers (walls) between the clean and soiled linen areas.

Sorting must be carefully performed because soiled linen (large drapes and towel drapes) from the operating room or other procedure areas occasionally contain sharps (e.g., scalpels, sharp-tipped scissors, hypodermic and suture needles and sharp-tipped towel clips). In addition, bedding from patients' rooms may contain soiled dressings and be bloodstained or wet with other body fluids.

15.3 LAUNDERING LINEN

All linen items (e.g., bed sheets, surgical drapes, masks, gowns) used in the direct care of a patient must be thoroughly washed before re-use. Decontamination prior to washing is **not necessary,** unless linen is heavily soiled and will be hand washed (repeat soaking of linen in chlorine, even dilute solutions, will cause the fabric to deteriorate more quickly). In addition, workers should not carry wet, soiled linen close to their bodies even if they are wearing a plastic or rubber apron.

Handwashing linen

Step 1: Wash heavily soiled linen separately from non-soiled linen.

Step 2: Wash the entire item in water with liquid soap to remove all soils, even if not visible.
Use warm water if available.
Add bleach (e.g., 30-60 mL, about 2-3 tablespoons, of 5% chlorine solution) to aid cleaning and bactericidal action.
Add soap (a mild acid agent) to prevent yellowing of linen, if desirable.

Step 3: Check the item for cleanliness. Rewash if it is dirty or stained.

Step 4: Rinse the item with clean water.

Machine washing linen

Step 1: Wash heavily soiled linen separately from non-soiled linen.

Step 2: Adjust the temperature and time cycle of the machine according to manufacturer's instructions and the type of soap or other washing product being used. Both cold and hot water washing cycles that include bleach reduce bacterial counts in the linen.

Note: hot-water washing linen:

- Use hot water above 71ºC (160ºF) and soap to aid in loosening soil.
- Add bleach and soap as above.
- Adjust the time cycle of the machine according to the manufacturer's instructions.

Step 3: When the wash cycle is complete, check the linen for cleanliness. Rewash if it is dirty or stained. (Heavily soiled linen may require two wash cycles).

Drying, checking and folding linen

For both hand and machine washed linens, the steps are the same.

Step 1: Completely air or machine dry before further processing. Air dry in direct sunlight, if possible, keeping the fabric off the ground, away from dust and moisture.

Step 2: After linen items are totally dry, check for holes and threadbare areas. If present, the item must be discarded or repaired before re-use or storage. (If there are any holes or many repaired areas, the item should not be used as a drape. It can be cut into pieces to be used a cleaning rags.)

Step 3: Clean and dry linen should be ironed as needed and folded. For example, if a clean, dry drape is acceptable, the drape can be ironed before placing it on a shelf or in a container for storage.

15.4 STORING, TRANSPORTING AND DISTRIBUTING CLEAN LINEN

Storing Clean Linen

- Keep clean linen in clean, closed storage areas.
- Use physical barriers to separate folding and storage rooms from soiled areas.
- Keep shelves clean.
- Handle stored linen as little as possible.

Transporting Clean Linen

- Clean and soiled linen should be transported separately.
- Containers or carts used to transport soiled linen should be thoroughly cleaned before used to transport clean linen; if different containers or carts are used to transport clean and soiled linen, they should be labeled.
- Clean linen must be wrapped or covered during transport to avoid contamination.

Table 12: Guidelines for Processing Linens and Personal Protective Equipment (PPE)

ITEM	DECONTAMINATION	CLEANING	HIGH-LEVEL DISINFECTION	STERILIZATION
Protective eyewear (plastic goggles and face shields)	Wipe with 0.5% chlorine solution. Rinse with clean water. After each procedure or when is visibly soiled.	Wash with liquid soap and water. Rinse with clean water, then air or towel dry.[1] After each procedure or when visibly soiled.	Not necessary	Not necessary
Linens (caps, masks, scrub suits or cover gowns)	Not necessary. (Laundry staff should wear plastic aprons, gloves, and protective foot and eyewear when handling soiled items.)	Wash with liquid soap and water, removing all dirt particles. Rinse with clean water, air or machine dry.[1] Air- dried attire can be ironed before use.	Not necessary	Not necessary
Aprons (heavy plastic or rubber)	Wipe with 0.5% chlorine solution. Rinse with clean water. Between each procedure or each time they are taken off.	Wash with liquid soap and water. Rinse with clean water, air or towel dry at the end of the day or when visibly soiled.[1]	Not necessary	Not necessary
Footwear (rubber shoes or boots)	Wipe with 0.5% chlorine solution. Rinse with clean water. At the end of the day or when visibly soiled.	Wash with liquid soap and water. Rinse with clean water, air or towel dry at the end of the day or when visibly soiled.[1]	Not necessary	Not necessary
Surgical gowns, linen drapes and wrappers	Not necessary. (Laundry staff should wear plastic aprons, gloves and protective foot and eyewear when handling soiled items.)	Wash with liquid soap and water removing all particles. Rinse with clean water, air or machine dry.[1]	Not practical	Preferred
Paper or disposable plastic items	Place in plastic bag or leak-proof, covered waste container for disposal.			

[1]If tap water is contaminated, use water that has been boiled for 10 minutes and filtered to remove particulate matter (if necessary), or use chlorinated water—water treated with a dilute bleach solution (sodium hypochlorite) to make the final concentration 0.001%

Source: Tietjen, L., D. Bossemeyer and N. McIntosh. (2003) *Infection Prevention - Guidelines for Healthcare Facilities with Limited Resources*. JHPIEGO Corporation. Baltimore, Maryland. p. 13-7.

Distributing Clean Linen

- Protect clean linen until it is distributed for use.
- Do not leave extra linen in patients' rooms.
- Handle clean linen as little as possible.
- Avoid shaking clean linen. It releases dust and lint into the room.
- Clean soiled mattresses before putting clean linen on them.

16.0 HOUSEKEEPING

Housekeeping refers to the general cleaning of hospitals and clinics, including the floors, walls, and certain types of equipment, furniture and other surfaces. Cleaning entails the removal of dust, soil, and microbial contaminants on environmental surfaces since they are the potential source of nosocomial infections. Effective and efficient cleaning methods and schedules are, therefore, necessary to maintain a clean and healthy environment in healthcare settings.

The purposes of general housekeeping are to:

- reduce the number of microorganisms that may come in contact with patients, visitors, staff and the community
- provide a clean and pleasant atmosphere for patients and staff

In high-risk areas where heavy contamination is expected, such as toilets and latrines, or for blood or body fluid spills, a disinfectant such as 0.5% chlorine or 1% phenol should be added to the cleaning solution (SEARO 1988). Using a disinfectant in addition to soap and water is also recommended in other high-risk areas such as operating rooms, pre- and postoperative recovery areas and intensive care units (ICUs).

If the purpose of housekeeping as stated above is to be achieved, it is important that house keeping staff be trained to perform their assigned tasks and are supervised on a regular basis. As part of their training, it is important that housekeeping staff:

- understand the risk of exposure to contaminated items and surfaces when performing environmental cleaning procedures
- follow recommended policies and guidelines, including the use of appropriate personal protective equipment

16.1 DEFINITIONS

Cleaning solution. Any combination of soap (or detergent) and water, with or without a chemical disinfectant, used to wash or wipe down environment surfaces such as floors, chairs, benches, walls and ceilings.

Disinfectants. Chemicals that destroy or inactivate microorganisms. Disinfectants are classified as low-, intermediate- or high-level depending on their ability to kill or immobilize some (low- or intermediate-level) or all (high-level) microorganisms (but not all spores). Phenols, chlorine or chlorine-containing compounds and QUATs are classes of disinfectants frequently used to clean non-critical surfaces such as floors, walls and furniture.

Disinfectant cleaning solution are products that are a combination of a detergent (soap) and a chemical disinfectant. Not all detergents and disinfectants are compatible. Several combinations are available commercially or can be prepared, such as alkaline detergents with chlorine compounds, alkaline detergents with quaternary ammonium compounds (QUATs) or other nonionic surfactants, and acid detergents with iodophors.

Environmental controls are standards specifying procedures to be followed for the routine care, cleaning and disinfection of environmental surfaces, beds, bedrails, bedside equipment and other frequently touched surfaces.

Sanitizers include chemicals that reduce the number of bacterial contaminants to safe levels on inanimate objects based on public health requirements (i.e., a chemical that kills 99.999% of the specific test bacteria in 30 seconds under the conditions of the test).

Soaps and **detergents (terms used interchangeably).** These are cleaning products (bar, liquid, leaflet or powder) that lower surface tension, thereby helping remove dirt, debris and transient microorganisms from hands. Plain soaps require friction (scrubbing) to mechanically remove microorganisms; antiseptic (antimicrobial) soaps kill or inhibit the growth of most microorganisms.

Table 13: General Principles for Cleaning Hospitals, Clinics and other Healthcare Facilities

- **Scrubbing** (frictional cleaning) is the best way to physically remove dirt, debris and microorganisms.
- **Cleaning** is required prior to any disinfection process because dirt, debris and other materials can decrease the effectiveness of many chemical disinfectants.
- **Cleaning products** should be selected on the basis of their use, efficacy, safety and cost.
- **Cleaning** should always progress from the least soiled areas to the most soiled areas and from high to low areas, so that the dirtiest areas and debris that falls on the floor will be cleaned up last.
- **Dry sweeping, mopping and dusting** should be avoided to prevent dust, debris and microorganisms from getting into the air and landing on clean surfaces. Airborne fungal spores are especially important as they can cause fatal infections in immunosupressed patients (Arnow et al., 1991).
- **Mixing (dilution) instructions should be followed** when using disinfectants. (Too much or too little water may reduce the effectiveness of disinfectants).
- **Cleaning methods and written cleaning schedules** should be based on the type of surface, amount and type of soil present and the purpose of the area.
- **Routine cleaning** is necessary to maintain a standard of cleanliness. Schedules and procedures should be consistent and posted

Source: Tietjen, L., D. Bossemeyer and N. McIntosh. (2003) *Infection Prevention - Guidelines for Healthcare Facilities with Limited Resources*. JHPIEGO Corporation. Baltimore, Maryland, p. 16-2.

16.2. HOW TO SELECT CLEANING PRODUCT

Different types of cleaning products are available: liquid soap and detergents, disinfectants, combinations (detergent and disinfectant) and sanitizers. Each type has different properties.

An ideal cleaning product should accomplish the following.

- Suspension of fats (suspend fats in water)
- Saponification of fats (make fats water-soluble)
- Surfaction (decrease surface tension of water and allow greater penetration of the agent into the dirt or soil)
- Dispersion (break up of soil into small particles)
- Protein destruction (break up protein)
- Softening the water (removal of calcium and magnesium)

16.3 CLEANING METHODS

Cleaning should start with the least soiled area and move to the most soiled area and from high to low surfaces. Common methods of cleaning are briefly described below.

Wet mopping is the most common and preferred method to clean floors.

- **Single-bucket (basin) technique:** One bucket of cleaning solution is used. The solution must be changed when dirty. (The killing power of the cleaning product decreases with the increased load of soil an organic material present).

- **Double-bucket technique:** Two different buckets are used, one containing a cleaning solution and the other containing rinse water. The mop is always rinsed and wrung out before it is dipped into the cleaning solution. The double-bucket technique extends the life of the cleaning solution (fewer changes are required), saving both labor and material costs.

- **Triple-bucket technique:** The third bucket is used for wringing out the mop before rinsing, which extends the life of the rinse water.

Flooding followed by wet vacuuming is recommended in the surgical suite, if possible. This process eliminates mopping, thus minimizing the spread of microorganisms, and increases the contact time of disinfectants with the surface to be cleaned. But it is necessary to leave the floor wet for several minutes. (Flooding is best done at night or at times when foot traffic is minimal).

Dusting is most commonly used for cleaning walls, ceiling, doors, windows, furniture and other environmental surfaces.

- Clean cloth or mops are wetted with cleaning solution contained in a basin or bucket. The double-bucket system minimizes the contamination of the cleaning solution.

- Dry dusting should be avoided, and dust cloths and mops should never be shaken to avoid the spread of microorganisms.

- Dusting should be performed in a systematic way, using a starting point as a reference to ensure that all surfaces have been reached.

Note: *When doing high dusting (ceiling tiles and walls), check for stains that may indicate possible leaks. (Leaks should be repaired as soon as possible because moist ceiling tiles provide a reservoir for fungal growth).*

16.4 USE OF PERSONAL PROTECTIVE EQUIPMENT

Table 14: Recommended Personal Protective Equipment for Housekeeping Tasks

TYPE OF PPE	WHEN USED
• Gloves (preferably household utility gloves) • Boots	• Handling disinfectant cleaning solutions • Cleaning patient care areas • Cleaning heavily contaminated areas • Handling soiled linen • Handling soiled items and instruments • Handling or disposing of waste
• Plastic apron, mask and protective eye wear	• When spills or splashes are expected

Source: Tietjen, L., D. Bossemeyer and N. McIntosh. (2003) *Infection Prevention - Guidelines for Healthcare Facilities with Limited Resources*. JHPIEGO Corporation. Baltimore, Maryland, p. 16-6.

16.5 SCHEDULE AND PROCEDURES FOR SPECIFIC AREAS

Housekeeping schedules should be planned, written and closely followed. Cleaning schedules should be developed according to the needs of each area.

- **Walls, windows, ceilings and doors, including door handles:** Spot clean when visibly dirty with a damp cloth, detergent and water. In general, routine damp dusting is adequate for these areas (disinfection is unnecessary). These surfaces are rarely heavily contaminated with microorganisms, as long the surfaces remain dry and intact (Russell, Hugo and Ayliffe, 1982).
- **Chairs, lamps, tables, tabletops, beds, handrails, grab bars, lights, tops of doors and counters:** Wipe daily and whenever visibly soiled with a damp cloth, containing disinfectant cleaning solution. Treat blood or other body fluid spills as described below.
- **Non-critical equipment (e.g., stethoscopes and blood pressure cuffs):** Wipe daily and whenever visibly soiled use a damp cloth, detergent and water. If the equipment is visibly soiled with blood or other body fluids, or if the patient is under contact precautions, it should be cleaned and disinfected before it is reused.
- **Floors:** Clean floors at least three times daily and as needed with a wet mop, detergent and water. A disinfectant should be used when contamination is present, such as blood or other body fluid spills as described below.

- **Sinks:** Scrub frequently (daily or more often as needed) with a cloth or brush and a disinfectant cleaning solution. Rinse with water.
- **Toilets and latrines:** Scrub frequently at least three times daily and as needed with a separate mop, cloth or brush and a disinfectant cleaning solution (HARPIC).
- **Patient rooms:** Clean at least three times daily and after patient discharge. The same cleaning process applies to rooms of patients who are under isolation precautions. Any cleaning equipment used in the rooms of patients under isolation precautions should be cleaned and disinfected before used in another room.
- **Procedure rooms:** Wipe horizontal surfaces, equipment and furniture used for procedures with a disinfectant cleaning solution after each procedure; whenever visibly soiled decontaminate before cleaning.
- **Examination rooms:** Wipe horizontal surfaces with a disinfectant cleaning solution after each procedure and whenever visibly soiled. Linen or paper on the examination table should be changed after each patient. Decontaminate before cleaning blood or other body fluid spills.
- **Laboratory:** Wipe countertops with a disinfectant cleaning solution after each shift; whenever visibly soiled decontaminate before cleaning.
- **Curtains:** Change and clean curtains weekly and when soiled.
- **Carpets:** Vacuum carpets daily in patient rooms, in offices and conference rooms.
- **Soiled linen**: Collect soiled linen three times daily (or more often as needed) in closed, leak-proof containers.
- **Waste:** Collect waste from all areas at least three times a day (or more frequently as needed). **Avoid overflowing.**
- **Waste containers:** Disinfect and clean contaminated waste containers after emptying each time and non-contaminated waste containers when soiled. Use a disinfectant cleaning solution and scrub to remove soil and organic material.

16.5.1 Schedule and Procedures for the Operating Room

- At the beginning of each day, all flat (horizontal) surfaces (table, chairs, etc.) should be wiped with a clean, lint-free moist cloth to remove dust and lint that may have collected overnight.
- Total cleaning is not necessary between each case for surgical procedures.
- Total cleaning or terminal cleaning (mopping floors and scrubbing all surfaces from top to bottom) of the operating room should be done at the end of each day.

16.6 TOTAL CLEANING

Step 1: Move covered decontamination buckets to the central supply or processing room. A clean bucket containing fresh 0.5% chlorine solution, or other locally available and approved disinfectant, should be provided at the beginning of each day and after each case.

Step 2: Remove covered contaminated waste container and replace it with a clean container. Arrange for burning (incineration) or burial as soon as possible.

Step 3: Close and remove sharps containers when three quarters full.

Step 4: Remove soiled linen in closed leak-proof containers.

Step 5: Soak a cloth in disinfectant cleaning solution and wipe down all surfaces, including counters, tabletops, sinks, lights, etc. Wash from top to bottom, so that any debris that falls on the floor will be cleaned up last.

Walls and ceilings: Wipe with a damp cloth, detergent and water as needed for visible soil.
Chairs, lamps, sinks, tabletops and counters: Wipe with a damp cloth and disinfectant cleaning solution.
Operating room lamp: Wipe with a damp cloth and disinfectant cleaning solution.
Operating room table. Wipe with 0.5% chlorine solution (or other approved disinfectant) to decontaminate. Then clean top, sides, base, legs and any accessories (e.g., leg stirrups) with a damp cloth and disinfectant cleaning solution.
Floors. Clean with a wet mop using a disinfectant cleaning solution.
Vents (heating or air conditioning). Wipe with a damp cloth, soap and water.

Between each patient, do the following:

Spills. Clean spills with 0.5% solution or other locally available and approved disinfectant.
Operating room bed. Wipe all surfaces and mattress pads with a disinfectant cleaning solution.
Instrument tables (trolley and Mayo stand) and other flat surfaces. Wipe all flat surfaces that have come in immediate contact with a patient or body fluids with a disinfectant cleaning solution.
Center of operating room surrounding the operating room bed. Mop with a disinfectant cleaning solution (if visibly soiled).
Waste. Collect and remove all waste from the operating room in closed, leak-proof containers.
Sharps containers. Close and remove containers from the operating room when they are three quarters full.
Containers with 0.5% chlorine solution for decontamination. Remove covered containers with instruments from the operating room and replace them with clean containers with fresh 0.5% chlorine solution.
Soiled linen. Remove soiled linen in leak-proof, covered waste containers.

16.7 HOW TO CLEAN SOILED AND CONTAMINATED CLEANING EQUIPMENT

Step 1: Decontaminate cleaning equipment that has been contaminated with blood or body fluids by soaking it for 10 minutes in 0.5% chlorine solution or other locally available and approved disinfectants.

Step 2: Wash cleaning buckets, cloths, brushes and mops with detergent and water daily, or sooner if visibly dirty.

Step 3: Rinse in clean water.

Step 4: Dry completely before re-use. (Wet cloths and mop heads are heavily contaminated with microorganisms).

PREVENTING INFECTIONS IN SPECIAL SETTINGS/CONDITIONS

17.0 PREVENTING NOSOCOMIAL INFECTIONS

Nosocomial infections, or hospital-acquired infections, are those that a patient develops with in 24 hours of admission in a hospital, or infections that a patient is incubating at the time he or she comes to the hospital. Nosocomial infections are a significant problem throughout the world and are increasing (Alvarado 2000). Although the exact data for the transmission of nosocomial infections in Tanzanian health facilities are yet to be determined, these infections are important contributors to morbidity and mortality. Nosocomial infections are an important focus of infection prevention in all countries, but especially in developing countries.

The most important nosocomial infections are:

- maternal and newborn infections
- infections following surgery
- infections related to intravascular interventions
- urinary tract infections
- pneumonia
- infectious diarrhea

The organisms causing most nosocomial infections usually come from the patient's own body (endogenous flora). They also can come from contact with staff (cross-contamination), contaminated instruments and needles, and the environment (exogeneous flora). Key contributing factors are:

- inadequate standards and practices for operating blood transfusion services
- increasing use of invasive medical devices (e.g., mechanical ventilators, urinary catheters and central intravenous lines) without proper training or laboratory support
- use of contaminated intravenous fluids, especially in hospitals making their own IV solutions.
- antibiotic resistance due to overuse of broad spectrum antibiotics
- unsafe and frequently unnecessary injections
- increasing numbers of people in healthcare facilities, overcrowding in wards, sharing beds
- more frequent impaired immunity (age, illness and treatments)
- new microorganisms, such as HIV, SARS and Ebola

17.1 IMPACT OF NOSOCOMIAL INFECTIONS

Nosocomial infections add to functional disability, emotional stress and may, in some cases, lead to disabling conditions that reduce the quality of life. In addition, nosocomial infections have now become one of the leading causes of death (Ponce-de-Leon, 1991). The impact of nosocomial infections takes on even more significance in resource-poor countries, especially those affected most by HIV/AIDS; recent findings strongly suggest that unsafe medical care may be an important factor in transmitting HIV (Gisselquist et al., 2002).

Nosocomial infections increase the cost of healthcare in the countries least able to afford it through increased:

- length of hospitalization
- treatment with expensive medications (e.g., antiretroviral drugs for HIV/AIDS and antibiotics)
- use of other services (e.g., laboratory tests, X-rays and transfusions)

17.2 PREVENTING NOSOCOMIAL INFECTIONS

Isolation Precaution Guidelines for Hospitals

Isolation Guidelines issued by CDC in 1996 involved a two-level approach.

1. **Standard Precautions** – These apply to all clients and patients attending healthcare facility

2. **Transmission-Based Precautions** – These apply primarily to hospitalized patients

Note: *In all situations, whether used alone or combined, Transmission-Based Precautions must be used in conjunction with Standard Precautions. (Garner and HiCPA, 1995)*

Airborne Precautions

Airborne precautions are designed to reduce the nosocomial transmission of particles that can remain in the air for several hours and be widely dispersed. They are used in addition to Standard Precautions for a patient known or suspected to be infected with microorganisms transmitted by the airborne route, such as tuberculosis.

Table 15: Airborne Precautions

PATIENT PLACEMENT

- Private room
- Door closed
- Room air is exhausted to the outside (negative air pressure) using fan or other filtration system
- If private room not available, place patient in room with patient having active infection with the same disease, but with no other infection (cohorting)
- Check all visitors for susceptibility before allowing them to visit

RESPIRATORY PROTECTION

- Wear surgical mask
- If TB known or suspected, wear particulate respirator (if available).
- If chicken pox or measles:
  - Immune persons—no mask required
  - Susceptible persons—do not enter room
- Remove mask after leaving the room and place in a plastic bag or waste container with tight-fitting lid

PATIENT TRANSPORT

- Limit transport of patient to essential purposes only.
- During transport, patient must wear surgical mask.
- Notify area receiving the patient.

Adapted from: ETNA Communications 2000.

Droplet Precautions

Droplet precautions reduce the risks for nosocomial transmission of pathogens spread by droplets. Examples of pathogens include viruses such as influenza, and rubella (german measles) and mumps, and bacteria such as Mycoplasma pneumoniae, Corynebacterium diphtheriae (diphtheria), Hemophilus pertussis (whooping cough), Pasteurella pestis (pneumonic plague) and Streptococcus pharyngitis sometimes causing scarlet fever.

Note: *Droplet infections are simpler to prevent than airborne precautions, because the particles remain in the air for a short time, and travel only a few feet. Therefore contact with the source must be close for a susceptible host to become infected.*

Table 16: Droplet Precautions

Use in addition to Standard Precautions for a patient known or suspected to be infected with microorganisms transmitted by large-particle droplets (larger than 5 μm).

PATIENT PLACEMENT

- Private room; door may be left open
- If a private room is not available, place the patient in a room with a patient having active infection with the same disease, but with no other infection
- If neither option is available, maintain separation of at least 3 feet between patients

RESPIRATORY PROTECTION

- Wear a mask if within 3 feet of patient

PATIENT TRANSPORT

- Limit transport of the patient to essential purposes only
- During transport, the patient must wear surgical mask
- Notify the area receiving the patient

Adapted from: ETNA Communications 2000.

Contact Precautions

Contact precautions reduce the risk of transmission of organisms from an infected or colonized patient through direct or indirect contact. They are indicated for patients infected or colonized with enteric pathogens. Examples include herpes simplex and haemorrhagic fever virus (Ebola) and multi-drug (antibiotic) resistant bacteria.

Table 17: Contact Precautions

Use in addition to Standard Precautions for a patient known or suspected to be infected or colonized with microorganisms transmitted by direct contact with the patient or indirect contact with environmental surfaces or patient care items.

PATIENT PLACEMENT

- Private room; door may be left open
- If private room not available, place patient in room with patient having active infection with the same microorganism, but with no other infection

GLOVING

- Wear clean, non-sterile examination gloves when entering room
- Change gloves after contact with infective material (e.g., fecal materials or wound drainage)
- Remove gloves before leaving patient room

HANDWASHING

- Wash hands with antibacterial agent or use a waterless, alcohol-based antiseptic hand rub after removing gloves
- Do not touch potentially contaminated surfaces or items before leaving the room

GOWNS AND PROTECTIVE APPAREL

- Wear clean, non-sterile gown when entering patient room if you anticipate contact with patient or if the patient is incontinent, has diarrhea, an ileostomy, colostomy or wound drainage not contained by a dressing
- Remove gown before leaving room. Do not allow clothing to contact potentially contaminated surfaces or items before leaving the room

PATIENT TRANSPORT

- Limit transport of patient to essential purposes only
- During transport, ensure precautions are maintained to minimize risk of transmission of organisms

PATIENT CARE EQUIPMENT

- Reserve non-critical patient care equipment for use with a single patient, if possible
- Clean and disinfect any equipment shared among infected and non-infected patients

Adapted from: ETNA Communications 2000.

18.0 PREVENTING MATERNAL AND NEWBORN INFECTIONS

Pregnant women in developing countries are at a much higher risk for acquiring nosocomial infections following delivery than their counterparts in developed countries.

The rate of postoperative infection after cesarean section is high (15 – 60%). With the emergence of HIV, up to 12% of pregnant women have been found to be HIV sero positive. As a result pregnant women in developing countries are at a higher risk for acquiring nosocomial infections following delivery.

Other than maternal tetanus toxoid immunization during pregnancy, and treatment to prevent congenital syphilis, few other preventive measures to protect the fetus and newborn are routinely available. For example, with the exception of prenatal HIV testing and antiretroviral treatment in a few countries, screening and treatment for infectious diseases (e.g., gonorrhea and Chlamydia) are not available because of the cost and lack of laboratory capability.

18.1 DEFINITIONS

Endometritis. Acute postpartum infection of the lining (endometrium) of the uterus with extension into the smooth muscle wall (myometrium). Clinical features include fever, usually developing on the first or second postpartum day, uterine tenderness, lower abdominal pain and foul-smelling vaginal discharge (lochia).

Episiotomy. Surgical cut made in the perineum just prior to delivery. The purpose is to facilitate delivery of the presenting part and minimize the risk of injury to the perineal area.

Intra-amniotic infection syndrome (IAIS) also referred to as amnionitis or chorioamnionitis. Acute detectable infection in the uterus and its contents (fetus, placenta and amniotic fluid) during pregnancy. It is usually related to colonization of the uterine cavity with organisms present in the cervix and vagina after prolonged ruptured membranes and obstructed labor.

Invasive group B streptococcal sepsis. Newborn infection characterized by bacteremia, pneumonia, meningitis and death in up to 25% of infants with the infection. It occurs most commonly following IAIS.

Nosocomial infection in newborns. Infection occurring after birth but excluding those infections known to have been transmitted across the placenta such as congenital syphilis, cytomegalovirus, rubella, varicella (chicken pox) and the protozoan parasite, *Toxoplasmosis gondii*.

Nosocomial infection in obstetrical patients. Infection that is neither present nor incubating at the time the patient is admitted to the hospital. Most urinary tract infections and endometritis are nosocomial even though the causative organism may be endogenous (i.e., present in the maternal lower genital tract prior to delivery).

Septic pelvic thrombophlebitis. Thrombosis (blockage) of the deep pelvic veins due to inflammation and blood clots. Predisposing factors include cesarean section after prolonged labor, premature rupture of membranes, difficult delivery (forceps or vacuum extraction), anemia and malnutrition.

18.2 EPIDEMIOLOGY

18.2.1 Maternal Infections

- Cesarean section is the most important factor contributing to both the frequency and severity of postpartum infection. Patients who have cesarean sections are at least 10 times more likely to become infected than patients who deliver vaginally (Minkoff and Schwarz, 1980).

- Predisposing factors for wound infection; Intra-partum factors (factors during delivery)
  Women who:
  - have bacterial vaginosis *(Gardnerella vaginalis)* isolated from the endometrium.
  - have a cesarean section during the second stage of labor

- Maternal factors
  - diabetes mellitus
  - malnutrition
  - immunodeficiency
  - anaemia
  - infection of the fetal membranes (chorioamnionitis) diagnosed prior to delivery (Mead 1993).

- Other obstetrical infections including:
  - nosocomial urinary tract infections
  - episiotomy infections
  - nosocomial pneumonia
  - septicemia
  - breast infection (mastitis) in postpartum nursing women

18.2.1 Fetal and Newborn Infections

Fetal and newborn infections are classified based on whether they were acquired in *utero* (transplacentally), during passage through the birth canal (vertical transmission) or in the neonatal period (i.e., during the first 28 days following birth).

Strictly speaking, only newborn infections acquired during passage through the birth canal or in the neonatal period are considered nosocomial. Determining whether an infection is nosocomial or was present or incubating prior to admission to the hospital is extremely difficult – and often not useful.

18.3 MICROBIOLOGY

18.3.1 Causes of Maternal Infections

Most postpartum infections are caused by endogenous flora – microorganisms that are normally present in the genital tract but usually cause no disease until labor, delivery or postpartum. Nearly 30 types bacteria have been identified as being present in the lower genital tract (vulva, vagina and cervix) at any time (Faro 1990). While some of these, including several fungi, are considered nonpathogenic under most circumstances at least 20, including *E. coli, Staphylococcus aureus, Proteus mirabilis and Klebsiella pneumoniae*, are pathogenic.

18.3.2 Colonization and Infection in Newborns

Most infants are delivered from a sterile environment inside the uterus. During and after birth, however, they are rapidly exposed to numerous microorganisms that colonize their skin, nasopharynx and gastrointestinal tract. Sick newborns, subjected to multiple invasive procedures (e.g., endotracheal tubes or umbilical artery catheters) may be colonized at multiple sites with numerous other organisms, particularly gram-negative bacteria.

The skin of the newborn is a major initial site of bacterial colonization, particularly for *S. aureus*, which is most often acquired from within the nursery rather than from the mother. Any break or cut in the skin provides an opportunity for infection to develop with this pathogenic organism. Therefore, to minimize the risk of infection in the newborn period, all sites must be cared for using aseptic technique.

Although severe infection in a full term infant is uncommon, when it occurs it often is secondary to group *B. streptococci, E. coli, Listeria monocytogenes, Citrobacter diversus,* salmonella, chlamydia, herpes simplex virus (HSV) or enteroviruses. All of these organisms can be transmitted to other infants in the nursery on the hands of hospital staff unless Standard Precautions are strictly followed, especially those for handwashing (or use of antiseptic hand rub) and gloves.

18.4 PREVENTING FETAL AND NEWBORN INFECTIOUS DISEASES

Prevention against most fetal and newborn infectious diseases has been achieved through improved maternal immunization, antenatal treatment of maternal conditions and prophylactic use of medications, e.g., postnatal eye drops to prevent conjunctivitis and, recently, the use of antiretroviral drugs to prevent mother-to-child transmission of HIV.

Reducing the Risk of Maternal and Newborn Infections

To minimize the risk of exposure to HIV and other blood-borne viruses during labor, childbirth and resuscitation of the baby, strict use of Standard Precautions, especially hand washing and use of gloves, face shields and plastic or rubber aprons, is mandatory.

Factors Increasing the Risk of Infection during Labor and Vaginal Childbirth

Vaginal deliveries are associated with a number of factors that increase a woman's risk of endometritis or urinary tract infection. These include:

- prolonged ruptured membranes (more than 18 hours)
- trauma to the birth canal (episiotomy, vaginal or perineal lacerations and urethral tears)
- manual removal of the placenta due to retained placenta or placental fragments
- prolonged labor

18.4.1 Decreasing the Risk of Infection during Vaginal Childbirth

Steps that can be taken to decrease the risk of maternal infection **before** and **during** childbirth include:

Step 1: Make sure the following items are available:

- two pairs of surgical gloves
- pair of clean gloves for washing the perineum
- basin of clean warm water, soap, a face cloth and clean dry towel
- plastic or rubber apron and face shield (or a mask and goggles)
- waterless, alcohol-based antiseptic hand rub or antiseptic solution (e.g., 2%
- chlorhexidine gluconate or 10% povidone-iodine)
- high-level disinfected or sterile blunt scissors (Mayo)
- high-level disinfected or sterile cord clamp or cloth to tie off the cord
- injectable oxytocin (with or without egormetrine)
- sterile syringe and needle
- sterile urinary catheter
- package of gauze squares
- clean basin for the placenta
- clean drape or cloth for wrapping the baby
- second Clean drape or cloth for wrapping the baby
- clean perineal pads
- light source (a flashlight or lamp) if needed
- puncture-resistant sharps container (within arm's reach if possible)
- plastic bucket with a tight fitting lid, filled with 0.5% chlorine solution for decontamination
- plastic bag or a leak-proof, covered waste container for disposal of contaminated waste items.

Prior to Childbirth

Step 2: Wear protective equipment including a plastic or rubber apron and face shield (or a mask and goggles) because splashing of blood and blood-tinged amniotic fluid can be expected.

Step 3: Once the patient is positioned for childbirth, put examination gloves on both hands and wash the perineal area (vulva, perineum, and anal region) with soap and clean water.

- use a downward and backward motion when washing the perineal area so that fecal organisms will not be introduced into the vagina
- clean the anal area last and place the washcloth or towel in a plastic container
- shaving perineal (pubic) hair prior to delivery is discouraged since it increases the risk of infection (Landry and Kilpatrick, 1997)

Step 4: Immerse both gloved hands in 0.5% chlorine solution, remove gloves by inverting, and place them in the plastic bag or leak-proof, covered waste container.

Step 5: Thoroughly wash hands, especially between the fingers, and forearm up to the elbows with soap and clean water and dry with a clean, dry towel or air dry.

Step 6: Put sterile (or HLD) surgical gloves on both hands.

During Childbirth

If resuscitation of the infant is required, use a Dee Le mucus trap or mechanical suction if available to avoid backflow of newborn secretions.

If manual removal of the placenta is required, use elbow length gloves if available, or improvise by using a second pair of fingerless surgical gloves, which should be used to avoid contaminating the forearm with blood.

After Childbirth

Step 7: Before removing gloves, put the placenta in the clean basin and place all waste items (e.g., blood-stained gauze) in the plastic bag or leak-proof, covered waste container.

Step 8: If an episiotomy or vaginal or perineal tears require surgical repair, use forceps to hold suture needle and then place sharps (suture needles) in the puncture-resistant sharps container.

18.5 MINIMIZING THE RISK OF INFECTION DURING CAESAREAN SECTION

Cesarean sections should be performed using the same standards as for any general surgical procedure. In addition the following procedures are observed.

- The surgeon and assistant should wear a face shield (or mask and goggles) and a plastic or rubber apron over their scrub suits because splashing of blood and blood-tinged amniotic fluid can be expected.
- Double gloving is recommended.
- Prophylactic antibiotics should be given before caesarean section when infection is suspected
- The health worker receiving the infant should wash her/his hands and put on clean examination gloves before handling the baby.
- The baby should be placed on a clean towel after being passed to the health worker caring for the infant.

- Change surgical gloves before manually removing the placenta. (If available, use elbow-length surgical gloves or a combination of fingerless gloves and a new pair of surgical gloves.)
- With prolonged ruptured membranes or with documented intra-amniotic infection syndrome (chorioamnionitis), avoid spillage of amniotic fluid into the abdominal cavity.
- Do not explore the peritoneal cavity unless absolutely necessary, and then only after closure of the uterine incision and surgical gloves have been changed.
- To minimize postoperative wound infections:
  - patients should not be shaved prior to surgery (if it is necessary to remove pubic or abdominal hair, clip the hair with scissors just prior to surgery)
  - make sure skin incision is done with a scalpel rather than with electrocautery
  - whenever possible, do not place drains in the subcutaneous layer
  - apply a sterile dressing and care for the wound

18.6 POSTPARTUM CARE OF THE MOTHER FOLLOWING CAESAREAN SECTION

Minimizing the risk of nosocomial infection in mothers during the postpartum period includes the following.

- Ensure urine is flowing and the urine collection system is intact.
- Follow the "Tips for Preventing Infections".
- Remove the catheter as soon as possible within 24 hours.

18.7 POSTNATAL CARE OF THE NEWBORN

Minimizing the risk of nosocomial infection in the newborn involves the following.

- Wash hands before holding or caring for the infant. Alternatively, a waterless, alcohol-based antiseptic hand rub can be used.
- Wear gloves and plastic or rubber apron when handling the infant until blood, meconium or amniotic fluid has been removed from the infant's skin.
- Careful removal of blood and other body fluids using a cotton cloth, not gauze, soaked in warm water followed by drying the skin may minimize the risk of infection.
- Bathing or washing the newborn should be delayed until the baby's temperature has stabilized (usually about 6 hours).
- Cover gowns or masks are not required when handling infants.
- The following general instructions for cord care can be applied:
  - wash hands, or use an antiseptic hand rub, before and after cord care
  - keep the cord stump clean and dry
  - do not cover the cord stump with a dressing or bandage
  - fold the diaper or baby napkin below the cord stump
  - if the cord stump gets soiled or dirty, gently wash it with boiled soapy water, and rinse with boiled water and dry with a clean cloth
  - explain to the mother that if the cord stump becomes red or is draining pus of blood she should bring the baby to a clinic or hospital equipped to care for newborns as soon as possible

18.8 MANAGEMENT OF OUTBREAKS IN THE NURSERY OR NICU

If an epidemic or outbreak of a particular disease such as diarrhea is suspected, the first step is to assess it promptly and carefully to:

- identify the source of the diarrhea (e.g., patients, staff or visitors) and the means of transmission (e.g., contamination via hands of staff, parents or visitors)
- decide on the type of control measures required to prevent the spread of the infection and determine the need for laboratory or epidemiologic studies (if available)

19.0 PREVENTING SURGICAL SITE INFECTIONS

Despite improvements in operating room practices, instrument sterilization methods, better surgical technique and the best efforts of infection prevention practitioners, surgical site infections (SSIs) remain a major cause of nosocomial infections and rates are increasing glob ally (Alvarado 2000).

To reduce the risk of nosocomial SSIs in developing countries, a systematic but realistic approach must be applied with awareness that this risk is influenced by characteristics of the patient, the operation, the healthcare staff and the hospital.

Among surgical patients, SSIs are the most common nosocomial infection, accounting for about a third of all such infections. On average, having an SSI increases a patient's hospital stay by 7-10 days, with organ/space and deep incisional SSIs accounting for the longest stays and highest costs.

Exogenous sources of SSI pathogens are occasionally responsible. These include the following.

- Organisms from members of the surgical team (e.g., hands, nose or other body parts)
- Contaminated surfaces in the operating room, even the air
- Contaminated instruments, surgical gloves or other items used in the surgery

Exogenous organisms are primarily aerobic staphylococci or streptococci species (with the exception of tetanus endospores).

19.1 DEFINITIONS

Surgical site infections (SSI): Either an incisional or organ/space infection occurring within 30 days after an operation or within 1 year if an implant is present.

Superficial SSI: involves only the incised body wall.

Organ/Space SSI: Any part of the body other than the incised body wall parts that were opened or handled during an operation.

19.2 CLASSIFICATION OF SURGICAL WOUNDS

The surgical wound classification system includes four categories.

- **Class 1 – clean.** Uninfected operative wound with no inflammation and in which the respiratory, gastrointestinal (GI), genital and urinary tracts were not entered. Clean wounds are closed at surgery and, if necessary, drained with closed drainage.

- **Class II – clean-contaminated**. Wound in which the respiratory, GI, genital or urinary tract was entered under controlled conditions but without unusual contamination or spillage of contents.

- **Class III – Contaminated.** Open, fresh accidental wound or an operation with a major break(s) in aseptic technique (e.g., open cardiac massage) or gross spillage from the GI tract. Also included are incisions in which acute, non-purulent inflammation is found.

- **Class IV – Dirty or infected.** Old wounds with dead tissue and those that involve existing clinical infection or a perforated bowel, suggesting that the pathogens causing the postoperative infection were present in the wound before the surgery.

19.3 PATHOGENESIS

By the end of an operation, bacteria and other microorganisms contaminate all surgical wounds, but only a small number of patients actually develop a clinical infection. Infection does not develop in most patients because their defense mechanisms effectively eliminate the contaminating organisms at the surgical site. Whether a potential infection occurs depends on several factors. The most important include the following.

- Number of bacteria entering the wound
- Type and virulence (ability to cause infection) of the bacteria
- Host defense mechanisms (e.g., effectiveness of inflammatory response and status of the immune system)
- External factors, such as being in the hospital several days before surgery or the operation lasting more than 4 hours

19.4 PREDISPOSING FACTORS

- **Obesity** increases risk substantially when the subcutaneous abdominal fat layer exceeds 3cm (1.5inches) (Nystorm et al 1987). The risk is increased by the need for a larger incision, decreased circulation to the fat tissue or the technical difficulty of operating through a large fat layer.
- **Infection at another site** may increase the risk of spreading infection through the bloodstream.
- **Immunocompromised patients** (e.g., those with HIV/AIDS, those with chronic corticosteroid use such as occurs with asthma and heavy smokers or users of other tobacco products) are at significantly greater risk of SSIs.

- **Malnutrition** may or may not be a contributing factor. Unfortunately, most studies have not been conducted in developing countries where severe malnutrition is more common.
- **Age, race, socioeconomic status and chronic diseases**, such as diabetes and malignancy, are difficult to assess because they are frequently associated with other factors that independently contribute to risk. For example, age over 70 may be accompanied by decreased defense mechanisms, poor nutrition and anaemia.

  Note: *When possible, the effects of conditions that might complicate surgical recovery should be corrected or stabilized preoperatively.*

Remember: Wash hands or use an antiseptic hand rub, before putting on gloves and after taking them off to avoid exposure to blood and other potentially infected body fluids and to decrease the risk of cross contamination.

19.5 OTHER FACTORS

These factors, coupled with the experience and skill of the surgeon and assistant, are known to reduce the risk of SSIs.

- **Prolonged preoperative hospitalization** exposes patients to hospital flora, including multidrug-resistant organisms. Completing pre-surgical evaluations and correcting underlying conditions before admission to the hospital decreases this risk. Also, performing elective surgery, where feasible, in ambulatory surgery centers rather than acute care hospitals decreases the risk of exposure to hospital flora.
- **Preoperative hair removal** should be avoided if it is unnecessary. If hair must be removed, clip it with scissors just before the surgery. Shaving is a proven risk factor for SSIs (Cruse and Foord, 1980).
- **Wide prepping of the proposed incision site with** antiseptic solution preoperatively helps keep microorganisms from migrating into the wound (breakthrough) if the site towels or drapes become wet during surgery.
- **Good surgical technique** minimizes tissue trauma, controls bleeding, eliminates dead space, removes dead tissue and foreign bodies, uses minimal sutures and maintains adequate blood supply and oxygenation. Specifically, it is important to:
  - handle soft tissue gently to avoid crushing that can result in tissue death (necrosis)
  - use electrocautery sparingly to control bleeding because it leaves behind dead tissue that is more likely to become infected
  - use absorbable sutures whenever possible because permanent sutures, especially silk sutures, reduce the number of bacteria necessary to cause infection (James and MacLeod, 1961)
  - use closed suction drains that exit through a separate stab wound to help prevent accumulation of tissue fluid in the dependent portion of the wound; preventing this is especially important in obese patients and may reduce SSIs (Fry, 2003). (Passive drains, such a Penrose drain, exiting through the bottom of the incision should not be used.)
- **Increased length of surgical procedures** is associated with increased risk of SSIs. It is estimated that the infection rate nearly doubles with each hour of surgery (Cruse and Foord, 1980).

- **Prompt discharge postoperatively,** provided patients are able to return to home care, reduces the risk of infection as well.

  Note: *Putting topical antibiotic ointments on closed skin incisions does not decrease the risk of SSIs (Fry 2003). Healthy tissue growth is damaged when dry gauze is removed; moisten the dry gauze with sterile normal saline before removing it.*

19.6 ANTIBIOTIC PROPHYLAXIS IN SURGEY

The use of antibiotics preoperatively can reduce the rate of infection, particularly wound infections, after certain operations. The benefit, however, must be weighed against the risks of toxic and allergic reactions, the emergence of resistant bacteria, drug interactions, super infection and cost (Nichols, 2001). For example, it is estimated that 5% of patients receiving an antibiotic will have a serious reaction to the drug. In general, antibiotic prophylaxis is recommended only for procedures with high infection rates and those in which the consequences of infection are especially serious.

19.7 GUIDELINES FOR CHOOSING A PROPHYLACTIC ANTIBIOTIC

Ideally the prophylactic drug(s) should be directed against the most likely infecting organisms, but need not kill or inactivate all pathogens.

Due to frequent development of resistance of drugs the following are recommended.

- Use drugs with a moderately long half life.
- Use drugs with broad spectrum activity.
- Avoid the use of drugs for prophylaxis when it is in use for post-op treatment.
- Use drugs according to the sensitivity pattern of the area or health facility.

In most instances, a single intravenous (IV) dose of an antibiotic completed 30 minutes or less before the skin incision provides adequate tissue levels throughout the operation. If surgery is prolonged (more than 4 hours), or if major blood loss occurs or an antibiotic with a short half – life is used, one or more additional doses should be given during the procedure.

20.0 PREVENTING INFECTIONS RELATED TO USE OF INTRAVASCULAR DEVICES

The use of intravascular devices, both venous and arterial, to deliver sterile fluids, medications and nutritional products, as well as for central monitoring of blood pressure and other hemodynamic functions, has dramatically increased during the past decade, creating a large population at risk for local and systemic blood stream infections.

Intravascular devices inserted into the venous or arterial bloodstream bypass the normal skin defense mechanism, and provide a way for microorganisms to enter the bloodstream from:

- the device at the time of insertion
- subsequent contamination of the device or attachments (e.g., tubing connected to the
- blood monitoring apparatus or the fluids being administered)
- pathogens on the skin surrounding the insertion site

20.1 MICROBIOLOGY

Both gram-negative bacteria and staphylococci are primary causes of catheter-related infection; however, with the advent of the HIV/AIDS epidemic, infections with fungi are increasingly being reported (Jarvis and Hughes, 1993). Some microorganisms, especially coagulase-negative *Staphylococcus aureus* and pseudomonas and acinetobacter species, adhere to the fibrin film that forms on the inside of catheters within days after insertion. As a consequence, infection with these organisms is quite common, especially if the infection occurs within 10 days of insertion (Raad et al., 1993). For devices left in place longer than 30 days (e.g., tunneled CVCs), bloodstream infections are more likely due to the contamination of the hub of the catheter, especially if frequent handling of the hub occurs (Schaberg, Culver and Gaynes, 1991).

20.1.1 Risk Factors

Individual related factors

- Burns
- Surgical wounds
- Malnutrition
- Immuno compromises (by HIV/AIDS or chronic corticosteroid treatment)

Person-to-person contact-related factors

- Cross-contamination with other infected areas of the patient's body either by the patient or on the hands of the health worker
- Cross-contamination from another patient via the hands of the health worker
- Cross-contamination from the patient when the health worker comes in contact with the patient's blood during insertion, care of the insertion site or removal of the catheter
- Poor insertion or dressing change technique

Device-related factors

Before insertion:

- Cracks in infusion bottles
- Punctures in plastic containers
- Contaminated infusion fluid or additives
- Leaky IV administration sets with multiple connections
- Non-sterile preparation of intravenous infusion fluid

During use:

- Multiple changes of IV fluid containers while using the same IV administration set
- Multiple injections and irrigations of the system
- Central venous pressure measurement apparatus

20.2 REDUCING THE RISK OF NOSOCOMIAL INFECTIONS WITH INTRAVASCULAR DEVICES

20.2.1 Hand hygiene and gloves

- Wash hands thoroughly with soap and running water before touching any of the IV set components.
- Examination gloves should be put on just before touching the insertion site or the hub of the needle or catheter.
- Wash hands with soap and running water after removing gloves.

20.2.2 Site Care and Dressings

- If the site for inserting the catheter is dirty, wash it with soap and clean water and dry it before applying the skin antiseptic.
- If using povidone-iodine (PVI) as the antiseptic agent, allow it to dry after applying or wait at least 2 minutes before insertion.
- Transparent, adherent dressings allow inspection of the site.
- Dressings can be left in place for up to 72 hours if they are kept dry. (They should be changed immediately if they get wet, soiled or loose).
- Gauze and tape dressings need to be changed if an inspection of the site is necessary.
- The catheter or needle site should be gently palpated daily for tenderness.
- The insertion site should be inspected if the patient develops tenderness or fever without an obvious cause (CDC and HICPAC 1996).

20.3 PERIPHERAL CATHETERS (VENOUS AND ARTERIAL)

20.3.1 Site Selection and Change

- For adults, hand veins are preferred over arm veins, and arm veins over leg and foot veins. (Needles and catheters inserted in leg and foot veins are more likely to cause inflammation at the insertion site, or phlebitis.)
- Changing sites at 72 – 96 hours will reduce phlebitis and local infection. “Cannulas” are preferred over steel needles because they are less apt to perforate the vein with movement.
- If only short-term (less than 48 hours) IV infusion is planned, straight or butterfly needles are less irritating than plastic catheters and have lower rates of infection.
- Because straight and butterfly needles frequently infiltrate, they should not be used with solutions that could cause tissue necrosis.

20.3.2 Central Venous Catheters

Site Care and Dressings

- If the site for inserting the catheter is dirty, wash it with soap and clean water and dry it before applying the skin antiseptic.
- Use 2% chlorhexidine gluconate, 10% PVI or 60 – 90% alcohol for skin preparation. Insertion should be done using full barrier precautions (sterile gloves, gown, mask and site drape) in a procedure area, not at the bedside.

20.3.3 Changing Fluids and Infusion Sets

- Change infusion bottles or plastic bags with parenteral solutions every 24 hours.
- Change infusion bottles or plastic bags with lipid emulsion given alone within 12 hours.
- Infusion (administration) sets should be changed whenever they are damaged and at 72 hours routinely. If the tubing becomes disconnected, wipe the hub of the needle or plastic catheter with 60 – 90% alcohol and connect it to a new infusion set.
- Tubing used to administer blood products or lipid emulsions should be replaced within 24 hours (CDC and HICPAC 1996).

20.4 INSERTION, MAINTENANCE AND REMOVAL OF PERIPHERAL VENOUS LINES

Insertion Procedure for Establishing an Intravenous (IV) Line

Step 1: Make sure all items are available:

- IV solution bag or bottle
- Straight or butterfly needle or plastic catheter (steel needle inserter covered with soft plastic tubing that is left in place after the needle is withdrawn)
- Antiseptic solution (e.g., 2% chlorhexidine, 60 – 90% alcohol or 10% povidone–iodine) and sterile or clean gauze squares (2 x 2 or cotton swabs)
- Surgical tape or transparent dressing
- Clean tourniquet
- Clean arm board
- Towel to place under patient's hand or forearm
- IV pole (drip stand)
- Clean pair of single-use examination gloves
- Basin of clean warm water, soap, face cloth and clean dry towel
- Plastic bag or leak-proof, covered waste container for disposal of contaminated items.

Step 2: Explain the procedure to the patient.

Step 3: Prior to starting the procedure, identify the best vein(s) for inserting IV needle or plastic catheter.

Step 4: If the venipuncture site is dirty, first wash it with soap and clean water and dry with a clean cloth before applying a skin antiseptic.

Step 5: Wash hands with soap and clean water and dry with a clean dry towel.

Step 6: Check the IV solution (bottle or plastic bag) to be sure it is the correct infusion.

Step 7: Open the infusion set and assemble the parts using aseptic technique (e.g., don't touch the ends of tubing).

Step 8: Insert the infusion set into solution bottle or bag.

- Remove the protective cover from the solution bottle or bag without touching the opening.
- Remove the protective cap covering the insertion spike without touching the spike and insert the spike into the stopper of the IV bottle or opening of the IV bag.

Step 9: Fill the infusion tubing.

- Compress the drip chamber and release.
- Remove the protective cover of the IV tubing and release the roller clamp to allow fluid to fill the tubing; close the roller clamp and replace the protective cover. (Check to be sure tubing is clear of air bubbles.)

Step 10: Put clean examination gloves on both hands.

Step 11: Cleanse insertion site with antiseptic solution using a circular motion outward from the insertion site. (If using povidone-iodine, allow it to dry, about 2 minutes, because it releases free iodine, the active antiseptic agent, slowly).

Step 12: Insert the needle or catheter with the bevel up using the dominant hand. Look for blood return in the tubing and carefully advance the needle or butterfly until the hub rests at the venipuncture site. (With catheters, after getting blood return, advance the needle about 1 cm ($^1/_2$ inch), withdraw the inner insertion needle and then advance the plastic catheter to the hub.)

Step 13: While stabilizing the needle or catheter, release the tourniquet and roller clamp to permit a rate of flow sufficient to keep the IV line open.

Step 14: Secure the needle or catheter by placing a narrow piece of tape (1 cm or $^1/_2$ inch) under the hub with the adhesive side up and cross tape it over the hub. Then place a second piece of narrow tape directly across the hub of the needle or catheter.

Step 15: Place a sterile gauze square (2 x 2) over the venipuncture site and secure it with two pieces of tape. (Alternatively, place a transparent dressing over the venipuncture site.)

Step 16: Prior to removing gloves, place any blood-contaminated waste items (cotton or gauze squares) in a plastic bag or leak-proof, covered waste container.

Step 17: Remove gloves by inverting and place them in a plastic bag or waste container.

Step 18: Wash hands or use antiseptic hand rub as above.

Step 19: Secure the wrist or forearm to the arm board by applying two strips of tape directly across wrist or forearm. (To minimize discomfort when removing the arm board, attach a shorter piece of tape to the longer piece, adhesive side to adhesive side, that will cover the wrist or arm.)

Step 20: Adjust the flow rate to the correct number of drips per minute.

20.5 MAINTENANCE OF IV LINES

Step 1: Check the line every 8 hours for phlebitis or evidence of infection.

Step 2: Change the infusion site at 72-96 hours, when practical, to reduce the risk of phlebitis and local infection.

Step 3: The infusion (administration) sets (including the piggybacks) should be changed whenever they are damaged and at 72 hours routinely.

Step 4: If the tubing becomes disconnected, wipe the hub of the needle or the plastic catheter with 60 – 90% alcohol and connect to a new infusion set.

Step 5: Mark the site on the plaster with time and date of insertion of the I.V. line and make sure the site is dry.

Removal Procedure

Step 1: Make sure all items are available:

- Clean pair of examination gloves
- Antiseptic solution (2% chlorhexidene gluconate, 60-90% alcohol or 10% povidone-iodine)
- Gauze squares (2x2) and surgical tape or a sterile, wide (1 inch) band aid
- Puncture-resistant sharps container within arm's reach if a straight or butterfly needle was used
- Plastic bag or leak-proof, covered waste container for disposing of the contaminated items

Step 2: Wash hands with soap and running water.

Step 3: Stop the infusion by closing the roller clamp.

Step 4: Put clean examination gloves on both hands.

Step 5: Remove the dressing and discard it in a plastic bag or leak-proof, covered waste container.

Step 6: Check the patient's hand or wrist for phlebitis or evidence of an infection (an area of swelling, redness, warmth and tenderness of the skin around the site where the intravascular catheter comes out of the skin, the exit site). If phlebitis is associated with other signs of infection, such as fever or pus coming from the exit site, it is classified as a clinical exit site infection.

Step 7: Carefully remove the needle or the plastic catheter with one hand and with the other hand cover the insertion site with a sterile gauze square (2x2).

Step 8: Press firmly for about a minute, or alternatively place two pieces of narrow tape, about 1cm or $^{1}/_{2}$ inch wide, directly across the gauze square. Or alternatively, after pressing on the gauze square, remove it and cover the insertion site with a sterile band-aid.

Step 9: Prior to removing gloves, discard the needle or plastic catheter in a sharps container and place the IV tubing and any blood-contaminated waste items (cotton/gauze square) in a plastic bag or leak-proof covered waste container.

Step 10: Remove gloves by inverting and place them either in a plastic bag or a leak- proof, covered waste container.

Step 11: Wash hands with soap and running water.

20.6 INJECTION SAFETY - BEST PRACTICES

A safe injection does not harm the client or patient, does not expose the healthcare provider to any avoidable risk and does not result in any waste that is dangerous for other people.

Eliminating unnecessary injections is the highest priority towards preventing injection-associated infections. When injections are medically indicated they should be administered safely. The following best practices are measures that have been determined through scientific evidence or expert consensus to effectively protect patients, healthcare providers and communities.

20.6.1 Use Sterile Injection Equipment

- Use a sterile syringe and needle for each injection and to reconstitute each unit of medication.
- Use new, quality-controlled disposable syringes and needles.
- Inspect packaging for breaches in barrier integrity, and discard a needle or syringe if the package has been punctured, torn or damaged by exposure to moisture.
- Use single-use syringes and needles.

20.6.2 Prevent Contamination of Injection Equipment and Medication

- Prepare each injection in a clean designated area where blood or body fluid contamination is unlikely.
- Use single dose vials rather than multi-dose vials.
- If multi-dose vials must be used, always pierce the septum with a sterile needle. Avoid leaving a needle in place in the stopper of the vial.
- Select pop-open ampoules rather than ampoules that require the use of a metal file to open them.
- If using an ampoule that requires a metal file to open, protect fingers with a clean barrier (e.g., small gauze pad) when opening the ampoule.
- Inspect medications for visible contamination or breaches of integrity (e.g., cracks, leaks); if any are found, discard the medication.
- Discard a needle that has touched any non-sterile surface.

20.6.3 Prevent Needle-Stick Injuries to Healthcare Providers

- Anticipate and take measures to prevent sudden patient movement during and after injection.

- Avoid recapping and other hand manipulations of needles.
- Collect used syringes and needles at a point of use in a sharps container that is puncture and leak-proof and can be sealed when $^{3}/_{4}$ full.

20.6.4 Prevent Access to Used Needles

- Seal sharps containers for transport to a secure area in preparation for disposal. After closing and sealing sharps containers, do not open, empty, reuse, or sell them.
- Manage sharps waste in an efficient, safe, environment-friendly way to protect people from voluntary and accidental exposure to used injection equipment.

20.6.5 Other Practice Issues

- **Provider's hand hygiene and skin integrity.**
  Perform hand hygiene (wash or disinfect hands) prior to preparing injection material, and after injections. The need for hand hygiene between each injection will vary based on blood or body fluids. Avoid giving injections if skin integrity is compromised by local injection or skin condition (e.g., weeping dermatitis, skin lesions, and cuts). Cover any small cuts.

- **Gloves**
  Gloves are not needed for injections. Single-use gloves may be indicated if excessive bleeding is anticipated.

- **Swabbing of vial tops or ampoules**
  Swabbing of vial tops or ampoules with antiseptic or disinfectant is not necessary. If swabbing with an antiseptic is done, use a clean, single-use swab and maintain product-specific recommended contact time. Do not use cotton balls stored wet in a multi-use container.

- **Skin preparation prior to injection**
  Wash skin that is visibly soiled or dirty. Swabbing of the clean skin prior to giving an injection is not necessary. If swabbing with an antiseptic is done, use a clean, single- use swab and maintain product-specific recommended contact time. Do not use cotton balls stored wet in a multi-use container.

- **Engineered technology**
  Whenever possible, use devices designed to prevent needle-stick injuries that have been shown to be effective for patients and healthcare providers. Auto-disable (AD) syringes should be used to prevent reuse of injection equipment including immunization services.

21.0 PREVENTING NOSOCOMIAL PNEUMONIA

Pneumonia is a complex infection that is often difficult to distinguish from other lung diseases, especially adult respiratory distress syndrome, bronchitis, emphysema and congestive heart failure. Most commonly accepted criteria for nosocomial pneumonia include fever, cough, decreased breath sounds or dullness in a specific area of the lungs and

production of purulent (infected) sputum in combination with X-ray evidence suggestive of an infection.

Nosocomial pneumonia is the infection most likely to be fatal and is the most expensive to treat.

Most nosocomial pneumonias occur by aspiration of bacteria growing in the back of the throat (oropharynx) or stomach. Intubation and mechanical ventilation greatly increase the risk of infection because they:

- block the normal body defense mechanisms – coughing, sneezing and the gag reflex
- prevent the washing action of the hair (cilia) and mucus-secreting cells lining the upper respiratory system
- provide a direct pathway for microorganisms to get into the lungs

Other procedures that may increase the risk of infection include oxygen therapy, intermittent positive pressure breathing (IPPB) treatment and endotracheal suctioning.

Most nosocomial pneumonias occur after surgery, especially if mechanical ventilation is needed postoperatively, and most reported cases of nosocomial pneumonia are due to bacteria. The combination of severe illness, the presence of multiple invasive devices (IVs, urinary catheters and mechanical ventilators) and frequent contact with the hands of personnel often leads to cross-contamination.

21.1 RISK FACTORS

Many risk factors for nosocomial pneumonias are not alterable, e.g., age over 70, chronic lung disease, severe head injuries with loss of consciousness, other serious medical conditions, such as end-stage renal disease, cirrhosis, cigarette smoking, alcoholism, obesity, major cardiovascular or pulmonary surgery and patients with endotracheal tubes or on venti lators. Although it is impossible to change these risk factors, knowing about them is valuable in terms of anticipating problems and limiting the use of invasive devices (e.g., intravenous lines and urinary catheters) as much as possible.

21.2 REDUCING THE RISK OF NOSOCOMIAL PNEUMONIA

Pre-operative Pulmonary Care

Healthcare workers should:

- limit the use of narcotics (for a short duration)
- prevent colonization and infection with new organisms
- prevent transfer organisms among hospitalized patients
- prevent cross-contamination from healthcare staff to patients
- teach patients about
  - deep breathing, moving in bed and frequent coughing
  - early ambulation

Minimizing contamination of respiratory therapy equipment

To minimize cross-contamination when suctioning patients on ventilators:

- wash hands or use an alcohol-based antiseptic hand rub before putting on gloves
- wear clean examination gloves and a protective face shield or mask
- remove gloves immediately after therapy and discard them in a plastic bag or leak- proof, covered waste container
- suction catheters should be decontaminated, cleaned and high-level disinfected by boiling or steaming

Note: *Mechanical ventilation should be used only when necessary and only for as long as necessary.*

To reduce the risk of contamination and possible infection from mechanical respirators and other equipment:

- drain and discard any fluid in the tubing, taking care not to allow the fluid to drain toward the patient
- use small nebulizer bulbs because they produce aerosols that can penetrate deep into the lungs (large volume nebulizers are associated with gram-negative pneumonia and they should not be used)
- decontaminate, clean and high-level disinfect breathing circuits by steaming or soaking in a chemical high-level disinfectant
- reprocess resuscitation devices, such as Ambu bags, promptly

21.3 PREVENTING GASTRIC REFLUX

- Avoid prolonged use of nasal gastric tubes for feeding.
- Feed small, frequent amounts rather than large amounts.
- Raise the patient's head on the bed so that the patient in a semi-sitting position.

21.4 POSTOPERATIVE MANAGEMENT

Surgical units should have effective plans for:

- optimizing the use of pain medication to keep the patient comfortable enough to cough effectively
- regularly moving and exercising patients
- encouraging deep breathing in the immediate postoperative period and for the next few days

22.0 PREVENTING URINARY TRACT INFECTIONS

Urinary tract infections (UTIs) are the most common type of nosocomial infections, accounting for 40% of all infections in hospitals per year (Burke and Zavasky, 1999). In addition, several studies have reported that about 80% of nosocomial UTIs occur following

instrumentation, primarily catheterization (Asher, Oliver and Fry, 1986). Because nearly 10% of all hospitalized patients are catheterized, preventing UTIs is a major factor in decreasing nosocomial infections.

Organisms attacking any portion of the urinary system cause urinary tract infections: the kidneys (pyelonephritis), bladder (cystitis), prostate (prostatitis), urethra (urethritis) or urine (bacteriuria). Once bacteria infect any site, all other areas are at risk.

22.1 MICROBIOLOGY

Most nosocomial UTIs are caused by gram-negative coliform bacteria, particularly *Escherichia coli*, pseudomonas species, and organisms from the enterobacter group. Collectively they account for more than 80% of culture-positive UTIs (Haley et al., 1985). While the most common organism is *E. coli*, infections with fungi, such as the candida species, have increased with the advent of HIV/AIDS and widespread use of broad-spectrum antibiotics.

Factors that can lead to bacteriuria and UTIs include:

- passage of organisms from the urine bag to the bladder (retrograde contamination) that occurs in patients with indwelling catheters (i.e., those left in place for several days or weeks)
- ability of some organisms to grow on the outside or inside of the tubing and even in the urine itself

22.2 REDUCING THE RISK OF NOSOCOMIAL URINARY TRACT INFECTIONS

Except for the end of the urethra or penis, the urinary system is normally sterile. The ability to completely empty the bladder is one of the most important ways the body has to keep the urine sterile and prevent UTIs. If the bladder empties completely during the voiding process, bacteria do not have the chance to infect tissue or grow and multiply in the bladder. Therefore, the normal defenses against a UTI are an unobstructed urethra, the voiding process and normal bladder mucosa. The insertion of a catheter, however, bypasses these defenses, introduces microorganisms from the end of the urethra or penis, and provides a pathway for organisms to reach the bladder.

Organisms may reach the bladder in two ways: through the inside of a catheter (i.e., the backward flow of urine) or by traveling up the space between the outer surface of the catheter and the urethral mucosa. Therefore, once the catheter is inserted, any back-and-forth movement of the catheter (e.g., raising the collection bag above the level of the bladder), or allowing urine to be collected in an open drainage system (bag or container) should be avoided because each of these activities potentially enables organisms to enter the bladder. The first way (backward flow of urine in the catheter) is the more common infection in men. The second (organisms migrating into the bladder along the outside of the catheter) is more common in women in part because of their shorter urethra. As a consequence, women are more likely to develop a UTI from organisms located in the vagina (Garibaldi et al., 1980).

Placement of an indwelling catheter should be performed only when other methods of emptying the bladder are not effective, and it is particularly important to limit the duration as much as possible.

Other methods for management of urinary tract problems include: intermittent catheterization using a sterile straight catheter, condom catheters for male patients, adult diaper pads, bladder retraining and the use of drugs to stimulate urination. Loss of control (incontinence) or inability to void (retention) may be managed better by straight (in and out) catheterization several times daily rather than by putting in an indwelling catheter.

22.3 PROCEDURES FOR INSERTION, REMOVAL, AND/OR REPLACEMENT OF URINARY CATHETERS

Before inserting a catheter, check to be sure that it is being inserted for the right reason. For example, if a catheter is being inserted because of urinary retention, ask the patient if she/he has voided, the time of voiding, and measure the height of the bladder. Also, before removing a catheter, check to be sure the doctor's orders are correct to avoid an error.

Indications for Catheterization

The indications for catheterization include but are not limited to the following:

- for short-term (days) management of incontinence (the ability to control urination) or retention (the inability to pass urine) not helped by other methods
- to measure urine output over several days in critically ill patients
- to instill medications
- for treatment of urinary outlet obstruction (blockage of the tube leading from the bladder to the outside, the urethra)
- for postoperative management of surgical patients with impaired bladder function (the most common routine use)

Insertion Procedure:

Step 1: Make sure that all of the following items are available.

- A sterile indwelling urinary catheter with a closed continuous drainage system, or a high-level disinfected or sterile straight catheter and a clean urine collection container
- Sterile syringe filled with boiled or sterile water for blowing up the balloon of an indwelling catheter
- Pair of sterile gloves
- Antiseptic solution (2% chlorhexidene gluconate or 10% povidone-iodine)
- Sponge holding forceps with gauze squares (2x2) or large cotton applicators
- Single-use packet of lubricant
- Light source (flashlight or lamp) if needed
- Basin of clean warm water, soap, a face cloth and paper towel
- Plastic bag or leak-proof covered waste container for disposal of contaminated items

Step 2: Prior to starting the procedure:

- have women separate their labia and gently wash the urethral area and inner labia
- have men retract their foreskin and gently wash the head of the penis and foreskin

Step 3: Wash hands with soap and clean water and dry with a clean dry paper towel. Apply about 1 teaspoonful, of a waterless, alcohol-based antiseptic hand rub to both hands and vigorously rub the hands and between the fingers until dry.

Step 4: Put surgical gloves on both hands.

Step 5: Use a small a catheter as consistent with good drainage.

Step 6: For health workers who are right-handed (dominant hand) stand on the patient's right side (and on the left side if left-handed).

Step 7: For women: separate and hold the labia apart with the non-dominant hand and prep the urethral area two times with an antiseptic solution using either cotton applicators or sponge forceps with gauze squares.

Step 8: For men: push the foreskin and hold the head of the penis with the non-dominant hand; then prep the head of the penis and urethral opening two times with an antiseptic solution, using cotton applicators or a sponge forceps with gauze squares.

Step 9: If inserting a straight catheter, grasp the catheter about 5cm (2 inches) from the catheter tip with the dominant hand and place the other end in the urine collection container.

Step 10: For women, gently insert the catheter until urine flows. For children insert only about 3cm (1.5inches).

Step 11: For men, gently insert the catheter about 18-22cm (7-9 inches) or until urine flows. For children insert only about 5-8cm (2-3 inches).

Step 12: If inserting an indwelling catheter, push another 5cm (2inches) after urine appears and connect to the urine collection tubing if not using a closed system.

Step 13: For an indwelling catheter: inflate the balloon, pull out gently to feel resistance and secure the indwelling catheter properly to the thigh or lower abdomen.

Step 14: For straight (in and out) catheterization, allow the urine to slowly drain into the collection container and then gently remove the catheter.

Step 15: Place soiled items, including the straight catheter if it is to be disposed of, in a plastic bag or leak- proof, covered waste container.

Step 16: Alternatively, if a straight catheter is to be reused, place it in 0.5% chlorine solution and soak it for 10 minutes for decontamination.

Step 17: Remove gloves by inverting and place them either in a plastic bag or waste container.

Step 18: Wash hands or use an antiseptic hand rub as above.

Removal and or Replacement

Step 1: Make sure all items are available (as step 1 above if replacing an indwelling catheter).

- Pair of examination gloves
- Empty sterile syringe for removing the fluid from the catheter balloon
- Sponge holding forceps with gauze square (2x2) or large cotton applicators
- Plastic bag or leak-proof, covered waste container for disposal of contaminated items

Step 2: Have the patient wash the urethral area (women) or the head of the penis (men), or do it for them wearing a pair of clean examination gloves.

Step 3: Wash hands with liquid soap and running water.

Step 4: Put clean single examination gloves on both hands.

Step 5: With the empty syringe, remove the water from the catheter balloon.

Step 6: For women, separate and hold the labia apart with the non-dominant hand, then prepare the urethral area twice with antiseptic solution using cotton applicators or a sponge forceps with gauze squares, and gently remove the catheter.

Step 7: For men, push back the foreskin and hold the head of the penis with the non-dominant hand, then prepare the head of the penis and the area around the catheter two times with an antiseptic solution, using cotton applicators or sponge holding forceps with gauze squares and gently remove the catheter.

Step 8: If you are just removing the catheter, then follow steps 15,17 and 18 of the Insertion Procedure.

Step 9: If you are replacing the indwelling catheter, follow steps 4 through 18 of the Insertion Procedure.

22.3.1 Tips for Preventing Infections in Catheterized Patients

- Remove the catheter as soon as possible within 24 hours.
- The catheter collection system should remain closed and not be opened unless absolutely necessary for diagnostic or therapeutic reasons.

- Caution the patient against pulling on the catheter.
- Urine flow through the catheter should be checked several times a day to ensure that the catheter is not blocked.
- Avoid raising the collection bag above the level of the bladder.
- If it becomes necessary to raise the bag above the level of the patient's bladder during transfer of the patient to a bed or stretcher, clamp the tubing.
- Before the patient stands up, drain all urine from the tubing into the bag.
- The urine drainage (collection) bags should be emptied aseptically; touching the tip of the emptying tube to the side of the collection bag or permitting the tip to touch the urine in the vessel should be avoided. Replace bags with new or clean containers when needed.
- If the drainage tubing becomes disconnected, do not touch the ends of the catheter or tubing. Wipe the ends of the catheter and tubing with an antiseptic solution before reconnecting them.
- Wash the head of the penis and urethral opening (men) or the tissue around the urethral opening (women) after a bowel movement or if the patient is incontinent.
- If frequent irrigation is required, the catheter should be changed.
- Never re-use disposable catheter materials.

23.0 PREVENTING NOSOCOMIAL DIARRHEA

Controlling the spread of nosocomial diarrhea from contaminated food is an ongoing concern in hospitals and nursing homes. Frequently this is due to poorly trained food- handling staff using unsafe practices involving the storage, preparation and handling of raw meat, chicken, fish, fresh eggs and vegetables.

23.1 DEFINITION

Nosocomial diarrhea: On at least 2 consecutive days having at least three loose or watery stools with the onset more than 72 hours after admission to the hospital (or more days than the incubation period if the agent is known).

23.2 CAUSES OF DIARRHEA

Outbreaks of diarrhea in hospitals, nursing homes and NICUs have been associated with a wide variety of organisms including salmonella, shigella, *Clostridium difficile,* vibrio (cholera), *Candida albicans*, *Staphylococcus aureus,* cryptosporidium, rotavirus and other enteroviruses.

Some of the most common bacterial and viral agents causing infectious diarrhea, their incubation period and most prominent clinical characteristics are listed below.

23.3 COMMON AGENTS

- **Salmonella:** Salmonellosis is a common cause of diarrhea secondary to food poisoning. The incubation period is less than 72 hours (3 days) when large doses of organisms are eaten in contaminated food or drinks.

- Rotavirus: Is the causative agent of sudden onset of vomiting and diarrhea within 2-3 days after exposure. It is the commonest cause of diarrhea in children under five years. The virus may be present in sputum or other secretions and survives well on inanimate surfaces. It may become endemic in hospitals.

23.4 RISK FACTORS

Risk factors for nosocomial diarrhea include the following.

- Old age
- Patients with burns
- Trauma
- Decreased immunity
- Decreased gastric acidity
- Altered flora in the stomach and gut occurring with antibiotic treatment
- Lack of hand hygiene, especially by food handlers
- Non-compliance with glove use

23.5 IMPORTANT CONSIDERATIONS

For staff in diarrhea wards

- Clean and wipe bedpans and bathroom equipment that are regularly handled by patients and staff with a disinfectant (0.5% chlorine solution or 1% Lysol) daily and whenever they have been used.
- Immediately disinfect and clean all soiled articles if soilage occurs.
- Staff who sort linen should wear utility or heavy-duty gloves. Also, soiled linen should be bundled so that leakage does not occur, and all linen should be handled as if fecal contamination were present.
- Wear gloves when handling linen soiled with moist body substances, used diapers or toilet paper, and place in a plastic bag or leak-proof, covered waster container.

For food service personnel

Food handlers with diarrhea should be immediately removed from handling foods. They should not return to food handling or work with immuno-compromised patients or intensive care patients or patients undergoing transplant until all symptoms are over for 24-48 hours.

For patients with diarrhea

- Patients with diarrhea from any cause should be managed according to Standard Precautions with Transmission-Based Precautions.
- Other precautions include moving roommates to another room in the hospital if fecal contamination is likely.
- Infants born to mothers with diarrhea should not enter the regular nursery. In addition, rooming-in should be provided for mother and infant. The mother and other caretakers should be taught good hygiene.

23.6 OUTBREAK MANAGEMENT

Refer to integrated disease surveillance and response guidelines (IDSR)

24.0 HEALTH LABORATORY

Health laboratory personnel, especially the staff of microbiology, work with infectious organisms and materials that do or may contain microorganisms. Some of these organisms are pathogenic and potentially dangerous.

Avoidance of infection is thus an essential element of the professional expertise of the workers. It is necessary to protect not only themselves but also to protect their materials from possible cross-contamination that may invalidate their work by giving false results. Health laboratory workers are also at risk of chemical, fire and radiation hazards.

The World Health Organization has given guidance regarding hazards of infective microorganisms by risk group, and thus laboratories are designated by level according to their design features, construction and containment facilities (safety precautions and equipment) as Basic Biosafety Level 1, Basic Biosafety Level 2, Containment – Biosafety Level 3 and Maximum Containment – Biosafety Level 4. Depending on the laboratory level, the health worker is referred to the WHO safety guidelines (Biosafety Level (BSL) guidelines).

24.1 DEFINITIONS

Biological safety cabinets (BSCs) are devices that provide protection for personnel, the agent being processed and the environment. They range in complexity from Level I (general research cabinets for use with low- to moderate-risk microorganisms) to Level III (totally enclosed cabinets with gas-tight construction that provide maximum protection to workers and the environment).

Laboratory-acquired infections are nosocomial infections resulting from the performance of laboratory activities by staff, regardless of how they occurred.

24.2 TYPES OF EXPOSURE RESULTING IN LABORATORY ACQUIRED INFECTIONS

Inhalation. Mixing, grinding or blending an infectious agent or flaming a transfer loop can generate aerosols that can be inhaled by unprotected workers.

Ingestion. Workers may be exposed through:

- unconscious hand-to-mouth actions
- placing contaminated articles (pencils) or fingers (when biting fingernails) in the mouth
- eating, drinking or smoking in the laboratory or failing to use proper hand hygiene (neglecting to wash hands or to use a waterless, alcohol-based antiseptic hand rub before and after eating)

- pipetting (13% of accidental laboratory-acquired infections are associated with mouth pipetting)

Puncture wounds. Accidental injury with sharps (suture needles, scalpel blades and contaminated broken glassware) is the leading cause of laboratory-acquired infections.

Contamination of skin and mucous membranes. Splashes and sprays of contaminated fluids onto mucous membranes of the mouth, nasal cavity and conjunctivae of the eyes, and hand-to-face actions can lead to the transmission of pathogenic organisms.

24.3 BIOSAFETY LEVEL (BSL) GUIDELINES.

A combination of primary and secondary containment and safety guidelines are designed for use in microbiology laboratories and bacteriology research units functioning at four levels (BSL-1 to BSL-4) of increasing risk.

- **BSL-1** is the lowest level of containment and microbiologic safety guidelines and is entirely based on standard laboratory practices. These guidelines are recommended for those working with microorganisms, such as *Bacillus subtilis*, that are not known to cause infections in healthy adults.

- **BSL-2** is generally applied in bacteriology laboratories working with agents (e.g., *Samonella species)* associated with human diseases of varying severity. When standard microbiologic practices are applied, the agents may be handled on open benches, especially if personal protective equipment, such as facemasks, gowns and examination gloves are used when appropriate. The use of biologic safety cabinets and safety centrifuges may be necessary.

- **BSL-3** is aimed at containing hazardous microorganisms primarily transmitted by the airborne route (aerosols and droplets), such as tuberculosis or varicella (chicken pox). Laboratory staff who work in these situations must be trained in the use of appropriate equipment, including suitable ventilation systems and the use of BSCs.

- **BSL-4** is designed for use where agents causing life-threatening or untreatable diseases that can affect the laboratory worker via the airborne route are present, such as hemorrhagic fever viruses. Trained workers using Level III BSCs or wearing full-body, air-supported positive pressure suits must perform all procedures in these laboratories. In addition, the facility itself must be totally isolated from other laboratories and have specialized ventilation and waste management systems.

24.3.1 General Biosafety and Infection Prevention Guidelines

- Wear new examination gloves when handling blood, body fluids and/or specimens containing pathogenic microorganisms, and do not touch telephones, pens, lockers, etc., with gloves on.
- Eating, drinking or smoking is prohibited in the laboratory.
- Food should not be stored in refrigerators used for clinical or research specimens.

- Mouth pipetting is prohibited; use proper mechanical devices (e.g., suction bulbs).
- Do not open centrifuges while still in motion.
- Always cover the end of blood collection tubes with a cloth or paper towel, or point them away from anyone's face when opening.
- Decontaminate work surfaces daily or when contaminated, such as after spills, with 0.5% chlorine solution.
- Wear protective face shields or masks and goggles if splashes of blood, body fluids, or fluids containing infectious agents are possible.
- Wear heavy-duty or utility gloves when cleaning laboratory glassware.
- Use puncture-resistant, leak-proof containers for sharps.
- Place infectious waste materials in appropriate plastic bags or containers.
- Wear a laboratory coat while in the laboratory and remove it when leaving the laboratory (coats should not be worn in non-laboratory areas such as offices, libraries canteens, etc.).
- Secure the lid of the specimen container tightly.
- Label the specimen clearly with name, date, time of collection and type of specimen at the site of collection.
- The laboratory should be kept neat clean and free of materials that are not pertinent to the work.

24.4 BLOOD DRAWING (PHLEBOTOMY)

Blood drawing (phlebotomy) is considered to be one of the highest-risk sharps procedures. This is because the most frequently used needles are large bore (18 to 22 gauge), and a considerable amount of blood is left in the needle after use.

When collecting a blood specimen (phlebotomy) be sure to:

- wear single use examination gloves
- have assistance when patients might be uncooperative (children, mentally impaired, etc.)
- have assistance for holding children when doing heel sticks

25.0 BLOOD BANKS AND TRANSFUSION SERVICES

Blood banks and transfusion services collect, process, store and provide human blood intended for transfusion, perform pre-transfusion testing and, finally, infusion into a patient. The transfusion service, in turn, is responsible for maintaining an adequate supply of needed blood and blood products, blood-typing and cross matching patients, and releasing the blood for transfusion.

Once the blood is collected, contamination can be avoided by:

- maintaining appropriate storage conditions
- testing the blood unit without entering the closed collection system
- infusing or discarding the blood unit within a short period once the closed system has been opened

In this section, the guidelines for the safe provision of blood bank and transfusion services are summarized from the perspective of:

- screening the blood donor
- ensuring the safety of the donor
- testing to make sure the blood or blood product is safe for use
- protecting the patient receiving the transfusion
- ensuring the safety of laboratory and clinical staff

25.1 DEFINITIONS

Blood bank. Facility or hospital unit that performs the collection, processing, storage and distribution of human blood or blood products.

Clinically significant antibody. An antibody capable of producing an adverse reaction to transfused blood or blood product obtained from a donor (allogenic antibody) or recipient (autologous antibody).

Closed system for obtaining blood. System in which the blood is not exposed to air or outside elements during collection and processing, including separation of components (e.g., platelets) if required prior to transfusion. It is the safest way to collect, process and store blood.

Donor-Patient. Person whose blood is collected for possible transfusion to another person (allogenic transfusion).

Donor-Recipient. Person whose own blood is collected for possible transfusion to herself/himself (autologous transfusion).

Look-back system. Process of identifying persons who have received a blood transfusion from donors who are subsequently found to have infections with HCV, HIV (and often HBV), and notifying them if appropriate.

Recipient transfusion reaction. Adverse reaction to infusing blood or blood products into a patient (recipient). It may occur at any time during the transfusion but often happens shortly after starting it. The reaction may be mild or severe and is rarely fatal.

Transfusion service. Facility or hospital unit that provides storage, pre-transfusion testing and cross matching, and infusion of blood or blood products to intended patients (recipients).

Unit of blood. Sterile plastic bag in which a fixed volume of blood is collected in a suitable amount of anticoagulant. (The collection system should be a closed system, usually consisting of a sterile hypodermic needle connected by tubing to a collection bag that has one or two sterile ports for inserting a sterile blood administration set).

Urticarial reaction. Allergic reaction consisting of itching (pruritis), hives, skin rash and/or similar allergic condition occurring during or following a transfusion blood products.

25.2 INDICATIONS FOR BLOOD TRANSFUSION

- Acute severe anaemia
- Massive blood loss
- Blood disorders

25.3 PROVISION OF BLOOD TRANFUSION SERVICES

Blood bank and transfusion services involve selecting donors; assuring that they are informed; collecting blood from screened donors; testing for blood components, antibodies and infectious diseases; storing and transporting blood; pre-transfusion testing of patient (recipient) blood; and transfusing patients.

25.3.1 Donor Selection

The donor selection process is one of the most important steps in protecting the safety of the blood supply. It is intended to identify medical problems, behaviors (e.g., intravenous drug use) or events that put a person at risk of being infected and transmitting a serious disease to the person receiving the transfusion.

25.3.2 Informed Consent

Prior to collection of blood, the elements of the donation process should be explained in simple, easily understandable terms using the patient's primary language if possible. The explanation should include information about the risks of venipuncture (phlebitis or local infection and rarely bacteremia or septicemia) and potential adverse responses to having 400 – 500 mL of blood removed.

A donor who wants to know his/her HIV status should be informed with appropriate pre- and post-test counselling.

25.3.3 Blood Storage and Transportation

Blood units must be stored in a refrigerator that can be maintained at temperatures between 1-6°C (34-46°F). There must be a system to monitor temperatures continuously and record them at least every 4 hours. In addition, the refrigerator should have an alarm system that signals by sound before the blood reaches unacceptable storage temperatures.

Blood units exposed to a temperature above the accepted level for an unknown period should be discarded. To do this:

- wear examination or utility gloves and protective eyewear
- pour contents down a utility sink drain, into a flushable toilet or latrine
- place empty blood bags and tubing in a plastic bag or leak-proof, covered waste container
- dispose of plastic bags or contents of the container according to hospital or facility or facility waste management guidelines

Blood units transported a short distance (e.g., from the blood bank or transfusion service to the ward or operating room) require no special handling. Blood should not, however, be allowed to reach temperatures outside the acceptable range.

25.4 BLOOD COMPONENTS AND INFECTIOUS DISEASE TESTING

ABO blood group is determined by testing the donor's red cells with anti-A and anti-B reagents and by testing the donor's serum or plasma A and B red cells.

Rh type is determined by testing with anti-D reagent. If the initial test with anti-D is negative, the blood also should be tested using a method designed to detect weak D.

Blood from donors with a history of transfusions or pregnancy should be tested for unexpected antibodies to red cell antibodies using methods to demonstrate clinically significant antibodies.

Note: In addition, blood should be tested for several infectious diseases. Blood should not be released for transfusion unless the results are negative for all tests, with the exception of the test for syphilis that has been shown to be a biologic false positive.

The recommended tests include:

- Syphilis by screening with a standard antibody test such as the rapid plasma reagin (RPR) test

- Hepatitis B virus by testing for the hepatitis B surface antigen (HbsAg) and HBV core antigen (anti-HBc)
- Hepatitis C virus by testing for anti-HCV
- Human immunodeficiency virus by testing for type 1 (HIV-1) antigen and antibodies to HIV-1 and HIV-2 antigens
- Malaria parasites

PART V

INFECTION PREVENTION AND CONTROL MANAGEMENT

26.0 INFECTION PREVENTION AND CONTROL — PROGRAM MANAGEMENT AND MONITORING (SURVEILLANCE)

Successful programs for preventing the spread of infectious diseases by any route (blood, body fluids, air, droplet or contact) in healthcare facilities are based on understanding the scope of the problem, prioritizing activities and effectively using available resources. Careful planning, implementing and monitoring of activities on a regular basis are all essential.

In Tanzania functioning infection surveillance systems lack laboratory backup to identify the cause of nosocomial (hospital-acquired) infections and treatment options. Thus infection prevention is the most cost-effective option, to limit the spread of disease within healthcare facilities. To implement infection prevention, control and surveillance requires staff at all levels to take an active role in preventing the spread of infections to patients, fellow workers and themselves.

Most nosocomial infections in healthcare facilities can be prevented with available, inexpensive strategies. To make this happen, healthcare facility administrators, managers and staff at all levels must be totally committed to support and use recommended infection prevention guidelines and practices.

26.1 PROGRAM DEVELOPMENT

In order for healthcare facilities to become safer places to work, changing behavior is necessary. It is the responsibility of Infection Prevention and Control teams to:

- set standards and monitor staff performance
- enhance consistent support by all hospital administrators, managers and staff
- regularly provide feedback and reward appropriate behavior
- encourage physicians, and other senior staff and faculty to be role models for recommended infection prevention and control practices

26.2 PROGRAM MANAGEMENT

Organization principles

It is important to identify and bring together key hospital staff to form an infection prevention and control working team. The purpose of the team is to guide and support the use of recommended practices and to review and resolve related problems that may arise. The team should include representatives from a variety of patient care areas such as surgery, central services, housekeeping, laboratory, purchasing and administration.

The responsibilities of the team include:

- conducting an initial assessment
- establishing the relative importance of the problem using Spaulding's categories of potential risk, including critical, semi-critical and non critical
- establishing an infection prevention and control committee coordinated by the infection prevention and control officer
- identifying and analyzing the reasons for poor or incorrect performance and estimating the costs and benefits
- formulating an action plan with costing, budgeting and financing
- developing an IEC strategy for healthcare workers and strengthen supervision.
- conducting staff orientation before new guidelines, recommendations or procedures are started and providing follow-up training when reinforcement is needed
- ensuring continuous availability of supplies and equipment for patient care management
- confirming value through monitoring, providing data and measuring the impact of interventions
- ensuring effective and regular communication and feedback at all levels

26.3 MAKING MANAGEMENT DECISIONS

Throughout these guidelines, evidence is presented to help managers make better informed decisions and recommendations regarding:

- compliance with hand hygiene guidelines
- appropriate selection and use of gloves for various healthcare tasks
- appropriate selection of effective antiseptic agents or chemical disinfectants, ones that are affordable and usually locally available
- consistent use of personal protective equipment (PPE), especially gloves and other items
- design of safer surgical operations
- use of safety checklists for making the operating room safer for patients and staff
- proper waste management particularly difficult problems.

In making these decisions, managers often must strike a balance between the importance of the problem and providing acceptable levels of safety for specific healthcare tasks.

26.4 STAFF TRAINING

Initially, all levels of healthcare workers (e.g., nurses, physicians, housekeepers and cleaners) need to know why infection prevention is important. Key topics to be taught should include:

- the disease transmission cycle, routes of infection and how to break the cycle
- use of Standard Precautions when dealing with all patients
- methods of minimizing disease transmission as well as "hands on" demonstrations about Standard Precautions

To have long-term effects, the initial training should be followed up, and monitoring should be targeted toward identifying and solving specific problems related to introducing new processes or procedures. General reminders regarding the importance of maintaining an infection-free environment for safer delivery of services should be repeatedly emphasized.

26.5 INFECTION MONITORING (SURVEILLANCE)

Keeping records of infections that occur in health facilities is a time-honored way of monitoring the effectiveness of infection prevention practices.

26.5.1 Definition

Surveillance is the systematic collection of relevant data on patient care, the orderly analysis of the data and the prompt reporting of the data to those who need it.

Active surveillance consists of collecting information directly from patients or staff.

Passive surveillance includes examining reports, laboratory information and data from other sources.

26.5.2 Purposes of Surveillance

The purposes of surveillance are to:

- determine baseline rates of nosocomial infections
- evaluate infection control measures (e.g., management of multidrug-resistant infections)
- monitor good patient care practices
- meet the safety standards required by regulatory agencies
- detect outbreaks and exposures

26.5.3 When to Consider Performing Surveillance

Surveillance should begin only after all recommended steps for Standard Precautions have been followed.

26.5.4 Conducting Surveillance

Clinical review of medical records should include collecting basic demographic information (e.g., name, age, date of birth, admission diagnosis), checking for fever, new antibiotic use, and new cases of diarrhea, clinical sepsis or the presence of an inflamed surgical wound, drain or IV site.

Discussions with patients (or parents of newborns in this example) should focus on the their health, the health of other young children at home, general hygiene, food handling and sanitation.

Discussions with staff working in the affected area should deal with ensuring that recommended patient care activities are being performed both correctly and at the appropriate times.

Laboratory information to be checked should include a review of positive cultures and other diagnostic findings if available.

26.5.5 Detecting and Managing Outbreaks

In case of an outbreak, proceed according to the guidelines on outbreaks in Integrated Disease Surveillance and Response Guidelines, Health Sector Guidelines & Protocol on Emergency Disaster Managementand cholera guidelines.

26.5.6 Challenges of Conducting Surveillance

- An assumption that an outbreak exists when it really does not. Where possible, confirm the diagnosis, search for additional cases and determine whether the increase is real before concluding that an outbreak is occurring.
- The isolation of an organism rarely explains an outbreak.
- The presence of organisms from multiple sites or personnel usually suggests that these sites became colonized from another source and were not the cause of the outbreak.
- Negative cultures do not justify concluding that the site (e.g., staff or inanimate objects) was not responsible for the outbreak.
- Prevention measures are not implemented immediately.
- Other similar practices are not evaluated. When a problem with reprocessing instruments or specific patient care practices is identified, often the same faults exist elsewhere in the hospital; all similar situations should be evaluated and corrected as soon as possible.

26.6 ADMINISTRATIVE PRIORITIES IN ALL HEALTHCARE FACILITIES

- Ensure recommended infection prevention practices are adhered to, such as sterilization, or where appropriate HLD, of all items that come in contact with normally sterile tissue.
- Ensure patient care practices are performed according to the Standard Precautions.
- Monitor compliance with recommended practices for certain high-risk procedures, such as inserting central venous catheters.
- Work to eliminate unnecessary and unsafe practices, e.g., unsafe injections.
- Routine surveillance should not outweigh investigating outbreaks, or providing safe water, food and sanitation within the hospital or healthcare facilities.

PRIMARY SOURCES

Reid U. 2001. *Infection Prevention and Control, Policies and Guidelines - Manual*. World Health Organization/Regional Office for Africa, Commonwealth Regional Health Community Secretariat, and East, Central and Southern African College of Nursing.

Tietjen, L, D. Bossemeyer and N. McIntosh. 2003. *Infection Prevention - Guidelines for Healthcare Facilities with Limited Resources*. JHPIEGO Corporation. Baltimore, Maryland.

ADDITIONAL REFERENCES

Alvarado CJ. 2000. The Science of Hand Hygiene: A Self-Study Monograph. U. of Wisconsin Medical School and Sci-Health Communications. March

AORN. 1990. Clinical Issues. AORN *Journal* 52:613-615.

Arnow P et al. 1991. Endemic and epidemic aspergillosis associated with in-hospital replication of Aspergillus organisms. *J Infect Dis* 164(5): 998-1002.

Asher EF, BG Oliver and DE Fry. 1986. Urinary tract infections in the surgical patient. *Am Surg* 54(7): 466-469.

AVSC International (1999). Infection Prevention Curriculum, Teachers Manual. New York

Baumgardner CA et al 1993. Effects of nail polish on microbial grown of fingernails. *AORN J* 58(4): 84-88.

Bessinger CD Jr. 1988. Preventing transmission of human immunodeficiency virus during operations. *Surg Gyn Ob* 167(4): 287-289.

Boyce JM and D Pittet. 2002. Guidelines for hand hygiene in healthcare settings: recommendations of the Healthcare Infection Practices Advisory Committee and the HICPAC/SHSA/APIC/IDSA Hand Hygiene Task Force. *Infect Control Hosp Epidemiol* 23(Suppl): S3-S40. Available at http://www.cdc.gov/handhygiene.

Burke JP and D Zavasky. 1999. Nosocomial urinary tract infections, in *Hospital Epidemiology and Infection Control,* 2nd ed. Mayhall CG (ed). Lippincott, Williams and Wilkins: Philadelphia, 173-187.

Centers for Disease Control (CDC). 1985. Recommendations for preventing transmission of infection with human T-lymphotropic virus type III/lymphadenopathy-associated virus in the workplace. *MMWR* 34(45): 681-686; 691-695.

Centers for Disease Control (CDC). 1989. Guidelines for prevention of transmission of human immunodeficiency virus and hepatitis virus to healthcare and public-safety workers. *MMWR 38(S-6):* 5- 6.

Chin J (ed). 2000. Viral hepatitis B, in *Control of Communicable Diseases Manual,* 17th ed. American Public Health Association (APHA): Washington DC, 247-251.

Cruse PJE and R Foord. 1980. The epidemiology of wound infection.: A 10-year prospective study of 62,939 wounds. *Surg Clin North Am* 60(1): 27-40.

Davis MS. 2001a. Choices of effective personal protective equipment , in *Advanced Precautions for Today's OR: The Operating Room Professional's Handbook for the OR: Injuries and Bloodborne Exposures,* 2nd ed. Sweinbinder Publications LLC: Atlanta, 39-48.

Davis MS 2001b. Obstetrical Procedures in *Advanced Precautions for Today's OR: The Operating Room Professional's Handbook for the Prevention of Sharps Injuries and Bloodborne Exposures,* 2nd ed. Sweinbinder Publications LLC: Atlanta, 85-92.

ETNA Communications. 2000. Infection Control Signs, Available on: www.etnacomm.com.

Faro S. 1990. Infections in Pregnancy. Gilstrap L and S Faro (eds) John Wiley & Sons, Inc.: New York.

Fry DE. 2003. Surgical site infection: Pathogenesis and prevention. *Medscape* (February 19). Available at www.medscape.com/viewprogram/2220_pnt.

Fox V. 1992. Passing surgical instruments sharps without injury. *AORN J* 55(1): 264.

Garibaldi RA et al. 1980. Meatal colonization and catheter-associated bacteriuria. *N Engl J Med* 303(6): 316-318.

Garner JS and MS Favero. 1986. CDC guideline for hand washing and hospital environmental control, 1985. *Infect Control* 7(4): 231-243.

Garner JS, and The Hospital Infection Control Practices Advisory Committee (HICPAC). 1996. Guideline for isolation precautions in hospitals. *Infect Control Hosp Epidemiol* 17(1): 53-80 and *Am J Infect Control* 23(4): 24-52; 225-236.

Gershon R and B Vlahov. 1992. Assessing and reducing HIV risk to the critical care nurse. *Critical Care Nursing Currents* 3: (No 3).

Gershon R and B Zirkin. 1995. Behavioral factors in safety training, in *Laboratory Safety, Principles and Practices*, 2nd ed. Flemming DO et al (eds). AMS Press: Washington DC, 269-277.

Girou E et al. 2002. Efficacy of hand rubbing with alcohol based solution versus standard handwashing with antiseptic soap: Randomized clinical trial. *BMJ* 325(7360): 362-365.

Gisselquist D et al. 2002. HIV infections in sub-Saharan Africa not explained by sexual or vertical transmission. *Ind J STD AIDS* 12(10): 657-666.

Haley RW et al. 1985. The nationwide nosocomial infection rate: A new need for vital statistics in U.S. hospitals. *Am J Epidemiol* 12(2): 182-295.

Hedderwick SA et al. 2000. Pathogenic organisms associated with artificial fingernails worn by healthcare workers. *Infect Control Hosp Epidmiol* 21(8): 505-509.

Jacobson G et al. 1985. Handwashing: Ring-wearing and number of microorganisms. *Nurs Res* 34(3): 186-188.

James RC and CJ MacLeod. 1961. Induction of staphylococcal infections in mice with small inocula introduced on sutures. Br J Exp Pathol 42: 266-272.

Jarvis WR et al. 1991. Nosocomial infection rates in adult and pediatric care units in the United States. *Am J Med* 91(Suppl3B): 185S-191S.

Jarvis WR and JM Hughes. 1993. *Nosocomial gastrointestinal infections, in Prevention and Control of Nosocomial Infections,* 2nd ed. Williams & Williams: Baltimore, Maryland. 708-745.

Landry Kand D Kilpatrick. 1997. Why shave a mother before she gives birth? *Modern Child Nurs* 2:189-190.

Larson EL. 1988. Guideline for use of topical antimicrobial agents. *Am J Infect Control* 16(6): 253-266.

Lynch P et al. 1997. Infection Prevention with Limited Resources. ETNA Communications: Chicago.

Lynch P et al. 1990. Implementing and evaluating a system of generic infection precautions: Body substance isolation. *Am J Infect Control* 18(1): 1-12.

McCormick RD et al. 2000. Double-blind, randomized trial of scheduled use of a novel barrier cream and an oil-containing lotion for protecting the hands of healthcare workers. *Am J Infect Control* 28(4): 302-310.

McGinley LK, EL Larson and JJ Leydon. 1988. Composition and density of microflora in the subungual space of the hand. *J Clin Microbiol* 26(5): 950-953.

Minkoff HL and RH Schwarz. 1980. The rising cesarean section rate: Can it be safely reversed? Obstet Gyn 56(2): 135-143.

Moolenaar RL et al. 2000. A prolonged outbreak of Pseudomonas aeruginosa in a neonatal intensive care unit: did staff fingernails play a role in disease transmission? *Infect Control Hosp Epidemiol* 21(2):80-85.

Nichols RL. 1991. Surgical wound infection. *Am J Med* 91(Suppl 3B): 54S-64S.

Nichols RL. 2001. Preventing surgical site infections: A surgeon's perspective. *Emerg Infect Dis* 7(2): 220-224.

Olsen RJ et al. 1993. Examination gloves as barriers to hand contamination in clinical practice. *JAMA* 270(3): 350-353.

Patterson JE. et al. 1991. Association of contaminated gloves with transmission of *Acinetobacter calcoaceticus var. anitratus* in an intensive care unit. *Am J Med* 91(5):479-483.

Pierce M. 1990. Personal reference. Johnson and Johnson Medical, Inc., Research Division, Arlington, Texas.

Ponce de Leon S. 1991. The needs of developing countries and the resources required. *J Hosp Infect* 18(Suppl A): 376-381.

Raad I et al. 1993. Ultrastructural analysis of indwelling vascular catheters: A quantitative relation ship between luminal colonization and duration of placement. *J Infect Dis* 168(2): 400-407.

Russell AD, WB Hugo and GA Ayliffe. 1982. *Principles and Practice of Disinfection, Preservation and Sterilization*. Blackwell Scientific Publications: Oxford, England.

Rutala WA. 1996. APIC guideline for selection and use of disinfectants. *Amer J Infect Control* 24(4): 313-342.

Schaberg DR, DH Culver and RP Gaynes. 1991. Major trends in the microbial etiology of nosocomial infection. *Am J Med* 91(Suppl 3B): 72s-75s.

Seropian R and BM Reynolds. 1971. Wound infections after preoperative depilatory versus razor preparation. *Am J Surg* 121(3): 251-254.

South East Asia Regional Office SEARO/WHO. 1988. *A Manual on Infection Control in Health Facilities*. SEARO: New Delhi, India, 72-88.

Tokars JI et al. 1995. Skin and mucous membrane contacts with blood during surgical procedures: Risk and Prevention. *Infect Control Hosp Epidemiol* 16(12): 703-711.

Tokars JI et al. 1992. Double gloving cuts blood contacts 70%. *Ob/Gyn News* 26(23): 1.

WHO. 1999. Safe Management of Wastes from Healthcare Activities. Geneva.

WHO. 1989. Guidelines on Sterilization and High-Level Disinfection Methods Effective Against Human Immuno-deficiency Virus (HIV). AIDS Series 2. Geneva.

APPENDIX I

CORE INFECTION PREVENTION AND CONTROL INTERVENTIONS FOR HEALTHCARE FACILITIES AT A GLANCE

Specific interventions	Target groups	Equipment and supply needs	Clinical process indicators for monitoring
Hand hygiene	• All healthcare workers • Visitors • Patients	• Clean running water • Soap (preferably mounted) • Towels • Alcohol-based solutions	• Proportion of staff observed performing hand hygiene before attending patients
Personal protective equipment	• All health workers	• Gloves • Gowns	• Proportion of staff observed wearing gloves when exposure to blood or body fluids is anticipated
Isolation precautions	• Nurses • Physicians • Nursing Aids • Others	• Gloves • Gowns • Masks • Eye protection	• Average time between admission and isolation for tuberculosis patients
Aseptic techniques	• Nurses • Physicians • Laboratory technicians • Dental surgeons	• Antiseptics • Sterile gloves • Sterile devices and instruments • Sterile barrier devices	• Proportion of intravenous lines inserted using aseptic technique
Cleaning and Disinfection	• Nurses • Nursing Aides • Housekeeping staff • Laboratory staff	• Cleaning fluids • Cleaning equipment • Disinfectant	• Proportion of rooms appropriately disinfected after patient's discharge
Sterilization	• Sterilization staff • Nurses • Laboratory technicians • Dental surgeons	• Autoclaves and steam sterilizers • Test strips • Chemicals	• Proportion of sterilized devices whose sterility is documented with test strips
Waste management	• Healthcare workers • Waste handlers • Logistics	• Sharp boxes and other collection containers • Storage space and container for interim storage • Final disposal options • Personal protection equipments for waste handlers	• Presence of healthcare waste in the surroundings of the healthcare facility
Protocol for Antibiotic Use	• Physicians	• Essential list of antibiotics	• Proportion of prescriptions including an antibiotic
Immunization and exposure management	• All healthcare workers*	• Hepatitis B vaccine and other appropriate vaccines	• Three-dose hepatitis B vaccine coverage among nurses, physicians and laboratory technicians

Source: World Health Organization Aide-Memoire, 2004.
* Include nursing staff, physicians, dental staff, laboratory staff, housekeeping staff, waste management staff and morgue staff

APPENDIX II

LIST OF PARTICIPANTS FOR THE DEVELOPMENT OF NATIONAL INFECTION PREVENTION AND CONTROL GUIDELINES FOR HEALTHCARE SERVICES IN TANZANIA

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