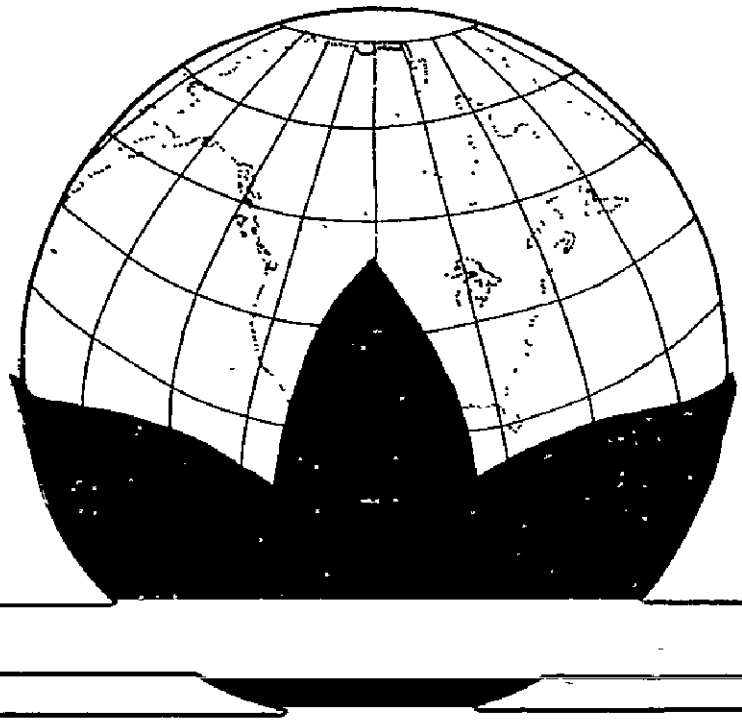
~~PN-AAK-813~~
4311 $56.00
PN-AAK-817
ISN 2069

This Five-Chapter Reprint Concerning Sorghum Research Is from MP-1451—

Biology and Breeding for Resistance

To Arthropods and Pathogens in Agricultural Plants



The Texas Agricultural Experiment Station, Neville P. Clarke, Director, The Texas A&M University System, College Station, Texas
AND
University of California--Agency for International Development, Pest Management Project, — Ray Smith, Director

AID/TA-C-1195
9310930

DSAN-G-0017
931 1156

This Five-Chapter Reprint Concerning
Sorghum Research Is from MP-1451—

Biology and Breeding for Resistance
to Arthropods and Pathogens in Agricultural Plants

Proceedings of a short course entitled
"International Short Course in Host Plant Resistance"

Texas A&M University
College Station, Texas
July 22-August 4, 1979

Edited by M.K. Harris

Short Course Committee and Subject Matter Associate Editors

R.A. Frederiksen	(Plant Pathology)
G.A. Niles	(Soil and Crop Sciences)
G. L. Teetes	(Entomology)

Table of Contents

BIOLOGY OF SORGHUM
K. F. Schertz. 124

BREEDING OF SORGHUM
F. R. Miller. 128

BREEDING FOR DISEASE RESISTANCE IN SORGHUM
R. A. Frederiksen and D. T. Rosenow. 137

BREEDING FOR ARTHROPOD RESISTANCE IN SORGHUM
J. W. Johnson and G. L. Teetes. 168

OVERVIEW OF PEST MANAGEMENT AND HOST PLANT RESISTANCE IN U.S. SORGHUM
G. L. Teetes. 181

Lecturers

Frederiksen, Dr. R. A.	Dept. of Plant Sciences Texas A&M University College Station, Texas 77843
Johnson, Dr. J. W.	Texas A&M Research Center Route 3 Lubbock, Texas 79401
Miller, Dr. F. R.	Dept. of Soil & Crop Sciences Texas A&M University College Station, Texas 77843
Rosenow, Dr. D. T.	Texas A&M Research Center Route 3, Lubbock, Texas 79401
Schertz, Dr. K. F.	USDA-SEA Texas A&M University College Station, Texas 77843
Teetes, Dr. G. L.	Department of Entomology Texas A&M University College Station, Texas 77843

PSAN-6-0017

FOREWORD

Crop productivity in the world must double in the next 25 years to meet the food and fiber requirements for a rapidly expanding world population. In order for Agriculture to meet this challenge, better varieties of crops carrying resistance to primary pests must be developed at a faster pace world wide. This must be done in an atmosphere of increasing energy shortages, inflation, and world environmental and health concerns. As we look at these future needs and the severe constraints that world agriculture will be under, breeding for resistance to major pest species, coupled with biological control, offers the most promise, especially to the developing countries and to small farmers. Much of the increased productivity needed must come from cropland in current food-deficient countries. This presents a great challenge to agricultural researchers to develop pest control strategies that do not rely heavily on costly and often unavailable pesticides.

The Host Plant Resistance International Workshop conducted at College Station during July 22-August 4, 1979, and sponsored by UC/AID and Texas A&M University, was a successful endeavor to promote increased awareness and efforts toward breeding for resistance to pests which will, in the years ahead, help meet the challenge to Agriculture for increased food production. In addition, the proceedings of this workshop presented herein provides a valuable contribution to existing literature in the field of Host Plant Resistance and will provide a valuable supplement to the recent text and resource book in the field "Breeding Plants Resistant to Insects" edited by F. G. Maxwell and Peter Jennings. Dr. Marvin Harris and the contributing authors to this Workshop publication are to be congratulated for a task well planned and executed.

Fowden G. Maxwell
Professor and Head
Department of Entomology
Texas A&M University
College Station, Texas 77843

Dr. Perry L. Adkisson
Deputy Chancellor for Agriculture
Texas A&M University
College Station, Texas 77843

Dr. Ray F. Smith, Director
UC/AID Pest Management Project
University of California
Berkeley, California 94720

BIOLOGY OF SORGHUM

K. F. Schertz

USDA-SEA
and Dept. Soil and Crop Sciences
Texas A&M University

ABSTRACT

Sorghum bicolor (L.) Moench is a cereal crop that is of special importance in regions of marginal moisture. Sorghum seedlings are relatively slow growing, but after a few weeks development is rapid and sturdy culms and abundant leaves are produced. Seed are produced on a panicle at the apex of the culm. Disease and insect problems often increase as sorghum is more intensively grown in an area, but sources of resistance are being located and offer opportunities in host-plant resistance.

INTRODUCTION

Sorghum is an important feed and food crop used for grain, forage, and other purposes. Its ability to produce under conditions of limited moisture has resulted in its use in marginal areas. Sorghum is attacked by its share of insects and diseases. Such problems cause considerable loss in sorghum production areas. The attacks may not be severe during the first few years that sorghum is grown in an area. Insects and diseases usually then become more prevalent, and we must know how to minimize the losses due to their attacks. Host-plant resistance is one approach to resolving this problem. A knowledge of its biology will help us understand host-plant resistance of sorghum. We will consider the characteristics of sorghum that relate to its response to pests and to breeding for resistance to those pests.

GENERAL CHARACTERISTICS

Sorghum is a member of the Gramineae (grass) family. Within the genus *Sorghum*, *S. bicolor* (L.) Moench is the most important agronomically. Within *S. bicolor* there are many types. The diverse types in the species are all diploids with a 2N = 20 chromosome number, and they will intercross. There are related grassy species with the same chromosome constitution and also tetraploids with 2N = 40.

Sorghum has the general structural and developmental characteristics of grasses. Roots are fibrous. Stems (culms) may be single to several per plant and each is divided into a series of internodes and nodes. Leaves consist of

sheaths and blades with parallel veins. At the apex of the culm is the inflorescence, a panicle that bears spikelets. The seed or caryopsis has a pericarp, endosperm, and a monocotyledonous embryo. The specifics of these and other characteristics will be discussed in later sections.

Before considering the details, however, let us observe how a sorghum plant grows. We will follow the whole plant through its development from a seedling to a seed. After observing the whole plant, we can then better understand its parts. The growth stages of sorghum are important to our understanding of the biology, pest control, and breeding of this species. Vanderlip (1972) provided a comprehensive presentation of this subject in which 10 stages were described and depicted.

We will simplify our discussion by considering only critical stages 3, 6, and 9. From emergence to growth stage 3 a sorghum plant is completely vegetative. It is differentiating leaves, nodes, and internodes, and is also developing roots. Approximately 30 days pass during this period when the activity of the growing point (apical meristem) is entirely vegetative. The time at which the growing point changes from vegetative to reproductive is called stage 3. By this time all of the leaves and internodes of the culm have been initiated. No more are possible. Although no new leaves will be initiated, leaves then present will expand and leaf area will increase. Subsequently, leaf area will decrease as the lower leaves die.

At stage 3, as the growing point becomes reproductive, its surface changes from smooth to rough. This can be detected by splitting a culm and observing the growing point with a hand lens. From this time on, differentiation at the growing point will be of reproductive organs. The panicle and its parts will be developed.

During the time from stage 3 to stage 6 (about 30 days) the size of the panicle and the number of potential seed is determined. It is also during this time that the internodes elongate and the apex of the culm is raised from a very few centimeters above the surface of the soil to heights from 50 cm to several meters.

At stage 6 flowering (anthesis) occurs. Pollen is shed, eggs are fertilized, and seed begin to form. Development and maturation of the seed takes about 35 days. At the end of this period the grain reaches physiological maturity (stage 9). The seed have accumulated maximum dry matter and will be ready to harvest whenever moisture content is sufficiently low.

A few general comments are in order. Growth stages of sorghum are defined and numbered in various ways by different scientists. The numbers are not important. The easily observed stages are 3) growing point differentiation, 6) anthesis, and 9) physiological maturity. These are critical stages for the sorghum plant and for scientists working with sorghum. It is important to remember that sorghum varies in developmental time intervals. Many lines are photoperiod sensitive and initiate panicles (stage 3) only in short days.

SPECIFIC CHARACTERISTICS

Morphological and anatomical characteristics of sorghum have been discussed by several authors. Included are reports by Artschwager and McGuire (1949), Artschwager (1948), Freeman (1970), Doggett (1970), Hector (1936), Leonard and Martin (1963), and Poehlman (1959). It is suggested that the reader refer to the photographs and detailed descriptions in those reports to supplement the overview here presented. The points discussed in this report will be those that have the most evident relationships to host-plant resistance.

Seedling. Sorghum seed germinate in three to seven days with appropriate conditions. Cool temperatures or dry soil can cause a delay. On germination, a primary seminal root is produced. Lateral branching and development of a few secondary roots from the base of the mesocotyl may occur. The succeeding roots are adventitious and develop in succession from the basal nodes situated just below to just above ground level. Root development of the young sorghum seedling

is usually very slow. However, after a few weeks the roots grow rapidly, branch profusely, and develop into an efficient fibrous root system.

So, too, the early development of the above ground portion of the plant is relatively slow. The mesocotyl elongates, the coleoptile pushes through the surface of the soil, and the leaves emerge. The blades of the first leaves (except for the seedling leaf) are usually narrow. The young sorghum seedling with limited root system and leaf area grows rather slowly. It is during this stage that relatively minor attacks by insects or diseases can cause considerable damage.

Stem. During the vegetative phase of the sorghum plant (until stage 3) internodes and nodes are developed. By the time the growing point differentiates to reproductive (stage 3), 15 to 18 nodes are present and a leaf sheath will be attached to each. Although internodes have formed, the total length of the stem is only a few centimeters; the internodes being short. The growing point is not far above the soil surface. At this stage the stem comprises a very small part of the above ground dry weight. Leaves are the main part of the plant. After growing point differentiation, the internodes begin to elongate. Remember, no additional internodes are formed after growing point differentiation. Internode elongation is most rapid in the few days just prior to anthesis. The internode just below the panicle, called the peduncle, elongates dramatically during this time. The stems are erect and solid and slightly furrowed on alternate sides. Vascular bundles are scattered throughout the fundamental parenchyma. The vascular bundles are each enclosed by a sheath. Pith forms in the center and may extend outward to the peripheral zone. There is a lateral bud at each node. In some varieties these buds, especially those near the base of the main culm, develop into tillers.

Leaf. Although all of the leaves (usually 15 to 18) have been initiated prior to the differentiation of growing point, they are not yet fully developed. From that time to anthesis the sheaths and blades of successively higher leaves continue to elongate. This leaf growth and development, coupled with internode elongation, unfurls the leaves. Maximum exposed leaf-blade area is reached near the time of anthesis. Leaves near the base of the plant are dying. Also, the environment of the lower leaves is changing. Changes in light, humidity, and air movement affect these leaves. These factors also affect insects and diseases that can cause problems.

Leaves are arranged alternately. Sheaths are attached to the nodes and are usually long, exceeding the length of the internodes. The margins on the sheaths are membranous and overlapping. The blades of young leaves are rather erect but they become more near horizontal and curved as the plant develops. The leaf has a prominent midrib and parallel veins. Stomata occur on both sides of the leaf blade. A wax (called bloom) develops on the blades and sheaths.

Inflorescence. The "head" or "ear" of sorghum is a panicle. It consists of a central axis with whorls of main branches each with secondary branching. The lengths of the main axis and branches vary, giving the panicles of each variety their characteristic shape from long and loose to short and compact.

The panicle emerges from the upper leaf sheath ("boot"). In a few days the spikelets near the apex open and those successively lower open during the next five to six days. Each sessile spikelet contains two florets, one fertile, the other sterile. The fertile floret contains a pistil with feathery stigma and three stamens, each with an anther. Anthesis occurs in early morning when spikelets open and anthers shed pollen. Although sorghum is mainly self pollinated, pollen is carried by wind and about 6% crossing occurs. Cytoplasmic-genetic male steriles have been developed and are useful as females in hybrid seed production and in breeding programs.

Caryopsis. Seed of sorghum begin to form shortly after pollination and can be observed on a panicle by seven days thereafter. By about 35 days from pollination the seed are mature. The main components of the seed are the pericarp, endosperm, and embryo. The pericarp, the outer layer of the seed, varies from dark brown to nearly colorless. The next layer, the testa, is sometimes brown.

Endosperm varies from floury to vitreous and from white to yellow. The embryo contains a scutellum, mesocotyl, coleoptile, plumule, and radical. Variations in the characteristics of the seed affect their susceptibility to damage, particularly by diseases. Seed of sorghum are quite exposed, being on the apex of the plant and little protected by glumes.

SUMMARY

The sorghum plant is at the same time well and poorly adapted for host-plant resistance. Sorghum is relatively tolerant of heat and drought, is a perennial in the warm climates, and is fast growing after the first few weeks. Sorghum is, however, slow growing as a seedling, not well suited to cool weather, has an abundance of closely spaced leaves (in short varieties) and develops panicles and seed exposed to rain and pests. Sorghum presents both a challenge and an opportunity in host-plant resistance.

LITERATURE CITED

Artschwager, E. 1948. Anatomy and morphology of the vegetative organs of Sorghum vulgare. USDA Tech. Bull. No. 957.

Artschwager, E. and R. C. McGuire. 1949. Cytology of reproduction in Sorghum vulgare. J. Agr. Res. 78. 659-673.

Doggett, H. 1970. Sorghum. Longmans. London.

Freeman, J. E. 1970. Development and structure of the sorghum plant and its fruit. p. 28-72. G. S. Wall and W. M. Ross (Ed) Sorghum Production and Utilization. AVI. Westport, Conn.

Hector, J. M. 1936. Introduction to the botany of field crops. Vol. 1. Cereals. Central News Agency Ltd. Johannesburg, South Africa.

Leonard, W. H. and J. H. Martin. 1963. Cereal Crops. Macmillan. New York.

Poehlman, J. M. 1959. Breeding Field Crops. Henry Holt & Company, Inc. New York.

Vanderlip, R. L. 1972. How a sorghum plant develops. Kansas Agr. Expt. Sta. C-447.

5

THE BREEDING OF SORGHUM

Frederick R. Miller
Associate Professor - Sorghum Breeder
Department of Soil & Crop Sciences
Texas A&M University
College Station, Texas

Sorghum, Sorghum bicolor (L.) Moench, is a large stemmed tropical grass and has the ability to grow to great heights. The species is extremely diverse and present collections contain over 17,000 distinct cultivars. These materials readily intercross and produce fertile offspring. From a breeder's interest, the greatest variability in both cultivated and wild sorghums exist in the Northeast quadrant of Africa, but all of Africa is a fertile source of diversity.

Murty, et al. (1) following the lead of Snowden developed a very workable and useful taxonomic classification of sorghum types. This system separates the major types of sorghum into recognizable working groups and has become widely used. Harlan and de Witt (2) suggested a system using only five basic races and hybrid races among these.

Rather than develop the extent of diversity which exists in the species, suffice to say this has been reported and described. Because sorghum is a short-day plant and is very sensitive to photoperiod and temperature, regional breeding efforts should consider these limitations. Generally sorghum hybrids, whether for grain or forage, require 100-140 days from planting to maturity. However, in tropical areas the crop is generally planted prior to the onset of rains and harvest is completed after the rains have subsided. Maturity of sorghums in this situation is regulated by photoperiod manipulation. Yields generally increase as time to maturity increases up to a point where the requirements for growth become limiting, then yields decrease. There is also a positive correlation between increased height and increased yield. As height is increased up to approximately 1.5 m, increased yield can be expected. Lodging is of serious consideration and minimum heights, which maximize yields, should be used where possible.

Because height and duration of growth are of such significance to any breeding effort, a further observation is given. Height in sorghum is a variable trait, but is under simple genetic control. In most areas of the world taller plants are preferred, but in those areas where mechanical harvesting is practices, short stature is required. Quinby and Karper (3) have shown height to be controlled by four recessive, non-linked, brachytic dwarfing genes. A single recessive gene may reduce height by 50 cm or more. Most grain sorghum hybrids developed in the U.S. are recessive at 3 height loci - 3 dwarfs - and are generally $\underline{dw_1}\underline{Dw_2}\underline{dw_3}\underline{dw_4}$. The dw_3 gene is unstable and results in a higher than normal frequency of tall plants. Most breeders recognize the positive correlation between height and yield and develop sorghum hybrids which are as tall as possible to withstand the local hazards of production. Maximum yields are generally recognized at aobut 1.5 to 1.75 m height.

Maturity in sorghum has been used to regulate the time of harvest to escape grain deterioration, seed mold and insect damage and to maximize yield. There is a wide array of maturity differences ranging from 60 to 300+ days among cultivars. When sorghum differs in maturity, it is the result of a response to temperature and photoperiod. Quinby (4) has shown that differences in numerous varieties are controlled by four genes and an allelic series at each locus. Rate of growth is reflected through maturity differences and leaf production appears at a similar rate. Rate of leaf production varies between 2.8 and 3.5

days per leaf, but both height and rate of growth are limited under stress conditions.

Sorghum is a self-pollinating species, but is handled in advanced agricultural situations as a cross-pollinating crop. The perfect flowers of sorghum allow from 0 to 10 percent outcrossing depending on variety with an average of about 2 percent. In much of the sorghum growing world there are only selected or improved varieties for use, while in high intensity agriculture areas F_1 hybrids predominate. Hybrid sorghums for either grain or forage have a recognized yield improvement. Grain sorghum single cross hybrids express 20 to 50 percent yield improvement.

The parents of sorghum hybrids are inbred lines, but, unlike maize inbreds, are reasonably vigorous. These inbred lines have been often widely grown varieties. Until recently there has been only variety selection as a major crop improvement program effort. Present parental lines have the same characteristics of improved varieties except that they were specially developed for hybrid utilization. There is a good association between the desirability of an inbred parental line and the yield of the resulting F_1 hybrid. Good lines make good hybrids.

CYTOPLASMIC GENETIC MALE-STERILITY

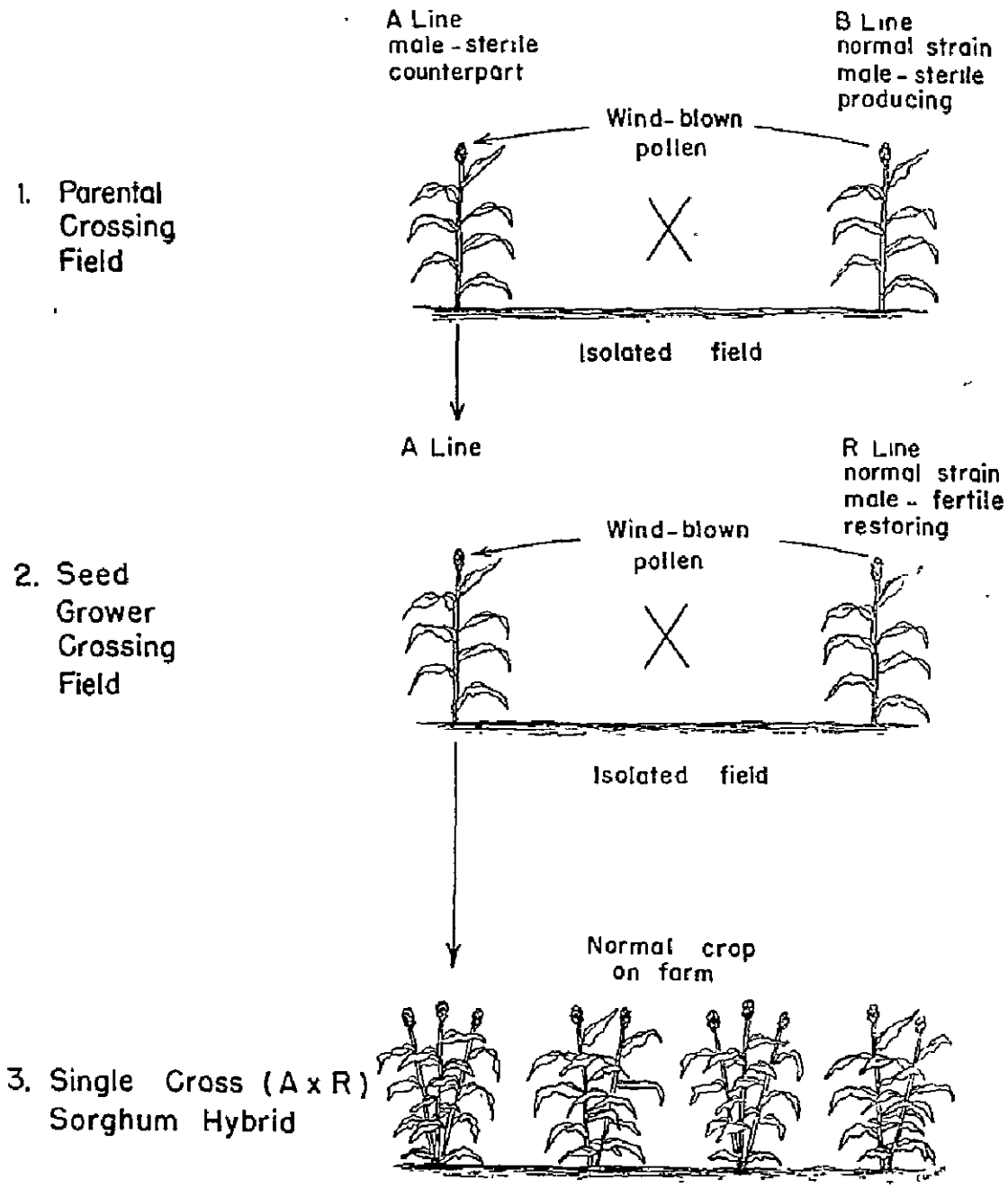
Cytoplasmic genetic male sterility is extremely important to hybridization in sorghum. Therefore we will discuss the development of male sterile but female fertile parents. Male sterility suggests normal function of the female portion of the flower and inhibited male response. Because the male-sterile plants do not disseminate viable pollen, these plants can be fertilized only by pollen from otherwise normal pollen producing plants. Both cytoplasmic and genetic male-sterility in sorghum cause poorly developed anthers and a lack of pollen production. The important difference between the two types of male sterility is their mode of inheritance. Genetic sterility is inherited normally and influence of the male is seen in the progeny. However, in cytoplasmic male-sterility, inheritance is maternal. All the progeny of a cytoplasmic male-sterile female pollinated by its normal counterpart will be sterile like the female parent. The mode of inheritance allows a cytoplasmic male-sterile to be maintained easily by growing A- (cytoplasmic male steriles) and B- (male sterile producing) lines together in parental crossing fields. Fertile sorghum hybrids can be produced by growing A- and R- (genetic pollen restoring) lines together in seed grower crossing fields. This type crossing is shown in Figure 1.

Cytoplasmic male-sterility is thought to result from imcompatability between the cytoplasm of the female and nuclear factors contributed by the male parent. Since the male parent passes little or no cytoplasm to the next generation with the male gamete, the female parent furnishes the cytoplasm to the offspring. Cytoplasmic male sterility was found in sorghum when the milo cytoplasm was used with nuclear factors from Kafir in a backcross breeding procedure. Figure 2 shows the cytoplasm and nuclear behavior in the development of a cytoplasmic sterile inbred parent.

Recently USDA and TAES released a second cytoplasmic genetic sterility system which differs from the Milo-Kafir system. This second system (A_2) was developed using Caudatum-Nigricans-Roxburghii where the former was the cytoplasm source. The development of new sterile sources from this cytoplasmic interaction should follow the same procedure as that outlined in Figure 2.

A- and B- lines are similar in their genetic make-up, but A-lines have sterile cytoplasm while B-lines have normal cytoplasm. Restorers (R-lines) must always carry the fertility restoring gene or genes and may have either normal or sterile cytoplasm.

FIG. 1. Procedure to maintain A-lines and produce hybrid seed of sorghum under field scale operations (number of A-line rows vary from 4 to 16 for each set of 2 rows of pollen source; either the B-line or R-line).



8

RANDOM-MATING POPULATIONS AND GERMPLASM STORAGE

Development of random-mating populations in sorghum has gained some importance within the past 5 years. Very little progress in yield improvement can be attributed to this method of breeding up to this time. Sorghum is a self-pollinated species and there are obvious large blocks of elite genes accumulated in superior inbred parental stocks. These are used to produce present F_1 single cross hybrids. The basic breeding stocks, parental lines, etc. which are in use today by the U.S. sorghum industry have resulted from pedigree breeding or backcross breeding with selection for the desired trait.

Random-mating populations are useful as storage facilities where large inventories of stocks would otherwise be required. In order to develop a random-mating population in sorghum, a self pollinated species, one needs the use of a genetic male sterile gene. Several such genes have been described in sorghum with the most widely used being $\underline{ms_3}\underline{ms_3}$ and $\underline{ms_7}\underline{ms_7}$. The gene "antherless" was used in a few early populations. The recessive gene is easily backcrossed into a number of stocks. When these stocks are grown as F_2 or selected progeny any number of materials can be outcrossed onto the male sterile plants. These crosses are grown as F_1's and can be either self-pollinated or left open-pollinated. Generally equal quantities of seed from each F_1 is then massed to form the original population. This equal quantity mass of F_2 seed is then grown in isolation and allowed to random mate. Generally there will be 3:1 segregation for fertile to sterile plants. The male sterile plants are marked so that they may be recognized at maturity. Each male sterile plant is threshed separately and an equal quantity outcrossed seed is again massed to form the next generation of random mating. By 3 random matings the population should be segregating approximately 1:1 fertile to male sterile plants and will be at equalibrium for that trait. Any of several improvement procedures may be used to evaluate and select the population at this time. Some are full-sib, half-sib, recurrent selection, reciprocal recurrent selection methods. These are described by other authors working with cross-pollinated species such as maize.

To store or add material to a population it is only necessary to plant the additions in alternate rows between rows of the base random mating population and either use natural pollination or hand crossing to incorporate the material.

In the event of a major disease or insect problem a large sample of the random mated population (germplasm storage facility) could be planted to screen for a source of resistance. This resistance could be extracted and used then in a rapid backcross or pedigree procedure to correct the deficiency in the elite inbred parental stocks.

Many states, especially Texas, and other agencies, such as USDA - Mayaguez, Puerto Rico, National Seed Storage Laboratory - Ft. Collins, Colorado and ICRISAT - Hyderabad, India, maintain large collections of inbred varieties to support and supplement population storage of germplasm.

DEVELOPMENT OF A-LINES

The A-lines used as female parental stocks in sorghum are male sterile counterparts of the normal strains or B-lines from which they derived. All the selection which can be practices on a new female must necessarily be done on the fertile counterpart, the B-line. Therefore, it is extremely important to understand the procedure of female development in sorghum.

Any strain that has the desired agronomic characteristics and produces only sterile progenies when crossed to cytoplasmic male-sterile stocks can be con-

verted to an A-line by the procedure called, "paired progeny selection." Paired progeny selection is necessary because male-sterility is expressed only in the crossed progeny, while effective selection can occur only in the recurrent parent. The original crosses of the strain to be converted should be made on an agronomically similar male-sterile (A-line) in order that segregation for undesired plant characteristics will be reduced as much as possible. The progenies of these crosses and the recurrent male parent are grown in adjacent rows. A number of panicles are bagged in the cross progenies and at least part of them should resemble the male parent. After anthesis is completed or is well advanced so that male sterility of the bagged panicles can be estimated, one or more of the panicles with the best sterility and greatest agronomic similarity to the recurrent male parent are crossed with selected plants from the male row, i.e., paired pollen parent row. This new cross and its male parent are then planted the next generation. This paired progeny selection is continued through successive backcrosses until the male-sterile strain is considered to be essentially like the normal fertile strain. See Figure 2 for further understanding. The pair are then designated A- and B- lines respectively, and can be maintained or increased either by hand pollinations or in isolated parental crossing blocks (Figure 1). Few cytoplasmic genetic steriles are always 100 percent male sterile. Seed certification standards allow low levels of these genetic x environmentally fertile plants to exist in hybrid seed. Quality control is of major concern to the commercial producer of hybrid seed. He must insure that less than .01 percent self-pollinated plants occur in his hybrid seed.

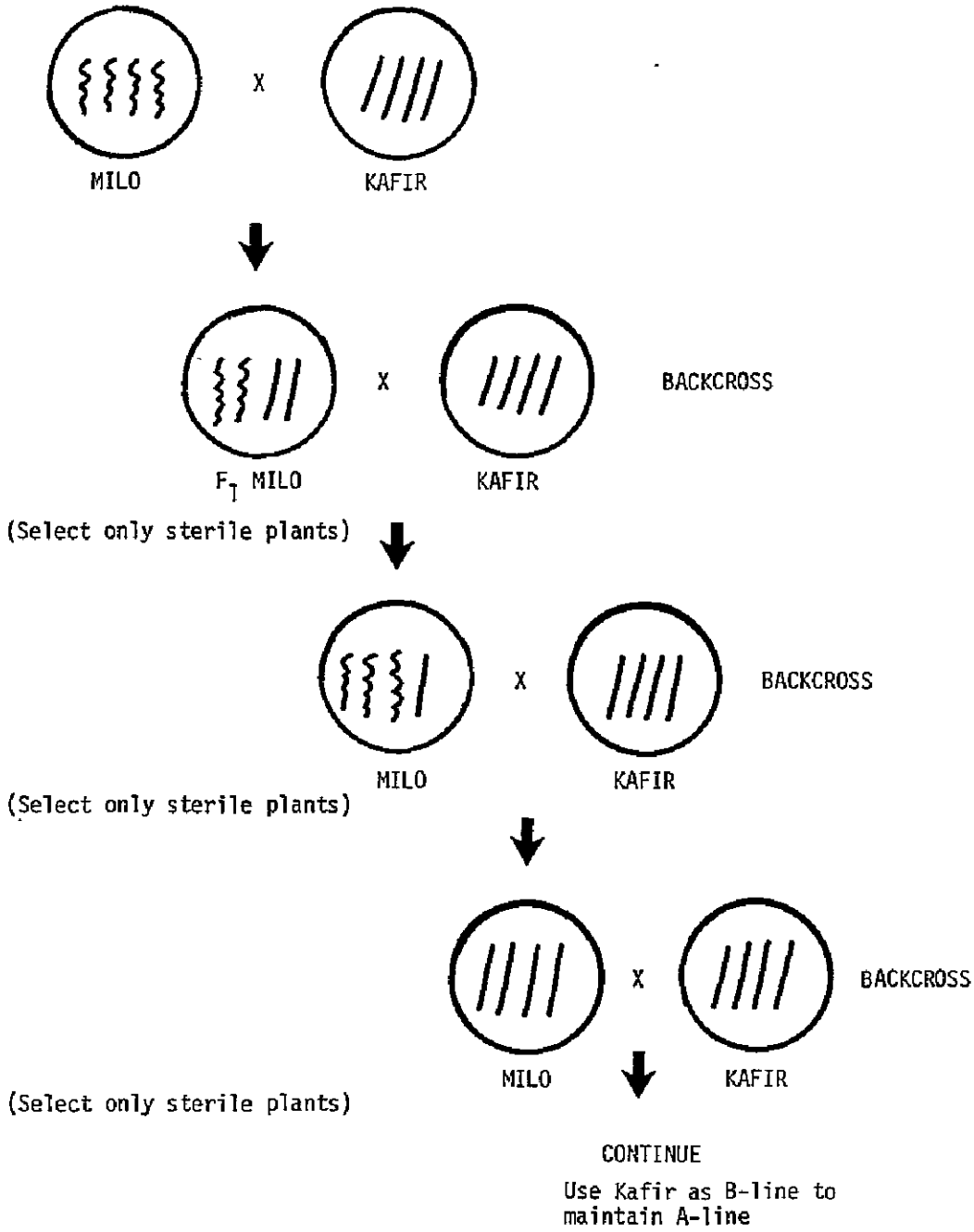
It is a matter of judgement and depends on the purity or homozygosity of the B-line stock just how many crosses will need to be made between parents and selected paired progenies. Obviously, if the parent strains were completely homozygous, there would be no need to make more than one cross or backcross per generation. Complete homozygosity cannot be assumed, but strains that have been evaluated as self-pollinated varieties or have been in nurseries maintained by single bagged panicles are essentially pure lines and usually offer little basis for selection between different paired progenies. The opportunity for effective selection increases with increased genetic variability between parental strains. One major gene has been recognized which separates good parental stocks from those which cannot be sterilized. There are several modifiers or partial sterility factors in some stocks which results in their non-usefulness. As a general rule, use as parental stocks only those which give complete male sterility when first crossed to a cytoplasmic-genetic male-sterile tester.

From this discussion, it is obvious that the development of new females (A-lines) in sorghum is a long and expensive process. This explains why there are so few A-lines in use today.

DEVELOPMENT OF R-LINES

Many varieties and strains give male-fertility to the progeny of crosses with cytoplasmic-genetic male-steriles (A-lines). There are approximately 3/4 of the World Collection of Sorghum which are of this type. These strains are called R-lines and are male-fertility or pollen restorers. If a pollen restorer is essentially a pure line, little improvement can be made with further selection. If the strain is somewhat variable in plant characteristics or fertility restoration, paired progeny selection similar to that used in producing A-lines will be effective in obtaining uniformity and in isolating good pollen restoration. The selected line is designated as an R-line and is crossed with appropriate A-lines to produce F_1 hybrids as in Figure 1.

FIG. 2. Diagramitic procedure to develop a cytoplasmic male-sterile inbred line in sorghum



The development of elite R-lines through backcross or pedigree breeding methods has been phenomenal in sorghum. Those breeders who have had the foresight to recognize limitations in elite materials and have used the above mentioned procedures to incorporate additional eliteness have made major contributions to this commodity. Some of these have been disease resistance - downy mildew, head smut, periconia, bacterial stripe, rust, anthracnose, etc,; insect resistance - chinchbugs, greenbugs, sorghum midge; quality - high protein, balance amino acid content, flavor, texture, white pericarp color; growth - maturity, height, drought tolerance, tropical adaptation, temperate adaptation, heat tolerance, and many more eilteness traits which increase productivity.

THE SORGHUM CONVERSION PROGRAM

The cornerstone to much of the present crop improvement in sorghum is the imaginative Sorghum Conversion Project. This joint TAES-USDA project changes tall, late or non-flowering sorghums from the tropics into short, early forms which can be used in all areas of the world, but especially in the temperate zones. This is done by substituting up to 8 genes which control height and maturity to obtain the desired genotypes. The procedure is outlines in Figure 3.

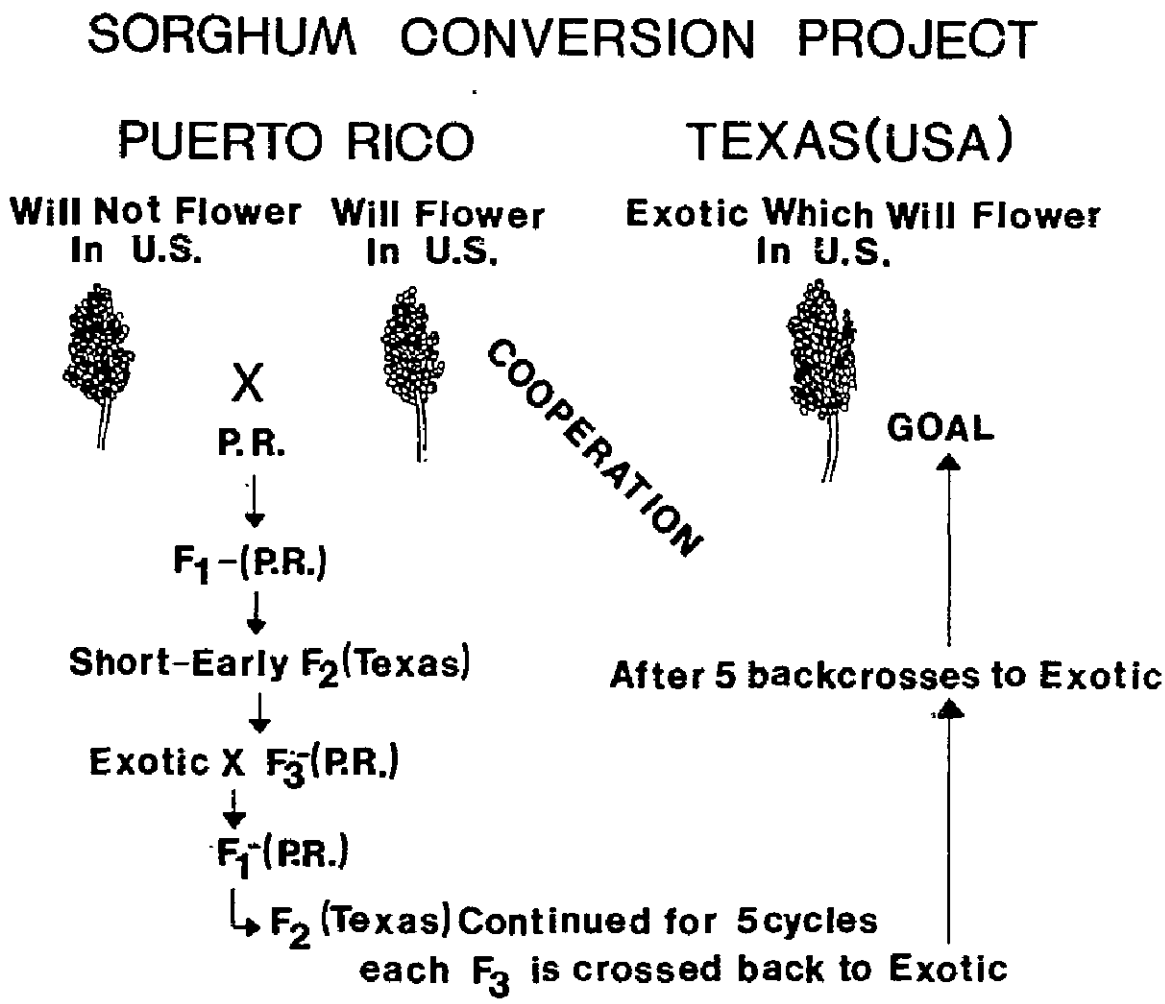
Because sorghum originated near the equator in Northeastern Africa, it is sensitive to the length of day. Knowing the genetics of height and maturity and the response to daylength, it was possible to use the facilities of the USDA's tropical research station at Mayaguez, Puerto Rico and a temperate selection site at Chillicothe, Texas to make vast amounts of germplasm available for further plant improvement. Selections were made from the World Collection which were judged to offer the greatest diversity and eliteness. Because of limited manpower and resources, it was possible to convert only those with outstanding characteristics. At present, there are 1,216 items in the program. The original cross and four backcrosses with selection in each generation have allowed the recovery of over 98% of the germplasm in each entry. During the last backcross which is done by hand emasculation the cross is made using the alien line as the female. This allows the recovery of the cytoplasm of the converted line also.

Materials from this program are dramatically changing the sorghum industry. Some of the important economic characteristics obtained from this project are: (a) new sources of disease resistance - downy mildew, head smut, maize dwarf mosaic virus, foliar diseases, stalk rots, kernel rots and anthracnose; (b) insect resistance - sorghum midge, greenbug, corn leaf aphid, white flies, and Bank's grassmites; (c) improved plant characteristics - drought, heat and salinity tolerance, stalk strength, twin-seed, easy threshing, erect leaves, lodging resistance, improved yield of grain, yield stability under diverse environments, greater root development, leaf area retention, increased grain filling rates, increased combining ability and new sources of cytoplasmic sterility; (d) outstanding kernel characteristics - thin pericarps, weather resistance, reduced discoloration of the endosperm, increase protein content, superior balance of amino acids, improved flavor, expanded diversity for food product development and greater digestibility; (e) reduced genetic vulnerability - expanded diversity to reduce narrow germplasm base.

In conclusion, S. bicolor and its related species offer the potential of major utilization in the future. Within the past 100 years this commodity has been improved dramatically, but the past 5,000 years of culture still hangs heavily over us. Yield stability in areas where sorghum is the major food source is of utmost importance. Yield increases and improvement of the quality of the grain must be critically evaluated for yield stability as well. There

12

FIG. 3. The Sorghum Conversion Project is a backcross breeding procedure which utilizes the knowledge of genetics of maturity and height to change tall, late photoperiod sensitive sorghums into short, early types which can be used throughout the world more easily.



13

are over 17,000 items in the World Collection of Sorghum and each is somewhat different from the other. This broad base of diversity is a plant breeder's dream. Know the species and know how it is used and then have the insight to see what limits its use - to correct those limitations is our challenge. The breeding procedures available to us and the diversity create a very envious situation for the sorghum worker of the future.

LITERATURE CITED

1. Murty, B.R. V. Arunachalam, and M.B.L. Saxena. 1967. Classification and Catalogue of a World Collection of Sorghum. Indian Journal of Genetics and Plant Breeding. 27 (Spec. No.): 1-312.

2. Harlan, J.R. and J.M.J. de Witt. 1972. A Simplified Classification of Cultivated Sorghum. Crop Sci. 12:172-176.

3. Quinby, J.R. and R.E. Karper. 1954. Inheritance of Height in Sorghum. Agron. J. 46:212-216.

4. Quinby, J.R. 1974. Physiology and Genetics of Maturity, Height, and Growth. In Sorghum Improvement and the Genetics of Growth. Texas A&M Press. Col. Sta., Tx.

SUPPORTING LITERATURE

1. Doggett, H. 1970. Sorghum. Longmans, Green and Co. Ltd., London.
2. Wall, J.S. and Ross, W.M. (Ed.) 1970. Sorghum Production and Utilization. The Avi Publishing Company, Inc. West Point, Conn.
3. Quinby, J.R. 1971. A Triumph of Research . . . Sorghum in Texas. The Texas Agricultural Experiment Station, College Station, Texas.
4. Miller, F.R. Sorghum Growth and Development (In Press). In Food and Agricultural Organization, Plant Protection Bulletin, Chapter 3.
5. Stephens, J.C., F.R. Miller and D.T. Rosenow. 1967. Conversion of Alien Sorghums to Early Combine Genotypes. Crop Sci. 7:396.
6. Miller, F.R. 1968. Some Diversity in the World Sorghum Collection. In Proceedings of 23rd Annual Corn and Sorghum Conference. American Seed Trade Association. Chicago, Ill.

BREEDING FOR DISEASE RESISTANCE IN SORGHUM

R. A. Frederiksen and D. T. Rosenow

INTRODUCTION

During the past decade, we have witnessed massive changes in the direction of research on sorghum diseases in North America. These changes were due in part to three factors: 1) repeated occurrences of damaging diseases; 2) the awareness of genetic vulnerability in our major commodities; and 3) the recognition of the importance of integrated pest management.

In southwestern USA, where sorghums are a principal agricultural commodity, serious disease problems have occurred repeatedly. During the past few years, there have been grave losses caused by head smut (Sphacelotheca reiliana), sorghum downy mildew (Peronosclerospora sorghi), maize dwarf mosaic (MDMV), anthracnose (Collectotrichum graminicola), charcoal rot (Macrophomina phaseolina), Fusarium stalk rot (Fusarium moniliforme) and a number of common foliage diseases. In Texas, we experienced anthracnose epidemics in 1966 and 1968, downy mildew and maize dwarf mosaic in 1967, head smut in 1969 and 1976, Pythium root rot in 1971, Fusarium head blight and stalk rots in 1972, serious downy mildew in 1973, and finally a major grain mold problem in 1974 and 1976. The outbreaks of these and other diseases suggest that disease problems vary from year to year for sorghum growers in Texas.

Thus, we elected to develop a broad-based multiple disease resistance approach in breeding Texas sorghum. Because of our varying weather and cropping patterns we must improve our levels of resistance to all of the major diseases of sorghum. Ignoring one disease or group of diseases could make us as vulnerable as the maize crop was to southern corn leaf blight in 1969 and 1970.

Sources of Genetic Diversity

Genetic vulnerability can be equated with genetic uniformity. It was estimated that, as recently as 1972, most of the grain sorghum hybrids grown in the United States were developed using closely related, and in some cases, identical pollinators and all were produced using the same cytoplasmic-genetic sterility system. To combat the narrowing base of elite germplasm, as early as 1963, the Texas Agricultural Experiment Station and the United States Department of Agriculture initiated a program whereby exotic sorghums from the World Sorghum Collection are converted from tall, late maturing, photoperiod-sensitive types to shorter, earlier maturing, less photoperiod-sensitive lines. During and following conversion we have been able to evaluate resistance in many partially converted and converted exotic sorghums to diseases prevalent in the United States. This screening has shown that the conversion materials are outstanding as sources of resistance to essentially all economically important diseases of sorghum. Of special significance was the finding of a

large number of agronomically desirable lines with high levels of resistance to downy mildew and anthracnose, two diseases for which agronomically desirable resistant sources were not available prior to screening conversion materials. Also, sources of resistance have been found to head smut, maize dwarf mosaic, grain mold, rust, zonate leaf spot, grey leaf spot, bacterial stripe, leaf blight and Fusarium head blight. Outstanding sources of resistance to the various types of lodging as well as resistance to charcoal rot have been identified. Recent advances in host plant resistance have been summarized in the Texas A&M, USAID Ta-c-1092 Annual Reports of 1974, 1975, and 1976.

Disease Nursery Program

General. Beginning as early as 1969, a group of partially converted sorghum lines were selected as potentially useful sources of disease resistance. These lines, most of which were zera-zera type sorghums from Ethiopia constituted our first "All Disease Nursery" (ADN), later called the ADIN because of certain selected insect resistant sorghums. Entries in the ADIN are selected from sorghums evaluated under natural or artificially induced disease epiphytotics. Initially these came from partially converted lines, but today they also are selected from converted lines, introductions from other programs, selections from random mated populations and from advanced breeding lines developed in the TAES sorghum improvement program. ADIN entries must have superior resistance to one or more of the disease groups. In the ADIN these entries are exposed to a vast array of hostile environments including all of the major disease nurseries in Texas and neighboring states. The number of locations at which the ADIN was evaluated grew from 12 in 1974 to 21 in 1976. The number of entries in the ADIN is 70 with 5 standard controls. Entries change from year to year as lines are improved agronomically, or with improved disease or insect resistance as they become available.

In 1971, a pilot test for distributing some of the better ADIN lines for international testing was initiated. The International Disease and Insect Nursery (IDIN) was the outgrowth of this and is comprised of the 25 best sources of disease and insect resistance in the ADIN along with 5 standard control cultivars (Table 1). By 1976, the IDIN was evaluated in about 20 locations throughout the world in addition to its exposure through plantings of the ADIN. Lines in the IDIN tend to remain in the nursery for several years; consequently, disease reactions from entries in the IDIN serve as good standards. At least one source of resistance for most of the known sorghum diseases is present in this nursery.

Specific Nurseries. In 1956, Stewart and Reyes grew the first head smut screening nursery near Bonneyview, Texas. Subsequently this nursery was moved to the Beeville Station where it has been grown continously. In 1969, head smut scoring began at Berclair for resistance to race 3, and for race 4 of *Sphacelotheca reiliana* at Edna in 1975. A program to monitor pathogen variation is continuing through the use of the Uniform Head Smut Nursery (UHSN) (Table 2). By growing these sorghum lines on a test field we can monitor the virulence patterns of the smut population. New races can be detected and the effectiveness of the resistance in genetic sources determined.

The downy mildew screening program began at College Station in 1965 and at Berclair in 1968. Downy mildew screening is conducted at several locations in South Texas by several private commercial firms as well. An international

Table 1. International Disease and Insect Nursery (IDIN) - 1979.

Entry No.	Designation	IS No.	Variety, Kind or Group	Plot No. RepI	Plot No. RepII
1	SC 103-12	2403	Caudatum	14	41
2	SC 110-14	12610 C	Zerazera	4	50
3	SC 170-6-17	12661	Zerazera	21	39
4	SC 170-14	12661 C	Zerazera	1	42
5	SC 173-12	21664	Zerazera	26	54
6	SC 175-14	12666 C	Zerazera	19	32
7	SC 326-6	3756	Nigricans	7	59
8	SC 414-12	2508	Cau-Kaf	11	49
9	SC 599-6 (9247)	17459	Cau-Nig (Rio)	28	46
10	SC 599-11E	17459	Cau-Nig (Rio)	3	38
11	SC 748-5	3552	Cau-Guin	15	53
12	SC 630-11E	1269	Caffr	6	40
13	R 5388	-	(SC599-6XSC110)der	25	34
14	6 BH 4613 (MR 4)	-	2566(3197X170-6)der	13	56
15	6 BH 4654 (MR 4)	-	2566(3197X170-6)der	18	31
16	B 447	-	B2752X(3197X170)der	2	43
17	Tx 2771 (MR 4-R line)	-	2566(3197X170)der	29	52
18	1790 E	-	(SC56XSC33)der	8	36
19	B Tx 623	12661 der	(B3197X170-6)der	23	48
20	R 1750	12661 der	(B3197X170-6)der	5	33
21	77 CS 1	-	(IS2930XIS3922)der	20	58
22	TAM 428	12610 der	Zerazera	10	45
23	Tx 430	12661 der	(Tx2536XSC170-6)der	27	60
24	GPR-148	-	CSV-5	17	35
25	CS 3541	-	CSV-4	22	55
26	QL3 sel	-	C. Kaf. der	30	44
27	B Tx 378	413	Redlan	9	57
28	B Tx 398	412	Martin	24	36
29	Tx 7078	415	Comb. 7078	12	47
30	Tx 2536	10542	Y. D. Fet. der	16	51

Table 2. Uniform Head Smut Nursery (UHSN) - 1979

Entry No.	Designation	Variety or Kind	Plot Numbers RepI	RepII
1	SA 281	Early Hegari	115	214
2	PI 48770	White Kafir	104	205
3	B TAM 618	Sm.Res.C.K.-60 der	111	219
4	Lahoma Sudan	Lahoma Sudan	120	204
5	Tx 7078	Combine	102	212
6	Tx 414	7078 der.	113	202
7	IS 12664C (Sc173-14)	Zerazera	108	218
8	SC 170-6-17(4267)(TAM2561)	Zerazera(IS12661 der)	106	213
9	SC 170-6 (FRM)(77CS2)	Zerazera(IS12661 der)	117	201
10	TAM 428 (SC110-9)	Zerazera(IS12610 der)	103	211
11	Tx 2536	Yel. End. Feterita	112	216
12	Tx 430	(2536X170-6) der	110	209
13	B Tx 3048	Redbine sel.	119	203
14	B Tx 3197	Comb. Kafir-60	107	215
15	SC 33-14 (IS12553C)	Durra	101	210
16	SC 324-12	Nigricans (IS2681 der)	114	207
17	SC 325-12	Nigricans (IS2562 der)	118	217
18	FC 6601	Spur Feterita	105	206
19	FC 8927	Dwf. Wh. Milo	116	220
20	SC 241-12E	Roxburghii (IS3911 der)	109	208

Number of packets are plot numbers - Plant in order by plot number in upper right hand corner, i.e., 101,102, etc. through 120, then 201 through 220. The test has been randomized and is to be planted in order of plot number on packets. Seed is treated with Captan and Chlordane.

downy mildew nursery program, containing a number of Texas sorghums is distributed by ICRISAT. Currently, the data suggests that different pathotypes of Peronosclerospora sorghi exist in different regions of the world and two are present in Texas.

Anthracnose resistance screening can be accomplished in Texas during some years, but has been done cooperatively at Georgia, Mississippi and Puerto Rico for about a decade. Anthracnose can be the most damaging disease of sorghum in the humid tropics. Fortunately some excellent sources of resistance are known. An International Sorghum Anthracnose Virulence Nursery was established in 1975 and through the use of this nursery we have found distinctly different host reactions in different geographic locations. Significant differences exist in host reactions in Georgia, Puerto Rico, Brazil, and West Africa.

Maize Dwarf Mosaic nurseries began in 1967 at College Station and San Antonio and are continued at Lubbock and College Station.

In 1978, there were 16 principle screening nurseries in Texas and cooperating states. Data were collected on a variety of foliage diseases following natural or artificial inoculation. Lodging nurseries which include damage caused by pathogens, environmental stress, insect and mite damage and genetic weakness in the host plant are evaluated annually in both line improvement programs and in the form of experimental hybrids.

D. Rosenow has developed novel and interesting approaches to the selection of lodging resistant sorghums. Resistance to stalk rots and lodging has increased dramatically in our trials during the past decade.

Sorghum Diseases

Among the better sources of information on sorghum diseases is the text by Tarr,(1962) Diseases of Sorghum, Sudangrass and Broomcorn. The Proceedings of the recent International Workshop on Sorghum Diseases, held at Hyderabad, India, (Williams et al 1980), will complement and update "Tarr". Two smaller handbooks on sorghum diseases are also of interest: Troublesome Grain Sorghum Diseases, Asgrow Seed Company (1974), and Sorghum and Pearl Millet Disease Identification Handbook by Williams et al (1978).

An annotated list of these diseases and their pathogens is presented in Table 4. However, we believe that it is more important to classify these diseases according to their potential vulnerability (Table 5). These vulnerability ratings are based in part on the known genetics of the host-parasite interaction (HPI), in some cases in the past history of the disease, and in others on other HPI's.

Stability of resistance can only be estimated. For example, resistance to Periconia root rot or Milo disease has not been lost in some 40 years of deployment. Similarly resistance to rust, leaf blight, and stalk rot appear quite stable, yet resistance to head smut and red rot has been overcome by changes in pathogen virulence. Consequently, for these diseases and their pathogens, we must continually monitor natural pathogen populations. This is done through Uniform Nurseries (See Tables 2 and 3). Current strategies anticipate changes in pathogen populations and complement host resistance by integration with other disease controls.

TABLE 3. International Sorghum Anthracnose Virulence Nursery (ISAVN) - 1979.

Entry No.	Designation	Plot No. Rep I	Plot No. Rep II
1	Brandes	101	212
2	Honey	102	211
3	MN 960	103	209
4	Rio	104	210
5	Wiley	105	213
6	B Tx 398	106	203
7	Tx 2536	107	214
8	TAM 428	108	206
9	SC 599-6 (9247)	109	201
10	SC 326-6	110	205
11	SC 167-14	111	204
12	SC 237-14	112	207
13	SC 328 C	113	202
14	SC 283 C	114	208
15	SC 748-5	115	215

Disease Nursery Design for Natural Inoculation

Foliar disease nurseries can be arranged in a number of ways; generally, we recommend frequent spreader rows or susceptible checks grown among candidate rows. Often spreader rows are planted before the test materials, which are then aligned perpendicularly in adjacent, leeward plots. By artificially inoculating these spreader rows, naturally occurring inoculum will be disseminated among the test materials. Examples of diseases examined in this manner include anthracnose, leaf blight, zonate leaf spot, downy mildew, and grey leaf spot.

Downy Mildew. High levels of oospores in the soil are required. To obtain these spores, a very susceptible sorghum or sudangrass is grown in the prior season. A rotation of susceptible sorghum with test materials in alternate years tends to keep high levels of inoculum uniformly distributed in the soil.

Head Smut. Virtually an identical program is possible for head smut. However, one should be cautioned that the growing of one susceptible sorghum cultivar tends to restrict the diversity of races in the smut fungus population. Diversity can be maintained by introducing smut sori collected from other host genotypes and by planting several sorghums with different reactions to different smut races in alternate years.

Epiphytotics of P. andropogonis. Where the objective is to screen sorghum varieties for disease resistance, difficulties in identifying,

2D

TABLE 4. Diseases and their pathogens.

Disease	Pathogen
Anthracnose (Foliage, Head)	Colletotrichum graminicola
Bacterial leaf spot	Pseudomonas syringae
Bacterial leaf streak	Xanthomonas holcicola
Bacterial leaf stripe	Pseudomonas andropogoni
Charcoal rot	Macrophomina phaseolina
Covered smut	Sphacelotheca sorghi
Crazy top	Scheropthora macrospora
Downy mildew	Peronosclerospora sorghi
Fusarium stalk rot	Fusarium spp.
Grey leaf spot	Cercospora sorghi
Head smut	Sphacelotheca reiliana
Leaf blight	Helminthosporium turcicum (Exserohilum sp.)
Leaf spot	Phoma insidiosa
Leaf spot	Ramulispora sorghicola
Long smut	Tolyposporium ehrenbergii
Loose smut	Spacelotheca crenta
Maize dwarf mosaic (Foliage, Stand)	Maize Dwarf Mosaic Virus (MDMV)
Milo disease	Periconia circinata
Pink root	Pyrenochaeta terrestris
Pokkah-bong	Fusarium spp.
Phthium root rot	Pythium graminicola
Red rot	Colletotrichum graminicola
Rhizoctonia stalk rot	Rhizoctonia solani
Rough spot	Ascochyta sorghina
Rust	Puccinia purpurea
Sooty stripe	Ramulispora sorghi
Sugary disease	Sphacelia sorghi
Target leaf spot	Helminthosporium sorghicola
Witchweed	Striga hermonthica, S. asiatica
Zonate leaf spot	Gleocercospora sorghi
Grain mold	Fusarium spp., Curvalaria spp., Altemaria spp.

TABLE 5. Characteristics of certain key sorghum disease problems.

Major Disease Problems in Sorghum	Genetic Nature of Host-Parasite Interaction	Degree of Genetic Vulnerability
Downy Mildew	General and Specific	Intermediate
Head Smut	Specific	High
Maize Dwarf Mosaic	General	High
Stalk Rots:		
Charcoal	General	Low
Fusarium	Intermediate	Intermediate
Red Rot	Intermediate	Intermediate
Foliage Diseases:		
Anthracnose	Intermediate	High
Bacterial Stripe	General	Low
Cercospora Leaf Spot	General	Low
Leaf Blight	Intermediate	Low
Rust	Intermediate	Low
Zonate Leaf Spot	General	Low
Root Rot:		
Pythium	Unknown	Low
Grain Mold	General	Low

preserving, and inoculating with the pathogen can sometimes be avoided by the use of naturally-infected spreader rows. One must have a sorghum variety extremely susceptible to the disease of interest, not unduly susceptible to other diseases which occur locally, and available in quantity from a region where the disease of interest does not prevent its reproduction. In the case of bacterial stripe, such varieties are available.

Frequency of spreader rows among candidate and check rows may vary from 1 in 6 to 1 in 2, depending on the intensity of exposure desired. Test materials adjacent to spreader rows which for any reason do not show severe symptoms should be disregarded; if a statistical field plot design is used, it should allow for missing plots.

Culture of Plant Pathogens

Culture of C. graminicola. Insolations are made from diseased plant material (either leaves or stalks) by plating surface-disinfected tissue onto nutrient agar (pH 5 to 6 with 100 ppm streptomycin sulfate to inhibit bacteria). Cultures cannot be identified as Colletotrichum graminicola by gross inspection; though typically greenish-black, colonies range through white to grey and black with a few pink and orange. Texture may be thick, loose and cottony, or thin and felty; aerial growth is not necessarily abundant. Conidia, too, may be nearly absent; if present, they are usually pink en masse and often falcate. The wide variations in their form do not typify specific isolates. Spores tend to form at the margin of the colony and to germinate to form appressoria. Setae may occur. Temperature optima of 28°C (USA) and 30° to 32°C (India) have been reported. Therefore, cultures are identified and checked for purity by microscopic examination. Single sporing, which can be accomplished by dilution, helps maintain sporulating activity and pathogenicity. Cultures long maintained by mass transfer tend to lack these functions. Suspect isolates are checked for pathogenicity before large-scale screening tests are undertaken.

Culture of Gleocercospora sorghi. Use lima bean agar, or oatmeal agar diluted to half strength. Potato dextrose agar is also acceptable, giving white or pink colonies. V-8 juice agar gives excellent sporulation. (V-8 is a pasteurized puree of tomato, carrot, beet, parsley, watercress, and spinach with ascorbic acid added which is commercially available in the U.S.). Sorghum juice agar gives sclerotia but not conidia.

Surface-sterilize leaf fragments bearing sclerotia or punches from margins of ongoing infections in living leaves. Transfer at 3-day intervals at 28°C, drawing inoculum from the margin of the colony and checking microscopically for the Gleocercospora conidia. Once a pure culture is obtained, wash it with sterile water and transfer conidia rather than mycelium; this aids in retaining the capacity to produce conidia and sclerotia, which is easily lost (usually, but not always, conidia are lost before sclerotia; passage through sorghum does not restore ability to sporulate; pathogenicity is lost separately, but isolates producing all organs are more likely to be pathogenic). When a culture with abundant sclerotia is obtained, let it dry and put it away; the isolate may be preserved a year or more in this way.

Viable sclerotia can be produced on some culture media as well as on sorghum leaves. Experiments usually begin with dry material, though no requirement for drying has been established. Following the experimental stresses, the sclerotia may be placed on moist paper in closed dishes at 28°C and examined daily for sporodochia produced. Most germination occurred

on days 3 through 7 in one such experiment. Since results were more characteristic of the day a specimen was put in the moist chamber than of its previous treatment, one should replicate such experiments sequentially.

Culture of Exserohilum turcicum. E. turcicum grows well on V-8 juice agar. If the agar is autoclaved in water and the can of V-8 juice is then opened (with sterile technique) and added, hydrolysis may be minimized and a firmer gel obtained. Various recipes specify 2 to 5 g $CaCO_3$ per liter; this raises the pH (6.0 is suitable) and precipitates colloidal substances which would otherwise cloud the medium and impair observation. An inoculating needle may be scraped across a mature lesionaand then used to streak the conidia onto a plate of water agar, after which either conidia or hyphal tips may be transferred to separate V-8 agar plates to give isolates of single haploid genotypes. Each such isolate will show one of two mating types when used in an attempt to produce the perfect stage. If the fungus is to be isolated from leaf, debris, or soil specimens, the $CaCO_3$ may be omitted and 0.1 g streptomycin and 1.13 g rose bengal per liter may be added.

If a defined medium is desired for E. turcicum, the following is suitable. It is based on a medium of Malcha and Ullstrup known to support rapid growth if 0.3 per cent casein hydrolysate is present, but here the more prominent amino acids of casein are substituted for it. Some of them may be unnecessary. The 15 g of crude agar is replaced by 0.5 g of purified agar, which must be sterilized separately from the salts.

KH_2PO_4	1.	g	glutamic acid	0.7	g
$MgSO_4$	0.5	g	proline	0.3	g
$FeNO_3 \cdot 9H_2O$	0.0014	g	leucine	0.2	g
$ZnSO_4 \cdot 7H_2O$	0.0009	g	aspartic acid	0.2	g
$MnSO_4 \cdot 4H_2O$	0.0004	g	valine	0.2	g
lactose	37.5	g	serine	0.2	g
'Ionagar'	0.5	g	isoleucine	0.2	g
threonine	0.2	g	arginine	0.1	g
phenylalanine	0.1	g	methionine	0.1	g

(PH is about 4. Adjust to 6 using about 0.2g KOH).

When mating types of the imperfect stage of E. turcicum are separated and recombined into Setosphaeria ascospores, which form the perfect stage, there is a chance that some of the recombinants may lack one of the enzymes necessary to support growth on a minimal medium. To avoid loss of these auxotrophs (which would be useful as genetic markers), ascospores may be germinated on a 'catch-all' medium:

Difco Czapek's medium	35.0	g
agar	15.0	g
Difco malt extract	7.5	g
Schwartz sodium nucleate	0.01	g
hydrolyzed casein	0.025	g
Sanderson's trace elements	1.0	ml

(The trace elements of the defined medium above may be substituted). (Not all casein hydrolysates are thoroughly hydrolyzed; BBL 'Acidicase', Difco 'Casamino acids', and Nutritional Biochemicals acid and enzymatic casein hydrolysates each supported an isolate of Puccinia graminis tritici which was fastidious in this respect.

Setosphaeria turcica is thought to require a natural substrate: the culm (stem with sheath) of a grass host. Luttrell, who discovered it, surface-sterilized mature dry barley straw with propylene oxide (1 ml per liter of air, for 24 hr) to avoid loss of heat-labile components, then partially embedded segments in Sach's agar: (1 liter)

$CaNO_3$	1.0	g
K_2HPO_4	0.25	g
$MgSO_4$	0.25	g
agar	20.0	g

Chlamydospores germinate on water agar; further development requires abundant sucrose, thiamine, biotin, and suitable inorganic salts. Dispense it into tubes, and also apply small amounts to cover glasses, forming thin films that may be used to isolate individual sporidia.

Slit the agar along one edge of the coverslip, forming four square stalls. Streak a few chlamydospores along the opposite edge and let them germinate. Move one monosporidium into each stall (using a micromanipulator with a drawn glass needle if available, or using a hand tool made by breaking a mature sorghum peduncle, slitting it lenghwise, and whittling the broken end to leave a single vascular bundle). Cut off each square of agar and transfer (with sterile technique) to the slanted surface of the medium in a tube. After growth occurs, if the culture is not soon to be transferred, add sterile mineral oil to aid preservation of shelf life.

Culture of Bacteria

Unknown bacteria are usually grown on Nutrient agar (beef extract, 0.3%; peptone, 0.5%; and agar, 1.5%). Mixed cultures are separated by streaking (drawing a zigzag line on the agar surface with an inoculating needle, then selecting one of the smallest symmetrical colonies for transfer); or by dilution plating (suspending bacteria in sterile water or lukewarm, not over 40°C, agar medium, serially diluting this liquid (e.g., five ten-fold dilutions). and pouring some of each dilution in agar into a sterile Petri plate).

Detailed Identification of Bacteria Pathogens of Sorghum. Pure culture of one of the three important facterial pathogens of sorghum can be roughly distinguished from the other two. P. andropogonis has flagella at both ends of the cell, while the other two have them only at one end, and it alone does not liquefy a gelating medium. P. syringae alone fluoresces under ultraviolet light. Colonies of X. holcicola on nutrient agar are yellow, whereas those of P. andropogonis are white and those of P. syringae, greyish-white. This diagnosis cannot be considered final (see below).

Bacteria are noted for their inducible enzymes, which appear only after exposure to the substrate for a relatively long time; therefore, a culture should be 'conditioned' as a fresh (24 hr) transfer onto a specified medium prior to biochemical tests. For most of the following tests, this is King's Medium B:

Proteose peptone No. 3	2.00	per cent
Bacto agar	1.50	
Glycerol, C. P.	1.00	
K_2HPO_4	0.15	
$MgSO_4 \cdot 7H_2O$	0.15	

(Adjusted to pH 7.2)

TABLE 6. Reaction of sorghum bacterial pathogens on King's medium.

Pathogen	Gelatin liquefi-cation	Nitrate reduction	H_2S pro-duction	Tween 80 lipolysis	Malonate utiliza-tion	Acid from glucose
Pseudomonas andropogonis	-	-	-	-	-	-
P. sorghicola	+	+	+	-	-	?
P. alboprecipitans	+	-	-	+	+	?
P. rubrilineans	-	+	+	+	-	?
P. rubrilianeans, nitrate reductaseless	-	-	+	+	-	?
P. rebrisubalbicans	-	+	-	-	+	?
P. rubrisubalbicans, Haywood strain	-	-	+	-	+	?
P. floridana	(+)	-	+	+	+	?
P. syringae	+	(+)	?	?	?	+
X. holcicola	(+)	?	+	?	?	-

The YS broth, used in three of the tests, is as follows:

Yeast extract	0.50 per cent
(but only 0.08 per cent for the malonate test)	
NaCl	0.50 per cent
$(NH_4)H_2PO_4$	0.05
K_2HPO_4	0.05
$MgSO_4 \cdot 7H_2O$	0.02

Instructions for six tests follow, and the expected results are given in Table 6. Where the specimen is known to be a non-fluorescent *Pseudomonas* spp., the gelatin liquefication and acid from glucose tests may be omitted.

Gelatin liquefication. Make up nutrient gelatin (beef extract, 0.3 per cent; peptone, 0.5 per cent; and gelatin, 1.5 per cent) in tubes. Stab-inoculate, and observe two weeks for hydrolysis of the gel.

Nitrate reduction. Make up liquid medium; sterilize in flasks, at least three per specimen:

NaCl	0.50 per cent
yeast extract	0.50
sodium succinate	0.20
KNO_3	0.10
Kh_2PO_4	0.05
K_2HPO_4	0.05
$MgSO_4 \cdot 7H_2O$	0.02

Make up test solutions of sulfanilic acid and of N,N^1 - dimethyl - 1 - naphthylamine, each in 28.5 per cent (v/v) glacial acetic acid. Inoculate;

incubate with shaking. After 2, 5, and 7 days, add a few drops of each reagent to a culture. A red color indicates nitrate reduction.

Hydrogen sulfide production. To YS broth (see above), add 0.01 per cent cysteine hydrochloride (use a 1 per cent stock solution in ethanol) after sterilizing in flasks. Inoculate, and hang a sterile strip of lead acetate paper from the closure of each flask. Shake gently, so as not to wet the papers, for two weeks. Observe strips after the third day for a black residue of lead sulfide.

Tween 80 lipolysis. Prepare medium in plates: peptone (Difco), 1 per cent; NaCl, 0.5 per cent; $CaCl_2 \cdot H_2O$, 0.01 per cent; Tween 80 (Atlas), 1 per cent autoclaved separately; final pH 7.0 to 7.4. Streak or spot-inoculate and incubate 1 week at 30°C. Observe after the third day for an opaque halo or cleared zone indicating lipolysis.

Acid from glucose. Prepare medium in flasks: 1 per cent peptone, 0.5 per cent glucose, and 0.0001 per cent bromcresol purple (use a 1.5 per cent stock solution in alcohol). Incubate two weeks at 27°C with shaking. Observe after the third day for color shift from purple to yellow, indicating acid production.

Malonate utilization. Prepare YS medium in flasks, adding 0.2 per cent sodium malonate and 0.0016 per cent bromthymol blue, and adjusting pH to 6.8 with NaOH. Inoculate, incubate 10 days with shaking, and observe after the third day. Prepare a standard by adjusting an uninoculated flask to pH 7.3. Green to blue colors indicating pH 7.3 or higher demonstrate consumption of the malonate.

Inoculum storage. Isolates of *C. graminicola* appear to retain their pathogenicity when stored on potato dextrose agar (PDA) at 10°C, allowing repeated transfers without apparent loss of virulence. V-8 broth cultures may be grown and refrigerated for several weeks before use but should be used immediately upon return to room temperature.

Preservation of bacteria. Ordinary cultures of *P. andropogonis* do not survive freezing. Most bacterial species survive when cells are suspended in water containing 0.2 per cent sodium glutamate and are lyophilized. For freezing not followed by drying, no methods have been verified for these particular bacteria, but suspension in 50 per cent glycerol or 10 per cent dimethyl sulfoxide may be tried. To preserve bacteria in leaves, try letting the plants wilt (though not to the point of death), chopping them, vacuum-infiltrating with 50 per cent glycerol, and storing in a food freezer.

Inoculum of *C. graminicola* for foliar inoculation. Grow the pathogenic isolate in large Ehrlenmeyer flasks of V-8 broth. Cultures are agitated continuously at room temperature. After approximately five days, colonies begin to clump into round balls and broth clarifies. At this point, check microscopically for purity and an abundance of conidia. Strain cultures through several layers of cheesecloth and dilute filtrates to approximately one thousand conidia per milliliter.

Inoculum of *G. sorghi* (from agar culture). A whole, fresh culture in lima bean agar, diluted at least 20-fold with water, is more effective than a spore suspension. Dilution 80-fold or more increases varietal selectivity

without greatly decreasing inoculation efficiency on a susceptible variety. Addition of 2 per cent gelatin increases efficiency (with unknown effects on selectivity). Following inoculation, plants (or detached leaves) must be maintained in a fully water-saturated atmosphere for 20 hours.

Inoculum Preparation - Methods Not Requiring Pure Cultures

Inoculum of Colletotrichum graminicola is commonly maintained satisfactorily from season to season in dried, infected host tissue, particularly in leaves. While it may be impossible to separate C. graminicola from other foliar pathogens in this manner, merely grinding or chopping infected leaves into small bits (less than 1 cm square) and dropping them into whorls of sorghum seedlings as inoculum greatly simplifies inoculation of large numbers of plants in the field.

Air-dry infected leaves at room (or other moderate) temperature, preferably using an extremely sensitive sorghum cultivar so that anthracnose will develop before other foliar diseases. These leaves may be used immediately or stored in a dry condition for several months. Experimental confirmation that inoculum will survive under local conditions is prudent. In Texas, Mississippi, and Georgia, (USA), inoculum is routinely stored from one growing season to the next, a period of nearly 6 months. Prior to use, the inoculum may be ground in a mill or manually cut into small bits. These are dusted by hand into the whorls of test plants in the field. In this position on the plant, inoculum will sporulate and infect during the next favorable period.

Zonate leafspot (Gleocercospora sorghi), like most other foliar diseases, may be established artificially by mincing dry leaves (bearing necrotic areas and sclerotia) and sprinkling the fragments into the whorls of 30-day plants. Activation of the sclerotia requires at least 3 days of continuous high humidity.

Inoculation with G. sorghi-infected grain or meal requires a pure culture, is about as efficient as the liquid inoculation methods mentioned below, and may prove more manageable when a large crew is required to inoculate several hectares. Autoclaved sorghum or other grain should be inoculated with a pure culture of G. sorghi which produces all organs (see below), and incubated at 27°C with occasional agitation until all grains bear mycelium. Spread the grain out to dry at room temperature (contamination will occur, but other sorghum pathogens have no competitive advantage in this medium). Put the grain through a corn grinder, then sprinkle the meal into the whorls of 30-day sorghum plants. This procedure will work only if rains occur sufficiently often topprovide the proper environment for germination and penetration by G. sorghi. Inoculation may be carried out using whole grains. In this case, the permissible delay before an adequate wet period is extended somewhat because the grains, unlike the meal, tumble back down into the whorl as they are exposed by leaf growth.

Since no one has published studies on rough leaf spot or on A. sorghina under artificial conditions, no proven techniques can be described. Presumably, chopped infected leaves which have not been kept too long may be dropped into whorls as inoculum. A. sorghi, which appears to be very similar, has been studied; suitability of these determinations for A. sorghina should be verified.

A. sorghi survived a year or more on leaves in the laboratory, though the pycnidia lost infectiousness. Pycnidia survived 36 hours at 48°C. However, pycnospores did not survive 6 months at room temperature nor 10 minutes at 48°C.

Leaves, especially those on older plants, were infected when sprayed with suspensions of fresh pycnidia or pycnispores. When exposed to relative humidity over 97 per cent between 25° and 38°C (31° optimally), viable spores produced a germ tube from each cell. Mycelium developed on 2 per cent potato dextrose agar, provided the temperature was well below 42°C. Black structures, resembling pycnidia but twice as wide and without spores, formed in culture and could be used to infect leaves.

Leaf blight (E. turcicum), like other leafspotting diseases, can be transmitted by dropping fragments of infected leaves into the whorls of test plants. Inoculum may be stored at 0°C, with less than 50 per cent relative humidity, for 12 years; at 0°, 86 per cent, or 25°, 50 per cent, it lasts about 4 months, whereas at a tropical ambient (30°C, relative humidity over 77 per cent) transmissibility fails in less than a month.

Inoculation should be planned for a time of cool, humid weather, since disease development is rapid at 20°C. Penetration is completed in 18 hours, so if the humidity variable is to be removed, plants may be sprayed with water throughout this interval. At lower temperatures, pathogen performance (sporulation) will decrease. Minimum conditions for sporulation of E. turcicum from a lesion on sweetcorn are 7 hours at 15°C with humidity near 100 per cent.

Since the conidium (with chlamydospores) is the overwintering unit of E. turcicum, spores may be separated from leaves if this is convenient. Conidia may be washed off, or the leaves may be homogenized and strained, Cultures on agar may be homogenized and used without fractionation. Liquid inoculum is sprayed onto the plants.

As an obligate parasite, Peronosclerospora sorghi gives no opportunity for inoculum production in culture. Inoculum originates from leaves and consists of either oospores or conidia. (An exception is mycelial inoculum on seeds with glumes, which may persist up to a month on seed kept at 3°C with at least 18 per cent moisture). Either oospores or conidia can initiate systemic infection in germinating seeds; conidia deposited on older seedlings can initiate local infection, but systemic infection is possible only with germinating seeds. They are most susceptible at shoot emergence, about one day after hydration. Dr. Jeweus Craig has developed a conidia inoculation technique for identifying resistance to sorghum downy mildew (Craig 1974). For inoculation with conidia seed at shoot emergence are placed embryo side up on moist paper in Petri dishes; infected leaves showing sporulation (white downy mat on abaxial surface) are gathered, washed, and promptly stretched across the rim of the dish and secured with the top. The dishes are held at 21°C; about 8 hours after the leaves have been removed from their plants, conidia are released. After another 24 hours, the seedlings are ready to be removed and planted.

Emerged seedlings (arranged as rows in flats) may be inoculated for local infection by gathering infected, sporulating leaves in the late afternoon, resting these (abaxial surface downward) across the rows of seedlings, and covering with polyethylene sheets. After a night at 20° to 28°C, up to 7 out of 10 seedlings may be infected, but one cannot expect uniformity (see below).

The best conidial inoculations result from care to maintain completely water-saturated air throughout the system. Arrange, from bottom to top. a large plastic tray; potted 2-leaf sorghum seedlings; a second, identical tray, inverted and with most of its bottom cut away; hardware cloth (welded mesh with openings about 1 cm); a single layer of cheesecloth; infected, sporulating sorghum leaves, cut into 1-2 cm segments and arranged, abaxial side down,

to give thorough coverage; a layer of seed germination paper (equivalent to about 20 layers of facial tissue - do not substitute crude paper towels, which may be toxic) soaked with water; and a plywood lid. Incubate at 18°-22°C. Conidia are discharged on straight trajectories, sometimes giving images on the leaf segments and of the cheesecloth projected onto the seedlings. Uniform inoculation may be assured by agitation with water-saturated air. Arrange a vacuum-pressure pump; a needle valve, adjusted to five ca. 0.3 atm gauge pressure at the system outlet; a water bubbler; about 10 m of 1 cm copper tubing, coiled in a tank of water maintained at 20°C; a forked pipe; and two rubber hoses with ends plugged and walls drilled near the ends to discharge the air radially, extending through the lid and the hardware cloth to the tips of the seedlings. Use great caution in substituting mechanical agitation, since its work would appear as heat which could lower the humidity significantly. The layer of cheesecloth, through which the conidiophores protrude, is no barrier to conidial discharge and has been found essential for a good yield of conidia; this is probably because it provides a source of moisture within 1 mm of each conidiophore.

Oospores may be recovered directly from infected mature leaves or indirectly from infested soil. Leaves are used where the origin and age of the oospores must be known (e.g., for investigating conditions of oospore survival and germination), though one cannot expect to infect more than half of the seedlings this way. Since the oospore diameters fall in the 30 to 50 micron range, a Wiley Laboratory Pulverizing Mill is fitted with a 74-micron sieve (200 mesh). Infected leaves are shredded, then ground in this mill and mixed with seed to be planted. Though some infection results from inoculum placed at a distance from the seed (up to 7 cm), this is less effective, as is inoculum previously stored several months at room temperature. Frozen inoculum may be kept at least three years.

For most applied research, the whole infested soil is the best inoculum. Over 80 per cent of the plants become infected, and some infested soils may be diluted 100-fold without loss of potency. Soil temperature should be 18°C, maintained in a constant-temperature tank or found in the field at an appropriate plant date. It is prudent to plant on a series of dates at one- or two-week intervals. Where reproducibility is not required, seeds may be planted in pots kept in the lab; the lower and upper temperature limits for infection are approximately 12° to 30°C, respectively.

When germination of oospores from the soil population is to be studied (e. g., to evaluate the effects of weathering on spores), it is convenient to concentrate them. Dry soil is pulverized in a Waring blender and the spores are recovered in the dust, e.g., by drawing some dusty air into a large syringe and expelling it through a filter (Swinney adapter). For larger quantities, one might operate the blender under a large inverted box, then slide the box to a clean surface and let the spores settle.

Sphacelotheca reiliana, like many of the smuts, is intermediate between obligate and facultative parasites: its chlamydospores (teliospores) germinate on suitable media to yield sporidia and a yeast-like growth, but the tissues typical of the parasite (including the chlamydospores themselves) are not reproduced. The sexual process can be managed in vitro, but the resulting inoculum can be returned to the host only in a highly unnatural manner. Pathogenicity is easily lost in culture.

Inoculum Preparation - Methods Utilizing Pure Cultures

Foliar Inoculation With C. graminicola. These inoculations are usually done when humidity is high and sorghum is at the early boot stage of growth (40 to 50 days postemergence). A liter or larger capacity container which can be pressurized and fitted with a hypodermic syringe is recommended for field inoculations. A football pump may be substituted. Plants are inoculated by injecting the conidial suspension into the whorl so as to expose as many young leaves as possible to the inoculum.

Injection of pure C. graminicola inoculum through a needle puncture to the base of the whorl assures infection of all sorghum varieties, at least in the vicinity of the wound. When the inoculum is squirted into the whorl from the top without wounding, there are some escapes in every variety, but some varietal resistance is revealed statistically. Spray inoculation of seedlings is effective, provided the humidity requirement is strictly observed.

Inoculation With Sporidia of S. reiliana. Transfer each isolate to a flask of potato dextrose broth which can be mounted on a rotary shaker or other device to provide gentle, continuous motion. After three days at room temperature, make inocula by mixing broth from pairs of cultures in all possible double combinations (i.e., 4 monosporidial cultures yield 6 inocula, 5 yield 10, and n yield (n x (n-1)/2). Commonly, all sporidial cultures from 3 individually germinated teliospores are pooled for use as inoculum (See Edmunds 1963). Immediately after mixing, draw inoculum (1 ml per plant to be inoculated) into a syringe with a 24 or 26 gauge needle. Use 4-week-old plants (i.e., 7 to 10 leaves, whorl, and differentiated growing point). Place the inoculum immediately below the point of differentiation; rehearse this using a syringe with dye and plants which may be dissected. Always test inocula from new isolates on host plants of the variety which gave rise to those isolates. Retain only those inocula which do not cause smut when paired with certain others. Whole teliospore isolates cannot be saved; new isolates are needed for each trial.

Inoculation with Chlamydospores of S. reiliana. Immature chlamydospores germinate readily, but those which have fully matured in the sorus (the usual natural inoculum) respond to weathering and germinate sporadically throughout the year in the soil. Shelf life is about 5 years. Although spores mixed with soil and applied to dry seed have produced some infected seedlings, spores mixed into the planting bed have shown much less infectivity to seeds sown one or two weeks later. Perhaps the greatest infective efficiency occurs when the chlamydospores have been in the soil 4 months, with considerably less at 8 months. Nonetheless, a field planted to susceptible sorghum once a year suffers inoculum buildup, and several years in non-susceptible crops do not disinfect it. With moderate soil moisture (15 per cent by weight), infection can occur at any temperature from 12° to over 36°C. Higher soil moisture narrows the permissible temperature range to 20° to 34°C. The optimal temperature is 28°C.

Inoculation of Stalk Rots (Macrophomia, Fusarium, Colletotrichum). Clear toothpicks by boiling in very dilute potassium hydroxide, rinse, add nutrients, sterilize and cool, add inoculum, and incubate about 5 days (with agitation, if possible). Picks are placed in stalks, 2 nodes above the ground line, approximately 10 to 20 days after flowering. Wounding to insert the picks can be done with a shortened ice pick. This can be made by driving a nail into a hand-sized wooden dowel, filing off the nail head, and sharpening the point. Evaluation can be on a 1-5 scale, based on the extent of damage within and

among internodes. Rot extending through 2 or more internodes generally indicates a susceptible reaction.

Charcoal and Fusarium stalk rots will develop under stress of low moisture and high temperature. Both diseases tend to develop more rapidly in dense plant populations.

Infested toothpicks bearing C. graminicola may be inserted into the peduncles of sorghum in the boot stage of growth, as an alternative to inoculating one of the lower internodes. If a yes-or-no answer is desired, head blight may be scored much more quickly than stalk rot, and the plant may be left to mature. Stalk rot is easier to quantitate. No gene is known to prevent blight in a head with a rotted peduncle, so the stalk rot score is useful even where peduncles are naturally infected directly. On the other hand, no such gene is likely to be found if head blight resistance is evaluated in the lower stalk. Toothpicks are used because they are available and inexpensive, because they can be prepared and stored, and because they can be found in the sorghum plants after a two-month interval.

The inoculation and culture methods given for Gleocerospora (see preceeding discussion) are also suitable for Ramulispora. In addition, because sclerotia of the latter survive in the soil, a method to cover them has been developed. Dry soil, 25 to 50 g, is ground in a mortar, then washed through a 0.25 mm (80 mesh) screen followed by a 0.047 mm (300 mesh) screen on which the sclerotia (and other particles of similar size) are retained. This residue is washed off the screen with a saturated solution of ammonium sulfate in water. The suspension is allowed to separate at 1 g in a 125 ml Ehrlenmeyer flask. Sclerotia come to the top. These are decanted, collected on a filter, washed with water, resuspended in about 1 ml, and placed atop a step-gradient consisting of 5 ml water atop 5 ml 70 per cent sucrose (w/w) in a tube for a swinging bucket rotor. When the rotor is spun to give 2000 g for 3 min, the sclerotia settle to the top of the 70 per cent sucrose layer and move no farther. They may be collected by introducing more 70 per cent sucrose to the bottom of the tube, or by decanting and filtering. Viable sclerotia of other species may be present along with Ramulispora. One can identify them, using the microscope. Treatment of sclerotia (or sporodochia) with one per cent NaOCL for 1 minute serves as both a surface sterilizing procedure and a germination stimulant.

Inoculation of Bacteria. Pseudomonas spp. may be inoculated to young sorghum leaves by suspending a fresh culture in water, drawing it into a 5 ml syringe fitted with a 2 cm length of 1 cm rubber tubing, and pressure-infiltrating the leaf from the abaxial side while supporting the opposite side with the thumb. Injury doe to water-soaking and bruising is evident immediately. Hold the inoculated plants, together with checks infiltrated with water or heated (60°C) inoculum ten days in an illuminated moist chamber. Local infection should be evident after 2 days, and spread through interveinal regions occurs later.

Status of International Nurseries

The Texas Agricultural Experiment Station developed an International Disease and Insect Nursery (IDIN) in 1972. This nursery contains our current elite sources of resistance to diseases, insects, lodging, and grain weathering. Lines are included with resistance to downy mildew, head smut,

anthracnose, maize dwarf mosaic, rust, Fusarium head blight, grey leaf spot, zonate leaf spot, charcoal rot, and grain mold. Insect and arthropod pests for which resistance sources are included are greenbug, midge, corn leaf aphid, and Bank's grass mite. The nursery is updated each year to include improved materials as they are developed and identified.

This nursery is distributed worldwide and provides information on performance of materials under different and harsh conditions throughout the world. It also serves as a germplasm source in which sorghum workers can select lines for use in their international or national sorghum improvement program.

In the Spring of 1977, a cooperative arrangement was agreed to between ICRISAT and TAES, whereby we could submit entries into some of the International Testing Programs of ICRISAT. We can enter materials either by sending seed to ICRISAT for later inclusion after seed increase, or by asking for entries and test locations and then sending the seed directly to each cooperator.

International ICRISAT nurseries for which such arrangements have been established are the ISLDN (leaf disease), ISDMN (downy mildew), ISGMN (grain mold, midge nursery, and yield nurseries. This agreement with ICRISAT allows the evaluation of our material in key international locations and a comparison with the other elite lines available internationally.

This past spring (1979) we began evaluation of selected hybrids and lines for yield and adaptation at several sites across Africa, Southeast Asia, and Central and South America.

Other nurseries we are involved with internationally are the Anthracnose (ISAVN) and Head Smut (UHSN) nurseries. Cooperative international work also exists with evaluation of food-type sorghums.

Close cooperative ties exist between TAES workers and sorghum researchers in developing countries, and visits have been made to evaluate nurseries, problems, and breeding materials. Some of the countries to which one or more visits have been made include: India, Thailand, Senegal, Upper Volta, Nigeria, Egypt, Niger, Sudan, Ethiopia, Mexico, Venezuela, Colombia, Brazil, and Guatemala.

Sorghum Disease Evaluation - General Consideration

A variety of systems for taking disease notes are suggested. Within each nursery a selected set of susceptible controls is needed. These controls provide the researcher with a valid basis for comparing data from one cropping season to the next. Lines used as controls vary in their disease reactions.

Date of flowering of each entry is very important to record, since in many cases, disease damage is related to maturity. The date when approximately 50% of the heads have begun to flower should be recorded as the date of flowering. Accurate flowering notes are best obtained by taking them every 3 to 4 days.

TABLE 7. Disease reactions of selected sorghum lines.

Sorghum Line	Susceptible to:	Resistant or Tolerant To:
Tx 7078	Head smut, races 1,2,3,4 Rust Zonate leaf spot Grey leaf spot Downy mildew	
BTx 398	Anthracnose	MDMV (tolerant)
BTx 378 (Redlan)	MDMV	
QL 3		MDMV (resistant)
TAM 2536	Downy mildew	Anthracnose
Sweet Sudan	Northern leaf blight Southern leaf blight Downy mildew (foliar phase)	

Notes on diseases not listed below should be taken and added to the data sheet. Also, notes on items such as bird damage may be made along with the species involved. Differences in reaction to other characteristics such as drought, heat, cold, flooding, and soil factors such as pH, and aluminum toxicity should be recorded if the opportunity arises. When a selection appears to segregate for disease reaction, this information is very useful to the breeder and should be recorded. For example, 4, 2s means that most individuals were rated 4 and most of the rest were rated 2. The apparent segregation ratio should be recorded in a comment column. The rating 1-4s means that individual ratings 1, 2, 3, and 4 occurred with more ones and twos than threes and fours. Explaining fully: let the rating format be a convenience rather than a restriction.

It should be pointed out that a "0" rating in the rating systems refers to any situation where a rating may be made on an individual plot and <u>does not</u> refer to a level of resistance.

Sorghum Disease Evaluation - Specific Considerations

<u>Downy Mildew</u>. If sorghum downy mildew is present, data should be collected on the incidence of systemically infected plants as well as the severity of foliar infection (local lesion phase).

Systemically diseased seedlings and plants will develop striped or streaked leaves with alternate green and yellow or chlorotic stripes. Down (asexual sporulation) is common-to-abundant on the lower leaves of these plants during humid weather with moderate-to-cool temperatures.

The local lesion phase develops extensively on sudangrasses under

favorable conditions and could develop to a lesser extent on many of the entries. Disease evaluations can be made for this phase as for other foliar diseases.

Head Smut. Determine the incidence of disease in each plot at or about the soft dough stage of growth.

Maize Dwarf Mosaic Virus (MDMV) or Sugarcane Mosaic. Data may be obtained at the boot stage of growth or after flowering, based on the following rating system. From flowering on, the mottling and chlorosis usually disappear; thus, one cannot determine 2 or 3 in late growth stages. Cool temperatures of 60°F or lower make symptoms much more severe, especially on susceptible lines. The red leaf symptom often does not develop in the absence of such cool temperatures. Note: Certain genotypes will show chlorotic and stunted plants without significant necrosis. These should be rated a 5. Incidence may be important and may be reported as a percentage. This is especially important when incidence is near 100% in some entries.

0 = No evaluation possible
1 = No apparent symptoms
2 = Leaves with mottling only
3 = Mottling and significant chlorosis
3.5 = Mottling with slight leaf necrosis
4 = Mottling with significant leaf necrosis (red leaf on most genotypes)
4.5 = As above with stunting
5 = The above accompanied by severe stunting or death

Foliar Diseases. A rapid evaluation of foliar disease incidence and severity can be made for the following diseases: grey leaf spot, sooty stripe, zonate leaf spot, rough spot, bacterial stripe, physiological or genetic spotting, leaf blight, rust, downy mildew, and others.

0 = No evaluation possible
1 = Resistant: disease inconspicuous or if present on an occasional plant
2 = Disease present (over 50% prevalence with low severity; apparently causing little economic damage)
3 = Disease severe (100% prevalent, estimated leaf area destroyed up to 25%; disease appears of economic importance)
4 = As in 3 but over 25% of leaf area destroyed
5 = Death of leaves or plants due to disease

Stalk Rot. Data should be collected at/about physiological maturity or when the grain has completely matured. (Note: At times, following artificial inoculation, little disease develops. Nevertheless, good comparative data can usually be obtained by recording differences among entries within inoculated internodes). Several longisections should be whittled away to reveal nodal anatomy. When rot extends into a node at 1 or 2 sites, it can be recorded as a 1.1 or 1.2. The x.5 ratings are often necessary. The following rating system can be used in evaluating the severity of anthracnose, Fusarium and charcoal rot following artificial inoculation:

0.0 = No evaluation possible
0.1 = Minimal reaction, indistinguishable from that to a sterile toothpick

0.2 = Discoloration centered about the wound, progressing farthest in the superficial parts of the stalk, but not reaching either node
0.5 = Extensive discoloration progressing farthest in the central part of the stalk
0.8 = Discoloration reaching one or both nodes superficially or forming a cylinder
1.0 = Most or all of one internode discolored with no penetration of nodal areas
1.1 = Slight penetration of one or both nodes
1.2 = Nearly complete penetration of one or both nodes
1.5 = Penetration of one node and slight invasion of the next internode
2.0 = More than 1 but not more than 2 internodes affected; infection must have spread through at least 1 internode
2.5 = Penetration of 2 nodes and slight invasion of distal internode
3.0 = Infection has passed through 2 or more internodes
4.0 = Extensive invasion of plant but not killed
5.0 = Death of plant due to stalk rot

Lodging. The ratings below, based on percentage of lodged plants, should be recorded. This lodging can be any one or a combination of: weak neck (breaking at base of peduncle), stalk breakage (due to stalk rots or weakened stalks due to stress, or very high wind), or root lodging (entire plant leans or falls due to wet soil and wind). The predominant type of lodging should be recorded. Use rating or actual %.

0 = No evaluation possible
1 = 2% or less lodging
2 = 3-10% lodging
3 = 11-30% lodging
4 = 31-70% lodging
5 = 71-100% lodging

Fusarium Head Blight. Infection and death of rachis branches, rachis and often peduncle (maybe even progressing downward to the base of the stalk) should be rated as below. The panicle branches of infected heads often droop severely after maturity. Infected rachis branches and peduncles have both external and internal discoloration as contrasted to MDM-induced discoloration which is limited to external discoloration only. The disease usually develops or becomes obvious at about physiologic maturity of the grain or later.

0 = No evaluation possible
1 = Resistant: no infection in rachis branches or head
2 = Infection limited to head, particularly rachis branches
3 = Whole head infected
4 = Both heads and peduncles affected
5 = Head blight resulting in death or lodging of plants

Anthracnose. Natural infection: Each of the following are rated from 1 to 5 based on 1 = resistant to 5 = death of tissue or plant. Numeric prefixes may be used to designate on what portion of the plant the evaluation was made by characterizing leaves (foliage) as 1, the stalk (peduncle) as 2 and the head as 3.

(Anthracnose). Artificial inoculation; Toothpick method, same as for stalk rot evaluation. Conidial injection, same as for natural infection.

Pythium or Other Root Rot. Ratings of root rot should normally be made at maturity, but before the plant has died. Ratings are based on the following scale:

0.0 = No evaluation possible
1.0 = Roots free from disease
2.0 = No root rot near crown or on major brace roots
2.5 = At least one dead major brace root
3.0 = More than one to 1/2 but not all brace roots dead
3.5 = More than 1/2 but not all brace roots dead
4.0 = All roots dead, but base of stalk is still alive
5.0 = All roots and base of stalk dead

Seed Weathering. A major environment and maturity related problem affecting both quality and quantity of seed is weathering. Should the opportunity present itself, data on seed mold and discoloration associated with weathering should be recorded on a 1 to 5 scale as noted. Ratings can be accurately made after the sample has been threshed. Established standards for reference are recommended.

0 = No evaluation possible
1 = Seed bright, free from mold damage
2 = Moderately resistant to weathering, seed slightly discolored
3 = Moderately susceptible, considerable discoloration
4 = Susceptible, extensive discoloration and deterioration of seed
5 = Very susceptible, seed essentially all dead. Embryos dead and endosperm deteriorated

Desirability. The overall desirability, adaptation or breeding potential ratings can be made near or at maturity. Many factors influence such a rating. However, each individual's overall appraisal of the desirability of entries in conjunction with the other ratings should give valuable information regarding the most useful material or germ plasm for certain areas of the world.

0 = No evaluation possible
1 = Very good
2 = Good
3 = Average
4 = Poor
5 = Very poor

SUMMARY

Comments regarding breeding for sorghum diseases, including inheritance of resistance, pathogen specificity, availability of sources of resistance, and screening procedures are grouped in the four categories as shown and addressed below:

A = Nature of inheritance of resistance
B = Specific pathogen differences
C = Sources of resistance
D = Screening procedure

1. Downy Mildew
   A. Dominant (field reaction)
   B. Some differences, e.g., India vs. USA
   C. Several resistant lines under field conditions in Texas. Only a few are resistant under artificial conidial inoculation, or in India under natural conditions.
   D. Field and lab

2. Head Smut
   A. Dominant (some intermediate and recessive)
   B. Rapid changes in pathogen in USA. (Sources have broken down).
   C. Several resistant lines
      1. Do not know if will hold under concentrated planting.
      2. Some low smutters are still holding.
      3. May be few to several major dominant genes.
      4. Are many modifier genes.
   D. Field (natural and artificial) plus lab. Needle inoculation breaks down some lines that are resistant in the field.

3. Maize Dwarf Mosaic (MDMV)
   A. Are several reactions
      1. Tolerant reaction (e.g., Martin) - dominant over "red leaf" (e.g., Redlan) (Tolerant lines take virus but not affected adversely).
      2. Rio reaction - dominant over "tolerant" and "red leaf" reactions. Plants severely mottled, chlorotic, and stunted, but do not have typical "red leaf". Mottling remains in older leaves. Most lines with Rio reaction have low incidence under field conditions, but can have high incidence under inoculation, or unusually high inoculum pressure in the field.
      3. QL (Krish source) is dominant (not infected).
   B. Significant differences
      1. Argentina and Australia - somewhat different than in USA
      2. Venezuelan strain (of sugarcane virus) - much different than MDM in USA based on host reaction
   C. 1. Very few resistant to infection
      2. Many are tolerant
      3. Several have Rio reaction, e.g., IS 2816C (SC 120), IS 12666C (SC 175), TAM 2566, Rio, several sweet sorghums
   D. Field (Natural - greenbugs and corn leaf aphids inoculate, and artificial - airbrush) plus lab.

4. Anthracnose
   A. Dominant
   B. 1. Some changes have occurred in USA
      2. Puerto Rico possibly different or due to environment
      3. Brazil definitely different
      4. West Africa different - lesions differ in appearance. Are circular in W. Africa

C. 1. Very few resistant in Brazil (but few good sources) (Most also good in US)
   2. Several resistant in USA
   3. Major differences in host reaction in Africa vs. USA
D. Field (Georgia, Mississippi, Puerto Rico, Brazil) - Natural and inoculated

5. Rust
   A. Dominant
   B. Some differences in host reaction between Texas, Puerto Rico, and Mexico, but probably due to environment.
      1. Rio, TAM 2566, get much rust in Puerto Rico and very little in USA
      2. Slow-rusting types such as TAM 428 and SC 170 derivatives hold up very well in Mexico
   C. Several resistant lines - distinct sources of resistance. Slow rusting trait is very useful.

6. Charcoal Rot
   A. Recessive
   B. No known differences
   C. Few lines with rather good resistance
      1. None are completely resistant if high yielding and severe moisture stress during grain development.
      2. Problems of relationship with grain yield, late maturity, and time of stress. Plants must be predisposed by moisture (and heat) stress in latter stages of grain development before they are susceptible.
   D. Field (moisture stress late) - Natural and artificial (toothpick inoculation). In Texas, screening is part of our lodging nurseries. We rate for senescence during the late grain development stage when plants are under moisture stress, and this predicts well their response to charcoal rot susceptibility. Non-senescing types possess good charcoal rot resistance.

7. Fusarium Head Blight
   A. Intermediate
   B. No evidence of major differences in US
   C. Only few with good resistance
   D. Field (natural and artificial)
      1. Artificial (infested toothpicks in peduncle) overcomes much of field resistance.
      2. Natural the best, but hard to screen, because plants must be at correct stage of growth (late grain development the most susceptible stage), and must have correct environment. Disease usually develops at or near maturity and often difficult to distinguish from natural drying and saprophytic invasion. Internal reddening of the pith the best diagnostic tool. Marbling of pith due to anthracnose is distinct from pith discoloration due to Fusarium.

8. Fusarium Stalk Rot
   Not much known. Often occurs when moisture stress is followed by wet conditions at or near maturity. Enters at nodal areas. We believe Fusarium head blight and Fusarium stalk rot may be related regarding resistance.

9. Grain Mold
   A. Some dominant, some overdominant in F_1 hybrids, some intermediate
   B. Several different pathogens involved. They differ in importance at different locations, under different environments, and at different stages of maturity of the grain when infection takes place.
   C. Only a few good sources of resistance (but are distinct heritable differences)
      1. None completely resistant
      2. Brown seeded (high tannin) lines generally more resistant
   D. Field (natural and inoculated)
      1. Rate for overall grain mold and grain weathering-delay harvest-plant so get rain or wet conditions at or after maturity. Here we rate for overall discoloration, amount of mold evident, and deterioration (including sprouting) of the grain. This rating obviously involves more than grain mold as such, but most types selected this way also show grain mold resistance under inoculation techniques.
      2. Inoculate in field with specific pathogens
      3. Water spray mechanism in field to create a wet environment to enhance grain mold

10. Zonate Leaf Spot
   A. Intermediate to recessive
   B. No known differences
   C. Only few resistant - None completely resistant
   D. Field - Somewhat maturity related - Generally plants become more susceptible after flowering and near maturity. Also, sterile plants are less susceptible.

11. Grey Leaf Spot
   A. Recessive
   B. No known differences - Some locations no beading type symptom
   C. Only few resistant - None completely resistant
   D. Field - Somewhat yield related - Sterile plants are less susceptible.

12. Bacterial Stripe
   A. Recessive
   B. No known differences
   C. Some very susceptible when others have little if any
   D. Field - Related to spacing as end plants or non-bordered plants may have much stripe

13. Bacterial Streak
   A. Recessive to somewhat intermediate
   B. Not known
   C. Some resistant lines
   D. Field

14. Leaf Blight
   A. Dominant
   B. No known differences
   C. Only few lines with good resistance
   D. Field (Mexico, Puerto Rico) - Natural and inoculation plus lab

TABLE 8. Summary of diseases, inheritance of resistance, and screening techniques on sorghum.

Disease	Inheritance Pattern	Screening Technique[a/]
Downy Mildew	Dominant	Field, Lab
Head Smut	Dominant	Field (N & A), Lab
Maize Dwarf Mosaic	Dominant	Field (N & A), Lab
Anthracnose	Dominant	Field (N & A)
Rust	Dominant	Field
Zonate Leaf Spot	Intermediate-Recessive	Field
Grey Leaf Spot	Recessive	Field
Leaf Blight	Dominant	Field (N & A), Lab
Bacterial Stripe	Recessive	Field
Bacterial Streak	Recessive	Field
Charcoal Rot	Recessive	Field (N & A)
Fusarium Stalk Rot	?	Field (N & A)
Fusarium Head Blight	Intermediate	Field (N & A)
Pythium Root Rot	?	Field, Lab
Grain Mold	Dominant-Intermediate	Field (N & A)

[a/] N = Natural, A = Artificial

TABLE 9. Some sources of sorghums with disease resistance (Most identified and selected within Texas - a few identified by ICRISAT).

Sorghum Resistant to Head Smut	
White Kafir (PI 48770)*	Lahoma Sudangrass*
SC 324-12 (IS 2861 der)*	SC 325-12 (IS 2462)*
SC 33-14 (IS 12553)	Tx 430
TAM 428	IS 2403C (SC 103)
Early Hegari (SA 281)	IS 12658C (SC 167)
Spur Feterita (Fe 6601)	IS 2508C (SC 414)
FC 8927 (D. Wh. Milo)	TAM 618**
Tx 3048**	Tx 7000 (Caprock)**
Tx 399 (Wheatland)**	(Other converted lines)

* Resistant under needle Inoc. also
** Low level of field infection (stable so far)

Sorghum Resistant to Anthracnose	
Brandes**	MN 960 (SC 972)**
Rio (IS 17459)	SC 326-6 (IS 3758 der)**
IS 7173C (SC 283)**	SC 748-5 (IS 3552 der)**
SC 328-14 (IS 8263 der)	TAM 428
Tx 430	Tx 623
IS 2403C (SC 103)	IS 12612C (SC 112)
IS 12610C (SC 110)	IS 1309C (SC 322)*
IS 3574C (SC 239)*	IS 7778C (SC 389)*
IS 12537C (SC 17)*	IS 12615C (SC 124)*
IS 2816C (SC 120)*	IS 7382C (SC 344)*
IS 12677C (SC 186)*	SC 60-14E (IS 2569 der)*
SC 490-14E (IS 6392 der)*	SC 589-14E (IS 6388 der)*
SC 37-14E (IS 12557 der)*	(Others resistant in USA

* Also resistant in Brazil (1 yr)
** Also resistant in Brazil (several yrs)

Sorghum Resistant to Downy Mildew	
QL 3 (India source)	IS 2816C (SC 120)
IS 2508C (SC 414)	IS 12664C (SC 173)
IS 12661C (SC 170)	IS 2403C (SC 103)
IS 12610C (SC 110)	IS 12666C (SC 175)
TAM 428 (SC 110 der)	Tx 430 (SC 170 der)
Tx 2519	SC 170-6-17
CS 3541 (CSV 4)	(Many other converted lines)

TABLE 9. Continued

Sorghum Resistant to Zonate Leaf Spot	
SC 326-6 (IS 3758 der)	77CS1 (IS 2930 x IS 3922)
R 1880 (SC 599 x SC 134)	SC 330-9 (IS 8187 der)
Sorghum Resistant to Fusarium Head Blight	
SC 599-6 (R 9188) (IS 17459 der)	SC 599-6 (R 9247) (IS 17459 der)
SC 630-11E (IS 1269 der)	SC 650-11E (IS 2856 der)
GPR-148 (CSV 5)	
Sorghum Resistant to Grey Leaf Spot	
77CS1 (IS 2930 x IS 3922)	R 1880 (SC 599 x SC 134)
SC 748-5 (IS 3552 der)	
Sorghum Resistant to Grain Mold	
SC 279-14 (IS 7419)	IS 7254C (SC 566)
SC 748-5 (IS 3552 der)	SC 630-11E (IS 1269 der)
SC 650-11E (IS 2856 der)	IS 9530
SC 719-11E (IS 7013 der)(Brown)	SC 170-6-17
CS 3541	IS 2367
IS 2368	
Sorghum Resistant to Charcoal Rot	
SC 599-6 (R 9188) (IS 17459 der)	SC 599-6 (R 9247) (IS 17459 der)
IS 12568C (SC 56-14)	SC 56-6 (IS 12568 der)
SC 170-6-17 (IS 12661 der)	R 1584 (SC 56 x SC 170)
SC 35-6 (IS 12555 der)	B4R (B 406 x Rio)
1790E (SC 56 x SC 33)	1790L (SC 56 x SC 33)
1778 (SC 56 x SC 33)	NSA 440
Sorghum Resistant to Rust	
SC 326-6 (IS 3758 der)	TAM 2566
IS 2816C (SC 120)	IS 12666C (SC 175)
SC 599-6 der (IS 17459 der)	SC 748-5 (IS 3552 der)
TAM 428*	Tx 623*

*Slow rusting

TABLE 9. Continued

Sorghum Resistant to Leaf Blight	
SC 326-6 (IS 3758 der)	IS 8337C (SC 574)
IS 12658C (SC 167)	IS 1335C (SC 418)
IS 6882C (SC 320)	IS 7254C (SC 566-14)
SC 325-12 (IS 2462 der)	

Sorghum Resistant to MDM	
QL1, QL2, QL3, QL4, QL11*	Rio Reaction***
Tx 398 (Martin)**	Rio (IS 17459)
Tx 399 (Wheatland)**	IS 12666C (SC 175)
(lot other lines)**	TAM 2566
	IS 2816C (SC 120)
	IS 12612C (SC 112)
	IS 2548C (SC 228)

*Resistant to infection
**Tolerant
***Some field resistance but infected plants severely damaged

In conclusion, here are some keys to a productive disease resistance breeding program.

1. Have a large amount of genetic diversity in the program.
2. Plant large amount of diverse breeding material in a few large prime disease screening locations. Use smaller nurseries in locations where only one disease is present.
3. Screen the same material for as many diseases as possible in the same year.
4. Test best sources of resistance extensively at additional locations and under different environments.
5. Do a lot of recombining of best sources of resistance, even among early generation sources. Selecting parents to complement each other, and at the same time make improvements agronomically, is a key item.
6. Pathologists and breeders must work cooperatively.
7. Breeder must be able to rate for disease resistance.
8. Pathologist must work in breeding material.
9. Must get in the field and look for resistance.

LITERATURE CITED

Asgrow Seed Company. 1974. Troublesome grain sorghum diseases - a management manual. Asgrow Seed Company. Des Moines, Iowa. 50310

Craig, T. 1976. An inoculation technique for identifying resistance to sorghum downy mildew. Plant Dis. Rep. 60:350-352.

Edmonds, L. 1963. Use of sporidial hypodermic injection to test sorghum for head smut resistance. Plant Dis. Rep. 47:909-913.

Tarr, S. A. 1962. Diseases of sorghum, sudangrass and broom corn. The Commonwealth Mycological Institute, Kew, Surrey, 380 pp. Printed in Great Britain at the University Press, Oxford.

Williams, R. T., R. A. Frederiksen, and G. O. Bengston. 1980. Internationally important sorghum diseases. Proc. of the International Sorghum Disease Workshop. ICRISAT, Hyderabad, India, Dec. 1978. (in press).

Williams, R. T., R. A. Frederiksen, and T. C. Girard. 1978. Sorghum and pearl millet disease identification handbook. ICRISAT Information Bul. No. 2.

BREEDING FOR ARTHROPOD RESISTANCE IN SORGHUM

Jerry W. Johnson and George L. Teetes

Texas Agricultural Experiment Station
Texas A&M University System
Lubbock, Texas 79401

ABSTRACT

Sorghums have been identified that exhibit resistance to the major arthropod pests of the crop. Sorghum lines resistant to the greenbug, Schizaphis graminum (Rodani), the sorghum midge, Contarinia sorghicola (Coquillett), and the Banks grass mite are presented. Appropriate breeding methods are presented that should facilitate a rapid transfer of resistance into acceptable agronomic types.

INTRODUCTION

Plant breeders, especially those working in the tropics, are increasingly concerned with insect problems in the initial planning of their breeding programs. Because the release of an insect susceptible, but otherwise superior variety may achieve little, breeding nurseries are now being established where no insecticides are used, thus providing early evaluation of breeding lines for insect susceptibility.

Several excellent reviews of sorghum insects, insect resistance in sorghum and its role in insect control have been published (1, 2, 3, 5, 6, 15, 27).

I will not attempt to discuss the many merits of resistance except to say that its proper utilization will be as one of several tools in a pest management program.

Breeding for insect resistance differs in no fundamental way from breeding for other characters. Consequently, any of the various methods of breeding appropriate for sorghum can be used in developing insect resistant varieties once resistance has been found and efficient evaluation techniques have been developed. We are concerned with a minimum of three objectives: (1) to develop agronomically suitable varieties resistant to insects of economic importance as rapidly as possible, (2) to continue to find new sources of resistance, and (3) to improve the level of resistance over that presently available.

This paper is intended to provide information on resistant sources and techniques to best accomplish these goals.

GREENBUG

In the United States the greenbug [*Schizaphis graminum* (Rodani)] has been recognized as a major pest of sorghum since 1968 and causes significant damage in Brazil in some years. Resistance to this pest has been reported in the seedling stage (7, 10, 18, 19, 20, 21, 23) and in the adult plant stage (8, 9, 10, 11, 22). Cultivars reported as resistant were not suitable for commercial use and considerable breeding work has been required to incorporate this resistance into commercially acceptable material. Lines that have been reported as resistant are given in Tables 1a and 1b. Agronomically improved lines can be obtained from the Agricultural Experiment Stations in Kansas, Oklahoma, or Texas.

The inheritance of greenbug resistance in lines that have been studied is dominant or incompletely dominant. The resistance of several breeding lines with resistance derived from *Sorghum virgatum* was reported to be conferred by dominant genes at more than one locus (7). Studies by Johnson (10, 13) indicate that resistance in PI 264453 is simply inherited and incompletely dominant and that resistance in IS 809, SA 7536-1, PI 220248 and PI 302236 is incompletely dominant. Studies by Weibel, et al. (23) of F_1 and F_2 populations from susceptible varieties and SA 7536-1, IS 809 and PI 264453 indicate the inheritance of resistance from three lines to be incompletely dominant and simply inherited.

TABLE 1a Sources of Resistance to the Greenbug

Designation	Species	Type
IS 809	*S. bicolor*	Grain
PI 264453	*S. bicolor*	Forage
KS 30	*S. virgatum*	Grassy
SA 7536-1	*S. nigricans*	Grassy
PI 302236	*S. hewisonii*	Grassy
PI 220248	*S. sudanense*	Grassy
PI 308976	*S. sudanense*	Grassy
PI 38108 (T.S. 1636)	*S. virgatum*	Grassy
Bloomless Combine Kafir 60	--	Grain

TABLE 1b. Agronomically Improved Sources of Resistance to the Greenbug

Designation	B or R Reaction	Source of Resistance
TAM BK-41	R	SA 7536-1
TAM BK-42	R	PI 264453
TAM BK-43	B	KS 30
TAM BK-44	B	KS 30
TAM 2567	B	SA 7536-1
TAM 2568	R	SA 7536-1
KS 41	R	T.S. 1636
KS 42	R	T.S. 1636
KS 43	R	T.S. 1636
KS 44	R	T.S. 1636
KS 56	B	T.S. 1636
KS 57	B	T.S. 1636
OK GP-1	-	SA 7536-1
OK GP-2	-	SA 7536-1
OK GP-3	-	SA 7536-1
OK GP-4	-	SA 7536-1
OK GP-5	-	SA 7536-1
OK GP-6	-	IS 809
OK GP-7	-	IS 809
OK GP-8	-	IS 809
Tx 2734	R	SA 7536-1
Tx 2735	R	SA 7536-1
Tx 2736	R	SA 7536-1
Tx 2737	R	SA 7536-1
Tx 2738	R	IS 809
Tx 2739	R	IS 809
Tx 2740	R	SA 7536-1
Tx 2741	R	SA 7536-1
Tx 2742	R	SA 7536-1
Tx 2743	R	SA 7536-1
Tx 2744	R	SA 7536-1
Tx 2745	R	SA 7536-1
Tx 2746	R	SA 7536-1
Tx 2747	R	SA 7536-1
Tx 2748	PB	SA 7536-1
Tx 2749	B	KS 30
Tx 2750	B	KS 30
Tx 2751	B	KS 30
Tx 2752	B	KS 30

Since it has been established that greenbug resistance is not complexly inherited and can be retained through several backcrosses, the primary need is an effective evaluation technique to identify resistant material in segregating populations.

SEEDLING EVALUATION

The technique described by Wood (26) for evaluating small grains has been modified to be used on sorghum. This technique consists of culturing greenbug biotype C on a susceptible sorghum hybrid grown in one-gallon cans. From 30 to 50 seed are planted per can in soil and covered with sand to a depth of one inch. Before plant emergence, they are covered with a cage inserted into the sand in each can to protect them from premature greenbug infestation and to prevent infestation by parasitic wasps. Cages are constructed of clear vinyl plastic 0.015 inches thick, 13 inches high and 5.5 inches in diameter. Ventilation is aided by cutting holes 2.5 inches in diameter in two sides of the cage. The ventilation holes and the cage top are covered with fine mesh nylon cloth. After the culture plants have reached a height of 6 to 8 inches, each can is infested with about 200 greenbugs. After about two weeks, these cultures are ready to use.

Breeding lines that are to be evaluated for resistance are planted in galvanized metal flats 14 x 20 x 3.75 inches. This flat will accommodate 10 entries in rows 14 inches long. A resistant and susceptible check is planted in each flat. Approximately 20 seeds of each entry are planted per row and thinned to 15 plants per row one week after emergence. These plants are infested from the culture plants by brushing approximately 1,500 greenbugs on each flat.

Rating for resistances begin when the susceptible variety is near death. A 1 to 9 rating system is used where 1 equals no damage and 9 equals dead plants. Rows segregating for resistance are recorded as such and a rating given on the most resistant plant in the row. This procedure permits evaluation to begin on seed from plants selected from F_2 populations.

ADULT PLANT RESISTANCE

Resistance of most lines are easily detected in the field if natural greenbug populations are at an adequate level. Natural populations averaging over 200 per plant are required to obtain reliable data on adult plants. The rating system given in Table 2 is useful in evaluating nursery material. When susceptible checks are rated as 6, resistant selections should have a rating no higher than 3. When susceptible checks are rated 3 or 4, resistant selections should have a rating no higher than 1. Evaluations should be made when greenbugs are present. If too much time elapses between maximum greenbug infestation and the time evaluations are made, it is difficult to distinguish natural leaf death of the lower leaves and death caused by greenbug feeding.

Because of the low number of greenbugs occurring in the late planted sorghum at Lubbock, a technique has been developed to artificially infest selected adult plants in the field. Materials needed in this procedure are clip-on cages described by Cate, Bottrell and Teetes (4) and a small artist paint brush. The cages are a clip-on type constructed of 1-inch square plastic boxes. Screen-covered holes, 3/4 inches in diameter in opposite sides of the boxes allow ventilation within the cages. Five adult greenbugs are transferred from culture cans into each cage with a small artist brush. Two cages are attached to a leaf of each plant to be evaluated. We normally use the third leaf from the top of the plant. Cages are inspected the day following attachment to the leaves to insure that five greenbugs are alive and feeding on the plant. Additional greenbugs are added as needed to bring the total to five. Resistant and susceptible checks are included in the test.

Snap cages can be used at any time, even on mature plants. This provides flexibility to the evaluation program and permits the breeder to determine when evaluations will be made.

TABLE 2 Proposed Rating Scheme for Rating Resistance of Sorghum to Greenbugs

Score	Description of Damage*
1	No red spotting on leaves
2	Red Spotting on leaves
3	Portion of a leaf killed by greenbugs
4	One entire leaf killed by greenbugs
5	Two entire leaves killed by greenbugs
6	Four entire leaves killed by greenbugs
7	Six entire leaves killed by greenbugs
8	Eight entire leaves killed by greenbugs
9	Plant killed by greenbugs

*Data may be taken at any plant growth stage when greenbugs are present. It is suggested that estimates of greenbug numbers be taken if possible.

Ratings of the leaf area under the cage begins about one week after infestation and are taken every two days if the greenbugs are reproducing well on the susceptible check. The rating system used with this technique is as follows: 0 equals no necrotic plant tissue in the caged area, 1 equals 10 percent necrosis, 2 equals 20 percent, 3 equals 30 percent, 4 equals 40 percent, 5 equals 50 percent, 6 equals 60 percent, 7 equals 70 percent, 8 equals 80 percent, 9 equals 90 percent, and 10 equals 100 percent. Plants that receive a mean rating of five or less on the same date the susceptible check receives an eight or more are considered to be resistant.

MIDGE

The sorghum midge [*Contarinia sorghicola* (Coquillett)] is a pest of sorghum in almost all regions of the world where the crop is grown. Varieties resistant to the insect have been reported from several countries (12, 14, 16, 17, 24, 25).

Identification of varieties resistant in the United States breeding programs in the past have been relatively fruitless primarily due to the limited amount of germplasm available in lines adapted to temperate regions. In addition, breeding nurseries were planted at a time designed to escape midge damage or were sprayed with an insecticide to control the midge if it became a problem, thus eliminating the opportunity to observe resistance if it were present.

Interest among breeders and entomologists at the Texas Agricultural Experiment Station was renewed in 1969 by independent observations by Dr. D. T. Rosenow at Lubbock, Texas and Dr. F. R. Miller at the Federal Experiment Station in Mayaguez, Puerto Rico. They reported that partially converted selections from IS 12610 had less midge damage than other lines in their nurseries. Since then, we have eliminated, or greatly reduced, insecticide application for midge control in our nurseries at Lubbock.

The absence of midge control provides the opportunity to select lines that have resistance. Using this approach, lines are identified for entrance in midge evaluation tests. Converted and partially converted exotic lines from the sorghum conversion program and their progenies from hybridization with elite U.S. material have been evaluated since 1971.

Resistant sources adapted to temperate areas of sorghum production are listed in Tables 3, 4, and 5. These lines were shown to be resistant by evaluation of replicated tests at several locations during 1972, 1973, and 1974. During this time, several rating systems have been tried, but we have now chosen the rating system shown in Table 6, and encourage others to utilize the same system to aid in compiling data from international nurseries or tests.

TABLE 3 Midge Damage Rating of Selected Sorghum Lines, 1973 and 1974

Line	SC No.	Group Name	Midge Damage Score		Days to Bloom	
			1973	1974	1973	1974
IS 2501C	52	Nigricans-Feterita	1.8	2.6	63	60
IS 12608C	108	Zerazera	3.8	4.8	65	65
IS 12612C	112	Zerazera	3.4	6.3	65	63
IS 2816C	120	Zerazera	3.9	6.1	64	63
TAM 2566	175-9	Zerazera	1.8	3.6	69	67
IS 12666C	175	Zerazera	1.2	4.2	67	65
IS 2549C	228	Zerazera	3.2	3.1	59	60
IS 7007C	268	Caudatum-Nigricans	4.0	8.4	63	57
IS 1309C	322	Nigricans	3.2	4.7	70	66
IS 2508C	414	Caudatum-Kafir	4.3	5.8	70	63
IS 2579C	423	Zerazera	2.1	4.6	69	62
IS 8100C	424	Caudatum-Nigricans	3.4	6.2	57	60
TAM 428	-	Zerazera	-	8.0	-	67
SGIRL-MR-1**	-	-	4.5	6.7	66	63
Tx 2536*	-	-	8.2	9.7	67	64
Tx 7000*	-	-	8.7	10.0	69	66
B Tx 378*	-	-	7.8	9.1	69	66
Tx 7078*	-	-	6.9	9.7	63	62
B Tx 3042*	-	-	6.2	9.6	64	57
B Tx 3197*	-	-	7.6	9.6	67	63
Tx 415*	-	-	8.6	9.5	76	67

**Resistant check

*Susceptible check

TABLE 4 Converted Sorghum with High or Moderate Levels of Resistance to the Sorghum Midge, 1978 Georgia and Texas

IS No. of Original Line	SC No.	Working Group No.	Working Group	Country Seed Received From	Midge Damage Rating
IS 12666	SC 175	39(1)	Zerazera	Ethiopia	1.6
IS 3071	SC 237	31(1)	Dobbs	Sudan	2.6
IS 12664	SC 173	39(1)	Zerazera	Ethiopia	3.1
IS 2579	SC 423	39(1)	Zerazera	Sudan	3.2
IS 12593	SC 84	47	Durra/Nigricans	Uganda	3.3
IS 8263	SC 328	33	Caudatum	India	3.3
IS 8337	SC 574	41	Durra	Pakistan	3.4
IS 12676	SC 185	39	Caudatum-Nigricans	Sudan	3.6
IS 7142	SC 564	33	Caudatum	Uganda	3.7
IS 8231	SC 645	23	Caffrorum/Darso	India	3.7
IS 2508	SC 414	38	Caudatum-Kafir	Sudan	4.0
IS 8233	SC 643	23	Caffrorum/Darso	Uganda	4.1
IS 2403	SC 103	33	Caudatum	S. Africa	4.2
IS 12609	SC 109	39(1)	Zerazera	Ethiopia	4.2
IS 2862	SC 655	22	Caffrorum	S. Africa	4.8
IS 12683	SC 221	41	Durra	India	4.9
IS 12610	SC 110	39(1)	Zerazera	Ethiopia	5.2
IS 6446	SC 586	46(1)	Nandyal	India	5.4
IS 2573	SC 64	31	Nigricans Feterita	Sudan	5.5
IS 2662	SC 114	33	Caudatum	Uganda	5.7
IS 7064	SC 420	38	Caudatum/Kafir(Heg.)	Sudan	5.7
IS 12612	SC 112	39(1)	Zerazera	Ethiopia	5.9
IS 8134	SC 590	47	Durra/Nigricans	India	5.9
IS 12577	SC 68	39	Caudatum/Nigricans	Kenya	5.9
IS 12608	SC 108	39(1)	Zerazera	Ethiopia	6.1
IS 2569	SC 60	33	Caudatum	Equatoria	6.2
IS 6394	SC 491	46(1)	Nandyal	India	6.2
IS 12535	SC 15	16	Dochna/Honey	Ethiopia	6.5
IS 8100C	SC 424	39	Caudatum/Nigricans	Japan	6.8
IS 12573	SC 63	39	Caudatum/Nigricans	Nigeria	6.9
IS 12674	SC 183	27	Caffrorum/Feterita	Mexico	6.9

TABLE 5 Agronomically Improved Midge Resistant Sorghum Lines

Designation	B or R Reaction	Source of Resistance
Tx 2754	B	*
Tx 2755	B	*
Tx 2756	B	*
Tx 2757	B	*
Tx 2758	B	TAM 2566
Tx 2759	B	TAM 2566
Tx 2760	B	TAM 2566
Tx 2761	B	TAM 2566
Tx 2762	R	TAM 2566
Tx 2763	R	TP8R
Tx 2764	R	TP8R
Tx 2765	R	TP8R
Tx 2766	R	TAM 2566
Tx 2767	R	TAM 2566
Tx 2768	R	TAM 2566
Tx 2769	R	TAM 2566
Tx 2770	R	TAM 2566
Tx 2771	R	TAM 2566
Tx 2772	R	TAM 2566
Tx 2773	R	TAM 2566
Tx 2774	R	TAM 2566
Tx 2775	R	TAM 2566
Tx 2776	R	TAM 2566
Tx 2777	R	TAM 2566
Tx 2778	R	TAM 2566
Tx 2779	R	TAM 2566
Tx 2780	R	TAM 2566
Tx 2781	R	IS 2508C (SC 414)
ISR 1	R	AF 28

*Derived from TP6BP, an unreleased population composed of exotic and partially converted sorghums.

TABLE 6 Proposed Rating Scheme for Rating Resistance of Sorghum to the Sorghum Midge

Score	Description of Damage
0	No damage
1	1-10% damaged seed
2	11-20% damaged seed
3	21-30% damaged seed
4	31-40% damaged seed
5	41-50% damaged seed
6	51-60% damaged seed
7	61-70% damaged seed
8	71-80% damaged seed
9	81-90% damaged seed
10	91-100% damaged seed

In order to provide a uniform infestation of midge over the test area, "spreader rows" should be planted throughout the nursery with not over 50 feet between rows. These rows should consist of a mixture of hybrids classed as early, medium and late in the location that the test or nursery will be grown.

The use of a large number of susceptible varieties as checks is required. Every effort should be made to include check varieties that overlap in their maturity and include the earliest and latest varieties adapted to the area. This is especially important in areas that have a large fluctuation in midge numbers throughout the blooming period. At Lubbock, the midge population generally reaches adequate levels by mid-August and stays high until frost. However, in other areas populations may be large one week and very low the next.

In our present program, varieties reported as resistant are grown at Lubbock to evaluate them for resistance to the midge and for their maturity in the U.S. Most midge resistant introductions are too late in blooming for proper evaluation of resistance and are entered into the conversion program. Partially converted selections from these lines are evaluated for midge resistance as they progress through the conversion program.

BANKS GRASS MITE

The Banks grass mite is normally a damaging pest only on adult plants. Evaluation of lines for resistance is difficult because of a maturity plant susceptibility interaction. Rarely does this pest damage sorghum until after bloom and damage generally occurs during the seed filling period.

Because the mite is normally a problem after the plants have bloomed, yield losses should be restricted to decreased seed size and losses due to loding. We have evaluated lines first for their ability to maintain green leaves in the presence of large mite populations. Lines that are above average in this respect are evaluated for their ability to maintain live stalks and normal seed size with mites present.

In general, grassy sorghum such as 'Sweet Sudan' and 'KS 30' do not receive much mite damage. However, observations indicate that the grassy types are not preferred by mites. Possibly because each plant usually has many tillers that have not reached the bloom stage. It is doubtful if this apparent nonpreference can be transferred to grain types.

Two lines, SC 599-6 - a partially converted 'Rio' - and a partially converted IS 12568 selection, have shown resistance to the Banks grass mite in preliminary tests. We are evaluating SC 599-6, its F_1 hybrid, and progeny from crosses with resistant sources in field planting. We are attempting to establish methods for evaluating material in the greenhouse.

SHOOT FLY

The sorghum shoot fly (Atherigona soccata Rond.) is not a pest of sorghum in the Americas. However, because of the importance of this insect in many sorghum producing areas and the demonstrated international usefulness of lines released by The Texas Agricultural Experiment Station, we are conducting work in the area.

Lines that are identified as resistant to the shoot fly or other important insects such as the stem borer (Chilo zonellus Swin) are being entered into the sorghum conversion program. Selections will be made of partially converted lines from the segregating F_2 populations that are adapted to temperate areas and evaluated for resistance by cooperators in the area that the insect is a problem. Lines that are resistant will be combined in a population with elite lines from the Texas program. Distribution of the population will be made to all interested breeders.

BREEDING METHODS

The breeding methods that seem appropriate for the arthropod pests discussed in this paper are outlined below:

Midge

Pedigree Method:

(1) The highest level of resistance should be transferred to agronomically acceptable types by hybridization and selection.

(2) Agronomically acceptable lines with the least susceptibility to midge should be used as the nonresistant parent, e.g., TAM 428.

(3) Grow a large F_2 population, at least 4,000 plants. Selection can be accomplished here without midge being present. Our limited experience has shown that selection for small-glumed types should increase the frequency of midge resistant types from the F_2 populations.

(4) Evaluate F_3 rows under large midge populations. To increase the probability of large midge populations during blooming of this material, it should be planted at more than one date or location.

(5) Evaluate F_4 selections in replicated progeny rows and backcross superior plants if necessary.

Population Breeding:

(1) Selection of the proper genetic male sterile may be critical in establishing the population. We have found ms_3 to be satisfactory. Populations containing genetic male sterility can be obtained from the Agricultural Experiment Stations in Kansas, Nebraska, or Texas. Antherless has not always been successful for use in the tropics.

(2) Four types of populations should be established:

a. for 'rapid' progress, B and R populations should be established combining acceptable agronomic types and high levels of resistance,

b. for long range improvement, establish B and R populations using elite varieties from the area the material will be utilized and all resistant sources available.

(3) Utilize the appropriate selection scheme best suited to the area, i.e., mass selection, reciprocal recurrent selection, etc.

Greenbug

Pedigree Method:

(1) Select lines with the highest level of resistance but suited agronomically to the area of intended use to be hybridized with elite adapted varieties.

(2) Backcross F_1 plant of cross between resistant and susceptible lines.

(3) Evaluate large F_2 populations of cross between F_1 and elite susceptible variety. Use either seedling evaluation, transplanting resistant types to the field or evaluate F_2 populations in the field.

(4) Backcross resistant F_2 plants or select for evaluation in F_3 rows depending on agronomic desirability of the plant.

Population Breeding:

The same basic plan outlined above for midge resistance breeding should be used for greenbugs.

Shoot Fly and Banks Grass Mite

The same basic procedure outlined for midge resistance should be an appropriate approach to use for these pests.

SUMMARY

A coordinated international effort to improve the insect resistance of sorghum is imperative. Interest in this endeavor is high and increasing, especially for midge resistance. We plan to increase our efforts in this area and request suggestions from participants in this program for ways that cooperation and information exchange can be improved.

LITERATURE CITED

1. Beck, S. D. 1965. Ann. Rev. Ent. 10:207-232.
2. Blum, A. 1972. In "Sorghum in Seventies." Oxford and IBH Publishing Co. New Delhi, 638 pp.
3. Bottrell, D. G. 1971. Entomological Advances in Sorghum Production. Texas Agri. Exp. Sta. PR-2940.
4. Cates, J. R., Jr., D. G. Bottrell and G. L. Teetes. 1974. Management of the greenbug on grain sorghum. J. Econ. Ent. In Press.
5. Dahms, R. G. 1972. In "Control of Sorghum Shoot Fly." Oxford and IBH Publishing Co. New Delhi, 324 pp.
6. Doggett, H. 1970. "Sorghum." Green and Co. LTD. London and Harlow. 402 pp.
7. Hackerott, H. L., T. L. Harvey and W. M. Ross. 1969. Greenbug resistance in sorghum. Crop Sci. 9:565-658.
8. Hackerott, H. L. and T. L. Harvey. 1971. Greenbug injury to resistant and susceptible sorghum in the field. Crop Sci. 11:641-647.
9. Harvey, T. L. and H. L. Hackerott. 1971. Research on greenbugs and resistance in sorghum. Proc. 7th Biennial Grain Sorghum Res. and Util. Conf., Lubbock, Texas.
10. Johnson, J. W. 1971. Evaluation of sorghum for greenbug resistance. Sorghum Newsletter. 14:114-116.
11. Johnson, J. W. and G. L. Teetes. 1972. Evaluation of sorghum hybrids and lines for adult plant greenbug resistance. Sorghum Newsletter. 15:137.
12. Johnson, J. W., D. T. Rosenow, and G. L. Teetes. 1973. Resistance to the sorghum midge in converted exotic sorghum cultivars. Crop Sci. 13:754-755.
13. Johnson, J. W., D. T. Rosenow, and G. L. Teetes. 1974. Response of greenbug-resistant grain sorghum lines and hybrids to a natural infestation of greenbugs. Crop Sci. 14:442-443.
14. Jotwani, M. G., S. P. Singh and S. Chaudhari. 1971. In "Investigations on Insect Pests of Sorghum and Millets (1965-70)." Final Technical Report of Project No. A-7-Ent-31. Indian Agricultural Research Institute. New Delhi-21. 157 pp.
15. Painter, R. H. 1968. "Insect Resistance in Crop Plants." The University of Kansas Press. Lawrence and Landon. 520 pp.
16. Parodi, R. A., R. D. Gamba, and J. L. Scantamburlo. 1974. "Huerin INTA" Grain sorghum variety tolerant to the "Sorghum Midge" (Contarinia sorghicola Coq.). Sorghum Newsletter. 17:1.
17. Santos, J. H. R., and C. M. Corma. 1974. Evaluation of resistance to Contarinia sorghicola by sorghum lines from the Cameroon, Africa collection, in Ceara, Brazil. Sorghum Newsletter. 17:10-11.
18. Starks, K. J., D. E. Weibel, E. A. Wood, J. W. Johnson, and A. J. Casady. 1971. Greenbug resistance in sorghum. Sorghum Newsletter. 14:97.
19. Starks, K. J., D. E. Weibel, and J. W. Johnson. 1972. Sorghum Resistance to the Greenbug. Sorghum Newsletter. 15:130.
20. Teetes, G. L. and J. W. Johnson. 1972. Mechanisms of greenbug resistance in sorghum. Sorghum Newsletter. 15:135-136.
21. Teetes, G. L., C. A. Schaefer and J. W. Johnson. 1974. Resistance in sorghum to the greenbug; Laboratory determinations of mechanisms of resistance. J. Econ. Ent. 67:393-396.

22. Teetes, G. L., C. A. Schaefer, J. W. Johnson, and D. T. Rosenow. 1974. Resistance in sorghum to the greenbug: Field evaluation. Crop Sci. 706-708.
23. Weibel, D. E., K. J. Starks, E. A. Wood, Jr., and R. D. Morrison. 1972. Sorghum cultivars and progenies rated for resistance to greenbugs. Crop Sci. 12:334-336.
24. Wiseman, B. R., W. W. McMillian, and N. W. Widstrom. 1973. Registration of SGIRL-MR-1 Sorghum. Crop Sci. 13:398.
25. Wiseman, B. R., W. W. McMillian, and N. W. Widstrom. 1974. International sorghum midge evaluations. Sorghum Newsletter. 80.
26. Wood, E. A., Jr. 1961. Description and results of a new greenhouse technique for evaluating tolerance of small grains to greenbugs. J. Econ. Ent. 54:303-305.
27. Young, W. R. 1970. In "Sorghum Production and Utilization." The AVI Publication Company, Inc. 702 pp.

OVERVIEW OF PEST MANAGEMENT AND HOST PLANT RESISTANCE IN U.S. SORGHUM

George L. Teetes

Department of Entomology
Texas A&M University
College Station, TX 77843

ABSTRACT

Within the last decade phenomenal research progress has provided the basis for implementation of successful sorghum pest management programs. A review of this research is presented. Monoculture produced sorghum necessitates sound, integrated insect pest management strategies. Multitactic approaches to deal with the intensified insect/mite pest problems are stressed, with special emphasis on the use of plant resistance. Resistance screening techniques are described.

INSECT AND MITE PESTS OF U.S. SORGHUM

Like most crops, sorghum is usually attacked by only one or two key insect pests in each agroecosystem. Key pests are serious, perennially occurring, persistent species that dominate control practices and, in the absence of deliberate human intervention, commonly attain population densities which exceed the economic-injury level each year, often over wide areas. The sorghum midge, *Contarinia sorghicola*, and greenbug, *Schizaphis graminum*, are key pests of sorghum in the United States.

Secondary pests, although often present in sorghum fields of surrounding areas, rarely occur in economically important numbers. Nevertheless, such pests can exceed the economic injury level as a result of changes in cultural practices or crop practices or crop varieties or because of injudicious use of insecticides applied for key pest. Spider mites, *Oligonychus* spp. are often regarded as secondary pests in sorghum.

A third group, occasional pests, cause economic damage only in localized areas or at certain times. Such pests are usually under natural control and exceed the economic injury level only sporadically. Most pests of sorghum are occasional pests.

A summary of information on the more common sorghum pests in the U.S. is given in Table 1, which includes the pests' scientific name, pest status (key, secondary, occasional), and nature of damage, and also a reference to whether or not plant resistance has been reported.

INSECT/MITE RESISTANT SORGHUMS

To feed and clothe the ever increasing human population will require sound judgement in dealing with the many factors which affect crop production. Host-plant resistance, either alone or more likely in combination with other pest-suppressing methods, is a feasible approach to dealing with insect and mite pests of sorghum. A pest's greatest vulnerability lies in the closeness with which it is associated with its host. A listing of some sorghums reported as insect or mite resistant is shown in Table 2.

Table 1. Common Insect and Spider Mite Pests Injurious to Sorghum in the U.S.

Common Name	Scientific Name	Pest status[a]	Nature and symptoms of damage	Resistance reported
Soil pests				
White grub	*Phyllophaga crinita*	Occ	pruning of roots, seedling death, stunting and/or lodging	no
Wireworms	Several species of true and false wireworms (*Eleodes*, *Conoderus*, *Aeolus*)	Occ	destroy planted seed, stand loss	no
Rootworms	*Diabrotica* spp.	Occ	pruning and tunneling of roots, stunting and dead heart	no
Aphids				
Greenbug	*Schizaphis graminum*	Key	suck plant sap, injects toxin that kills leaves, virus vector disease predisposer	yes
Yellow sugar-cane aphid	*Sipha flava*	Occ	suck plant sap, injects toxin that kills leaves	no
Corn leaf aphid	*Rhopalosiphum maidis*	Occ	suck plant sap, virus vector	yes
Armyworms				
Fall armyworm	*Spodoptera frugiperda*	Occ	leaf feeder in whorl or destruction of seed in head	yes
Beet armyworm	*Spodoptera exigua*	Occ	leaf feeder in whorl and on leaf margins	no

Table 1, cont.

Common Name	Scientific Name	Pest status[a]	Nature and symptoms of damage	Resistance reported
Stem Borers				
Southwestern corn borer	Diatraea (=Zeadiatraea) grandiosella	Occ	some leaf feeding, boring in stalk, stalk lodging	yes
Sugarcane borer	Diatraea saccharalis	Occ	some leaf feeding, boring in stalk, stalk lodging	yes
European corn borer	Ostrinia nubilalis	Occ	some leaf feeding, boring in stalk, stalk lodging	yes
Lesser Cornstalk borer	Elasmopalpus lignosellus	Occ	boring in stalk at soil surface of seedling plants	no
Sugarcane rootstock weevil	Anacentrinus deplanatus	Occ	boring in stalk and roots above and below soil surface causes lodging	no
Chinch bug	Blissus leucopterus	Occ	suck sap from leaves and stems	yes
Spider mite				
Banks grass mite	Oligonychus pratensis	Sec	suck plant sap causing discoloration and death of leaves	yes
Sorghum midge	Contarinia sorghicola	Key	destroys developing seed	yes
Head Caterpillars				
Sorghum webworm	Celama sorghiella	Occ	destruction of seeds in head	no
Corn earworm	Heliothis zea	Occ	leaf feeder in whorl or destruction of seed in head	no

Table 1, cont.

Common Name	Scientific Name	Pest status[a]	Nature and symptoms of damage	Resistance reported
Head bugs				
False chinch bug	Nysius raphanus	Occ	feed on developing seed causing smaller, lighter distorted seed	no
Stink bugs	Pentatomidae, e.g. Solubea pugnax	Occ	feed on developing seed causing smaller, lighter distorted seed	yes
Leaffooted bug	Leptoglossus phyllopus	Occ	feed on developing seed causing smaller, lighter distorted seed	no
Stored grain pests				
Rice weevil	Sitophilus oryzae	Key	consume whole grain in field and storage	yes
Maize weevil	Sitophilus zeamais	Occ	consume whole grain in storage	yes
Angoumois grain moth	Sitotroga cerealella	Key	consume whole grain in field and storage	yes
Lesser grain borer	Rhyzopertha dominica	Occ	consume whole grain in storage	no
Indian-meal moth	Plodia interpunctella	Occ	feed on cracked grain, a secondary feeder	no
Grain mite	Acarus siro	Occ	feed on cracked grain, a secondary feeder	no
Red flour beetle	Tribolium castaneum	Occ	feed on cracked grain, a secondary feeder	yes
Confused flower beetle	Tribolium confusum	Occ	feed on cracked grain, a secondary feeder	no

[a]Occ = occasional, Sec = secondary, and Key = key pest

Table 2. Sorghums Resistant to Insects and Mites.

Insect/Mite Species	Variety/Line	Location Reported
Schizaphis graminum	IS 809 (PI 221613)	USA
	KS 30	USA
	SA 7536-1 (Shallu)	USA
	PI 264453	USA
Rhopalosiphum maidis	Piper sudan 428-1	USA
	TAM 428	USA
Spodoptera frugiperda	Freed (PI 29166)	USA
Ostrinia nubilalis	Kafirs (several)	USA
	Feterita	USA
	Shantung Brown Kaoliang	China
Diatraea grandiosella	Y-4 (Tex 63 x Kaura)	USA
	NK x 3007	USA
Diatraea saccharina	King's Diamond	USA
Blissus leucopterus	Atlas	USA
	Axtel	USA
	Dwarf Kafir 44-14	USA
	Redline 60	
Oligonychus pratensis	Rio (SC 599-6)	USA
	IS 12568 (SC 56-6)	USA
Contarinia sorghicola	Nunaba	Africa
	Huerin Inta	Argentina
	AF 28	Brazil
	SGIRL - MR - 1	USA
	TAM 2566 (IS 12666C)	USA

Table 2, cont.

Insect/Mite Species	Variety/Line	Location Reported
	IS 25016 (SC 52)	USA
	IS 2508C (SC 414)	USA
	IS 2549C (SC 228)	USA
	IS 2579C (SC 423)	USA
	IS 3071C (SC 237)	USA
	IS 8100C (SC 424)	USA
	IS 12612C (SC 112)	USA
Solubea pugnax	White Darso (Kans. 33 - 378)	USA
Sitophilus oryzae	Double Dwarf Early Shallu	USA
	Sagrain	USA
Sitophilus zeamais	Double Dwarf Early Shallu	USA
	Early Kalo	USA
	Early Sumac	USA
Sitotroga cerealella	Double Dwarf Early Shallu	USA
	Early Kalo	USA
	Early Sumac	USA
Rhyzopertha dominica	Double Dwarf Early Shallu	USA
	Early Kalo	USA
	Early Sumac	USA
Tribolium castaneum	Double Dwarf Early Shallu	USA

Sorghum resistance to insect and mite pests is best viewed as a component of a pest management system which can interact with and influence other components in the system. Insect-resistant sorghum may inhibit a pest's ability to attain the economic injury level because of nonpreference or antibiosis. Or, it may increase the damage tolerance level of the crop because of a tolerance mechanism. Similarly, insect-resistant sorghums could create a situation where natural biological control agents are more efficient because of slower rates of increase of the pest. Of course, resistant plants must allow the pest to reach a "threshold" of predation or parasitism. Low densities of the pest species may provide a pivotal reserve food supply for beneficial organisms needed later in the growing season in sorghum or neighboring crops. Also, resistant varieties may contribute to the effectiveness of insecticides or make it possible to omit or reduce treatments.

RESISTANCE SCREENING TECHNIQUES

Procedures used to screen sorghums for resistance to insects and mites are based on the nature of damage and resulting injury symptoms caused by the pests. Progress in selection for resistant plant types is dependent upon uniform and sufficient selection pressure. Although selection can be done under natural infestation, frequently the pest population and/or crop must be manipulated to insure selection pressure and to avoid chance escape of susceptible plant types. For convenience general procedures are provided for pest groups or major pest species. Modification of existing screening and evaluation procedures are often required. The techniques described are a compilation of those reported in the literature and the amount of detail varies with the amount of emphasis the species or group has received.

Aphids. Several aphid species infest sorghum. Resistance of seedlings and mature plants can be successfully evaluated under natural pest infestation, provided populations reach uniform, high levels at appropriate times. Due to the fluctuation and seasonality of natural aphid populations, techniques are often required to evaluate seedling and adult sorghum plants for resistance in the absence of natural infestations. The following techniques are common for evaluation of sorghums resistant to the greenbug, *Schizaphis graminum* (Johnson *et. al.* 1976, Starks and Burton, 1977).

Greenhouse Seedling Screening. Aphids are reared in a greenhouse on culture plants, usually sorghum, grown in 20.3 cm plastic pots or 3.8 liter metal cans. A 3:1:1 mixture of soil, sand, and peat is the preferred growth medium. If soil alone is used, sand should be used to cover seed. A small amount of complete fertilizer is added to the soil medium. Soil mixtures are sterilized if plant diseases are a problem. Seed should be treated with a fungicide to control soil-born diseases. From 30 to 50 seeds per container are planted to a depth of 2.5 cm or covered with sand to that depth. Sand prevents excess water evaporation and aids in keeping cage bottoms clean. Prior to plant emergence, a bottomless, cylindrical cage is inserted into the soil or sand to protect emerging plants from premature aphid infestation and to exclude extraneous insects, especially predators and parasites. Cages are constructed of 0.0381 cm thick, clear, vinyl plastic or transparent, polished, nitrocellulose film, .05 cm thick. Cage size varies but cages approximately 33 cm high and 14 cm in diameter are commonly used. Ventilation is provided by cutting two 6.3 cm diameter holes in opposing sides of the cage. The ventilation holes and cage tops are covered with fine-mesh nylon cloth glued with rubber cement. These cages can be washed with a mild soap and reused. Once

plants attain a height of 15-20 cm (ca. 2 weeks), they are infested with ca. 200 aphids. The culture should have a maximum number of aphids two weeks later (Fig. 1).

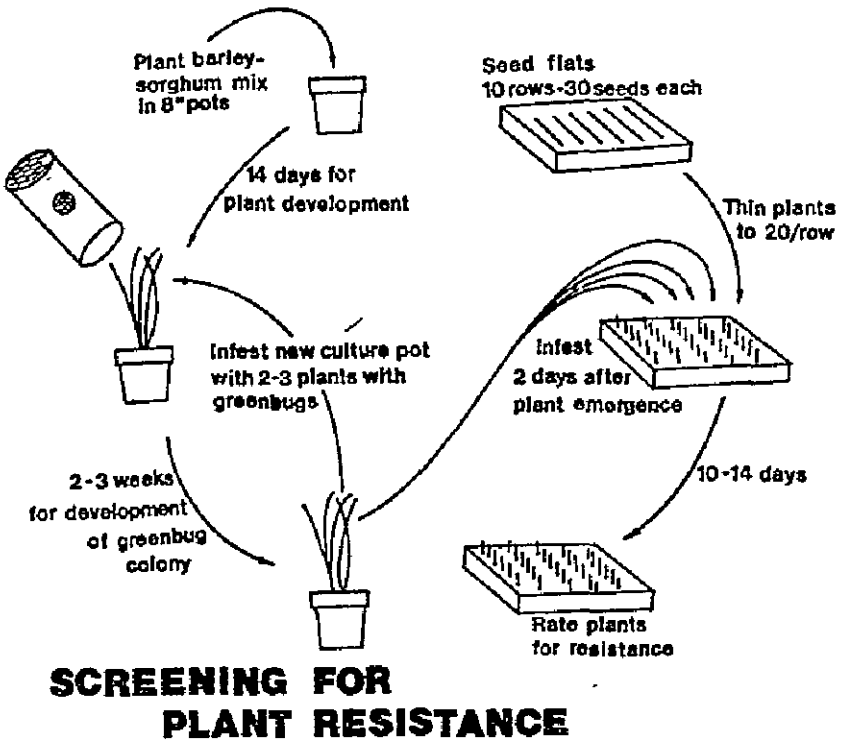


FIG. 1. Screening for plant resistance.

Temperature and humidity requirements are often dictated more by the aphid-culture plants than by aphids. Greenbugs, for example, reproduce in a temperature range of 15.6°-32.2°C, although fecundity maximizes at ca. 22.2°C at which temperature each ovoviviparous female produces an average of 100 nymphs over a 20-day period.

Breeding lines to be evaluated for resistance are planted in galvanized metal flats 35.6 wide x 50.8 long x 9.5 cm deep filled with soil to about 2.5 cm from the top. Ten, equally spaced rows about 2.5 cm deep are made in each flat by pressing a planting board on the top of the soil. Each flat may accommodate 10 entries if one entry is planted per row or 20 entries in rows 17.8 cm long. A test can contain any number of flats, depending upon the supply of aphids and available greenhouse space.

Approximately 20-30 seeds of each entry are planted per row and thinned to about two-thirds that number one week after plant emergence. Whenever possible, known resistant and susceptible lines should be planted in each flat as controls. If breeding selections from resistant crosses are to be evaluated, the resistant parent used in the cross should be included as the resistent control.

After thinning, plants are infested with aphids of all ages and instars by brushing or shaking aphids from culture plants fairly uniformly over flats, or by placing uprooted, infested, culture plants between rows and allowing the aphids to crawl to the test plants. Plants are examined about two days after being infested and additional aphids are applied to flats which have inadequate infestations. From four to ten greenbugs per plant are considered adequate. Test flats are left uncovered before and after infestation.

Generally, plants in each flat are rated for resistance when plant in the susceptible, control row are near death, usually ca. 10-14 days post infestation. A visual rating of an entire row is possible for nonsegregating material; in segregating rows, individual plants can be rated. A 0 to 9 rating system for seedling evaluation is feasible, where 0 = no damage, 1 = 10-20% plant necrosis, 2 = 21-30%, etc., and 9 = 91% necrosis or a dead or dying plant.

Adult Plant Screening. Leaf damage ratings are fairly easy to make and offer a good measurement of resistance if an adequate, natural, aphid infestation occurs. The following rating system is feasible for field evaluation: 0 = no damage; 1 = red spotting on leaves; 2 = portion of leaf killed; 3 = 1 leaf killed; 4 = 2 leaves killed; 5 = 4 leaves killed; 6 = 6 leaves killed; 7 = 8 leaves killed; 8 = 10 leaves killed; and 9 = dead plant. Data may be collected at any plant growth stage when aphids are present and aphid numbers should be estimated. Also, plant growth stage should be recorded at the time aphid counts and damage ratings are made. The growth stages described by Vanderlip (1972) are recommended. If aphid populations differ markedly among entries, an indication of the level of infestation on each entry can be made using the following code after the rating: 1 = low incidence, 2 = average incidence, and 3 = high incidence.

An alternative to natural infestations of aphids in the field is the use of cages. Cages can be relatively large to enclose groups of entire plants or they can be small plastic cages attached to a portion of a leaf. Large cages should be constructed of metal frames covered with small mesh screen to exclude parasites. In large cages aphid population levels increase rapidly, often to unnaturally high levels. Populations of aphids on artificially infested plants under cages usually increase rapidly and damage may be accentuated. Small plastic clip-on cages can be used in the field for evaluation of resistance. Small

cages (2.54 cm^3) clipped to leaf blades need cloth-covered ventilation holes (1.9 cm in diameter) on at least one side. Five to 10 aphids, usually adults, are put in each cage with a small artist's brush. The cages keep the aphids confined to a small area and exclude parasites and predators. Cages are inspected the day following attachment to the leaves to insure that all aphids remain alive and feeding on the plant. Additional aphids are added where necessary to ensure equal numbers per cage. Ratings of the damaged leaf area covered by the cage begin about one week after infestation and continue at two-day intervals until the caged area of the susceptible plants are near death. A feasible rating scheme is as follows: 0 = no necrotic plant tissue in the caged area, 1 = 10-20% necrosis, 2 = 21-30%, and so on to 91+% necrosis.

Armyworms, Panicle Caterpillars, and Borers. A large complex of caterpillars attack sorghum as foliage and grain feeders or as stalk borers. Different species are encountered in different geographic locations. Most sorghum screening trials involving lepidopterous species have been conducted in the field using natural pest infestations. Greenhouse screening techniques have been reported for a few of the species attacking sorghum, and for a somewhat larger number, a technique combining greenhouse or laboratory culturing and artificial inoculation in the field has been used (Mayo 1967). Compared to sorghum, considerably more progress has been made in screening for resistance to lepidopterous pest in corn.

Late planted sorghum is usually more heavily and uniformly infested with most lepidopterous species than in early planted sorghum (Chada 1962). Field screening trials planted where stubble exists from previously planted sorghum helps ensure natural infestations of stalk borers. Plant escape is a problem associated with field screenings by natural infestation.

Several caterpillar species can be reared on artificial diet, sometimes allowing mass rearing for use in screening when natural populations of the pest species cannot be relied upon. Using diet-produced insects, it is possible to ensure uniform test-plant infestation at desired plant growth stages. The plants are infested with eggs and/or larvae. In greenhouse screening trials, with foliage feeding caterpillars, 2nd or 3rd instar larvae are placed on plants grown in flats as described earlier for aphid screening (McMillian and Starks 1967). Generally, the severity of damage is recorded by visual classification of damage using a rating system of 1-9 (Wiseman et. al. 1966) or 0-5 (Hormchong 1967). Resistance to panicle feeding larvae is based on relative amounts of damaged seed (Buckley and Burkhardt 1962, 1963).

Commonly, especially for several borer species, egg masses in the "black head" stage are placed on the underside of the top leaves of individual test plants (Dicke et. al. 1963). Reaction to borers is generally based on percentage infested stalks (cavities in the peduncle area), length of tunnel and percentage stalk breakage and lodging. Early leaf feeding by borers may or may not be highly correlated with stem tunneling, and length of tunneling may not be related to yield.

Sorghum Midge. Greenhouse techniques for screening sorghums for resistance to the sorghum midge are not available because techniques for artificially rearing the insect have not been developed. Presently, naturally occurring infestations in field plantings, must be relied upon. The unreliability and/or fluctuations of midge population levels and variation of maturity in test plants, as in segregating rows, are inherent problems associated with field screening sorghums for this pest.

Damaging midge population levels are best attained by delayed planting, multiple plantings of the same test materials, and/or the use of earlier planting of susceptible sorghums in which midge populations reach high levels by the time the test plants are blooming. In the latter technique, susceptible sorghums are planted at 10 day to two-week intervals beginning as early in the season as practical, thus providing a continuous supply of blooming sorghum over an extended period. Also, bulks (mixtures) of sorghum seed of varying maturity lines may be planted as spreader rows between blocks of test plants.

Midge damage is usually rated as percent "blasted" seed. Generally, plants cannot be rated sooner than 20 days after bloom. Individual heads in a row are rated and a mean damage rating calculated; or the entire row is rated by visual observation. A feasible rating scheme is as follows: 0 = no damage, 1 = 10-20% blasted seed, 2 = 21-30%, etc., and 9 = 91+% blasted seed. A more objective evaluation can be obtained by "protecting" portions of the test plants with pollinating bags or insecticides. Seed yield comparisons of protected and unprotected heads are then made. Standard resistant and susceptible varieties should be included as controls.

Adult midge population levels should be estimated. This is accomplished by visual examination of blooming heads or by placing a large mouth jar or plastic bag over the heads to capture adults. Days to 50% bloom of each entry should be recorded and correlated with any fluctuation in midge adult abundance. Care should be taken to differentiate midge damage from bird damage and from sterile florets. The presence of a midge cocoon at the tip of the glumes is proof that a seed was damaged by midge.

Chinch Bug. Infestations of chinch bugs in sorghum generally result from mass migration of the pest from winter-grown small grains. Screening for and evaluating chinch bug resistant sorghums is most commonly accomplished by field trials under natural infestations (Snelling et. al. 1937). Field infestations are increased by growing sorghums adjacent to infested small grains. Growing sorghums of mainly milo germ plasm in the test may assist in increasing the infestation level and these same sorghums can serve as susceptible checks. Greenhouse screening of seedling plants for chinch bug resistance might be accomplished by slightly modifying the technique previously described for aphids, if the pest is reared artificially (Parker and Randolph 1972). Field-collected bugs might be used for greenhouse screening purposes, although the use of this method has not been reported.

Chinch bug damage to sorghum is recorded as percentage of plants killed or injured (Snelling et. at. 1937). The degree of stunting and yield reduction are additional criteria for measuring relative resistance. Although no damage rating schemes have been reported, one based on the amount of reddish discoloration on sorghum stems could be used. Rating damage to sorghum is complicated by date of planting, earliness, drought, and varietal adaptation (Painter 1951, 1958). Seedlings are more readily damaged than more mature plants, and the latter may not be killed but will show the characteristic stem discoloration.

Spider Mites. Screening sorghums for resistance to spider mites is relatively difficult because many factors affect mite abundance and plant reaction to infestation. The more serious species generally require hot, dry, climatic conditions for injurious populations to develop. Also, mite infestations tend to be most damaging to sorghum that has attained the reproductive

growth stage (Ehler 1974). All these factors hinder greenhouse screening (Owens et. al. 1976).

Field evaluations depend on the availability of injurious mite densities. Manipulating planting to coincide with dry environmental conditions during sorghum reproductive stages often increases mite abundance. Moisture stressed plants are generally more heavily infested with mites than nonstressed plants. Plant maturity influences the reaction of plants to mites.

Rating schemes are generally based on percent leaf area damaged by mites (Foster et. al. 1977). Mites generally begin feeding in colonies adjacent to the midrib of the leaf. Feeding is accompanied by webbing and desiccation of the leaf tissues beginning at the tip and margins of the leaves. Mites generally cause damage to sorghum after it has bloomed; however, ratings are made at any stage of plant growth at which there are sufficient mite infestations. Stalk death ratings may be made when severe damage occurs. A feasible rating scheme is as follows: 0 = no damage, 1 = 10-20% leaf necrosis or stalk death, 2 = 21-30%, etc., and 9 = 91+% or dead plant. Plant lodging and differential seed weight reduction among varieties can also be used. To record mite abundance, a population density rating is used where 1 = no mites, 2 = few individuals above midrip only, 3 = colonies along midrib, 4 = mites spreading away from midrib, and 5 = entire leaf covered with mites (Foster et. al. 1977).

Stored Grain Pests. A common practice in searching for resistance to stored grain insects among varieties of sorghum is to infest a grain sample of each variety with a specific number of insects for a period of feeding and oviposition. Resistance is then evaluated by counting damaged grains and/or 1st generation progeny (Stevens and Mills 1973). Parent insects, in some cases, are given free choice of all samples tested; in others, they are confined to specific samples of grain (Rogers and Mills 1974a). Equilibration of the relative humidity and seed moisture content of the test samples is necessary for valid results (Roger and Mills 1974b).

Evaluations of groups of varieties involve small samples, typically 50-100 kernels contained in small plastic boxes measuring 48x48x18 mm (Mills 1976). Three to five samples of each variety are included in each tests. In no-choice tests six females and three males are used to infest each sample, allowed a 5-7 day ovipositioning period, and then removed. The relative resistance ranking of the entries is based on numbers of emerged progeny/sample. Resistance is evaluated by recording the numbers of larvae that develop to adults.

In free-choice tests, samples of grain are arranged in a circle equidistant from the center of the circular test chamber (Stevens and Mills 1973). An appropriate number of adult test insects are dropped into the center of the chamber and allowed free access to all the grain samples. Insects within each sample are then counted in 4-7 days for nonpreference evaluation and then removed. Grain samples are then held to evaluate for progeny emergence. Free-choice tests probably are more practical where large numbers of entries are to be evaluated because obviously susceptible entries can be quickly eliminated.

As stated previously, the most commonly used measure of resistance has been progeny production. Other factors have been used such as developmental periods and weights of progeny. Dobie (1974) proposed an index of susceptibility based on progeny numbers and development periods. Soft X-rays have been used in resistance screening trial to detect developing larvae within the grains (Russell 1962).

DETERMINATION OF RESISTANCE MECHANISMS

Plant resistance to insect and mite pests commonly is divided into three basic types of mechanisms: nonpreference, antibiosis, and tolerance (Painter 1958). Resistance mechanisms involved in host-pest relationships can be determined, but the evaluation techniques differ from those normally used in screening diverse germ plasm. Resistance is usually a result of more than one mechanism.

Differential preference reactions of insects and mites to resistant and susceptible sorghums is usually determined by allowing the pest a choice of resistant and susceptible plants. In the laboratory or greenhouse, this is done by randomly planting resistant and susceptible selections in circular arrangement in pots or metal cans. After emergence, each entry is thinned to one plant. Pest insects are then placed on the soil surface in the center of the circle of plants at a rate five times the number of test plants. Test plants are covered with a cage. Insects present on individual test plants are counted at 24 hr intervals for at least four days after initial infestation. Frequently some insects die or are lost during the testing procedure.

Antiobiosis is expressed as an adverse effect of the plant on the biology of the insect or mite. Techniques are used which allow the experiment to the compare the fecundity, size (weight), longevity, and increased mortality of the pest on resistant versus susceptible plants. Some "conditioning" of the insect on the resistant plant is normally required.

Of the three resistance mechanisms, tolerance is perhaps the most difficult to quantify. It basically involves a comparison of pest numbers to subsequent plant damage. Consequently, pest numbers must be determined and related to visible damage, and, eventually, to yield.

SORGHUM INSECT PEST MANAGEMENT

The role of host plant resistance in sorghum pest management in the USA and the implications of this vital component can be seen in the examples that follow.

Chinch Bug. Painter (1951), in discussing insect resistant sorghums, devoted most of his attention to the chinch bug, as it was the most important pest of sorghum at that time. However, for almost 20 years this species was largely an insignificant consideration in sorghum production. It is only speculation, but possibly chinch bug resistant sorghums were at least partly responsible for the decreased severity of the pest. In some areas, during most years, resistant sorghum varieties proved to be a most practical means of control and largely replaced the creosote barriers which were once recommended for control of populations migrating from small grains.

Research conducted prior to the introduction of combine-type sorghum hybrids revealed that most milo varieties of sorghum are highly susceptible to chinch bug damage (Hayes 1922). Several of the sorgo varieties are relatively resistant, Aglas sorgo being one of the most resistant. Most of the kafir varieties also exhibit resistance; whereas, the feteritas are susceptible or intermediate in reaction (Dahms 1943). Data collected for the period 1870 to 1923 showed that considerably more kafir than milo sorghum was grown in the chinch bug infested areas of Oklahoma, Kansas and Texas, whereas more milo sorghum was grown in areas where the pest was less serious (Snelling et. al. 1937).

Factors influencing resistance among varieties tested are tolerance of plants, differences in food values for the bugs, preference of bugs for susceptible varieties, and specific morphological characters of plants, such as loose fitting leaf sheaths which tend to be associated with increased susceptibility. Sorghums which produce a dense canopy tend to be less infested. Hybrid vigor reflected in rapid and heavy plant growth also tends to reduce chinch bug damage.

The relative chinch bug resistance of current sorghum hybrids is largely unknown and recent literature on the subject is lacking. Based on the early literature, sorghum hybrids possessing kafir germ plasm such as Combine Kafir 60 should exhibit a relative level of resistance. In contrast, sorghum hybrids possessing milo germ plasm could be relatively susceptible. The same may be true for Wheatland which is also susceptible (Dahms and Sieglingen 1954). In very recent years, chinch bugs have caused serious damage to sorghum adjacent to small grains and these situations point to the need for assessing currently used germ plasm.

Sorghum Midge. The sorghum midge is an introduced pest, though to have originated with the crop in Africa and spread with it around the world. Its major hosts are all members of the genus Sorghum. The midge is probably the most important sorghum insect pest causing losses amounting to millions of dollars in Texas. The midge infests sorghum in all areas of the state except the Northern Panhandle (Rummel and Daniels 1971).

The small orange-red colored female midge, less than 2 mm in length, oviposits in flowering spikelets. These small flies live for only a day or two, but may lay 50-250 eggs. The eggs hatch in about two days and the orange colored larvae complete their development in 9-11 days, pupating within the spikelet for about three days. The injury is caused by the larvae feeding on the ovary preventing normal development and resulting in "blasted seed." A generation may be completed in 14-16 days. This rapid developmental cycle permits 9-12 generations during a season, and permits the build-up of high midge densities where sorghum flowing times are extended by a wide range of planting or maturity dates.

Since the development of damaging infestations of midge is dependent on progressive buildup on Johnson grass and sorghum, a very effective cultural recommendation became a key component in the management strategy. This practice is early, uniform planting of sorghum so that flowing occurs throughout an area during a relatively short period, prior to the presence of damaging midge densities.

Relatively small acreages of sorghum are now planted late, but those that are may require insecticidal treatment to prevent loss. For these situations, judgement decisions for insecticidal control are based on economic threshold levels. Research has shown, that fields should be carefully inspected daily for adult midge, beginning when panicles first start to flower at the tip. Treatments should be applied when 25 to 30 percent of the heads have begun to flower and number of midge adults per panicle exceeds one. If adults are still present three to five days later, a second treatment should be made. Three applications at three to five-day intervals may be justified if yield potential is high and midges are abundant.

Another tactic may soon be added to the midge management strategy. Sorghum lines from the Texas Sorghum Conversion program were discovered that are resistant to the pest (Johnson et. al. 1973) (Table 3). These resistant sorghums will

provide not only a means of reducing midge damage, but also will provide flexibility for planting time and reduce insecticidal treatments. This would be advantageous particularly during seasons when weather conditions are unfavorable for early planting. In some areas of the state, especially the High Plains, sorghum could be planted later in the season which would allow for more efficient use of natural rainfall or permit sorghum to develop during the cooler, wetter part of the season. This is advantageous yield-wise, reduces irrigation requirements and provides another potential approach to dealing with the Banks grass mite which increases in abundance after the heading stage of sorghum during periods of hot, dry weather.

Present research in Texas is directed toward the development of commercially acceptable midge-resistant sorghum hybrids and the determination of resistance mechanisms. Resistance is not a dominant inherited character, and is apparently polygenic in nature. Both parental lines must possess the resistant character and experimental hybrids of resistant x resistant parents have exhibited high and effective levels of midge resistance. Preliminary research on resistance mechanisms indicates antibiosis to be the major resistance mechanism (Table 4 & 5), but levels of nonpreference also are exhibited (Table 6).

TABLE 3. Midge damage ratings of eight sorghum selections from converted exotic lines and five US cultivars; Lubbock, Texas, 1972. (Johnson et. al. 1973).

Designation	SC no.[2/]	Group[3/] no. and name	Mean[4/] score	Days to bloom for June 20 planting
IS 12612C	112	39(1) Zerazera	3.0	66.0
IS 12666C	175	39(1) Zerazera	3.0	66.5
IS 2508C	414	38 Caudatum/Kafir	3.0	68.0
IS 2816C	120	39(1) Zerazera	3.5	70.0
US 3574C	239	39(1) Zerazera	3.5	70.0
IS 12608C	108	39(1) Zerazera	4.5	68.0
IS 12664C	173	39(1) Zerazera	4.5	69.5
IS 2597C	423	39(1) Zerazera	4.5	68.0
Tx 2536			6.0	63.0
Tx 09			6.5	66.0
B Tx 378			7.5	74.5
B Tx 398			8.9	66.5
Tx 7078			9.0	60.5

1/ an IS number is given to all sorghums in the world collection; the C following IS number denotes that the sorghum lines has been converted.

2/ the SC number denotes the code by which these sorghums were identified during conversion.

3/ the group number and name of the exotic variety based on a Modified Snowden's classification. All selections are in the race Caudatum using classification by Harland and deWet.

4/ 1 = <5% damage, 2 = >5<10%, 3 = >10<20%, 4 = >20<30%, 5 = >30<60%, 6 = >60<70%, 7 = >70<80%, 8 = >80<90%, and 9 = >90%

TABLE 4. Number of sorghum midge adults emerging from sorghum panicles.

Line	Mean no. of adults/sample	% difference in emergence compared to Tx 7000
TAM 2566	63.6	98.6
SC 423	394.7	91.3
TAM 428	1421.8	68.6
Tx 7000	4533.8	—

TABLE 5. Percentage of cage spikelets of TAM 2566 and Tx 7000 found to be infested with each life stage of the sorghum midge.

Stage	Line		Difference between lines infestation level (%)
	Tx 7000	TAM 2566	
Egg	22.2	7.8	64.8
Larva	10.3	5.1	50.9
Pupa	6.6	0.1	98.8
Adult	0.5	0.1	85.2

TABLE 6. Comparison of sorghum midge adult members per panicle collected by plastic bag sampling to determine non-preference among resistant and susceptible sorghums.

Line	Mean no. adults collected daily	
	1976	1977
TX 7000	107.6	35.6
TAM 428	44.7	19.0
SC 423	50.3	11.3
TAM 2566	11.8	2.5

Greenbug and Other Aphids. The first outbreak of the greenbug in sorghum occurred in 1968 in almost all areas of the United States where sorghum is produced. This greenbug was identified as biotype "C" and differed in host and variety response from the previous biotypes "A" and "B" which attacked small grains. Their migration pattern changed in 1968 (Daniels 1977).

The greenbug is approximately 1.6 mm long, light green in color with a darker green dorsal abdominal stripe. The distal leg segments and tips of the cornicles are black. Alate and apterous forms may be present in the same colony. Females produce living young parthenogenetically and under optimum conditions, the young begin reproduction in about 7 days, producing about 80 offspring during a 25-day period (Almand et. al. 1969).

Infestations are detectable by reddish spots on the leaves caused by toxins injected into the plant by aphids feeding in colonies on the underside of leaves. The reddened areas enlarge as greenbug numbers increase, and the leaf may die, turning brown from the outer edges toward the center. Also, greenbugs transmit maize dwarf mosaic virus (MDMV) and may predispose sorghum to charcoal rot (Daniels and Toler 1971, Frederiksen and Daniels 1970, Teetes et. al. 1973).

Greenbug is presently a key pest of sorghum in most areas of the United States where the crop is grown, especially the Great Plains. Small grains, primarily wheat, provide a winter host. Where the growing season of this crop does not overlap that of sorghum, grasses such as Johnson grass, Sorghum halepense, Linn., serve as interim hosts (Fig. 2) (Lopes and Teetes 1976). Greenbug may be a pest during the seedling stage of sorghum though commonly it does not reach damaging proportions until after heading. In either case, sorghum generally becomes infested soon after emergence. The aphid's rate of increase has been recorded in the field to be as high as 20 fold per week with an average of 5-6 fold per week throughout the season (Fig. 3) (Bottrell 1971). Spring rains and predators can suppress this increase.

Seasonal abundance profiles of the pest in sorghum in the Texas High Plains have shown peak density levels to occur in late July to early August (Bottrell 1971, Teetes 1971). Abundance of natural enemies has shown a characteristic lag time of about one-two weeks, and native aphid predators often do not hold greenbugs in check (Kirby and Ehler 1977). The major seasonal mortality factor of the greenbug has been parasitism by Lysiphlebus testaceipes (Cresson) which usually causes a rapid decline in greenbug population levels by mid-August (Fig. 3) (Teetes et. al. 1975, Walker et. al. 1973). Consequently, only about a two-week period exists in which greenbug numbers may exceed the damage tolerance level of sorghum.

High rates of persistent systematic insecticides were initially relied on as the sole controlling agent of the greenbug in sorghum. These treatments were highly effective, but at the same time were broadly toxic and ecologically disruptive. Consequently, a system of integrated control was developed. Ecological selectivity was achieved by dosage rate manipulation of several approved organophosphorous insecticides (Cate et. al. 1973). Extremely low dosage rates ($\leq$ .1 lb. AI/acre) provided greenbug control yet spared most of the naturally occurring beneficial species (Teetes 1972, Teetes et. al. 1973). Treatments with selective insecticide rates have the effect of throwing the balance back in favor of natural control by preserving natural enemies. Such selective treatments are applied when the aphid population reaches the economic threshold (Teetes and Johnson 1973, 1974). When properly timed, only one insecticide application is required and resurgence of the greenbug is prevented by parasites and predators.

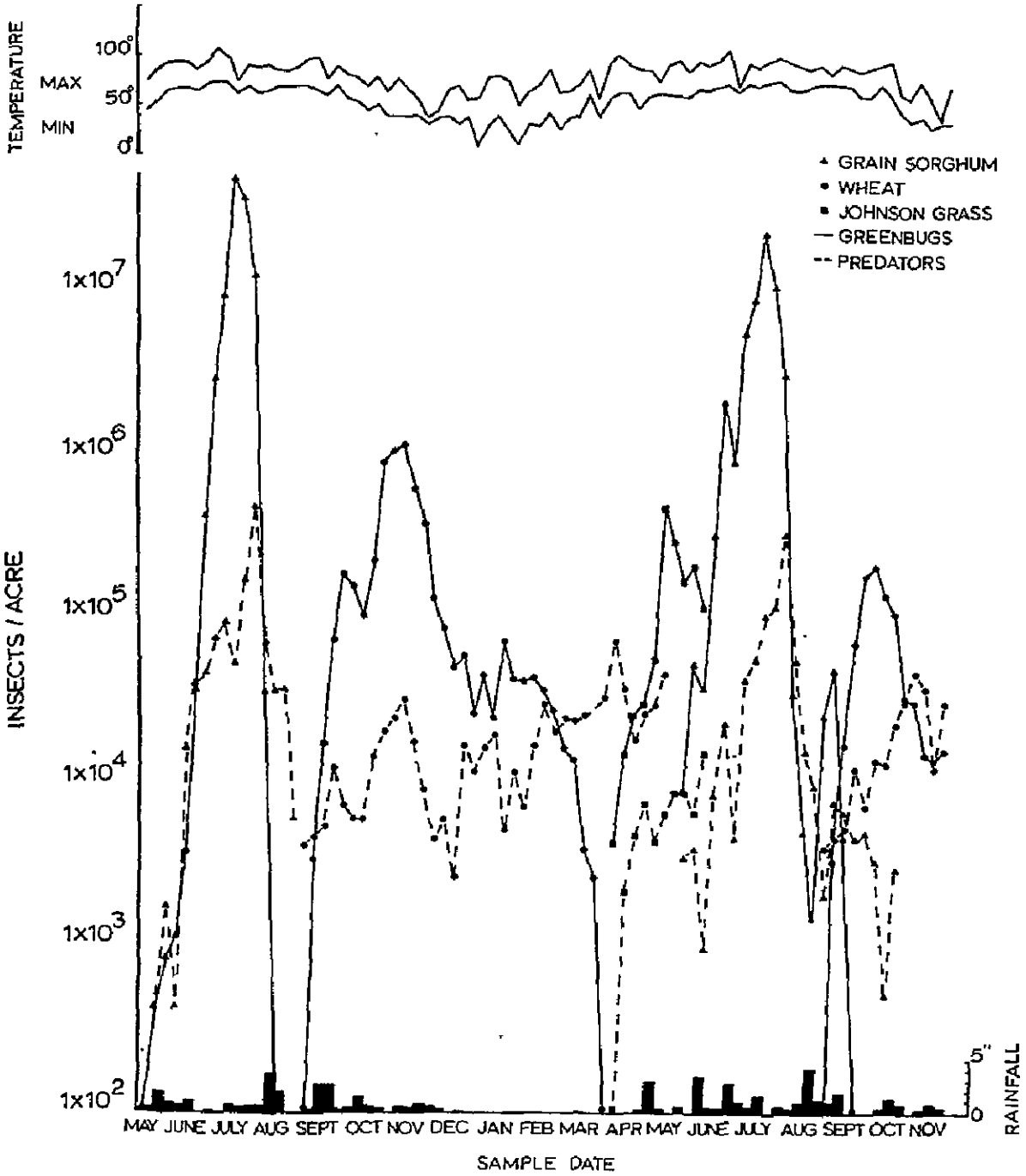


FIG. 2. Seasonal abundances of greenbug and selected beneficial species in sorghum, wheat and Johnson grass and temperature and rainfall, 1971-73.

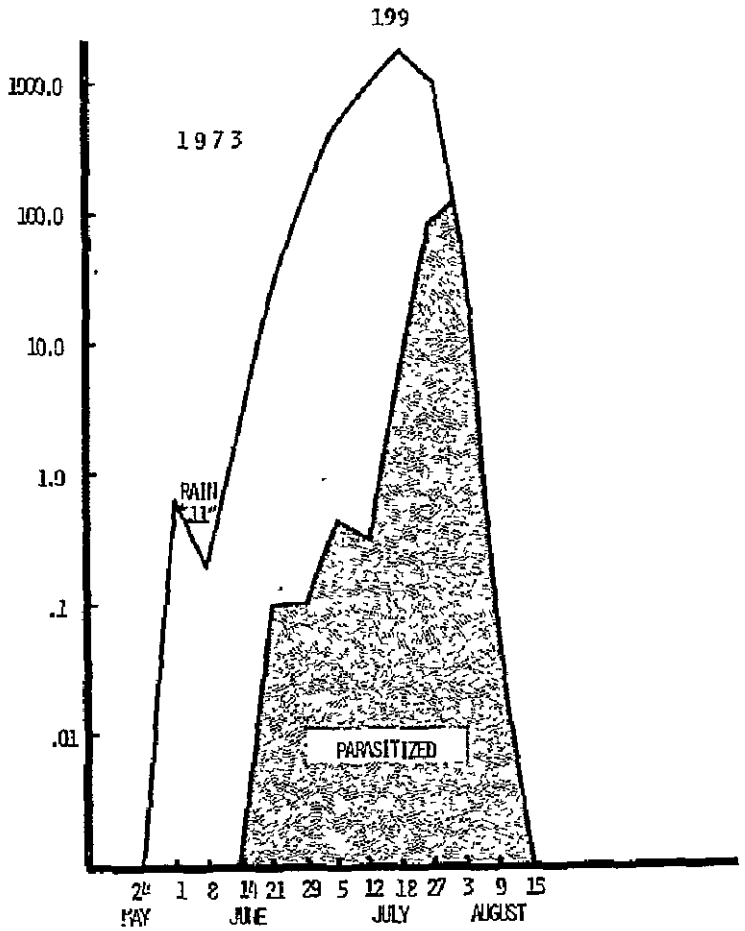


FIG. 3. Seasonal abundance and parasitism of the greenbug in grain sorghum in the Texas High Plains.

Recent development of insecticide resistance by the greenbug greatly hamper the use of selective insecticide rates. Laboratory tests confirmed field observations of resistance development at levels of about 30, 10, 3 and 5-fold for disulfoton, dimethoate, phorate, and parathion, respectively (Fig. 4) (Teetes et. al. 1975). A continuing effect is being made to identify alternate, effective insecticides (Daniels and Chedester 1975, 1976).

Soon after 1968, sources of greenbug resistant germ plasm were found in sorghums such as SA 75361-1, KS 30, IS 809, and PI 264453 (Johnson et. al. 1974). Releases of resistant breeding materials were made to commercial seed companies. Laboratory and field experiments have identified the mechanisms of resistance in these sorghums as moderate levels of nonpreference and antibiosis. Antibiosis is expressed as an increase in the duration of the developmental stages and a decrease in progeny per adult, adult longevity and duration of the reproductive period. The primary resistance mechanism has been shown to be tolerance (Teetes et. al. 1975). Greenbug resistant sorghum of Tunis grass background reduces greenbug numbers as a result of non-preference and antibiosis and increases the economic injury level through tolerance (Teetes et. al. 1975). Resistant sorghums are complemented by greenbug mortality caused by natural enemies.

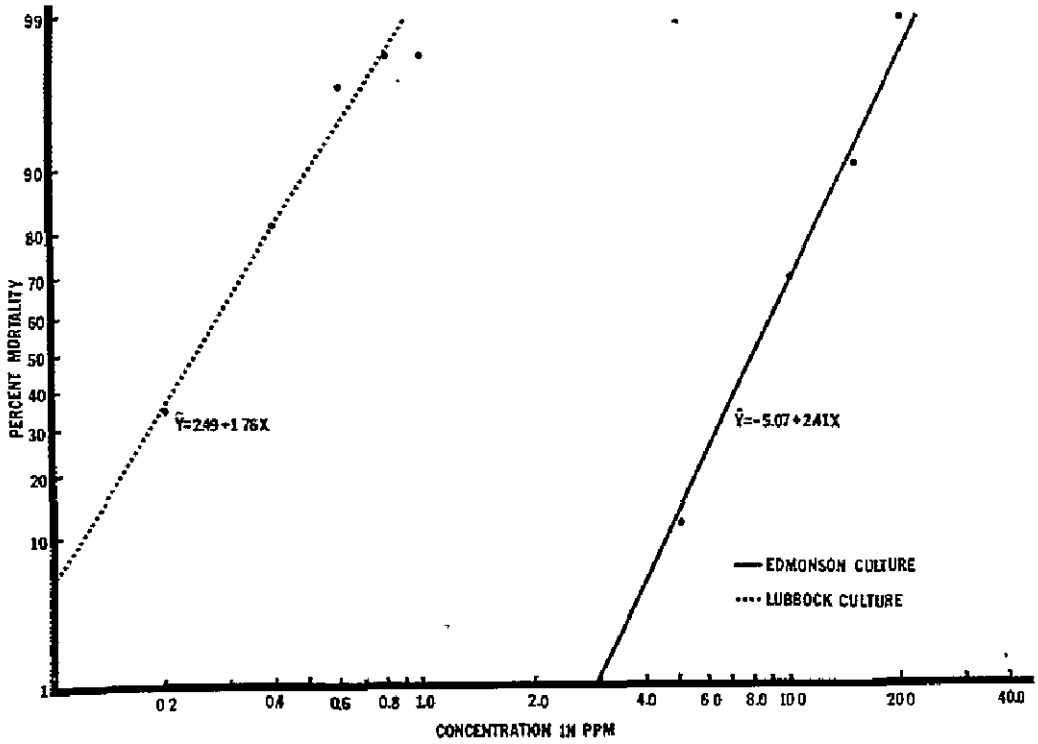


FIG. 4. Dosage-mortality after 48 h of 2 populations of the greenbug treated with disulfoton.

Research revealed that it took greater numbers of greenbugs feeding for a longer period of time on resistant sorghums than susceptible sorghums to cause an equal amount of yield loss (Table 7). However, the same amount of damage, that is, the same amount of leaf loss to resistant sorghums, resulted in similar amounts of yield loss when compared to susceptible sorghums. Under natural conditions, resistant sorghums were not severely damaged and only under artificially produced conditions such as caging to exclude predators and parasites was this accomplished.

Biologically, the resistant sorghums had been shown not to be ecologically disruptive. To determine this, a test was conducted in 1975 where resistant and susceptible sorghums were separately isolated in a cotton field where the relative number of predators were monitored (Teetes 1975). Results showed that, as in past experiments, greenbug numbers were lower in resistant sorghums than in susceptible sorghums (Fig. 5). Similarly, as would be expected, predator numbers were lower in resistant than susceptible sorghum (Fig. 6). Interestingly,

TABLE 7. Greenbug densities and damage to resistant and susceptible sorghum hybrids, Lubbock, Texas, 1975.

Hybrid	Greenbug/plant prior to treatment	Leaves/plants killed	Yield/ acre	Seeds/gm
Susceptible	640a	0.8a	5667a	27a
(A Tx 399 x TX 2536)	1510a	3.7ab	4700a	33a
	2938b	4.3b	5067a	34a
	4767c	4.7b	3317b	36a
Resistant	19a	0.0a	6167a	26ab
(A Tx 399 x TAM 2568)	88a	0.0a	5850a	26ab
	300b	0.3a	6033a	24b
	700b	0.6	5900a	27a
Resistant	5a	0.0a	5866a	26a
(A Tx 399 x TAM 2568)	543a	0.6ab	5250a	29a
	937b	1.0ab	5350a	30ab
	2487c	3.3b	4766n	33b

however, when the ratio of predators to greenbugs was compared, the ratio was about equal and sometimes greater in the resistant sorghum than in the susceptible (Fig. 7). Based on predator-prey ratios, it appears that resistant sorghum and biological control are complementary. Tests utilizing cages to exclude predators and parasites provided a dramatic example of the value of natural enemies in assisting greenbug resistant plants.

Biological and ecological studies have been conducted on both native and exotic species of the major predators and parasites of the greenbug (Archer et. al. 1973, 1974, Daniels and Chedester 1972) to improve the contribution to biological control to the pest management program. Indigenous primary parasites include Lysiphlebus testaceipes (Cresson), Aphelinus nigritus (Howard) and Diaretiella rapae (M'Intosh) and secondary parasites Charips sp., Pachyneuron siphonophorae (Ashmead), Asaphes lucens (Provancher), Aphidencyrtus aphidovorus (Mayr), and Tetrastichus minutus (Howard).

Because indigenous beneficial insects have not always held the greenbug below damaging levels, the introduction of exotic parasites such as Aphelinus asychis (Walker), A. varipes (Forester), Ephedrus plagiator (Nees), Praon gallicum (Story) and Aphidius avenae (Haliday) and predators Propylea 14-punctata (L.) and Menochilus sexmaculata (Fab.) has been attempted (Starks et. al. 1975). Basic studies have led to biological and ecological knowledge and provided methods of mass production (Archer et. al. 1973).

In Texas, sorghum and cotton are the two major crops and are ecologically related. Intensive and widespread use of insecticides in sorghum could upset the balance of predators and parasites creating widespread and damaging pest infestation in cotton. Also, the advantages of habitat diversification by planting adjacent alternate crops for increasing predator densities are well known. Sorghum served as the major source of natural enemies which later are active in cotton. By experimentation it was shown that there was a seasonal relationship in abundance of greenbug and corn leaf aphid, Rhopalosiphum

FIG. 5. Seasonal greenbug populations on resistant and susceptible sorghum hybrids.

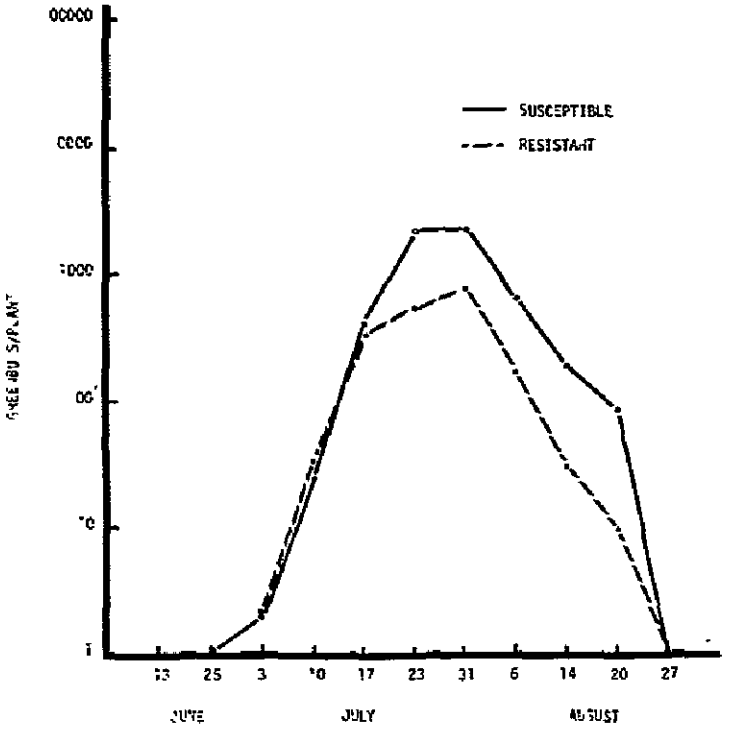


FIG. 6. Seasonal predator populations on greenbug resistant and susceptible sorghum hybrids.

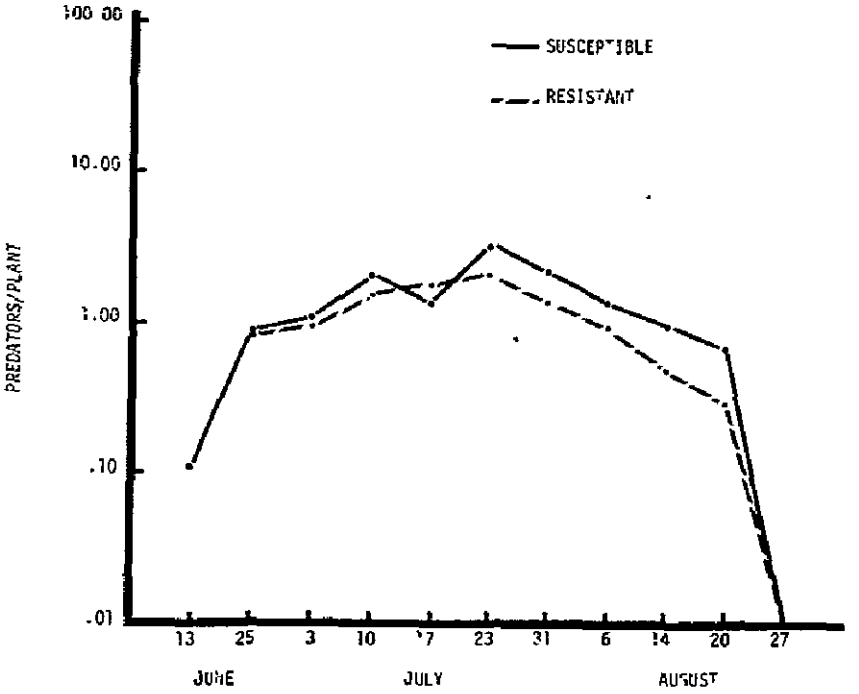


FIG. 7. Seasonal predator-to-greenbug rations on greenbug resistant and susceptible sorghum hybrids.

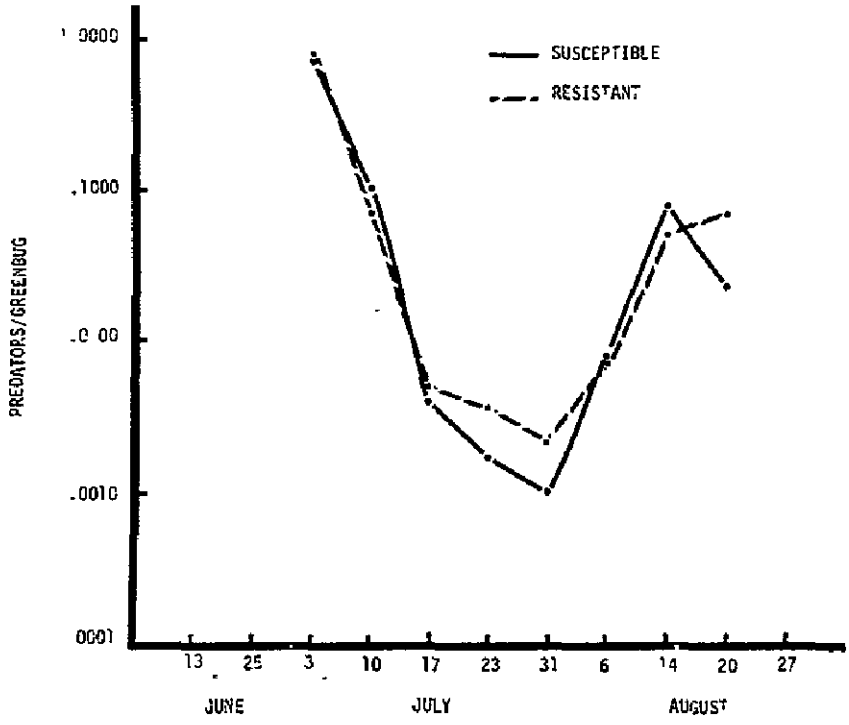
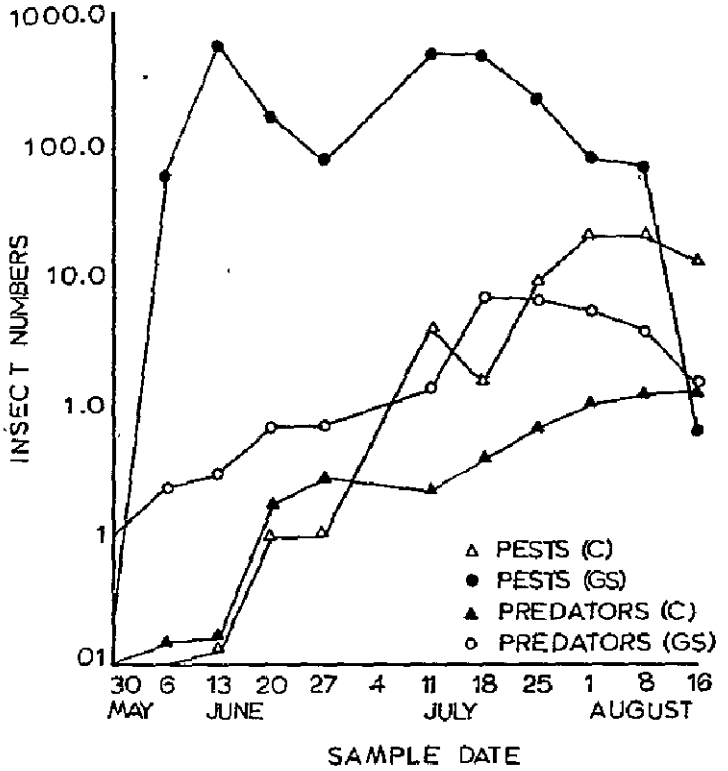


FIG. 8. Predator and pest abundance in grain sorghum and cotton, 1972.



maidis (Fitch), and several selected predators on field planted sorghum, wheat, and volunteer Johnson grass. As aphid numbers increased on any host, predators also increased. As parasitism decreased aphid population levels in grain sorghum, predator density decreased in that crop. Predator population levels in cotton began to increase at about the same time that predator density began to decrease in sorghum (Fig. 8) (Lopez and Teetes 1976). Based on the relative similarity of the 2 habitats in terms of insect species composition, the 2 different plant communities became more intimately related as the growing season progressed (Fig. 9). Of ca. 500,000 predators captured, marked and released in grain sorghum, 51 or 0.01% were recaptured in nearby cotton.

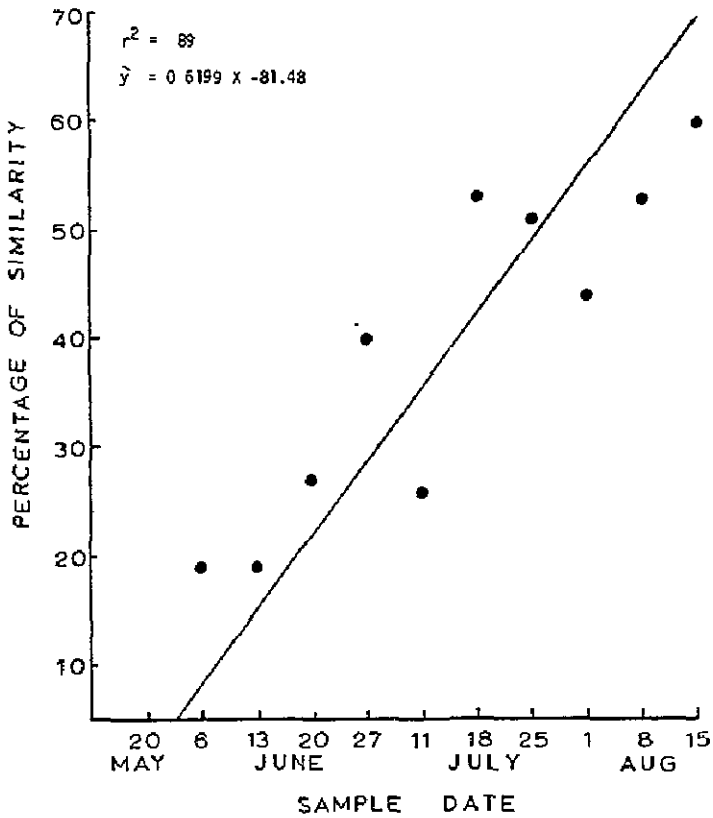


FIG. 9. Relative similarity of grain sorghum and cotton in terms of species composition, 1972.

Other aphids that infest grain sorghum include the corn leaf aphid, Rhopalosiphum maidis (Fitch), and the yellow sugarcane aphid Sipha flava Forbes. The corn leaf aphid is greenish-blue in color and generally feeds within the plant whorl. Larger plants can tolerate large numbers of this insect without suffering serious damage (Harding 1965). However, large populations may damage seedling plants, causing plant death and stand loss. Large populations infesting plants during the booting stage may cause poor head exertion. Heavy plant head infestations prior to harvest have resulted in harvesting difficulties because of the stickly honeydew deposited by the insects. The corn leaf aphid is also a vector of MDMV.

Since 1968, the presence of the corn leaf aphid in sorghum has attracted increased attention as a result of confusion with the greenbug. Research has shown that the species is generally not a serious pest of sorghum, but since some producers apply insecticides to control it, it does become an important consideration in sorghum pest management.

Some commonly used sorghum parental lines are more susceptible to corn leaf aphid damage than others. For example, B "Redlan", a common parent of sorghum hybrids, is extremely susceptible. However, several sorghum lines, especially some Zerazera types are highly resistant to damage by corn leaf aphid.

The yellow sugarcane aphid is lemon yellow, covered with setae and has two double rows of dark tuberacles down the dorsum. Feeding aphids secrete a plant toxin and relatively light populations have been known to kill sorghum in the pre-boot stage. This aphid has been observed damaging sorghum in the Gulf Coast counties and Blackland areas of Texas, although it also occurs in the Texas Panhandle.

Symptoms of damage in seedling sorghum are expressed as purpling of the leaves, while in more mature plants, the leaves generally turn yellow. Preliminary data indicate that severe stunting of sorghum results from aphid feeding and toxin injection. No resistant sorghum lines have been identified although control tactic and economic threshold levels are being established.

Spider Mites. Several species of spider mites, Oligonychus spp., and Tetranychus spp., infest sorghum in Texas. The Banks grass mite (BGM) O. pratensis (Banks), is the most frequently encounter species (Ehler 1973, Owens et. al. 1976).

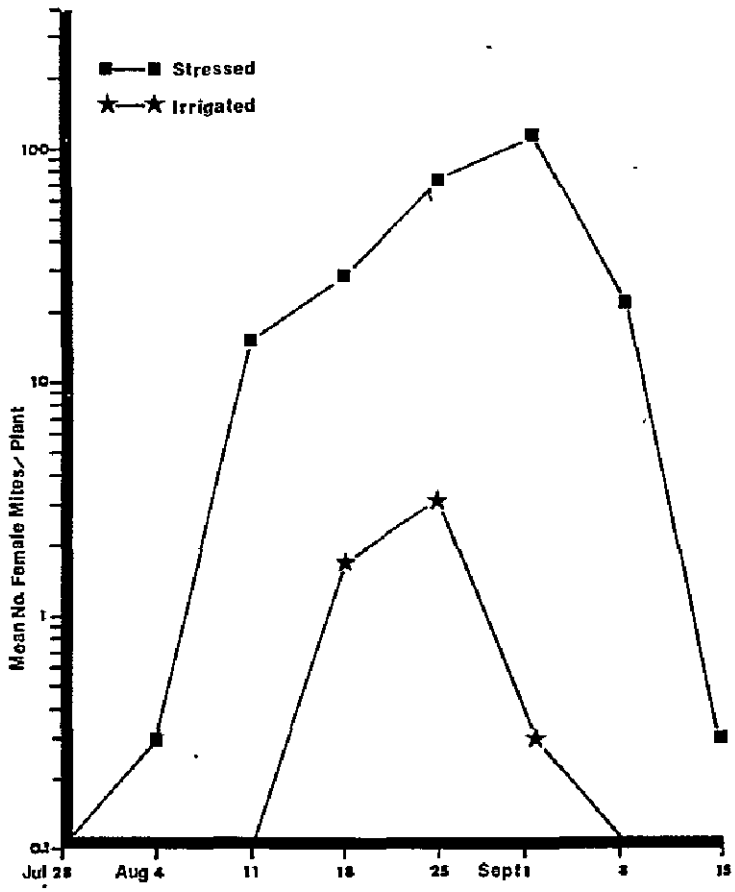
Adult BGM exhibit marked sexual dimorphism. After feeding, both sexes become a deep green, with the exception of the palpi and first two pairs of legs which remain light salmon. The female, which is much large than the male reaches an overall body length of about 0.40-0.45 mm. The life cycle requires 11 days at 78-80°F and as many as 61 days under less favorable conditions.

Spider mites are usually along the midrib on the underside of the lower functional leaves. The infested areas of the leaves are pale yellow initially and later take on a reddish color on the top side of the leaves. If mite numbers continue to increase, the entire leaf may turn brown. As mite numbers increase on the lower leaves, the infestation spreads upward through the plant. The underside of heavily infested leaves will be densely webbed by the mites. In the final stages of infestation, the mites may invade and web sorghum heads. Plant lodging may occur.

Based on distribution records, the BGM is native to North America, although this conclusion remains tentative until the tetranychid fauna of South American, Africa and Asia are more thoroughly described (Ehler 1974). The pest is generally restricted to monocotyledenous plants, particularly grasses. Over 80 species of grasses in 17 genera have been recorded as hosts (Ehler 197

Spider mite outbreaks are closely correlated with reproductive maturity of the host plant (Ehler 1974, Kattes and Teetes 1978) with rapid increases generally beginning after heading (Cate and Bottrell 1971). There is a positive correlation between mite density and hot, dry climatic conditions and moisture stressed plants (Fig. 10) (Kattes and Teetes 1978). Mite infestation appear to be separated temporally and spatially from populations of effective natural enemies (Ehler 1974).

FIG. 10. Mean adult female spider mite population levels per week on water stressed and non-stressed grain sorghum.



Natural enemies of spider mites include several species of general predators as well as some which are prey-specific for phytophageous mites, i.e., Scolothrips sexmaculatus (Pergande), Stethorus punctum LeConte, S. atomus Casey, Amblyseius fallacis (Garmen), A. mesembrinus Dean, Pronematus ubiquitis (McGregor), and cecidomyiids (Dean 1957, Ehler 1973). However, this predator complex has not provided adequate control of the tetranychid mite pests in grain sorghum.

Presently, chemical control is the only available method of suppressing outbreaks of mites in some regions but grower experience and research data indicate varying degrees of success (Cate and Bottrell 1971, Pate and Neeb 1971, Teetes 1973, Ward et. al. 1972). Insecticide resistance was accounted for control failures in some areas (Owens et. al. 1976, Ward et. al. 1972, Ward and Tan 1977).

In some areas of Texas the BGM acts as a secondary pest where insecticidal treatments, especially parathion, applied for aphid control increased the severity of the mite (Fig. 11) (Kattes and Teetes 1978). Parathion treatments result in the dispersel of mite populations, consequently releasing the reproductive inhibitory effect of crowded mited colonites (Fig. 12) (Kattes and Teetes 1978).

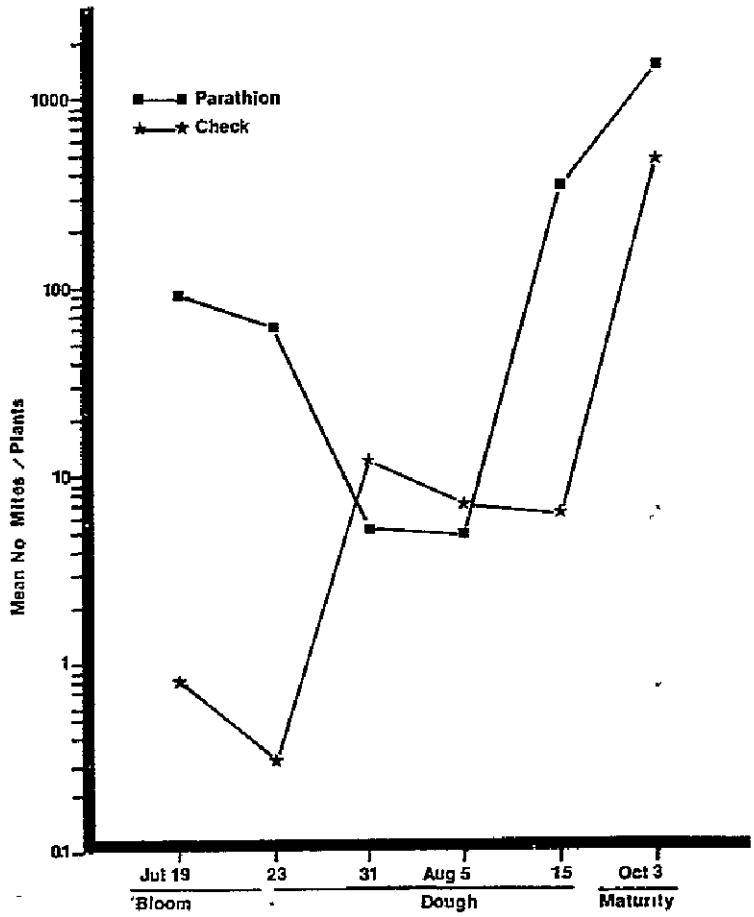


FIG. 11. Seasonal abundance of spider mites infesting parathion treated and non-treated sorghum.

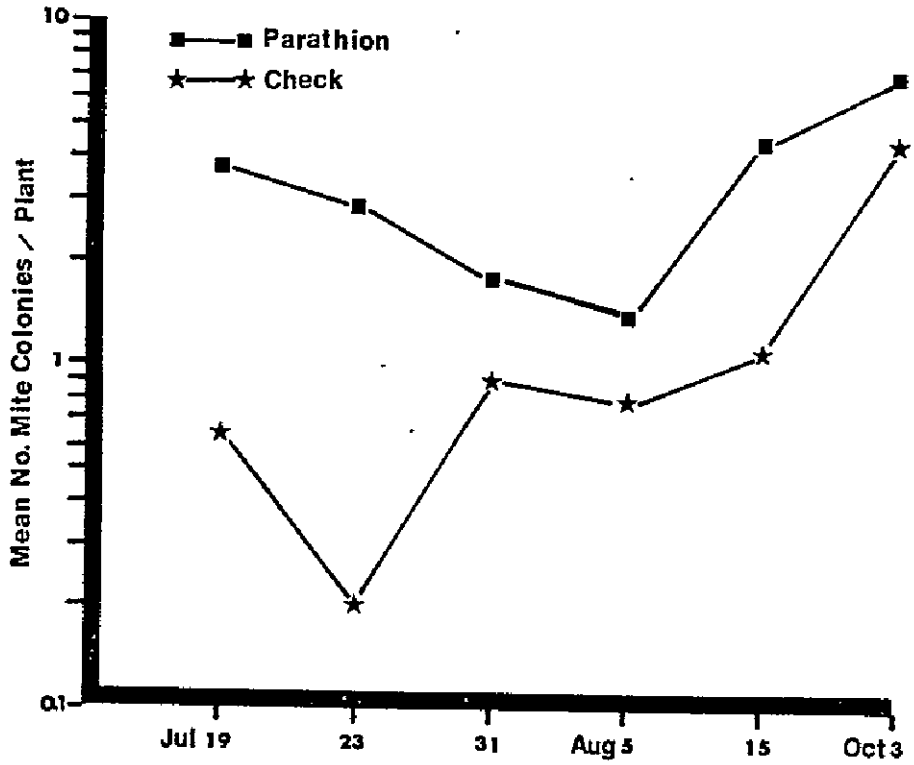


FIG. 12. Mean number of spider mite colonies per plant on parathion treated and non-treated grain sorghum.

Sorghum germ plasm resistant to mites has been identified from the Texas sorghum breeding program (Foster et. al. 1977b). SC 599-6, a partially converted "Rio" selection from the sorghum conversion program appeared especially promising during several years of testing at Pecos, Texas (Table 8). This line is a "non-senescing" type which maintains green leaves and healthier stalk much longer than most lines. Interestingly, it continues to maintain green leaves even when infested with mites, consequently, the resistance mechanism appears to be of the tolerance type. The line is higher in total sugars than standard grain sorghums and this may be involved in the resistance mechanism (Foster et. al. 1977a).

TABLE 8. Mite damage rating of selected sorghum lines, 1975.

Entry	Days to 50% bloom	Sugar content (%)	Mean # mites	Damage leaf	Rating[1] stalk	%[2] loss in seed weight
SC 599	78	11	1213	4.7	1.0	29
BTX 618	71	6	2671	6.0	4.8	24
BTX 378	85	6	2706	7.5	7.3	50
TX 2536	78	7	1980	7.8	8.5	51
ATX 618 x SC 599	71	8	1934	4.7	2.9	30
ATX 378 x SC 599	78	7	1974	7.1	7.6	56
HT 124	77	6	2015	7.7	8.5	53

[1] 1 - no leaf or stalk death, 2 - 1-10% of leaf area dead, 3 - 11-20%, 4 - 21-30%, 5 - 31-40%, 6 - 41-60%, 7 - 61-80%, 8 - 81-90%, 9 - 91-100%.

[2] Difference in seed weight of each line grown at Pecos with high mite populations and the same line grown at Lubbock with no mites present.

Wireworms. The economic importance of false wireworms (Tenbrionidae) and true wireworms (Elateridae) as pests of sorghum planting seed is not well known. The seriousness of these seed pests has been masked because of the extreme effectiveness of certain chlorinated hydrocarbon insecticides, such as dieldrin, which have been applied to seed before planting usually by commercial seed companies. Wireworms have been reported as sporadic pests of sorghum in the Texas High Plains and control has been possible using several organochlorine insecticides applied as seed or soil treatments or by rotation (Daniels 1969). Soil treatments of thiofanox, Dasanit[R], and carbofuran were more effective in controlling wireworms in grain sorghum than disulfoton, aldicarb, phorate, or a one-to-one combination of disulfoton-Dasanit[R] (Daniels and Chedester 1975).

The current action by the Environmental Protection Agency toward banning certain chlorinated hydrocarbon insecticides for use in soil pest control has created the need to find replacement insecticides in the phosphate or carbamated groups. Seed treatments versus in-furrow or band applications would reduce the amount of toxicant and would not require extra farming operations. Also, there is a need for a method to assess the severity of wireworms in the absence of treated seed and a monitoring tool for determining the need for control measures.

Recent research dealing with these issues has provided sufficient results. Both methods for monitoring wireworm populations prior to planting and alternate seed treatment chemicals have been studied.

Populations of the true wireworm, Aeolus mellillus (Say), were determined with baited traps of sorghum seed. Trap catches were highest 1 or 2 weeks after trap establishment. The number of wireworms found in traps was twice the number present in one linear row foot (Table 9). A population level of one wireworm per linear foot of row usually damaged approximately 10 percent of the planted seed, which caused a reduction in plant stand but not in grain yield (Teetes 1976).

TABLE 9. Comparison of numbers of wireworms caught in baited traps and wireworms per linear row ft. and subsequent plant populations and yield, Lubbock. 1975.

Treatment	Wireworms/baited trap after indicated week 1	2	3	Wireworms/ linear ft.	Percent damaged seed	Plant pop. /acre and (%) decrease[1]	Yield in lbs/acre and (%) decrease[1]
Dieldrin						47,103 a	3666 a
Untreated	2.1	2.3	1.0	1.0	10	39,087(17)b	3266(11)a

[1]Means followed by the same letter are not significantly different at the 5% level (Duncan's multiple range test).

Wireworms were effectively attracted to bait traps placed 12.7 cm (5 inches) in depth and which contained 120 gm of bait (Table 10) (Foster and Ward 1976). Traps baited with a 1:1 mixture of wheat and corn were equally as attractive as traps baited with sorghum. Wireworms did not appear in traps until certain soil moisture and temperature requirements were satisfied. Cultural practices of the fields studied did not eliminate wireworm populations but populations were generally below the economic threshold (Daniels and Chedester 1976, Foster and Ward 1976).

TABLE 10. Number of wireworms found in traps with different depth placements and different size sorghum bait samples, field 105, Lubbock, Texas, February 23 and April 27, 1975.[1/]

	Avg. number of wireworms/trap						
	Depth placement (cm)			Size of bait sample (gm)			
Date of trap placement	12.7	20.3	27.9	60	120	240	120+500 ml H_2O
February 23, 1976 (Test 1)	0	0	0	0	0	0	0
April 27, 1976 (Test 2)	2.19	1.63	1.25	1.67	2.17	1.83	1.58

Several insecticide seed treatments reduced wireworm numbers and damaged seed, and some resulted in increased plant stands but not yields. Lindane appeared to affect seed germination. Carbofuran and Dasanit[R] (0, 0-diethyl 0-P-[(methylsulfinyl-phynyl) phosphorothioate] appear to hold promise as replacement seed treatments for the standard chlorinated hydrocarbon insecticides (Table 11).

TABLE 11. Effect of insecticide seed treatments applied for control of wireworms in grain sorghum, Lubbock, 1975, Test 1.

Insecticide	Formulation	Oz. AI rate/100 lbs seed	Wireworms/ linear ft[1]	Percent damaged seed	Plant pop. /acre and (%) increase	Yield/acre and (%) increase[2]
Heptachlor	3#EC	1.5	0.5	0	48533 (21) bc	3700 (14) a
Chlordane	40% D	1.5	0.3	3	44633 (15) cd	3333 (4) a
Chlordane	8#EC	1.5	0.9	7	44200 (14) c	3600 (11) a
Carbofuran	75% WP	2.0	0.5	0	50267 (24) b	3633 (12) a
Carbofuran	75% WP	4.0	1.0	0	48967 (22) bc	4366 (27) a
Dasanit	6#EC	2.0	0.3	0	51133 (25) ab	3300 (5) a
Diazinon+Lindane	11+16.6% WP	1.5 + 2.3	1.3	0	37267 (0) e	3200 (0) a
Diazinon	4#EC	2.0	0.4	0	47233 (19) bcd	2800 (0) a
Dieldrin	50% WP	1.0	1.0	0	55033 (31) a	3900 (18) a
Check			1.4	16	38133 (-) e	3200 (-) a

[1] Wireworms/baited trap 2.1.

[2] Means followed by the same letter are not significantly different at the 5% level (Duncan's multiple range test).

White Grubs. Species of the genus Phyllophaga are injurious to a wide range of agricultural crops in Texas. Damage by the white grub, Phyllophaga crinita (Burmeister), to sorghum has increased in severity in some areas. Approximately 40,000 acres are damaged annually on the Texas High Plains.

Adults are brown to brownish-black, 13 to 19 mm long and are commonly referred to as May or June beetles. Larvae are C-shaped with brown heads and white bodies. Digested food can be seen through the shiny and transparent tip of the abdomen.

Damage to sorghum may occur in several different ways. The most obvious damage and perhaps the most significant, is death of seedling plants from larvae feeding on the roots. Seed germination may occur and a satisfactory stand established, but within a short period when plants are four to six inches tall, seedlings begin to die. Stand loss can occur within one week to ten days in severely infested areas. One grub is able to destroy all plants within one-two feet of row. Plants not killed as seedlings are severely stunted and in many cases never produce seed. A third type of damage results from root pruning by overwintered as well as current season larvae. Injured plants able to produce seedheads after such damage, frequently do not have sufficient roots to prevent lodging. Occasionally, lodging is increased by secondary stalk rot organisms.

Seasonal field data indicate delayed planting as a possible means to escape seedling damage by overwintering larvae (Fig. 13)(Teetes and Wade 1974). However, the crop remains susceptible to attack later in the season. These studies also have revealed the fact that some individuals have a life cycle longer than one year in contrast to the one year life cycle reported earlier. Light trap data have shown only one major peak in adult abundance and activity.

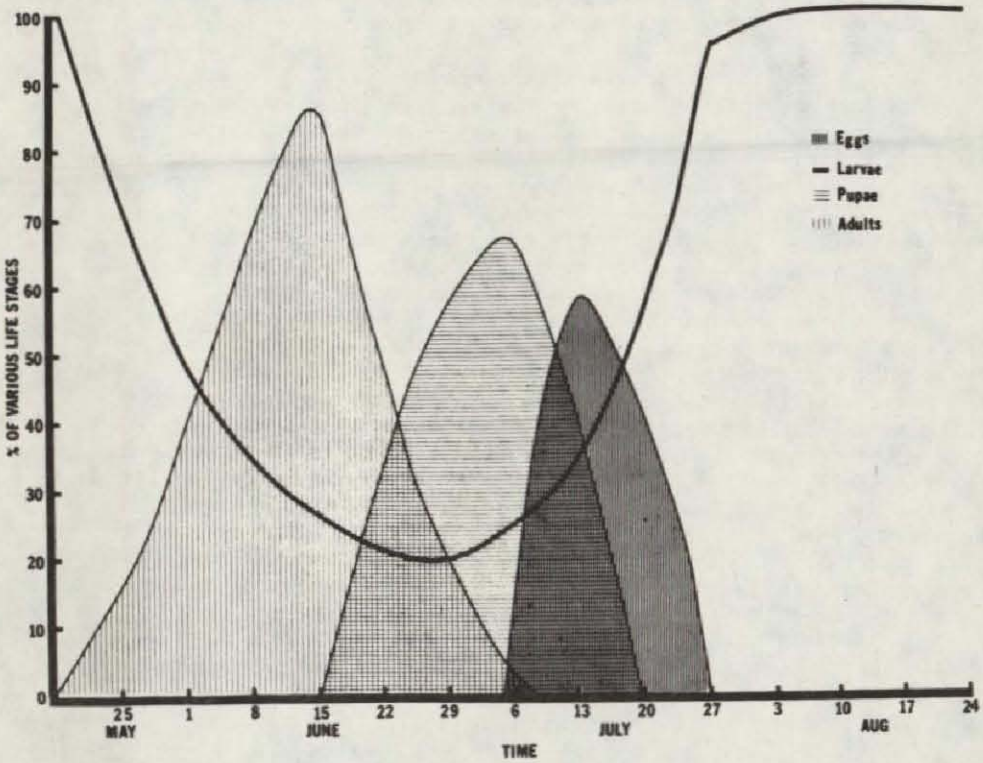
Damage assessment studies show that the economic injury level is two grubs per square foot and the economic injury has been set at one grub per square foot (Teetes 1973). Because of the nature of the grub infestations, a preplant broadcast, incorporated application of insecticide is required for control when the economic threshold is exceeded. Effective insecticides include diazinon, carbofuran and Dasanit^{R} (Teetes 1977).

Rootworms. Rootworms, especially the southern corn rootworm, Diabrotica undecimpunctata howardi, may prune the roots of sorghum. The damage usually results in plant stunting, lodging, or a "dead heart" condition. Little research has been conducted on this species attacking sorghum. In-furrow, at planting applications of insecticides have provided effective control.

Fall Armyworm. The fall armyworm, Spodoptera frugiperda (J. E. Smith), is one of several lepidopterous insects known as armyworms which attack sorghum in Texas. They often cause extensive leaf ragging to sorghum and commonly feed within the plant whorl. The leaves unfolding from the whorls are perforated with holes caused by the feeding of the insect. The corn earworm, Heliothis zea (Boddie), may cause similar damage. Damage rarely justifies control of these insects except with heavy infestations on small plants and generally those planted late in the season.

False Chinch Bug. Infestations of a lygaeid bug, Nysius raphanus (Howard), in sorghum in Texas are sporadic, but they cause considerable concern to producers because of the occurrence of extremely high population levels. Populations of the false chinch bug are normally concentrated in small areas of a field, and general infestations over an entire sorghum field are rare. They infest sorghum by immigration of mature and immature forms. Reproduction has not been observed on sorghum.

FIG. 13. Seasonal occurrence of the various life stages of white grubs.

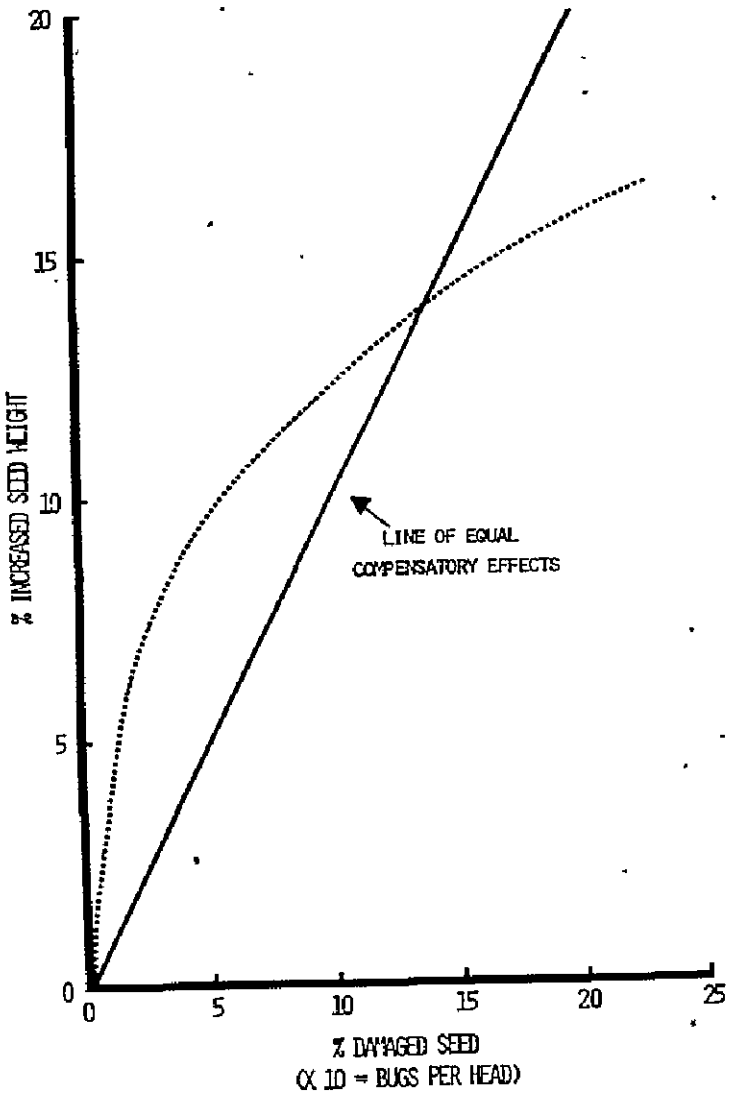


Damage results from bugs sucking juices from the immature developing seed. Commonly, the damaged seed are infected with a fungus (Alternaria sp.) which causes the seed to be black in color resulting in further deterioration of quality. Damaged seed rarely develop fully and are considerably smaller, softer, and lighter in weight than undamaged seed and are subject to loss in harvesting. False chinch bugs also feed in clusters on the leaves of sorghum; however, no apparent leaf damage resulted from the population levels experience in 1973 when most bugs were concentrated on sorghum heads.

Destructive infestations of the false chinch bug on grain sorghum and other crops have been reported from several parts of Texas (Daniels 1958, 1969). The pest is most injurious while the grain is in the dough stage and damaging infestations usually are concentrated in spots 5 to 200 feet in diameter within a field. From small plot and aerial field tests, malathion and parathion provided high levels of control. Toxaphene, phosdrin and dieldrin were less effective.

Damage to sorghum by the false chinch bug was assessed by confining different populations in cages on sorghum heads (Teetes et. al. 1974). Reductions in grain yield were evident only when 200 bugs per head damaged 23 percent of the seed. Increase in weight of undamaged seed apparently compensated for yield loss to a point because in most of the treatments percentage of damaged seed was less than the percentage increase in seed weight. Only when percentage of damaged seed was greater than the percentage increase in seed weight was there evidence of reduced yields. The point of equal compensatory effect occurred at 14 percent damaged seed or approximately 140 bugs per head (Fig. 14).

FIG. 14. Compensatory effects of sorghum seed at various levels of false chinch damage, 1973.



A number of other seed-sucking bugs attack sorghum, but little research has been done on their managment. These pests include several stink bug species, and the leaf footed plant bug, Leptoglossus phyllopus (L.).

Corn Earworm. The corn earworm, Heliothis zea (Boddie), is a general feeder and injures sorghum in two ways. It often causes extensive leaf ragging in sorghum and commonly feeds within the plant whorl. The leaves unfolding from the whorl are perforated by the feeding of the insect. The pest also feeds in the panicle (head) of sorghum where early instar larvae hollow out the kernels and later instars completely destroy the maturing grain. Small larvae appear to prefer less mature kernels for feeding while older larvae prefer more mature kernels.

Numerous insecticides have been evaluated for effectiveness in controlling corn earworm in sorghum. Effectiveness of the tested chemicals vary considerably, but in general carbaryl, mevinphos, methomyl, parathion, and toxaphene have been effective. Most chlorinated hydrocarbon insecticides present residue problems.

Nuclear-polyhedrosis viruses have been tested and are generally more effective in controlly corn earworm in sorghum than cotton. The viruses appear more persistent in sorghum than cotton.

Sorghums have been screened for panicle resistance to Heliothis with little success. It is known, however, that sorghum types with loose panicles are seldom damaged as severely as those with compact panicles. Also, open-panicle sorghum increase the effectiveness of chemical control.

The corn earworm presents a severe problem in sorghum breeding, in that, the use of paper bags placed over sorghum panicles to insure self-fertilization creates an ideal environment for the pest. As a result the insect damage is often so severe that the grain is entirely destroyed. It has been speculated that this situation results from protection of the larvae from their natural enemies. Insecticide treated pollination bags provide a means of controlling Heliothis in such instances.

Several reports have been published which relate population density of H. zea to damage in sorghum. Infestations of one to 16 larvae of varying size per panicle have been reported to result in 10-60% damaged grain (Burkhardt 1957a,b, Burkhardt and Breithaupt 1955, De Pew 1957). Based on regression analysis of data collected following artificial infestation, an equation was developed for predicting the number of kernels that a known number of larvae would destroy (Table 12) (Buckley and Burkhardt 1962). Estimates of sorghum grain losses due to earworms can be made using the predictive equation, $Y = 71 + 102X$, where X = number of larvae per panicle. The percentage of infested panicles must be determined, and that value multiplied by the answer to the equation.

Also by regression analysis, it has been shown that as the level of infestation increased one larvae per panicle, the yield decreased 3.9 g per panicle (Kinzer and Henderson 1968). The mean yield per panicle at population levels of 0, 4, 8, 12, and 16 larvae per panicle was 95.1, 79.5, 63.9, 48.1, and 32.8 g respectively, for one test and 121.3, 105.6, 90.0, 74.3, and 58.7 g for a second test. These data compare favorably with that of later research where for each additional H. armigera larvae, a decrease in yield of 5.4 g occurred (Wilson 1976). Consequently, it has been suggested that for 40 in. row spacing, populations of 1, 1.5, 2, 2.5 and 3.5 larvae per panicle would constitute economic thresholds with 4, 6, 8, 12, and 16 in. panicle spacing, respectively.

TABLE 12. Corn earworm damage to sorghum.

No. of larvae	No. of damaged kernels	% damaged kernels
1	166	6
2	274	10
3	370	13
4	497	16
5	632	20
6	690	23
7	785	27
8	915	30
9	1,010	33
10	1,068	37
11	1,219	40
12	1,406	44
13	1,549	48

There are several factors which must be considered in assessing Heliothis damage and the need for control: these include larvae competition for food, the numbers of predators and parasites, and larval size. Over 75 percent of the total feeding in sorghum panicles is done by the last two larval instars (Kinzer and Henderson 1968). Large larvae (passed third instar) are more difficult to control with insecticide than small larvae (Henderson et. al. 1965). Also there is a relationship between maturity of grain and maturity of larvae. Larvae are generally smaller (less than 1/2 grown) in immature panicles than mature panicles. The density of earworm larvae in sorghum panicles decisively affects their survival and, consequently, their feeding damage (Buckley and Burkhardt 1963). Apparently, both cannibalism, including combat mortality, and disease contributes to total mortality, as up to the third instar, 65 to 88 percent of crowded larvae survive (Barber 1936). Beyond that stage, larval mortality increased greatly because of cannibalism. Research has shown, that by increasing the infestation level by five larvae, from one to six, was sufficient to increase mortality (Buckley and Burkhardt 1963).

Where levels were increased by six, mortality was significantly affected in all cases. Larval mortality with 20 and 30 larvae per panicle was 71.6 and 83.2 percent, respectively.

Based on the research data available, and considering the factors discussed above, the economic threshold of Heliothis in sorghum would appear to be 1-2 larvae per panicle. The number of bushels of grain that must be saved to equal the cost of control is determined by dividing treatment cost by the value of a bushel of grain. If a market price of $2.20/bu and a treatment cost of $5.50/acre are assumed, a saving of at least 2.5 bu of grain/acre would be necessary to offset the cost of treatment. If a grower produced 50 bu/acre at $2.20/bu, the per acre value would be $110.00/acre. A 6% loss (1 larvae/panicle) and a 10% loss (2 larvae/panicle) would result in a monetary loss of $6.60 and $11.00/acre, respectively, provided that all larval damage could be prevented by one treatment. The losses express the maximum potential damage attributable to a given level of infestation if it is assumed that the larval stage is completed by all individuals and that no environmental factors adversely affect the larvae.

Direct control tactics applicable to managing Heliothis in sorghum include, growing open-panicle varieties, manipulating planting time, and the use of chemical controls based on the economic threshold level. Another, perhaps broader corridor of redress to Heliothis in sorghum would seem to lie in exploitation of the polyphagous feeding habit of these insects. Manipulation of the species by cultural, biological or chemical means in one crop host in the agroecosystem could mitigate infestations in other crops. Studies on the relative similarity of sorghum and cotton in terms of species composition, have shown a closely woven relationship, one that through the season, progressively acquires greater intimacy. An uncharted opportunity for pest management seems to lie here.

The fall armyworm may also feed in the panicles of sorghum. Their nature of injury is similar to that of the corn earworm.

Sorghum Webworm. The sorghum webworm, Celama sorghiella (Riley), occurs primarily in the more humid areas of Texas. It frequently occurs in large numbers in sorghum head where it eats circular holes in the seed and feeds on the starchy contents. Observation indicate that each larva may consume as many as 12 seeds in 24 hours, resulting in severe crop losses (Randolph and Garner 1961). Moths deposit from 100 to 300 eggs singley on the flowering parts or seeds of the host plant. There may be up to six generations annually. For making decisions for chemical control, heads should be inspected beginning in the bloom stage and continued until the hard dough stage has been reached. Chemical control is justified when heads are infested with an average of five larvae per head (Doering and Randolph 1960, Randolph et. al. 1960). Planting sorghum early often provides a means of escaping the pest. Also, "open-headed" sorghums are generally less infested than more compact sorghum panicles.

Stalk Borers. Stalk borers, namely the southwestern corn borer Diatraea grandiosella (Dyar), sugarcane borer, D. saccharalis (F.), and sugarcane rootstock weevil, Anacentrinus deplanatus (Say), may cause damage from extensive tunneling and girdling of the stalk which often results in stalk lodging. Only light infestations have in general been reported in sorghum. Although efforts at chemical control of borers in sorghum have generally given sporatic results, insecticides may be justified in late planted or ratoon sorghum. Soil applications are required to control sugarcane rootstock weevil.

Various cultural control practices appear to afford the most effective means of controlling these pests. Area-wide stalk destruction through practices such as double discing and deep breaking, destroy the plant crown which affords overwintering larval destruction. Research and observational data indicate that early plantings may escape infestation and are less susceptible to plant lodging. A reasonable plant population to insure large healthy stalks along with proper fertilization and adequate irrigation help prevent lodging of borer-infested stalks. Crop rotation, use of early maturing varieties, and early harvest with equipment designed to pick up lodged stalks aid in reducing yield losses.

CONCLUSION

Sorghum and its associated pests are in a dynamic state. The progress that has been made with regard to sorghum entomology will undoubtly continue at an accelerated rate, as it must, to meet the ever increasing pest problems. The area of greatest need lies in the establishment and refinement of economic threshold levels in order to eliminate needless insecticide treatments and to gain maximum benefits from the other components of pest management. This will require interdisciplinary, problem-oriented, integrated control approaches based on sound ecological principles.

Insecticides, at least in the foreseeable future, will remain a major management tool as they are generally effective, economical and can be administered quickly to curb pest populations in emergency situations. They must be used judiciously on the basis of the potential positive values weight against possible negative values occurring from hazards to non-target organisms.

Management components such as cultural practices, biological control, and resistant plant varieties must be given greater consideration in monoculture production of sorghum.

The current literature on sorghum entomology reflects the increasing attention being given to the crop as the world faces the human population - food supply problem. There is a tremendous potential in sorghum for increased production of food, feed and fodder in the tropics that can be achieved with intensified research input on all aspects of its production, including better management of pests.

LITERATURE CITED

Almand, L. K., D. G. Bottrell, J. R. Cate, Jr., N. E. Daniels and J. G. Thomas. 1969. Greenbugs and other aphids on sorghum and small grains. Tex. Agr. Ext. Ser. L-819:4pp.

Archer, T. L., C. L. Murray, R. D. Eikenbary, K. J. Starks and R. D. Morrison. 1973. Cold storage of Lypiphlebus testaceipes mummies. Environ. Entomol. 2:1104-8.

Archer, T. L., R. H. Cate, R. D. Eikenbary, and K. J. Starks. 1974. Parasitoids collected from greenbugs and corn leaf aphids in Oklahoma in 1972. Ann. Entomol. Soc. Amer. 67:11-4.

Barber, G. W. 1936. The cannibalistic habits of the corn earworm. U.S. Dept. Agri. B-499. p. 3-18.

Bottrell, D. G. 1971. Entomological advances in sorghum production. Tex. Agr. Exp. Stn. Consol. PR-2940. 28-40.

Buckley, B. R. and C. C. Burkhardt. 1962. Corn earworm damage and loss in grain sorghum. J. Econ. Entomol. 55:435-9.

Buckley, B. R. and C. C. Burkhardt. 1963. Notes on the biology and behavior of Heliothis zea (Boddie) on grain sorghum. J. Kansas Entomol. Soc. 36:127-32.

Burkhardt, C. C. 1957a. Population evaluation, survey methods, and interpretation of results of corn earworm surveys. Proc. N. Cent. Br. Entomol. Soc. Amer. 12:58-60.

Burkhardt, C. C. 1957b. Corn earworm control in grain sorghum. J. Econ. Entomol. 50:529-41.

Burkhardt, C. C. and M. P. Breithaupt. 1955. Chemical control of the corn earworm in sorghum heads. J. Econ. Entomol. 48:255.

Cate, J. R., Jr. and D. G. Bottrell. 1971. Evaluation of foliar sprays for controlling the Banks grass mite on grain sorghum in the Texas High Plains, 1969. Tx. Agr. Exp. Stn. PR-2870. 6pp.

Cate, J. R., Jr., D. G. Bottrell and G. L. Teetes. 1973. Management of the greenbug on grain sorghum. I. Testing foliar treatments of insecticides against greenbug and corn leaf aphids. J. Econ. Entomol. 66:945-51.

Chada, H. L. 1962. Reaction of sorghum varieties and hybrids to natural infestation by southwestern corn borer, Mangum, Oklahoma, 1961. USDA, ARS, Special Rep. W-160. 8pp.

Dahms, R. G. 1943. Insect resistance in sorghums and cotton. J. Amer. Soc. Agro. 35:704-15.

Dahms, R. G. and J. B. Sieglinger. 1954. Reaction of sorghum varieties to the chinch bug. J. Econ. Entomol. 47:536-7.

Daniels, N. E. 1958. Chemical control of a species of the false chinch bug group in grain sorghum. Tx. Agr. Exp. Stn. PR-2063. 3p.

Daniels, N. E. 1969. Chemical control of a species of the false chinch bug group on wheat and sugar beetes. Tx. Agr. Exp. Stn. PR-2667. 2p.

Daniels, N. E. 1971. Detection of insecticidal residue and control of soil insects. J. Econ. Entomol. 64:175-7.

Daniels, N. E. 1977. Seasonal populations and migration of the greenbug in the Texas Panhandle. Southwestern Entomol. 2:20-6.

Daniels, N. E. and L. D. Chedester. 1972. Development studies of the convergent lady beetle. Tx. Agr. Exp. Stn. PR-3109. 3pp.

Daniels, N. E. and L. D. Chedester. 1975. Experimental aphid control in grain sorghum. Tx. Agr. Exp. Stn. PR-3335. 16pp.

Daniels, N. E. and L. D. Chedester. 1975. Wireworm control in grain sorghum. Tx. Agr. Exp. Stn. PR-3306. 4pp.

Daniels, N. E. and L. D. Chedester. 1976. Aphid and mite control in grain sorghum. Tx. Agr. Exp. Stn. PR-3430. 18pp.

Daniels, N. E. and L. D. Chedester. 1976. Grug and wireworm populations after manure and nitrogen application. Tx. Agr. Exp. Stn. MP-1308. 3pp.

Daniels, N. E. and R. W. Toler. 1971. Transmission of maize dwarf mosiac by the greenbug. Tx. Agr. Exp. Stn. Consol. PR-2869:20-2.

Dean, H. A. 1957. Predators of Oligonychus pratensis (Banks), Tetranychidae. Ann. Entomol. Soc. Amer. 50:164-5.

De Pew, L. J. 1957. Control of corn earworms in sorghum heads by aerial spraying in southwestern Kansas. J. Econ. Entomol. 50:224-5.

Dicke, F. F., R. E. Atkins and G. R. Pesho. 1963. Resistance of sorghum varieties and hybrids to the European corn borer (Ostrinia nubilalis Hbn.). Iowa State J. Sci. 37:247-57.

Dobie, P. 1974. The laboratory assessment of the inherent susceptibility of maize varieties to post-harvest infestation by Sitophilus zeamais Motsch. (Coleoptera, Curculionidae). J. Stored Products Res. 10:183-97.

Doering, G. W. and N. M. Randolph. 1960. Field methods to determine the infestation of the sorghum webworm and the damage by the sorghum midge in grain sorghums. J. Econ. Entomol. 54:749-50.

Ehler, L. E. 1973. Spider-mites associated with grain sorghum and corn in Texas. J. Econ. Entomol. 66:1220.

Ehler, L. E. 1974. A review of the spider-mite problem on grain sorghum and corn in West Texas. Tx. Agr. Exp. Stn. B-1149. 15pp.

Foster, D. G. and C. R. Ward. 1976. Wireworm bait traps in grain sorghum. Tx. Agr. Exp. Stn. PR-3425. 4pp.

Foster, D. G., G. L. Teetes and J. W. Johnson. 1977a. Field evaluation of resistance in sorghums to Banks grass mite. Crop. Sci. 17:821-3.

Foster, D. G., G. L. Teetes and J. W. Johnson. 1977b. Resistance in sorghums to the Banks grass mite. J. Econ. Entomol. 70:259-62.

Frederiksen, R. A. and N. E. Daniels. 1970. The influence of greenbugs on stalk rots of sorghum. Tx. Agr. Exp. Stn. PR-2772. 7pp.

Harding, J. A. 1965. Effect of insecticidal phytotoxicity and aphids on grain sorghum yields. Tx. Agr. Exp. Stn. Pr-2350. 6pp.

Hayes, W. P. 1922. Observations on insects attacking sorghum. J. Econ. Entomol. 15:349-56.

Henderson, C. F., H. G. Kinzer and J. H. Hatchett. 1965. Effectiveness of insecticides against the corn earworm in sorghum heads. J. Econ. Entomol. 58:207-9.

Hormchong, T. 1967. Studies on the development of insect resistant sorghum varieties and hybrids. Ph.D. thesis. Oklahoma State Univ. 87pp.

Johnson, J. W., D. T. Rosenow and G. L. Teetes. 1973. Resistance to the sorghum midge in converted exotic sorghum cultivars. Crop. Sci. 13:754-5.

Johnson, J. W., D. T. Rosenow and G. L. Teetes. 1974. Reponse of greenbug-resistant grain sorghum lines and hybrids to a natural infestation of greenbugs. Crop Sci. 14:442-3.

Johnson, J. W., G. L. Teetes and C. A. Schaefer. 1976. Greenhouse and field technique for evaluating resistance of sorghum cultivars to the greenbug. Southwestern Entomol. 1:150-4.

Kattes, D. H. and G. L. Teetes. 1978. Selected factors influencing the abundance of Banks grass mite in sorghum. Tx. Agr. Exp. Stn. B-1186. 7pp.

Kinzer, H. G. and C. G. Hendersen. 1968. Damage by larvae of the corn earworm to grain sorghum. J. Econ. Entomol. 61:263-7.

Kirby, R. D. and L. E. Ehler. 1977. Survival of Hippodamia convergens in grain sorghum. Environ. Entomol. 6:777-80.

Lopez, E. G. and G. L. Teetes. 1976. Selected predators of aphids in grain sorghum and their relation to cotton. J. Econ. Entomol. 69:198-204.

Mayo, Z. B. 1967. Resistance of native sorghums from India to the corn earworm and fall armyworm. Ph.D. thesis. Oklahoma State Univer. 48 pp.

McMillian, W. W. and K. J. Starks. 1967. Greenhouse and laboratory screening of sorghum lines for resistance to fall armyworm larvae. J. Econ. Entomol. 60:1462-63.

Mills, R. B. 1976. Host resistance to stored product insects - II. Proc. Joint U.S. - Japan Seminar on Stored Product Insects. Manhattan, Kansas. 77-87.

Owens, J. C., C. R. Ward and G. L. Teetes. 1976. Current status of spider mites in corn and sorghum. Proc. 31st Ann. Corn and Sorghum Res. Conf. 38-64.

Painter, F. w. and N. M. Randolph. 1972. Mass rearing the chinch bug in the laboratory. J. Econ. Entomol. 65:894-5.

Painter, R. H. 1951. Insect resistance in crop plants. The MacMillian Company, New York. 520 pp.

Painter, R. H. 1958. Resistance of plants to insects. Ann. Rev. Entomol. 3:267-90.

Pate, T. L. and C. W. Neeb. 1971. The Banks grass mite problem in the Trans-Pecos area of Texas. Tx. Agr. Exp. Stn. PR-2871. 3pp.

Randolph, N. M., G. W. Doering and A. J. Buckholt. 1960. The sorghum webworm and sorghum midge on grain sorghum. Tx. Agr. Exp. Stn. PR-2130. 5pp.

Randolph, N. M. and C. F. Garner. 1961. Insects attacking forage crops. Tx. Agr. Ext. Serv. B-975. 26pp.

Rogers, R. R. and R. B. Mills. 1974a. Evaluation of a world sorghum collection for resistance to the maize weevil, Sitophilus zeamais Motsch. (Coleoptera: Curculionidae). J. Kansas Entomol. Soc. 47:36-41.

Rogers, R. R. and R. B. Mills. 1974b. Reactions of sorghum varieties to maize weevil infestation under three relative humidities. J. Econ. Entomol. 67:692.

Rummel, D. R. and N. E. Daniels. 1971. Sorghum midge surveys in the Panhandle and South Plains of Texas. Tx. Agr. Exp. Stn. Consol. PR-2864. 8-10.

Russell, M. P. 1962. Effects of sorghum varieties on the lesser rice weevil, Sitophilus oryzae (L.) I. Oviposition, immature mortality, and size of adults. Ann. Entomol. Soc. Amer. 55:678-85.

Snelling, R. O., R. H. Painter, J. H. Parker, and W. M. Osborn. 1937. Resistance of sorghums to the chinch bug. U.S. Dept. Agr. Tech. Bull. 585. 56pp.

Starks, K. J. and R. L. Burton. 1977. Greenbugs: determining biotypes, culturing, and screening for plant resistance with notes on rearing parasitoids. ARS, USDA. Tech. Bull. 1556. 12pp.

Starks, K. J., R. L. Burton, G. L. Teetes and E. A. Wood, Jr. 1976. Release of parasitoids to control greenbugs on sorghum. USDA, ARS-S-91. 12p.

Stevens, R. A. and R. B. Mills. 1973. Comparison of technique for screening sorghum grain varieties for resistance to rice weevil. J. Econ. Entomol. 66:1222-3.

Teetes, G. L. 1971. Research results on grain sorghum pests in the High Plains. Proc. 4th Annu. Tx. Conf. Insect, Plant Dis., Weed and Brush Control. Tx. A&M Univ. p. 89-98.

Teetes, G. L. 1972. Differential toxicity of standard and reduced rates of insecticides to greenbugs and certain beneficial insects. Tx. Agr. Exp. Stn. PR-3041. 6pp.

Teetes, G. L. 1973. Insecticidal control of a spider mite in grain sorghum on the Texas High Plains. Tx. Agr. Exp. Stn. PR-3178. 4pp.

Teetes, G. L. 1973. Phyllophaga crinita: Damage assessment and control in sorghum and wheat. J. Econ. Entomol. 66:773-6.

Teetes, G. L. 1975. Insect resistance and breeding strategies in sorghum. Proc. 31st Ann. Corn and Sorghum Res. Conf. 32-48.

Teetes, G. L. 1976. Damage assessment and control of wireworms in grain sorghum. Tx. Agr. Exp. Stn. B-1167. 4pp.

Teetes, G. L., C. A. Schaefer and J. W. Johnson. 1974a. Resistance in sorghums to the greenbug: laboratory determination of mechanisms of resistance. J. Econ. Entomol. 67:393-6.

Teetes, G. L., C. A. Schaefer, J. W. Johnson, and D. T. Rosenow. 1974b. Resistance in sorghums to the greenbug: Field evaluation. Crop. Sci. 14:706-8.

Teetes, G. L., C. A. Schaefer, J. R. Gipson, R. C. McIntyre and E. E. Latham. 1975. Greenbug resistance to organophosphorous insecticides on the Texas High Plains. J. Econ. Entomol. 68:214-6.

Teetes, G. L., D. T. Rosenow, R. A. Frederiksen and J. W. Johnson. 1973. The predisposing influence of greenbugs on charcoal rot of sorghum. Tx. Agr. Exp. Stn. PR-3173. 6pp.

Teetes, G. L., E. G. Lopez, and C. A. Schaefer. 1975. Seasonal abundance of the greenbug and its natural enemies in grain sorghum in the Texas High Plains. Tx. Agr. Exp. Stn. B-1162. 4pp.

Teetes, G. L., G. W. Brothers, and C. R. Ward. 1973. Insecticide screening for greenbug control and effect on certain beneficial insect. Tx. Agr. Exp. Stn. PR-3116. 6pp.

Teetes, G. L. and J. W. Johnson. 1973. Damage assessment of the greenbug on grain sorghum J. Econ. Entomol. 66:1181-6.

Teetes, G. L. and J. W. Johnson. 1974. Assessment of damage by the greenbug in grain sorghum hybrids of different maturities. J. Econ. Entomol. 67:514-6.

Teetes, G. L., J. W. Johnson and D. T. Rosenow. 1974. Damage assessment of a false chinch bug, Nysuis raphanus (Howard), in grain sorghum. Tx. Agr. Exp. Stn. PR-3260. 4p.

Teetes, G. L., J. W. Johnson and D. T. Rosenow. 1975. Response of improved resistant sorghum hybrids to natural and artificial greenbug populations. J. Econ. Entomol. 68:546-8.

Teetes, G. L. and L. Wade. 1974. Seasonal biology and abundance of the white grub, Phyllophaga crinita, in the Texas High Plains. Tx. Agr. Exp. Stn. PR-3261. 4p.

Vanderlip, R. L. 1972. How a sorghum plant develops. Coop. Ext. Serv. Kansas State Univ. C-477. 19pp.

Walker, A. L., D. G. Bottrell and J. R. Cate, Jr. 1973. Hymenopterous parasites of biotype C greenbug in the High Plains of Texas. Ann. Entomol. Soc. Am. 66:173-6.

Ward, C. R., E. W. Huddleston, J. C. Owens, T. M. Hills, L. G. Richardson and D. Ashdown. 1972. Control of the Banks grass mite attacking grain sorghum and corn in West Texas. J. Econ. Entomol. 65:523-9.

Ward, C. R. and F. M. Tan. 1977. Organophosphate resistance in the Banks grass mite. J. Econ. Entomol. 70:250-2.

Ward, C. R., L. G. Richardson, D. Ashdown, E. W. Huddleston and R. Gfeller. 1971. Development of pesticide resistance in Banks grass mite indicated in field studies. Tx. Tech. Univ. Ent. Sp. Rept. 71-1. 8pp.

Wilson, A. G. L. 1976. Varietal responses of grain sorghum to infestation by Heliothis armigera. Expl. Agric. 12:257-65.

Wiseman, B. R., R. H. Painter and C. E. Wassom. 1966. Detecting corn seedling differences in the greenhouse by visual classification of damage by the fall armyworm. J. Econ. Entomol. 59:1211-4.