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Plant Resistance to Insects in Sorghum

International Crops Research Institute for the Semi-Arid Tropics

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Abstract

Sorghum is one of the most important cereals in the semi-arid tropics. Nearly 150 insect species have been reported to damage the crop worldwide, causing an estimated loss of over US$ 1 000 million annually. Of these, shoot fly, stem borers, greenbug, sugarcane aphid, shoot bug, spider mites, armyworms, midge, head bug, and head caterpillars are the major pests. Plant resistance to insects is one of the most important components of pest management in sorghum. Thus, the 18 specialist scientists who have contributed the 25 papers included in this book, that is based on a training course for researchers involved in the development of insect-resistant cultivars for integrated pest management in sustainable agriculture, describe and discuss the theoretical and practical aspects of resistance-screening techniques, mechanisms and inheritance of resistance, breeding for resistance, statistical designs, and strategies for integrated pest management. Information has also been provided about the international sorghum insect resistance testing program and the role of networks in collaborative research and technology exchange.

Résumé

La résistance variétale aux insectes nuisibles chez le sorgho. Le sorgho est une des plus importantes cultures céréalières dans les zones tropicales semi-arides. Près de 150 insectes nuisibles seraient responsables pour les dégâts à cette culture à travers le monde, occasionnant des pertes annuelles de l'ordre de 1 000 millions de dollars. Les insectes les plus nuisibles sont la mouche des pousses, les foreurs des tiges, le puceron vert, le puceron jaune du mil, la cicadelle du maïs, les araignées rouges, les chenilles légionnaires, la cécidomyie du sorgho, les punaises et les chenilles des panicules. La résistance variétale aux insectes constitue l'une des composantes les plus importantes de la lutte contre les insectes nuisibles chez le sorgho. Cet ouvrage, comportant 21 articles présentés par les chercheurs spécialistes, est basé sur un cours de formation destiné aux chercheurs travaillant à la mise au point de cultivars résistants aux insectes dans le cadre de la lutte intégrée contre les ravageurs pour l'agriculture durable. Les auteurs décrivent et examinent les aspects théoriques et pratiques des techniques de criblage pour la résistance, les mécanismes et l'hérédité de la résistance, la sélection pour la résistance, les dispositifs statistiques, ainsi que les stratégies pour la lutte intégrée contre les ravageurs. L'ouvrage fournit également des informations sur le programme international d'essai de la résistance du sorgho aux insectes nuisibles ainsi que sur le rôle des réseaux dans la recherche collaborative et dans l'échange de la technologie.

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Contents

Part 3 Mechanisms and inheritance of resistance

Part 4 Breeding methodologies

Part 5 insect pest management strategies

Part 6 International insect resistance testing program

Appendices

Introduction

Sorghum is one of the most important cereal crops in the semi-arid tropics. Over 150 species of insects cause damage to sorghum. The sorghum shoot fly (*Atherigona soccata*), stem borers (*Chilo partellus* and *Busseola fusca*), greenbug (*Schizaphis graminum*), sorghum midge (*Stenodiplosis sorghicola*), and head bugs (*Calocoris angustatus* and *Eurystylus oldi*) are the most important pests worldwide. Avoidable losses due to insects have been estimated to be over 32% in India, 9% in the USA, and 20% in Africa. In monetary terms, the losses due to insects have been estimated to be over US$ 1000 million annually in the semi-arid tropics.

Recommendations for insect control on sorghum include such cultural practices as early and uniform sowing, biological control, breeding insect-resistant cultivars, and the use of insecticides. In rainfed agriculture, it is difficult to plant during the periods when insect damage can be avoided. The technology to utilize natural enemies for insect control in the classical sense needs to be developed. Insecticides are costly, and beyond the means of resource-poor farmers in the semi-arid tropics. Therefore, host-plant resistance can form an important component of pest management in sorghum. Using insect-resistant cultivars is not only compatible with other methods of pest control, but is also environment-friendly. There is no cost involvement for the farmers.

To develop insect-resistant cultivars, it is important to ensure adequate and uniform insect infestation over seasons. Hot-spots can serve as useful locations to screen and breed for resistance to insects. However, it is difficult to achieve uniform insect infestation over seasons/locations under natural conditions. To overcome this problem, several techniques can be employed to augment natural insect populations and to achieve adequate insect infestation: collecting insects by different methods and confining them with plants at the most susceptible stage using cage techniques, or rearing them in a laboratory and infesting the test material in greenhouse or field conditions. To share knowledge on these procedures and techniques, a training course on "Host-Plant Resistance to Insects in Sorghum" was conducted at ICRISAT-Patancheru from 24 Oct to 3 Nov 1995.

The papers presented during the course are published in this book which describes theoretical and practical aspects of resistance-screening techniques for important insect pests of sorghum. It also covers various aspects of insect rearing, collection, and augmentation for resistance screening, field and greenhouse screening techniques, and damage evaluation.

It is hoped that this book will be useful in developing standardized procedures for resistance screening and data collection across locations, and foster closer collaboration among national agricultural research systems (NARS) and between NARS and ICRISAT. This will ultimately help in developing insect-resistant cultivars for increasing and stabilizing sorghum production in sustainable agriculture.

Part 1

Principles of Host-plant Resistance to Insect Pests

Fundamentals of Entomological Research

K F Nwanze

Introduction

Insects have existed on earth for over 350 million years, while human beings evolved only 1 million years ago. Insects provide many useful products such as silk, honey, and shellac, besides being eaten for food (caterpillars, grasshoppers, ants, termites, etc.). There must have been a period of mutual existence in nature, in which animals (including insects) and man lived in harmony. What are today regarded as diseases, or calamities, were viewed as natural regulating mechanisms. They still are, although humans may view them differently. However, humans became intruders in nature, and the most upsetting factor in the balance of nature. This imbalance is enhanced by:

- development of agriculture, especially the cultivation of extensive monocultures;
- the domestication of animals;
- storage of foodstuffs; and
- the explosion of human population.

Entomology deals with the study of insects, e.g., agricultural entomology (insects of agricultural importance), medical entomology (insects of medical importance), veterinary entomology (insects of veterinary importance), etc.

An insect is an invertebrate organism with: a symmetrical body; jointed appendages (mouth parts, legs, antennae, etc.); a body divided into three parts: head, thorax, and abdomen; one pair of compound eyes and antennae; three pairs of legs; and two pairs of wings.

An insect that interferes in human welfare and activities is termed an insect pest. It can be harmful to crops, commodities, or livestock. What we do in order to understand and contain insect pest populations can be defined as applied entomology. This requires an understanding of the insects and the environment they live in. Classical entomological research involves the identification of the insect, its geographical distribution (pest occurrence), incidence and population dynamics, bioecology and economic importance, and identification/development of control strategies.

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Nwanze, K.F. 1997. Fundamentals of entomological research. Pages 5-8 *in* Plant resistance to insects in sorghum (Sharma, H.C., Faujdar Singh, and Nwanze, K.F., eds.). Patancheru 502 324, Andhra Pradesh, India: International Crops Research Institute for the Semi-Arid Tropics.

Insect Classification

The following is an example of the hierarchy used in classifying scientific and common insect names:

Kingdom	-	Animalia
Phylum	-	Arthropoda
Class	-	Insecta
Order	-	Diptera (flies)
Family	-	Muscidae
Genus	-	*Atherigona*
Species	-	*soccata*
Common name	-	Sorghum shoot fly

Butterflies and moths are placed in order Lepidoptera; aphids, leafhoppers, and scales in Homoptera; grasshoppers and crickets in Orthoptera; beetles and weevils in Coleoptera; bugs in Hemiptera; and ants. bees, and wasps in Hymenoptera. For more examples, see Table 1.

Causes of Insect Outbreaks

- Large-scale changes-in cultivation practices, e.g., monoculture, use of single cultivars, introduction of new crops, etc.
- Introduction of new insect species into a region, e.g., cassava mealybug in Africa.
- Changes in the weather.
- Changes in production and management practices, e.g., excessive use of insecticides.

The Insect Environment and Related Terminology

The environment is the space and conditions surrounding an organism.

Biotic conditions - food, natural enemies, and competitors.

Abiotic conditions - atmospheric and edaphic factors (air, rainfall, humidity, temperature, light, etc.).

Pest incidence - presence and/or damage by an insect pest on a plant, or host.

Population dynamics - the fluctuation in abundance of an insect pest over time in a given environment.

Bioecology - the study of the life history and the (biology+ecology) interrelations between an insect and its environment.

Economic importance - in relation to crop loss resulting from the damage caused by an insect.

Economic injury level - the lowest pest density that will cause economic damage, or the pest density that causes damage equal to the cost of preventing the damage.

Table 1. Classification of important insect pests of sorghum.

Chilo partellus

Order		Lepidoptera
Family	:	Pyralidae
Genus	:	*Chilo*
Species		*partellus*
Common name	:	Spotted stem borer

Busseola fusca

Order	:	Lepidoptera
Family		Noctuidae
Genus	:	*Busseola*
Species		*fusca*
Common name		Maize stalk borer

Schizaphis graminum

Order	:	Hemiptera
Family	:	Aphididae
Genus		*Schizaphis*
Species		*graminum*
Common name	:	Greenbug

Stenodiplosis sorghicola

Order	:	Diptera
Family	:	Cecidomyiidae
Genus	:	*Stenodiplosis*
Species	:	*sorghicola*
Common name	:	Sorghum midge

Calocoris angustatus

Order	:	Hemiptera
Family	:	Miridae
Genus	:	*Calocoris*
Species		*angustatus*
Common name		Head bug

Eurystylus oldi

Order		Hemiptera
Family		Miridae
Genus	:	*Eurystylus*
Species	:	*oldi*
Common name	:	Head bug

Economic threshold - the pest density at which control measures should be applied to prevent an increasing pest population from reaching the economic injury level.

Insect Control Methods

Cultural control. The reduction of insect populations by use of agricultural practices. This makes the environment unfavorable to the insect (crop rotations, trap crops, tillage, and date of sowing).

Biological control. The reduction of insect populations by means of artificially encouraged living organisms (predators, parasites, and pathogens). In natural control, the same factors are at play, but are not manipulated by humans.

Chemical control. The reduction of insect populations, or prevention of insect damage by the use of chemicals to poison, attract, or repel the insects from specified areas.

Legal control. Lawful regulation of areas to eradicate, prevent, or control infestation, or reduce damage by insects (mostly through quarantine).

Host-plant resistance. The genetic ability of a plant to prevent or reduce damage caused by an insect, in host-plant resistance, one or more of three mechanisms could be involved: nonpreference, antibiosis, and tolerance. The ability of a host-plant to escape damage due to its growth pattern is often referred to as pseudo-resistance.

Integrated pest management. Management of insect populations by the utilization of all suitable techniques in a compatible manner so that damage is kept below the economic injury level.

Insect Pests of Sorghum: Biology, Extent of Losses, and Economic Thresholds

H C Sharma and K F Nwanze

Introduction

Sorghum is an important cereal crop in Asia, Africa, the Americas, and Australia. Sorghum grain yields on farmers' fields in Asia and Africa are generally low (500-800 kg ha^{-1}) mainly due to insects, diseases, weeds, and drought. Nearly 150 insect species have been reported as pests on sorghum (Reddy and Davies 1979; Jotwani et al. 1980). Shoot fly (*Atherigona soccata*), stem borers (*Chilo partellus, Busseola fusca, Eldana saccharina, Sesamia* spp and *Diatraea* spp), armyworms (*Mythimna separata, Spodoptera frugiperda, and S. exempta*), shoot bug (*Peregrinus maidis*), aphids (*Schizaphis graminum,* and *Melanaphis sacchari*), spider mites (*Oligonychus* spp), grasshoppers and locusts (*Hiewglyphus, Oedaleus, Aiolopus, Schistocerca,* and *Locusta*), sorghum midge (*Stenodiplosis sorghicola*), mirid bugs (*Calocoris angustatus* and *Eurystylus oldi*), and panicle-feeding caterpillars (*Helicoverpa, Heliothis, Euhlemma, Cryptoblabes, Pyroderces,* and *Nola*) are the major pests of sorghum worldwide. This paper summarizes the information on biology and ecology of the major insect pests of sorghum in the semi-arid tropics.

Nature of Damage, Biology, and Population Dynamics

Considerable information is available on the biology of white grubs, shoot flies, stem borers, armyworms, aphids, and sorghum midge. There is limited information on the biology of wireworms, mirid bugs, panicle-feeding caterpillars (except *Helicoverpa* and *Heliothis,* which are pests of several crops), blister beetles, stink bugs, lygaeid bugs, and panicle-feeding scarabaeid beetles.

Information on population fluctuations, diapause, carry-over from one season to another, and the role of biotic and environmental factors on insect development and abundance is inadequate. Such information is important for the formulation of insect population prediction models for integrated pest management. With greater specialization in biological sciences, recent advances in the use of electronic devices for data recording.

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and biotechnological approaches for pest management, little attention is being given to studies on insect biology, ecology, behavior, and insect-host plant-environment interactions. These are vital components for developing an ecologically sound pest management program. Information on insect biology and population dynamics of sorghum insects has been compiled by Nair (1975), Gahukar and Jotwani (1980), Sharma et al. (1982), Teetes et al. (1983), Sharma and Davies (1988), Sharma and Lopez (1990), and Ratnadass et al. (1994).

White grubs (*Holotrichia* spp and *Phyllophaga crinita*)

White grubs are cosmopolitan and damage a range of crops. *H. consanguinea* and *H. serrata* are occasional pests of sorghum in India. The damage to seedlings occurs soon after germination. Plant stand can be reduced significantly if the crop is infested immediately after germination. One grub can destroy several plants within a row. Infested plants can be severely stunted, and may not produce any grain. Infested plants are also prone to lodging in later growth stages. Adults are brownish black, and nearly 13 mm long. Larvae are C-shaped with brown heads and white bodies. Fully grown larvae are over 20 mm long. Tropical and subtropical species have a life cycle of 1-2 years, while temperate species take 1-4 years. In India, adults emerge soon after the summer rains, mate, and lay eggs singly, 5-10 cm deep in the soil (Veeresh 1977). Eggs hatch in 2 weeks, and the larvae undergo three molts. First and 2nd larval instars last about a month each, while the 3rd-instar lasts 2-4 months. Pupation takes place in an earthen cell below the root zone, and the pupal period lasts about 2 weeks. The adults remain within the pupal cell until the next summer rains. Light and well-drained soils are favorable for white grub infestation.

Wire worms (*Gonocephalum, Eleodes, Conoderus,* and *Aeolus*)

Several species of true and false wireworms belonging to Elateridae and Tenebrionidae (*Gonocephalum, Eleodes, Conoderus,* and *Aeolus)* feed on seeds or seedlings soon after sowing. They can be recognized by their shiny, wire-like, yellow, or orange bodies. They feed on plant parts below the ground. *Gonocephalum* spp occur in Asia and Australia. *Eleodes, Conoderus,* and *Aeolus* are prevalent in North America. Life history of wireworms is similar to that of the white grubs. Eggs are laid singly 3-15 cm below the soil surface. A female may lay 50-300 eggs, which hatch in 3-4 weeks. Fully grown wireworms pupate about 15 cm below the soil surface, and the adults emerge a week later. The life cycle may take 1-3 years, and generations may overlap.

Southern corn rootworm (*Diabrotica undecimpunctata*)

This insect is known as the southern corn rootworm in the larval stage, while the adult is known as the spotted cucumber beetle in North America. The larva bores into the roots or the stalk just above the roots, eats the crown of the young plants, and kills the growing point. The symptoms of damage are stunting and deadhearts. The eggs are pale yellow.

and sculptured with hexagonal pits. The larvae are white, pale yellow, and 12 mm long. The adult is yellowish green with 11 black spots on the forewings. It overwinters as an adult in trash, but may be active during mild winters. There are 2 generations a year.

Sorghum shoot fly (*Atherigona soccata*)

Sorghum shoot fly is a key pest of sorghum in Asia, Africa, and Mediterranean Europe. Shoot fly females lay cigar-shaped eggs singly on the lower surface of the leaves, at the 1-7 leaf stage. Eggs hatch in 1-2 days. The larva cuts the growing point, resulting in wilting and drying of the central leaf, known as "deadheart". The deadheart can be pulled out easily, and it produces a bad smell. Normally, the damage occurs 1-4 weeks after seedling emergence. The damaged plants produce side tillers that also may be attacked. Larval development is completed in 8-10 days and pupation takes place mostly in the soil. The pupal period lasts for 8 days. The entire life cycle is completed in 17-21 days. Shoot fly numbers begin to increase in Jul, and peak in Aug-Sep. Infestations are high when sorghum sowings are staggered due to erratic rainfall. Shoot fly infestations are high in the postrainy season crop planted in Sep-Oct. Temperatures above 35°C and below 18°C and continuous rainfall reduce shoot fly survival. During the off-season, the insect survives on alternate hosts (*Cymhopogon* sp, *Echinochloa colonum, E. procera, Paspalum scrobiculatum,* and *Pennisetum glaucum*) and on volunteer or fodder sorghum.

Stem borers (*Chilo, Busseola, Eldana, Sesamia,* and *Diatraea*)

Several species of stem borers have been reported as pests of sorghum in different regions. The stem borer infestation is indicated by appearance of small elongated windows in young whorl leaves where the young larvae have eaten the upper surface of the leaves. Later, the plants present a ragged appearance as the severity of damage increases. The 3rd-instar larvae migrate to the base of the plant, bore into the shoot, and damage the growing point resulting in the production of a deadheart. Normally, two leaves dry up as a result of stem borer damage. Larvae continue to feed inside the stem. Throughout the crop growth, extensive tunneling in the stem and peduncle leads to drying up of the panicle, to a partially chaffy panicle, or to peduncle breakage. Stem borer infestation starts about 20 days after seedling emergence, and deadhearts appear on 30-40-day-old crop.

Spotted stem borer, *Chilo partellus* is common in Asia and eastern and southern Africa. A female can lay up to 500 eggs in batches of 10-80 near the midrib on the undersurface of the leaves. Eggs hatch in 4-5 days. The larvae move to the leaf whorl and feed on tender leaves resulting in leaf-scarification and shot-holes. Third-instar larvae move to the base of the plant and bore into the shoot. Damage to the growing point results in the production of a typical deadheart. In mature plants, the larvae tunnel inside the stem. The larval development is completed in 19-27 days. Pupation takes place inside the stem and the adults emerge in 7-10 days. During the off-season, the larvae undergo diapause in plant stalks and stubbles. With the onset of rains, the larvae pupate and the adults emerge in 7 days. In northern India, moth catches in light traps begin to increase during the last week of Jul and peak during Aug-Sep, while in southern India the peak in moth catches has been observed during Jan-Feb.

Maize stalk borer, *Busseola fusca* is a key pest of sorghum in Africa. Young larvae feed on the leaves, while the older larvae bore into the stems and produce deadhearts. Under severe infestation, plant growth is retarded, and panicle emergence and grain formation are severely affected. Damage symptoms are similar to those caused by *C. partellus.* Eggs are laid in batches of 30-150 on the inner surface of leaf sheaths. About 400-500 eggs are laid by a female over a period of 5-6 days. Eggs hatch in 5-6 days, and the young larvae remain in clusters inside the leaf sheaths. The larvae disperse the following night, and move to the leaf whorl for feeding. The larvae have buff to purple brown-colored bodies. There are 6-7 larval instars, and larval development is completed in 24-36 days. Larvae pupate in the plant stem, and cut an exit hole before pupation. Adults emerge in 9-12 days, and exhibit a wide variation in color. Its major infestations are encountered between Aug-Oct in western Africa. It is only the 1st generation that causes severe damage to the crop. Usually 3 generations are produced in a year. The 3rd-generation larvae enter diapause with the onset of the dry season, and complete development in 6-7 months.

Pink borer, *Sesamia inferens* is a pest of sorghum in Asia. Several other species of *Sesamia* are important pests of cereal crops including sorghum in Africa. The larvae bore into the stem and kill the central shoot, producing a deadheart. One larva may damage several plants in its life span. Other symptoms of damage are similar to those of C. *partellus.* The moth is fawn-colored, has dark brown streaks on the forewings, and white hindwings. Each female lays up to 400 eggs. Eggs are laid in batches of about 150, and arranged in two to three rows between the leaf sheath and the stem. Eggs hatch in 5-7 days in warm weather, but the incubation period may be longer in the winter, or dry season. The fully grown larva is pale yellow with a pink tinge, and a reddish brown head. The larval period lasts for 25 days, and may be prolonged to 75 days in cold months. Pupation occurs in the larval tunnel in the stem, and the adult emerges in 12 days. It completes 1 generation in 6-7 weeks.

African sugarcane borer, *Eldana saccharina* occurs throughout Africa south of the Sahara. Its infestations are characterized by the presence of frass at the point of penetration in the stem. Young larvae feed on the leaves, usually boring into the midrib. Fully grown larvae bore into the stems, and produce deadhearts. The larvae hang down with silken threads and are blown over to neighboring plants. A female lays about 400-600 eggs in 2 weeks in batches of 2-200 eggs. The egg incubation period is 5-7 days. The young larvae are orange, turn dark gray as they feed, become active, and produce large quantities of frass. There are 6-7 larval instars in the males, and 7-8 instars in the females. The larval period ranges from 20 to 60 days. Pupation takes place in a tough silken cocoon inside the stem. The adults emerge in about 8-13 days.

American sugarcane borers, *Diatraea saccharalis, D. grandiosella,* and *D. lineolata* are widely distributed in the Americas. They are major pests of sugarcane, and occasionally infest sorghum. Young larvae feed on the leaves, and then bore into the stems. Larvae tunnel the stalk extensively and cause plant lodging. Boring into the peduncle causes peduncle breakage. Larvae are white-yellow, and have black spots on most body segments. The spots fade, or are absent in the diapausing larvae. Pupation occurs in the spring, and the adults emerge a few weeks later. Eggs are elliptical to oval, flattened, covered with scales, laid in clusters, and hatch in 3-7 days. The larval stage lasts about 25 days, and the pupal stage for 10 days. There are 1-3 generations in a year.

Armyworms (*Mythimna separata, Spodoptera frugiperda,* and *S. exempta*)

Several species of armyworms are sporadic pests of sorghum crop. The oriental armyworm, *Mythimna separata* is widely distributed in Asia, Pacific islands, Australasia, and parts of Africa. The larvae feed on leaves, leaving only the midribs and panicles. It is also a serious pest on other cereal crops. Its outbreaks are sporadic. When the larvae are in the gregarious phase, they move in a band and feed on the foliage of most of the graminaceous plants they come across. Feeding takes place mostly at night, and the larvae hide in the plant whorls, or under the cover of vegetation during the day. Females lay 500-900 eggs on the lower surface of leaves, on dry leaves, and on grasses. The eggs hatch in 2-7 days. Larval development is completed in 14-22 days and the pupal stage lasts for 8-9 days. The adults live for 4-5 days. Mating occurs on the 3rd day and oviposition on the 4th day after emergence. The larvae feed mostly on leaves at night and migrate when the food is exhausted. Maximum larval abundance is in August in India. Peak moth catches occur in light traps during September. Trap catches are highest during a period of low rainfall, following a 2-4 week period of high rainfall, moderate temperatures, and high humidity.

The fall armyworm, *Spodoptera frugiperda* is an important pest of sorghum in southeastern USA and tropical America. The larvae feed on the tender whorl leaves. The unfolded leaves show a row of shot-holes or elongated feeding areas. After panicle emergence, the larvae feed on developing grain. The moth has variegated forewings. Eggs are usually deposited on the lower leaf surface. The egg masses are covered with scales and hairs of the moth. The young larvae are slightly greenish, with black heads. Mature larvae vary from greenish to grayish brown, have a predominantly white inverted Y-shaped suture on the head, and dorsal lines running lengthwise on the body. The life cycle is completed in 1 month. In subtropical and tropical America the adults migrate northward each year as the weather warms up.

The African armyworm, 5. *exempta* is an occasional pest of sorghum in Africa. Its outbreaks occur periodically. During outbreaks, it causes serious damage to pasture grasses and cereal crops. The larvae are gregarious, and feed on the leaves. Eggs are laid on the lower surface of the leaves in groups of 20 or more. Eggs hatch in 3-4 days. Larval development is completed in 10-21 days. Solitary larvae are green, but the gregarious forms are black with green undersides. Pupation occurs in soil and the adults emerge in about 1 week. Moths migrate long distances, giving rise to outbreaks away from the site of infestation. Outbreaks are associated with rains and, in eastern Africa, there is a general northward progression with infestations occurring earlier in the south than in the north of Africa.

Leaf roller (*Marasmia trapezalis*)

Leaf roller is a sporadic pest of sorghum and pearl millet in India and Africa. The larvae fold the tips of the tender leaves and feed inside the folded leaf. The larvae scratch the leaf tissue inside the folded leaf. As a result, the damaged leaves begin to dry up from the

tip. Broad-leaved and highly succulent varieties are more susceptible to this pest. The moths are brown with waxy brown markings on the forewings. The eggs are laid on the upper surface of the leaves. The larva is slender. yellowish green and 20 mm in length. It has small, oval, spiny patches scattered over the body from which stout bristly hairs arise. The larvae are fully grown in 11-20 days. Pupation occurs within the folded leaf, and the pupal period lasts for 6-8 days.

Grasshoppers and locusts (*Hieroglyphus, Oedaleus, Aiolopus, Locusta, and Schistocerca*)

Several species of grasshoppers and locusts damage sorghum in various parts of the world. *Hieroglyphus* and *Schistocerca* are important in India, while *Oedaleus, Aiolopus,* and *Locusta* are important in Sub-Saharan Africa. All stages of the plant may be attacked, but crop losses are higher when the damage occurs at the seedling stage, or on ripening panicles. Grasshoppers and locusts are polyphagous, and feed on several plant species.

Hieroglyphus nigrorepletus and *H. banian* are important grasshopper pests in India. Adults and nymphs feed on foliage and occasionally on panicles. A female may lay 3-6 egg pods, each containing 20-50 eggs that hatch the following Jun-Jul with the onset of monsoon rains. The eggs remain viable up to 3 years. The nymphs mature in 3-5 months. Adults hibernate among grasses or other wild hosts.

Oedaleus senegalensis adults are 30-40 mm long, and greenish brown. *Aiolopus simulatrix* is 25 mm long and pale brown. Adults migrate at night and increase in insect density can occur quickly. Nymphal development is completed in 2 months. Nymphs have the capacity to survive long dry spells. *O. senegalensis* can remain in the egg stage for over 1 year, while *A. simulatrix* survives in the adult stage. Egg laying in the latter is delayed till the onset of rains. Breeding is continuous in the rainy season, and the generation time is 40-50 days.

Schistocerca gregaria and *Locusta migratoria* are important locust species. They cause heavy damage to crops during outbreaks. Nymphs and adults feed on leaves, flowers, and developing grain. They feed on a number of graminaceous plants and occasionally on other plant species. They occur in solitary and gregarious phases. In the migratory phase locusts cause widespread damage.

The adults of desert locust, 5. *gregaria* in the solitary phase are light yellowish gray, while those of the gregarious phase are lemon yellow. It is widely distributed in semidesert regions of Asia and Africa. Most severe losses are caused by the young adults when they leave the breeding sites and invade new areas during outbreaks. Females lay eggs in damp soil at a depth of 6-8 cm. Eggs are laid in masses of 20-200, and a female lays 60-160 eggs. Egg incubation takes 10-15 days, but varies according to temperature. Nymphal development is completed in 6-7 weeks. Hoppers are able to fly for 2 days after the final molt. Rain and a decline in temperature causes the swarms to settle on the ground.

The migratory locust, *L. migratoria* is brownish and winged, and about 50 mm long in the migratory phase. The nymphs pass through 5 instars, and initially are all black, but become orange-black with age. Solitary nymphs are green. The gregarious nymphs occur

in dense bands, and can cover several hundred meters in a day. The adults form swarms, and migrate long distances. Each female lays 3-4 egg pods in the soil, and each egg pod has approximately 50 eggs. The total development time is 3 months under favorable conditions, and there may be up to 4 generations in a year. Under adverse circumstances, there may be only 1-2 generations in a year.

Aphids (*Schizaphis graminum, Rhopalosiphum maidis, Melanaphis sacchari,* and *Sipha flava*)

Greenbug, *Schizaphis graminum* is widely distributed in the Americas, Africa, and Asia. It is a key insect pest of sorghum in the Americas. Its infestations can be detected by the presence of red spots on the leaves caused by the toxins injected into the plant by the aphids. The reddened areas enlarge, and yellow leaves turn brown from the edges. The aphids feed in colonies on the underside of the foliage. It also transmits maize dwarf mosaic virus. The adult is nearly 2 mm long, light green, with a darker green dorsal abdominal stripe. Winged and wingless forms may be present in the same colony. Each female lays about 80 offspring in 25 days, and the life cycle is completed in 7 days.

The corn leaf aphid, *Rhopalosiphum maidis* sucks the sap from the whorl leaves during the vegetative stage of the crop. It also feeds on the panicles, and produces honeydew on which sooty molds grow. It also transmits maize mosaic virus. It is distributed in most sorghum-growing areas. However, its infestations rarely reach damaging proportions. Damage may result in the yellowing, tanning, and drying up of leaves. The aphids are bluish green, 2 mm long, with black legs, antennae, and cornicles. The females give birth to apterous forms that molt 4 times to become adults. Under crowded conditions, or when the host plants are under stress, aphids produce winged adults, which molt 5 times to become adults. Nymphal development is completed in 12 days. Reproduction begins 5 days after the final molt. Each female can produce up to 46 progenies, with an average of 19. The adults live for 11-18 days. One generation is completed in 12 days. In mild climates, this species is active throughout the year.

The sugarcane aphid, *Melanaphis sacchari* occurs in Asia, Africa, Australia, and America. It prefers to feed on the undersurface of older leaves. The damage proceeds from lower to upper leaves. The adults and nymphs are yellow. They suck sap from the lower surface of leaves, and this leads to stunted plant growth. Damage is more severe in a drought-stressed crop, resulting in drying of leaves, and plant mortality. The aphids secrete honeydew that falls on the ground, on which sooty molds grow. Their numbers increase rapidly at the end of the rainy season during dry spells. This insect multiplies by parthenogenesis. Each female gives birth to 60-100 nymphs in 13-20 days. The life cycle is completed in 6-7 days during the dry season. Its abundance is high during the postrainy season in India.

The yellow sugarcane aphid, *Sipha flava* is an occasional pest of sorghum in North America. It prefers to feed on older leaves, secretes a potent toxin, and causes plant mortality at the seedling stage. The initial symptom of damage is leaf purpling at the seedling stage. In older plants, feeding results in severe yellowing of the plants. The adults are lemon-yellow, 2 mm long, and have two rows of dark tubercles down the

dorsum. A female produces 18 nymphs over a period of 28 days, and the development is completed in 13-19 days. Both winged and wingless forms exist in the same colony.

Shoot bug (*Peregrinus maidis*)

Shoot bug is a common pest of sorghum in India, parts of Africa, the West Indies, and the Philippines. It sucks sap from the leaf whorls, and between the leaf sheath and the stem. Damaged plants become stunted. In cases of severe infestation, the top leaves start drying up first, extending gradually to the lower leaves, and the plant may die. The leaves curl and present a tanned appearance. They also secrete honeydew on which sooty molds grow. Infestation at the later stages of plant growth may twist the top leaves, and inhibit panicle emergence. Its infestations are more severe under drought conditions. It is a serious pest of sorghum in the postrainy season in India. Females are yellowish brown and males are dark brown. Wings may be longer or shorter than the abdomen. Long winged forms have transparent wings. Females are larger than the males. The nymphs and adults live in groups in plant whorls and on the inner side of leaf sheaths. The females make a slit in the upper surface of the midrib, and insert eggs in groups of 1-4, and cover them with a white waxy substance. A female lays up to 100 eggs in 7 days. The eggs are white, elongate, cylindrical, and taper at the ends. Egg incubation period is 1-7 days. There are 7 nymphal instars, and the development is completed in 16 days.

Chinch bug (*Blissus leucopterus*)

Chinch bug is widely distributed in North America. It destroys sorghum plants by sucking large quantities of sap from the stem and underground plant parts. Young plants are highly susceptible. Older plants become weak, reddened, stunted, and are prone to lodging. Eggs are laid in the lower leaf sheaths, on roots, or on the ground. Newly emerged nymphs are pale yellow, but soon become red except for the first two abdominal segments. The later instars become dark red, but retain a pale yellow band at the anterior part of the abdomen. The last instar is gray-black with a conspicuous white spot on the back between the wing pads. The life cycle is completed in 30-40 days, and there are normally 2 generations in a year. Chinch bugs overwinter in the adult stage in bunch grass. Migrations begin when temperatures reach 21°C.

Spider mites (*Oligonychus indicus* and *O. pratensis*)

Spider mites suck sap from the undersurface of the leaves, beginning along the midrib of lower functional leaves. The infested leaf areas become pale yellow initially, and later turn reddish on the top. The entire leaf may turn brown. As mite numbers increase on the lower leaves, the infestation spreads upwards through the plant. The underside of the infested leaves has a dense webbing. Under severe infestation, mites may web the sorghum panicles as well. Infestation generally increases after panicle emergence. Banks grass mite, *O. pratensis* exhibits marked sexual dimorphism. The females are larger than

the males. After feeding, both sexes become dark green, except the palpi and first two pairs of legs. Each female lays about 50 eggs on the undersurface of the leaves. Eggs hatch in 3—4 days. Six-legged nymphs are light-colored, and the eight-legged nymphs become progressively green. The life cycle is completed in about 11 days.

O. indicus infestations become severe under drought conditions and are thus generally more severe during the postrainy and dry summer seasons. Nymphs and adults feed on the leaves, and sometimes on the panicles during the postrainy season. Infestations generally spread from the field margins along the wind direction.

Sorghum midge (*Stenodiplosis sorghicola*)

Larvae of the sorghum midge feed on the developing ovary resulting in kernel loss. Females lay eggs within spikelets at flowering. Damaged panicles have a blasted appearance. Spikelets damaged by sorghum midge have a pupal case attached to the glumes, or have a small exit-hole of the midge parasite on the upper glume. Adults emerge between 0600 and 1100 in the morning. Mating takes place within 1 h after emergence. Generally, males emerge 1 h earlier than the females, and hover around the spikelets where the females are about to emerge. Males die after mating, while the females proceed in search of sorghum panicles at flowering for oviposition. Each female lays 75-100 eggs singly inside the spikelets during the morning hours, and dies after oviposition by afternoon. Eggs hatch in 1-4 days. The larvae ingest the contents of developing ovaries and complete development in 7-12 days. Larvae pupate inside the glumes. The pupal period lasts tor 3-8 days. Adults live for 2-48 h. A small proportion of the larvae enters diapause in spikelets in each generation, which may last as long as 3-4 years. The larval diapause is terminated by warm and humid weather (25-30°C and >60% relative humidity).

Head bugs (*Calocoris angustatus, Eurystylus oldi, Taylorilygus vosseleri, Creontiades pallidas,* and *Campylomma* spp)

The nymphs and adults suck the sap from the developing grain. Damage starts as soon as the panicle emerges from the boot leaf. Bug-damaged grain shows distinct red-brown feeding punctures which create quantitative and qualitative losses. Head bug damage spoils the grain quality, and renders the grain unfit for human food. Such grain also has poor germination. Bug damage also increases the severity of grain molds.

Calocoris angustatus females lay eggs inside spikelets from panicle emergence to post-anthesis. A female lays 150-200 eggs. The eggs hatch in 5-7 days. Nymphal development is completed in 15-17 days. Nymphs feed on milky and soft-dough grains resulting in pigmentation and shriveling of the grain. Its infestations are high during Aug-Sep in the rainy season in India. During the off-season, bugs feed on fodder sorghum. There is no evidence of diapause.

Females of *Creontiades pallidus* insert the eggs in the grain at the milk stage. The tip or the operculum of the egg can be seen outside the grain surface. The grain pericarp develops a red-brown ring around the egg. A female lays 45-250 eggs and the eggs hatch

in 6-8 days. Five nymphal instars complete development in 11—15 days. Males live for 11 days and the females for 13 days. The adults also feed on pearl millet and pigeonpea during the off-season.

Eurystylus bellevoyei occurs in India and is closely related to E. *oldi*—the predominant mirid bug species in western Africa. The eggs are laid inside the grain at the milk stage. The tip of the egg projects outside the grain surface. The eggs hatch in 7 days, and the nymphal development is completed in 7-8 days. The entire life cycle is completed in 14-16 days.

Eurystylus oldi lays 1-7 eggs in the grains. Egg incubation period lasts for 4-6 days. The five nymphal instars complete development in 6-11 days. The preoviposition period lasts for 2-3 days. Females survive for 4-18 days, and males for 6-20 days. On average, a female produces 24-136 eggs.

Campylomma spp lay the eggs inside the grain at the milk stage. The egg incubation period is 5 days. Five nymphal instars complete development in 11 days. Species belonging to this genus are polyphagous.

Panicle-feeding bugs (*Dolycoris, Nezara, Agonosceiis, Oebalus, Calidea, Chlorochroa, Spilostethus,* and *Leptoglossus*)

Panicle-feeding bugs suck sap from the developing grain and tender branches of the panicle. Bug-damaged grain also becomes more prone to grain mold infection that further diminishes the grain quality. Damaged grain shrivels and becomes softer and lighter than the undamaged grain. Such grain may be lost during threshing. The extent of damage depends on insect abundance, stage of grain development at the time of infestation, and the duration of infestation. Some of these characteristics may also differ with the species. Bug-damaged grain is unfit for food purposes, and shows poor seed germination and seedling establishment.

The sap-sucking bug, *Dolycoris indicus* is widely distributed in Asia. Adults are dull brown, or yellowish with black spots, and are 10 mm long and 6 mm wide.

The green stink bug, *Nezara viridula* is cosmopolitan. It is typically shield-shaped and 19 mm long. Males are smaller than the females. Females lay 300-500 eggs in clusters of about 30. Egg incubation period is 4-7 days, and the nymphal development is completed in 3-6 weeks. The life cycle is completed in 5-7 weeks. Adults live for 40-60 days. It overwinters in the adult stage.

Agonosceiis (versicolor) pubescens is a pest of grain sorghum in Africa. The adults are yellow-brown with hairs on the body.

The rice stink bug, *Oebalus pugnax* is a pest of sorghum in the Americas. It is straw-colored, shield-shaped, and 12 mm long. It lays 10-47 light green cylinder-shaped eggs, arranged in a cluster of two rows. Eggs hatch in 5 days. Nymphs complete development in 15-28 days.

The iridescent blue-green cotton bug, *Calidea dregii* is a pest on sorghum panicles in Africa. This bug is conspicuous because of its blue-green color. Spherical eggs are laid in batches of up to 40 in closed spirals. The eggs are white, and turn red as they develop. The nymphs resemble the adults in color. The life cycle is completed in 23-56 days.

Lygaeid bugs, *Spilostethus* spp are distributed in Asia and Africa. Under some circumstances they become abundant during the late stages of grain development.

Panicle-feeding caterpillars (*Helicoverpa, Heliothis, Eublemma, Cryptoblabes, Nola, and Euproctis*)

Head caterpillars feed on the developing grain. They destroy the grain mostly inside the panicle. Some species produce webs of silken threads inside the panicle, or make small holes in the grain. In cultivars with compact panicles, the inside of the panicle may be completely damaged and filled with frass while the panicle may look healthy externally.

The American bollworm, *Helicoverpa armigera* is a pest on a number of crops. Eggs are spherical, yellow, and laid singly all over the panicle. A female lays approximately 700 creamy white eggs, which hatch in 4-6 days. The larvae complete development in 3-4 weeks. Pupation occurs in the soil and adults emerge after 2-4 weeks. Moths are large, brown, or gray with specks that form a V-shaped mark on the forewings. Its infestations are high in sorghum cultivars with compact panicles.

The corn earworm, *Heliothis zea* is widely distributed in the Americas. Young larvae feed on tender folded leaves. The unfolded leaves present a ragged appearance. Feeding on the developing grain is more serious. Moths are dusty yellow, or gray to reddish brown. Females are active in the evening, and lay 350-3000 eggs. Eggs are flattened, spherical, ribbed, and hatch in 3-5 days. Newly emerged larvae are white and grow rapidly. The older larvae are pink, green, yellow, or almost black. Many are conspicuously striped. Pupation occurs in the soil. It overwinters in the pupal stage.

Eublemma silicula is a pest of sorghum in several parts of India. The caterpillars remain hidden in a small gallery formed of silken threads and anthers. The greenish white eggs are elongate and oval. The caterpillars are hairy and brownish yellow. The forewings of the moths are reddish buff with three dark spots on the anterior margin. The egg, larval, and pupal periods last for 4, 12-13, and 12 days, respectively. *E. gayneri* is closely related to this species, and is a pest of sorghum in Africa. *Pyroderces simplex* also infests sorghum panicles in Asia and Africa.

The sorghum webworm, *Nola sorghiella* is a serious pest in humid regions of the Americas. Young larvae feed on the floral parts, while the older larvae cut circular holes in the developing grain. The larvae do not spin webs over the panicles, but spin a silken thread when disturbed, and hang themselves from the panicles. Moths are active at night, and lay about 100 eggs singly on the panicle. Eggs hatch in 3-4 days. Mature larvae are 12 mm long, and larval development is completed in about 13 days. The larvae are flattened, yellowish, or greenish, and marked with four longitudinal reddish black dorsal stripes. The body is covered densely with spaced long hairs and spines. The pupal period lasts for 6 days. Adults live for 5 days. A generation requires about 1 month. Diapause occurs in the larval stage hidden in the host plant.

The earhead webworm, *N. analis* is of minor importance in Asia and Africa. The larvae feed on the developing grain. Eggs are creamy white and are laid on spikelets and the grain. Egg incubation period is 2-3 days. The young larva is dark gray with hairs on the body. The larval development is completed in 2 weeks. The larvae remain inside the

webs formed from excreta and silken threads. The pupa is short, conical, and fully covered with webbing. The pupal period is about 8 days. The adults are small and white. There are two prominent black spots on the anterior margin of forewings followed by zigzag dirty-white stripes that run vertically.

Cryptoblabes gnidiella is a pest of hybrids and high-yielding varieties in India. The eggs are laid on the spikelets and tender grain. Caterpillars are dark brown. Egg and larval periods last for 3-4 and 9-10 days, respectively. Creamy white, round, or conical eggs are laid on the spikelets and grains on the panicle. The freshly emerged larva is dirty white, with a brown head. The fully grown larva is dark brown and measures 12 mm. Pupation takes place inside the silken webs. It is fully covered by silken threads that are produced by the larva. The adult has dark gray forewings. Hindwings are fringed with hairs on the anterior margin, and are larger than the forewings. The life cycle is completed in 22-24 days.

The tent hairy caterpillar, *Euproctis subnotata* is an occasional pest on the sorghum panicle in India. The larvae infest sorghum panicles in large numbers and feed on hardening grain. The damage caused is relatively small. The hairs on the larvae can cause skin irritation. Adults have brown forewings with dark scales. The hindwings are yellow. Spherical white eggs are laid in batches of 6-24, and covered with orange yellow hairs from the anal tuft of the female. The larvae congregate on the panicle. The larva is dark brown with a wide yellow band dorsally on the abdominal segments I—VII, and IX. Pupation takes place in the ground, and also on the panicles. Egg, larval, and pupal stages last for 5-7, 15-40, and 10-17 days, respectively.

Extent of Losses

Assessments of sorghum grain yield losses caused by insect pests are scarce and difficult to obtain. Annual losses due to insect pests differ in magnitude on a regional basis. They have been estimated to be US$ 1 089 million in the semi-arid tropics, US$ 250 million in the USA, and US$ 10 million in Australia (ICRISAT 1992). Nearly 32% of the sorghum crop is lost due to insect pest infestation in India (Borad and Mittal 1983). Four to 84% of the sorghum grain in India is lost to panicle-feeding insect pests. Annual grain yield loss at the minimum infestation level of 4.6% is equivalent to US$ 100 million (Leuschner and Sharma 1983).

Economic Thresholds

Economic threshold levels (ETLs) have been established for many insect pests of sorghum. ETLs vary over seasons and locations. They are influenced by variations in the cost of inputs, the value of the produce, productivity potential of the crop, and relevant socioeconomic factors. One wireworm larva in 30 hand-sized clods/debris before sowing has been estimated to be the ETL for *Gonocephalum* sp in Australia (Passlow et al. 1985). For *A. soccata,* the ETL has been estimated to be 4-10, 3-9, and 6-15%

deadhearts in sorghum cultivars CSH 1, CSH 5, and Swarna, respectively (Rai et al. 1978). There can be a considerable compensation in grain yield by production of tillers in the damaged plants, and up to 20% deadheart formation may not cause a significant reduction in grain yield. A 1% increase in infestation leads to 89.1 and 30.5 kg ha^{-1} reduction in grain yield in CSH 5 and M 35-1 (Mote 1986a). ETLs for the spotted stem borer. *C, partellus* have not been computed. However, the relations between stem tunneling and loss in yield have been estimated by Mote (1986b). One larva $plant^{-1}$ has been reported as the ETL for the army worm (Giraddi and Kulkarni 1983). ETLs for sorghum midge, *S. sorghicola* have been estimated to be 0.6 adult sorghum midges $panicle^{-1}$ in Taiwan (Hong 1987), 0.4-3.0 $panicle^{-1}$ in the USA (Fuchs et al. 1993), 1.0 in India and Argentina (Karanjkar and Chundurwar 1978; Limonti and Villata 1980), and more than 6 in Australia (Passlow et al. 1985). ETLs for panicle-feeding bugs differ by cultivar and the stage of panicle development when the infestation occurs. ETLs for C. *angustatus* have been estimated to be 0.06-0.12 adults at the half-anthesis stage and 5.4-10.5 adults at the milk stage, or 7.9-15.0 nymphs at the milk stage (Natarajan and Sundara Babu 1988; Sharma and Lopez 1989). For *E. oldi,* the ETLs have been estimated to be 0.97-2.52 bugs $panicle^{-1}$ at the milk stage (O. Ajayi, personal communication). Hall et al. (1983) studied the insect density-to-yield loss relations for four species of panicle-feeding bugs: *O. pugnax, C. ligata, L. phyllopus,* and *N. viridula.* Largest reductions in grain yield occurred when panicles were infested at the milk-to-maturity stage of kernel development. Percentage yield reductions increased quadratically with an increase in bug abundance. At the milk stage, the ETLs were 2-6 bugs $panicle^{-1}$ for *N. viridula, Chlomchroa ligata,* and *Leptoglossus phyllopus,* and 3-8 for *O. pugnax.* ETLs for head caterpillars have been worked out for a range of production levels and costs of control for corn earworm in sorghum (Teetes and Wiseman 1979; Fuchs et al. 1993). For example, when the value of the crop is US$ 650 ha^{-1}, the ETL is 1 larva $panicle^{-1}$.

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Plant Resistance to Insects: Basic Principles

H C Sharma

Coevolution of Plants and Insects

Herbivorous insects and plants have coexisted for over 250 million years. Insects tend to select specific plants of a particular age to optimize nutritional intake, and secondary plant substances act as token stimuli for feeding and oviposition (van Emden 1978). Some of the secondary plant metabolites have a defensive function (Whittaker and Feeny 1971). The evolutionary processes that selected for the biosynthesis of specific secondary plant substances, and the counter adaptation by some phytophagous insects in using them as attractants or phagostimulants have been termed 'coevolution' (Ehrlich and Raven 1964). Close interrelationship between insect herbivores and their host plants have been defined as reciprocal (Fraenkel 1959) or sequential evolution (Jermy 1984). Various aspects of insect plant interrelations have been discussed by Fraenkel (1959), Beck (1965), van Emden (1978), and Sharma (1994).

Selection of Insect-Resistant Cultivars under Traditional Farming Systems

Cultivation of plant genotypes resistant to insects has been a principal method of insect control for a very long time. With the domestication of plants for agricultural purposes, farmers always selected the plants that withstood adverse environmental factors, including insects and diseases. The plants that were susceptible to pests generally died, and only resistant plants survived until crop harvest. This process led to the natural selection of plant varieties resistant to insect pests. Because of this unintentional but continuous selection of plants over several hundreds of years, many landraces selected by farmers evolved as having, or accumulating genes conferring resistance to insects. In sorghum, the best examples of this process are: shoot fly resistance in landraces cultivated during the postrainy season in India, sorghum midge resistance in genotypes originating from eastern Africa, and head bug resistance in guineense sorghums cultivated in western Africa.

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In spite of the importance of host-plant resistance (HPR) as a component of integrated pest management (1PM), breeding for plant resistance to insects has not been as rapidly accepted and developed as was the case in breeding disease-resistant cultivars. This was partly due to the relative ease with which insect control is achieved with the use of insecticides. Another lag for slow development of insect-resistant cultivars has been the difficulties involved in ensuring adequate insect pressure for resistance screening. Insect-rearing programs are expensive, the technology development may require several years, and may not produce the behavioral or metabolic equivalent of an insect population in nature.

However, with the development of insect resistance to insecticides, the adverse effects of insecticides on natural enemies, and public awareness of environmental contamination and conservation, there has been a renewed interest in the development of insect-resistant cultivars. The establishment of international agricultural research centers, and the collection and evaluation of existing germplasm for insect resistance, has given a renewed impetus to the identification and use of HPR in pest management worldwide. Such studies in crop improvement programs involve understanding the basic principles of HPR, establishing an insect-resistance breeding program, and its application in crop production and pest management.

Plant Resistance to Insects: Definitions

"Resistance of plants to insects enables a plant to avoid or inhibit host selection, inhibit oviposition and feeding, and reduce insect survival and development, tolerate, or recover from injury from insect populations that would cause greater damage to other plants of the same species under similar environmental conditions'' (Smith 1989). Resistance of plants to insects is the consequence of heritable plant characters that result in a plant being relatively less damaged than the plant without these characters. This property is generally derived from morphological and/or biochemical characters of the plants, which affect the behavior and biology of insects, and influences the relative degree of damage caused by the insects. From an evolutionary point of view, resistance traits are preadaptive and genetically inherited. Plants with such preadaptive genes withstand the selective pressure of herbivore populations, and thus increase their chances of survival and production. Plant resistance to insects is always relative, and the degree of resistance is based on comparison to susceptible plants that are more severely damaged under similar conditions. This is important, since expression of resistance is dependent on environmental factors both in time and space.

Pseudo-resistance or false resistance may occur in normally susceptible plants through avoidance of insect damage. Induced resistance may occur in plants because of variations in temperature, photoperiod, plant-water potential, and chemicals in the soil (e.g., potassium iodide, copper sulphate, and suboptimal doses of some herbicides) that induce the production and accumulation of secondary plant substances (phytoalexins) through increased activity of the phenylpropanoid metabolic pathway. Associate resistance occurs when susceptible plants grow in association with resistant plants, and derive protection from insect damage from resistant plants. Associate resistance indicates

that the diversion or delaying actions of mixtures of plant species can help in the slow development of an insect biotype that can overcome resistant cultivars. Various aspects of host-plant resistance to insects have been discussed by Painter (1951), Maxwell and Jenkins (1980), Smith (1989), and Kalode and Sharma (1993). Components of plant resistance to insects are antixenosis or nonpreference, antibiosis, and tolerance.

Antixenosis

Antixenosis is a Greek word, xenos meaning "guest". It describes the inability of a plant to serve as host to an insect herbivore. As a result, the insect is forced to change its host plant for feeding and oviposition. This term was proposed by Kogan and Ortman (1978) to replace the term nonpreference proposed earlier by Painter (1951). Antixenosis may be due to morphological or chemical plant factors that affect the insect behavior adversely, resulting in selection of an alternative host plant.

Insect sensory systems involved in host selection

Olfaction. This comprises chemical stimuli emanating from the host plant that affect the orientation of insects toward their host plants. These are perceived by sensilla basiconica. Chemicals that help in host identification/selection are called kairomones, and give adaptive advantage to the insects. Chemicals that repel the insects away from their host plants are called allomones, and these give adaptive advantage to the plants.

Vision. This involves the ability of insects to perceive spatial patterns using instinctive stimuli templates and the insects' ability to detect differences in color, e.g., brightness, hue, and the saturation of various wavelengths of light. Visual cues perceived by the insects during host selection are the result of the spectral quality of light, dimensions of the objects, and the pattern or shape of objects.

Thigmoreception. Contact stimuli are perceived by trichoid sensilla on the insect body, tarsi, head, and antenna. Such stimuli are received from leaf or stem trichome, epidermal ridges, leaf margins, and chemical stimuli.

Gustation. These stimuli are perceived by sensilla styloconica, maxillary palpi, and lateral gustatory receptors. Quantitative and qualitative differences in primary and secondary plant substances influence the gustatory processes that may in turn influence the host selection behavior of insects.

Plant factors affecting insect behavior

Triehomes. Dense growth of trichomes on the leaves affects the host selection by insects.

Surface waxes. Plant leaves are protected from pests by a layer of surface waxes over the epicuticle. When sense organs on the insect tarsi and mouthparts receive negative

chemical and tactile stimuli from the leaf surface, these stimuli play an important role in plant resistance to insects.

Tissue thickness. Foliar toughness adversely affects the host selection by several insect species. Resistance in sorghum to shoot fly is related to thickened cells that surround the vascular bundles of leaves (Blum 1968).

Antibiosis

Antibiosis includes the adverse effects of the physico-chemical characteristics of the plants on the biology of an insect attempting to use that plant as a host. Both chemical and morphological factors mediate antibiosis. Lethal effects may be acute, often affecting young larvae and eggs, and chronic effects lead to mortality of older larvae, pupae, and adults. Individuals surviving the direct effects of antibiosis may have reduced body size and weight, prolonged period of development, and reduced fecundity.

Plant defenses imparting antibiosis

Allelochemicals. Alkaloids, ketones, terpenoids, flavonoids, and organic acids produced by the plants are toxic to insects. Glycoalkaloids in potato, γ-tomatine in tomato, gossypol in cotton, and rutin and chlorogenic acid in tomato are toxic to insects.

Growth inhibitors. Insect growth inhibition due to presence of growth inhibitors or poor nutritional quality of the host plant are responsible for plant resistance in several crops. Maysin in maize silks, coumestrol in soybean, and terpenoids in pigment glands of cotton inhibit the growth of insects feeding on these crops. Imbalanced ascorbic acid content in maize plants, amino acid content in pea, lower quantities of glutamic acid and asparagine in rice, and low lysine content in sorghum impart resistance to some insects. The chronic effects of secondary plant substances affect the metabolism of insects that feed on resistant plants.

Morphological barriers. Hypersensitive growth responses of plants such as rapidly growing tissues of cotton bolls may kill bollworm larvae penetrating the bolls. Plant structures such as trichomes of many crop plants (e.g., in potato, tomato, soybean, cotton, etc.) kill the larvae and immature stages.

Tolerance

The ability of plants to withstand or recover from damage caused by insect abundance equivalent to that required to damage a susceptible cultivar is termed tolerance mechanism of resistance'. This expression of tolerance is determined by the inherent genetic capability to outgrow an insect infestation or to recover and add new plant growth after insect damage. From an agronomic perspective, the plants of a tolerant cultivar produce a greater yield than plants of a nontolerant susceptible cultivar; but tolerance often occurs in combination with antixenosis and antibiosis.

Quantitative measurements Of tolerance. Techniques used to measure tolerance to insects include increases in the size and growth-rates of leaves, stems, petioles, roots, and seeds or fruits. Seedling survival is a measure of tolerance in cereals. Production of tillers in plants damaged by shoot fly and stem borer, and increase in grain mass in midge-infested plants, are measures of tolerance in sorghum.

Factors affecting expression of tolerance. Environmental factors directly affect the expression of tolerance to insect damage in several crops. Temperatures affect the tolerance to greenbug in cereals. High levels of nutrients affect the tolerance by seedlings to greenbug (Schweissing and Wilde 1979). Fertility conditions and moisture availability affect tiller production in sorghum following damage by shoot fly and stem borers.

Techniques Used to Measure Plant Resistance

Manipulation of insect abundance

It is possible to manipulate insect abundance by field infestation, caging, artificial rearing and by evaluating insecticide-protected and unprotected plots.

Field infestation. Rarely is a researcher able to grow a group of plant genotypes and accurately evaluate insect damage. Without proper planning, either there will be insufficient insect numbers to cause adequate insect damage, or insects occur at an inappropriate phenological stage of crop growth. Field infestations are normally used to evaluate a large number of plant materials at early stages of the resistance evaluation program. Unmanaged insect populations may be too low or unevenly distributed to expose all test entries to a uniform level of insect density. Also, there are large differences in insect density over years and locations. Field evaluations are additionally influenced by nontarget insects which may interfere with plants damaged by the target insect. This makes it difficult to achieve dependable screening of plant material for resistance to insects in the field. Managed or augmented insect density ensures a uniform distribution of insects, but the insects are subjected to naturally occurring biological control agents and abiotic population regulation factors. To avoid interference by natural enemies, the crop can be treated with selective insecticides before the insect infestation takes place. Insects on infested plants can also be protected by cages from the natural enemies.

The objective of all these approaches should be to have an optimum insect density:damage ratio that allows the researcher to observe maximum differences among the resistant and susceptible plants. Several procedures can be employed to obtain adequate insect abundance for resistance screening. Hot-spots, where the insects are known to occur regularly in large numbers across seasons, can be used efficiently for a large-scale screening of the test material. Planting mixed or uniform susceptible plants as infester rows along the field borders, or at regular intervals in the field helps to increase insect abundance. The infester rows may be planted in advance so that the insect can have sufficient time to multiply on the infester rows. These rows can then be removed after

infestation of the test material has taken place. The crop can also be planted 2-3 times so that one of the plantings is exposed to adequate insect abundance, e.g., sorghum midge and head bugs. Such an approach also helps to reduce the chances of escape.

Pest abundance can be augmented by placing nondestructive light traps, pheromone traps, and kairomone traps. Indigenous insect populations can be collected from the surrounding areas and released in the test plots. In field-screening under natural infestation, known resistant and susceptible control plants should be grown at regular intervals in the screening nursery. When feeding insects are on reproductive parts of a plant, and if there are large differences in the flowering time of the genotypes, resistant and susceptible controls with different maturity should be included. The test material can also be tagged with different-colored labels or marked with paint. This will permit comparison of the test material with resistant and susceptible controls of similar duration.

Caging. Caging insects with test plants is one of the most dependable methods of screening for insect resistance. In this method, considerable control is exercised to maintain uniform insect pressure on the test entries, and to infest the test plants at the same phenological stage. This also prevents insects from migrating away from the test plants. The cage also keeps natural enemies away from the insects. Such tests can be carried out under greenhouse and field conditions. Small cages can be developed to cover the plant parts to be tested or whole plants can be put under a cage. Cage size and shape are determined by the type and number of test plants needed for evaluation. For valid conclusions, resistant and susceptible controls should also be included.

Supplementing natural abundance with artificially reared insects. Artificially reared insects can be made available throughout the year for screening tests. Artificial diets have been developed for several insect species (Singh and Moore 1985). In sorghum, the spotted stem borer can be reared on an artificial diet. But, if it is not possible to rear insects on an artificial diet, insect colonies can be maintained on natural hosts (shoot fly, head bugs, and midges) under greenhouse conditions.

Direct measurements of resistance

Direct-feeding injury. Measurements of insect damage to plants are often more useful than measurements of insect growth or development on plants. The plant damage and the resulting increase in yield or quality are the ultimate goals of most crop improvement programs. Often measurements of yield reduction indicate direct insect feeding injury to plants. Plant damage can also be determined by measuring the incidence of tissue necrosis, fruit abscission, and stem damage. Measurements of quality of produce can also be used to measure the effect of insect damage. Insect defoliation of plants is usually determined by rating scales that make use of visual estimates of plant damage based on percentages or numerical ratings. Several such rating scales have been developed to assess insect damage in crop plants. Direct measurements of leaf area are also used to measure insect damage. Indirect feeding injury measurements such as plant growth, photosynthetic rates, transpiration rates, ethylene production, and respiratory rates are also recorded. Feeding injury is measured as loss of yield under protected and

unprotected conditions. Different levels of insect infestations are created using different spray regimes. Less affected genotypes with low *b*-values (regression coefficient) are selected after comparison with high *b*-values of susceptible control plants.

Simulated feeding injury. Insect feeding injury can be simulated by mechanical defoliation. However, plants respond somewhat differently to artificial defoliation than to actual insect feeding. Therefore, relations between artificial and natural insect feeding should be determined before results on artificial defoliation are accepted. Insect injury is also measured by injection of toxic insect secretions into plant tissues, e.g., application of crude extract of greenbug in sorghum.

Correlation of plant factors with insect resistance. Chemical or mechanical resistance are measured by concentrations of allelochemicals or the density or size of morphological structures present in the tissues of resistant plants. This permits the rapid determination of potentially resistant plant material. This also removes the variations associated with insect density, and the effect of environmental influences on the expression of resistance to insects.

Indirect measurements of resistance

Sampling insect populations. Insect abundance can be estimated by sampling at the plant site where damage has taken place, and at the appropriate phenological plant stage and time. The population of immobile insects is measured visually, but this method is subjected to variations in colony size and pattern of insect distribution. Shaking the plants, use of sampling nets, use of traps, or actual counts are used to obtain an estimate of insect abundance.

Measurements of insect feeding and development. Insect development is monitored if antixenotic and antibiotic effects are exhibited by resistant plants. Several measures of consumption and use of food by the insects are used to determine the level of plant resistance to insects (Waldbauer 1968). Effect of plant resistance on insect feeding and development is measured in terms of amount of food consumed per unit body weight day^{-1} or leaf area consumed, duration of larval/pupal development, fecundity, and insect survival. Antibiosis effects are expressed in terms of weight and size of insects, sex ratio, and proportion of insects entering diapause.

Measurements of insect behavior. Several techniques for studying insect behavior are used to quantify the antixenosis mechanism of resistance. Responses of insects to volatile stimuli have been studied for several insects. Several designs of olfactometers have been used to observe insect behavior. Olfactory responses are also studied physiologically by electroantennograms, electroretinograms, and by electronic feeding monitors.

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Part 2

Screening for Resistance to Insects in Sorghum

Screening for Resistance to Sorghum Shoot Fly

K F Nwanze

Introduction

Screening of the world sorghum germplasm collection for resistance to sorghum shoot fly, *Atherigona soccata* began in India in 1962. This work was organized with the cooperative efforts of the Accelerated Hybrid Sorghum Project, Indian Agricultural Research Institute, and the Rockefeller Foundation. At ICRISAT-Patancheru, screening for resistance to sorghum shoot fly was initiated in 1974. The objective was to develop an effective, reliable, and repeatable resistance-screening technique, identify sources of resistance, and incorporate shoot fly resistance into high-yielding cultivars. Various techniques to screen for resistance to shoot fly have been described by Pradhan (1971), Jotwani (1978), and Taneja and Leuschner (1985).

Resistance-Screening Techniques

Interlard-fishmeal technique (field-screening)

Adequate shoot fly density for resistance screening can be achieved by manipulating the sowing date, using infester rows, and spreading fishmeal (which attracts the shoot flies) in the field (Fig. 1).

Shoot fly abundance can be monitored through fishmeal-baited traps to determine the periods of peak abundance of shoot fly. This information can be used for planting the test material during the susceptible stage of the crop's growth when it is exposed to optimum shoot fly pressure. Late-sown crops are subjected to high shoot fly abundance. At ICRISAT-Patancheru, sowing test material in mid-Jul in the rainy season, and during Oct in the postrainy season, is effective in screening for resistance to shoot fly.

The interlard-fishmeal technique, which is useful for increasing shoot fly abundance under field conditions, involves:

- Planting four rows of a susceptible cultivar (such as CSH 1, or CSH 5), sown 20 days before sowing the test material. These are referred to as interlards, or infester rows.

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Figure 1. The interlard fishmeal technique to screen for resistance to sorghum shoot fly under Held conditions.

- Moistening and spreading the fishmeal uniformly 1 week after seedling emergence, or keeping it in plastic bags in the interlards to attract shoot flies from the surrounding areas. One generation of the shoot fly is completed on the interlards, and the emerging flies infest the test material. The same procedure can also be adopted for the test material itself (Taneja and Leuschner 1985).

Cage-screening technique

To confirm resistance to shoot fly observed under field conditions, and to study resistance mechanisms, a cage-screening technique developed by Soto (1972) has been modified to simulate field conditions.

The modified technique requires no artificial rearing of shoot flies because they can be collected from fishmeal-baited traps in the field. After entering into the trap, the shoot flies move upwards into the collection jar due to positive phototactic behavior. The jar containing the shoot flies can be easily removed and emptied. To ensure a positive light gradient towards the collection jar, the container and the tunnel are made of colored (black or blue) plastic, whereas the collection jar is transparent. Shoot flies are collected in the morning and evening, and are separated from other species.

The cage-screening technique can be used for multiple- or no-choice tests. For a multiple-choice test, several genotypes are sown in the field in 3.4 x 2m beds, with a row spacing of 15 cm. Ten days after seedling emergence, the plants are covered with a 3.4 x 2 x 1 m screened cage. Then flies are introduced into the cage. Eggs and deadhearts are recorded after 1 week. For a no-choice test, only one genotype is sown in 1 x 1 m beds.

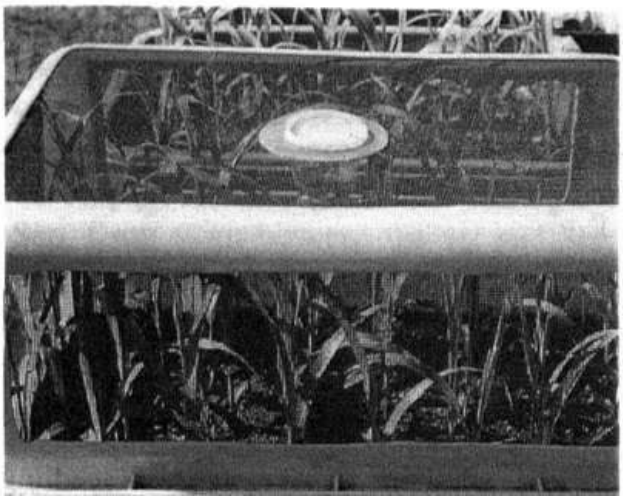

Figure 2. The cage technique to screen for resistance to sorghum shoot fly.

Six beds can be covered with a 2 x 3 x 0.5 m cage having six compartments. Ten days after seedling emergence, 20 flies are released into each compartment and observations are recorded as described above.

Rapid screening can also be carried out using a top-cage technique (Fig. 2). This system consists of two plastic trays (40 x 30 x 14 cm), one for sowing test material and the other (a top-cage fitted with fine wire-mesh) is clamped over the first tray, thus forming a cage. Ten days after seedling emergence in the plastic tray, the top-cage is assembled and 20 flies are released into each cage through an opening. Observations are recorded, as described above.

Damage evaluation for resistance screening

- Record the number of plants with eggs, plants with deadhearts, and the total number of plants at 14 and 21 days after seedling emergence.
- Record the number of tillers, and tillers with panicles at maturity as a measure of genotype's recovery resistance.
- Grain yield under protected and unprotected conditions can also be used as a measure of resistance to shoot fly.

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Screening for Resistance to Spotted Stem Borer

H C Sharma

Introduction

The spotted stem borer, *Chilo partellus* attacks sorghum 2 weeks after seedling emergence until crop harvest, and affects all plant parts except the roots. The first symptom of attack is leaf scarification and the presence of shot-holes caused by the early instar larval feeding in the leaf whorls. Infested plants show a ragged appearance. The older larvae leave the whorl and bore into the stem at the base. Stem boring by the larvae in young plants (up to 1 month old) damages the growing point and results in deadheart formation. In older seedlings, in which intermode elongation has started and the growing point has moved upwards, the larva feeds inside the stem, causing stem tunneling. Later infestations also result in peduncle tunneling and breakage. Both stem and peduncle damage sometimes lead to the production of completely- or partially-chaffy panicles.

Resistance-Screening Techniques

Techniques to screen for resistance to spotted stem borer have been described by several workers (Pradhan 1971; Jotwani 1978; Taneja and Leuschner 1985; Sharma et al. 1992). The following approach may be followed to screen for resistance to stem borer under natural and artificial infestation.

Screening under natural infestation

Hot-spots. Crop material may be tested at hot-spot locations where the pest populations are known to occur naturally and regularly at levels that often result in severe damage. Hot-spot locations for C. *partellus* are Hisar in Haryana, and Warangal in Andhra Pradesh, India; Agfoi and Baidoa in Somalia; Panmure and Mezarbani in Zimbabwe; Kiboko in Kenya; and Golden Valley in Zambia.

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Sowing date. To screen for resistance under natural infestation, especially at the hot-spot locations, adjust the sowing date of the crop such that the crop is at a susceptible stage when the stem borer abundance is at its peak. Determine the periods of maximum borer density through pheromone traps, light traps, or by monitoring borer infestation in the crop planted at regular intervals, e.g., at Hisar, C. *partellus* is most abundant in Aug-Sep. Crop sown between the 1st and 3rd week of Jul at Hisar suffers maximum stem borer damage. At ICRISAT-Patancheru, a maximum number of moths in the light traps has been recorded during Sep, followed by smaller peaks during Nov and Feb-Apr.

Mass rearing and artificial infestation

The efficiency of any resistance screening program depends on uniform and timely infestation of the test material. Artificial infestation with laboratory-reared insects has been successfully used for several pest species, including lepidopterous stem borers. Several diets have been used in the mass rearing of *C. partellus* (Dang et al. 1970; Siddiqui et al. 1977; Seshu Reddy and Davies 1979). An artificial diet to rear C. *partellus* has been developed at ICRISAT-Patancheru (Taneja and Leuschner 1985). Most of the ingredients of this diet (Table 1) are available in the local market. For preparing sorghum leaf powder, collect leaves from a susceptible cultivar (such as CSH 1)from 35-40-day-old plants. For inclusion in the artificial diet, wash, dry, and grind the leaves to a fine powder, and autoclave for 15 min at 120°C at 5 kg cm^{-2} pressure.

Table 1. Artificial diet used for mass rearing spotted stem borer, *Chilo partellus,* at ICRISAT-Patancheru, India.

Ingredient	Quantity[1]
Fraction A	
Water	2000 mL
Kabuli chickpea[2] flour	438.4 g
Brewer's yeast	32.0 g
Sorbic acid	4.0 g
Vitamin E (Viteolin capsules)	4.6 g
Methyl parahydroxy benzoate	6.4 g
Ascorbic acid	10.4 g
Sorghum leaf powder	160.0g
Fraction B	
Agar-agar	40.8 g
Water	1600 mL
Formaldehyde (40%)	3.2 mL

1. Amount used to prepare 15 jars of 300 g diet each.
2. A *Cicer arietinum* cultivar.

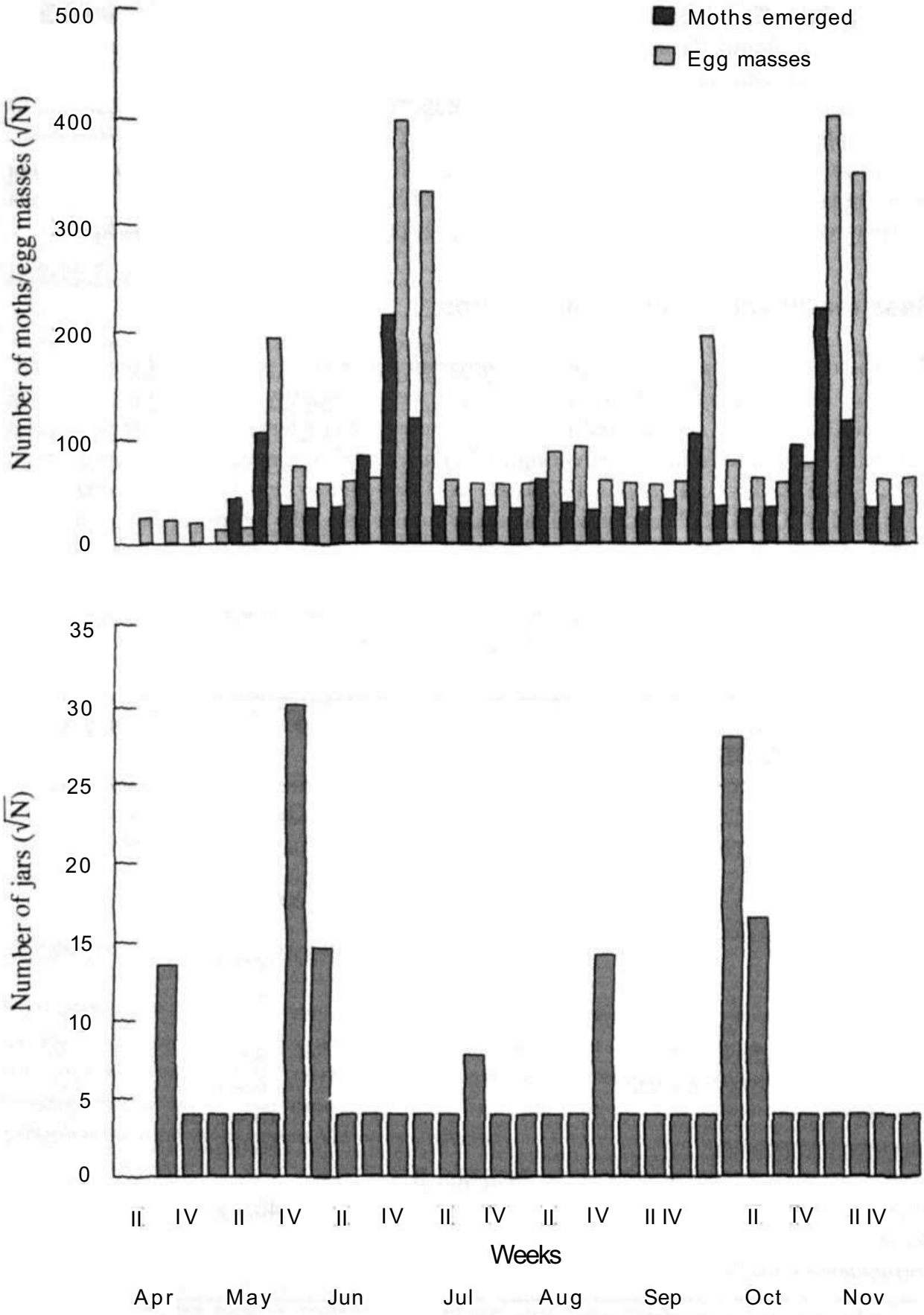

Figure 1. Rearing schedule to screen for resistance to spotted stem borer under artificial infestation.

Diet preparation

- Blend the ingredients of fraction A as per Table 1 (except the sorghum leaf powder) for 1 min.
- Soak the sorghum leaf powder in warm water (70°C) and blend with fraction A for 2 min.
- Boil agar-agar (fraction B) in 1.6 L of water, cool it to 40°C, combine with formaldehyde and fraction A, and blend for 3 min.
- Pour 300 g diet in a 1-L plastic jar.
- Allow the diet in the jar to cool to room temperature.
- Place about 100 eggs at the black-head stage in each jar, and keep the jars in a dark room for 2 days. This discourages the photopositive behavior of 1st-instar larvae, and they settle on the diet. The rearing room is maintained at 28±1°C, 60-70% relative humidity (RH), and 12 h photoperiod.

On artificial diet, the larval period lasts for 22-28 days and the pupal period for 5-6 days (Fig. 1). Moth emergence begins 30 days after larval inoculation, and continues up to the 40th day. Females emerge 2-3 days later than the males. The sex ratio is close to 1:1. Average moth emergence from this diet is 70-75%, with a maximum of up to 90%. Most of the moths emerge in 30-40 days after larval inoculation.

Moth collection. Collect the moths with the help of aspirators attached to a vacuum cleaner (a bifurcated tube is attached to the vacuum cleaner, which terminates in the collection bottles, or aspirator), or with the help of hand-held aspirators. Collect the male and female moths separately (males are smaller in size with dark forewings and pointed abdomen), and transfer them to the egg-laying cages.

Oviposition. The oviposition cage consists of an open cylinder (25 cm high and 25 cm in diameter) made of galvanized iron wire net with 36-mm openings (Fig. 2). A fine georgette cloth with 6 x 6 mm holes at regular intervals is fitted around the outer side of the cylinder, which is wrapped with a sheet of white glycine paper (25 x 80 cm) to serve as an oviposition site. Two plastic saucers covered with mosquito net are placed at the ends of the cylinder.

Release 50 pairs of moths in each oviposition cage. An average of 10-12 egg masses (500-600 eggs) are laid per female over a period of 4 days. Most eggs are laid on the 2nd and 3rd day after emergence. The eggs are laid in batches on the glycine paper through the holes in the wire cage. Replace the glycine paper daily. Feed the moths with water using a cotton swab.

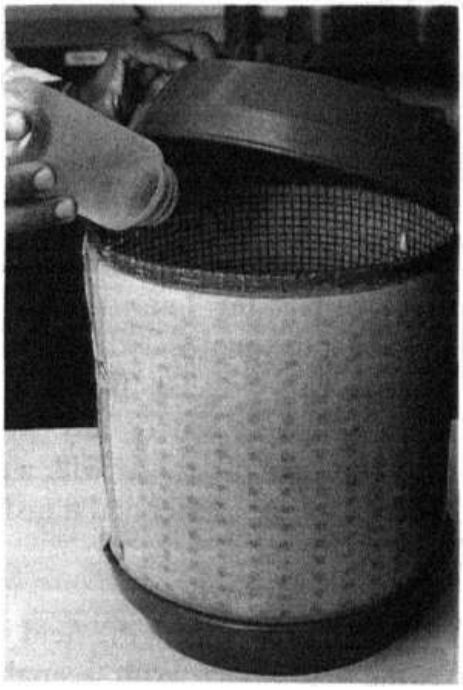

Figure 2. An oviposition cage for spotted stem borer.

Egg Storage. High humidity (80-90%) is needed for normal embryonic development. Hatching is drastically reduced when relative humidity falls below

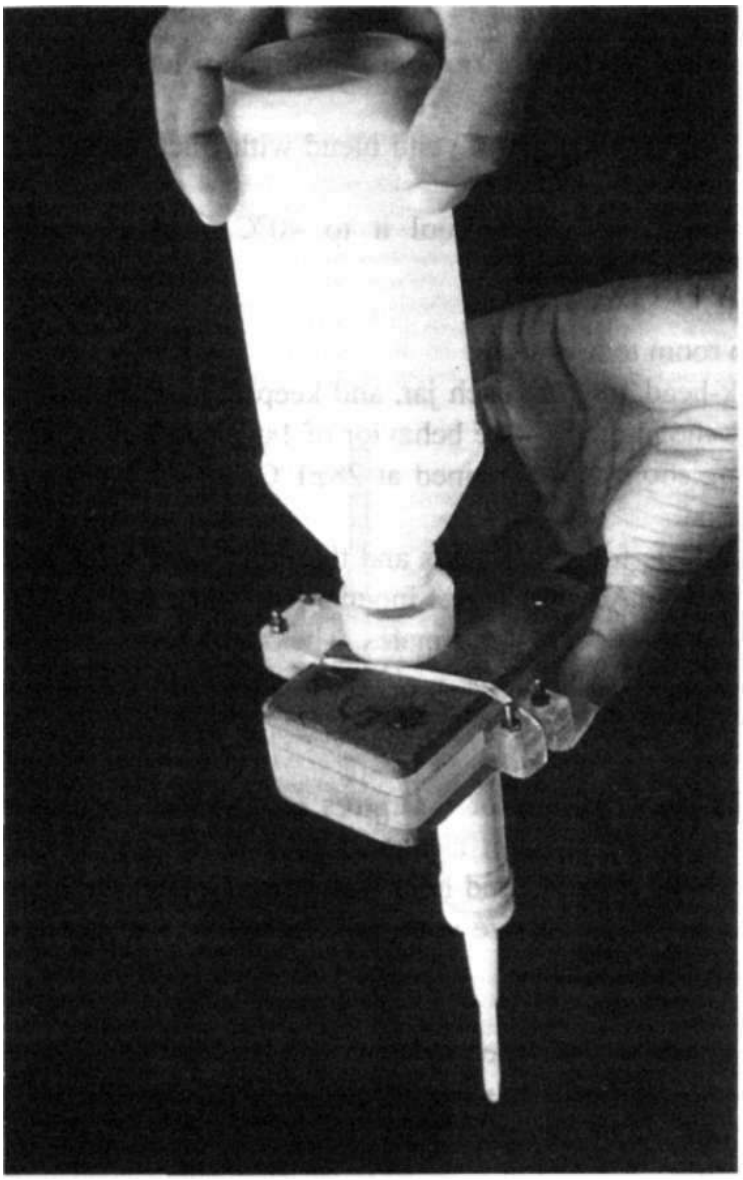

Figure 3. Bazooka applicator used to infest sorghum plants.

50%. To obtain high humidity, hang the glycine papers containing egg masses on a rod in a plastic bucket containing water. Cover the plastic bucket with a lid. Store the eggs at 26±1°C. Under these conditions, the embryo matures to the black-head stage within 4 days. For long-term storage, keep black-head stage eggs at 10°C. This delays egg hatching up to 10 days.

Rearing schedule. Efficient planning is required to produce sufficient numbers of insects to infest the test material at the proper growth stage. At ICRISAT-Patancheru, screening for stem borer resistance is carried out during the rainy and the postrainy seasons. The rainy season sowing is done in mid-Jun and the postrainy season sowing at the end of Sep. A schedule for diet preparation, crop sowing, and infestation is given in Figure 1. This schedule may be adapted in different locations with modifications as necessary.

Preparation Of a bazooka applicator. For field infestation, this applicator, developed at the International Maize and Wheat Improvement Center (CIMMYT) in 1976 to infest maize with corn earworm (CIMMYT 1977), was.modified to suit sorghum requirements (Fig. 3). Take 500 black-head stage egg masses, along with 85 g of poppy seeds *(Papaver* sp) or any small and light seed, or corn cob grits, and keep them overnight in a plastic jar with a tightly fitted lid. In the morning, mix the 1st-instar larvae gently with the carrier and transfer them into the plastic bottle of the bazooka.

Field infestation

- Infest the plants in the field individually by placing the nozzle of the bazooka close to the leaf whorl. With a single stroke, 5-7 larvae are released into each plant whorl. Generally 5-7 larvae plant^{-1} are sufficient to cause appreciable leaf feeding and deadhearts (>90% damage in susceptible genotypes).

- Infest 15-20-day-old plants. Deadheart formation decreases progressively as the infestation is delayed.
- For stem and peduncle tunneling, plants may be infested at a later stage (35-45 days after emergence).
- Infest the crop in the morning between 0800 and 1100 to avoid larval mortality due to high temperature. However, on cloudy days, infestations can be carried out at any time of the day.
- Rotate the bazooka applicator after every 10 strokes to ensure uniformity in larval distribution.
- There is often an accumulation of water in the plant whorl. To avoid drowning the larvae, tap the whorl gently before infestation. The number of larvae per plant can be regulated by varying the number of egg masses mixed with the carrier in each bazooka. A second infestation may be required if it rains immediately after first infestation.

Control Of Shoot fly. Shoot fly infestation interferes with the screening for resistance to stem borer. A selective insecticide can be used to control shoot fly without leaving any residual effect on stem borer establishment. Spray fenvalerate or endosulfan to suppress shoot fly infestation 1 week before artificial infestation with stem borer. Cypermethrin (a synthetic pyrethroid) applied through an Electrodyne sprayer 1 week before the borer infestation effectively controls the shoot fly without any detrimental effect on borer establishment. Also, it is helpful to sow the test material early in the season when shoot fly infestation is negligible.

Damage evaluation for resistance screening

Stem borer attack in sorghum causes leaf damage, deadheart formation, stem and peduncle tunneling, and production of chaffy panicles. These symptoms are not necessarily related to yield loss. Leaf injury is the first larval feeding symptom, found to be related to yield loss only under severe infestation. Stem tunneling adversely affects the quantity and quality of fodder, but is not correlated with reduction in grain yield. Peduncle damage could be critical if there are winds of high enough velocity to break the peduncle. Deadheart formation causes the most critical damage. This parameter is the most important criterion for differentiating degrees of resistance. The second criterion is the production of chaffy panicles. The following observations may be recorded for damage evaluation.

Leaf feeding. Record the rate of leaf feeding 1 week after artificial infestation, and 3 and 6 weeks after crop emergence under natural infestation. Record the total number of plants, the number of plants showing the leaf-feeding symptoms, and the leaf-feeding score evaluated on a 1-9 scale (based on plants showing leaf-feeding symptoms: see Fig. 4 and Table 2). Calculate the leaf-feeding index by multiplying the percentage of plants showing leaf-feeding symptoms with the leaf-feeding score.

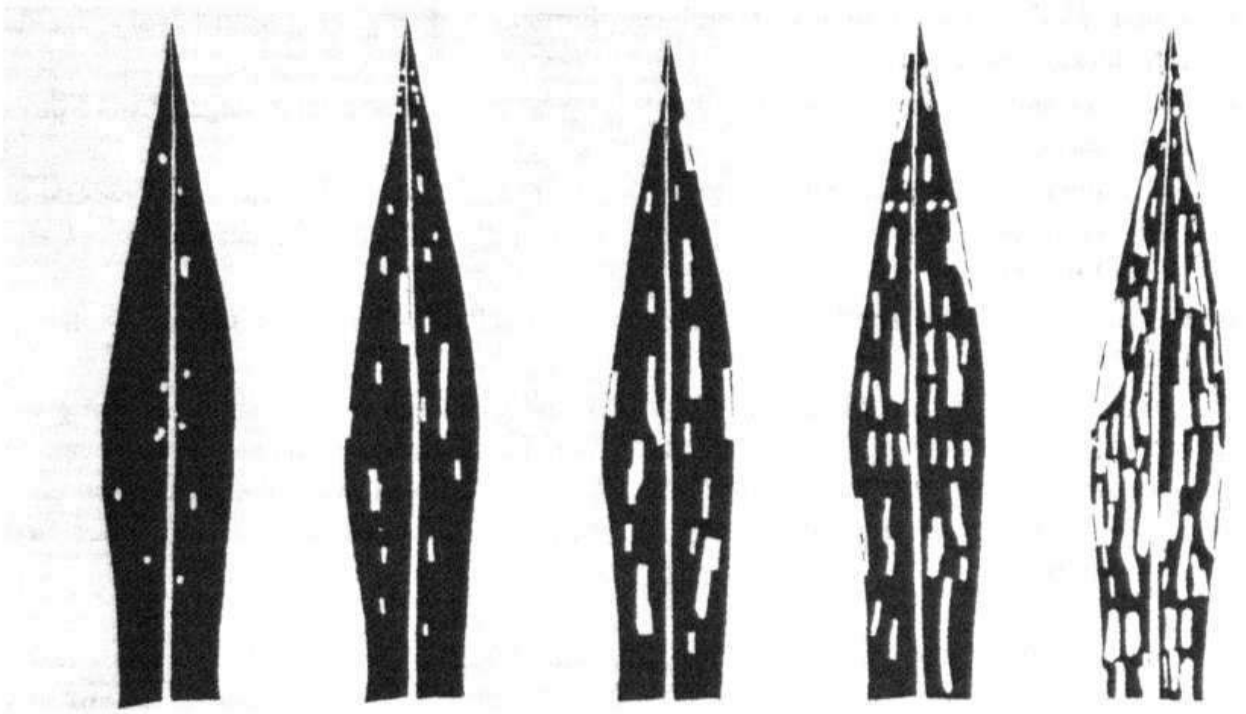

Figure 4. Leaf damage rating scale for spotted stem borer infestation.

Table 2. Visual damage rating scale for leaf feeding, deadheart formation, and chaffy and broken panicles caused by *C. partellus* infestation.

Score	No. of leaves with feeding symptoms	Leaf area eaten (mm^2)	Deadhearts/chaffy/broken panicles (%)
1	1-2	<150	<10
2	1-2	150-300	10-20
3	2-3	300-450	21-30
4	2-3	450-600	31-40
5	3-4	600-750	41-50
6	3-4	750-900	51-60
7	4-5	900-1050	61-70
8	4-5	1050-1200	71-80
9	5-6	>1200	>80

Deadhearts. Record deadhearts 3 weeks after artificial infestation, and 4 and 6 weeks after crop emergence under natural infestation. Record the total number of plants, plants showing borer deadhearts, and the visual score (1-9) for deadhearts.

Chaffy panicles. At crop harvest, record observations on the number of partial and complete chaffy panicles, the number of broken panicles, and the visual score (1-9) for chaffy/broken panicles and grain mass.

Recovery resistance. Record the number of plants with tillers and the number of tillers with productive panicles. Evaluate for recovery resistance, as explained under mechanisms of resistance.

Stem tunneling. At maturity, record plant height and the peduncle length of five plants at random in each plot. Measure the stem and peduncle tunneling separately and express it as a percentage of stem/peduncle tunneling.

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Effect of Genotypic Resistance on Avoidable Losses and Economic Thresholds for the Spotted Stem Borer

S P Singh

Introduction

Spotted stem borer, *Chilo partellus* is a key pest of sorghum in Asia and southern and eastern Africa. Stem borer damage affects both grain yield and fodder quality. It damages sorghum from 15 days after seedling emergence until crop harvest. It affects all plant parts, except the roots. The initial symptoms of borer infestation are leaf scarification, caused by the early instar larvae feeding in the plant whorl. The older larvae leave the leaf whorl and bore into the stem at the base, and damage the growing point. Damage to the growing point results in deadheart formation. The larva also feeds inside the stem, causing extensive stem tunneling. It also tunnels into the peduncle up to the rachis.

Assessment of crop losses caused by stem borer damage on different genotypes is essential for determining the status of the pest and the economic threshold levels, and for determining appropriate strategies for keeping the borer populations below threshold levels. Only limited information is available on quantitative and qualitative losses caused by stem borers in sorghum.

Effect of Genotypic Resistance on Avoidable Losses

Quantitative losses

Avoidable losses in sorghum caused by spotted stem borers have been estimated by using different levels of protection against the borer damage at different growth stages (by application of carbofuran 3G in the leaf whorls) under natural infestation during the 1993-1994 rainy seasons at Haryana Agricultural University, Hisar, India. Two sorghum cultivars (CSH 1 grain type and HC 260 fodder type) were sown during the second half of Jul in a randomized complete block design. There were three replications. The plot comprised six rows, each 3 m long and 45 cm apart. Carbofuran granules (2 g m^{-1} row) were applied at 15,

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Singh, S.P. 1997. Effect of genotypic resistance on avoidable losses and economic thresholds for the spotted stem borer. Pages 46-51 *in* Plant resistance to insects in sorghum (Sharma, H.C., Faujdar Singh, and Nwanze, K.F., eds.). Patancheru 502 324, Andhra Pradesh, India: International Crops Research Institute for the Semi-Arid Tropics.

Table 1. Effect of five protection levels and two sorghum genotypes on spotted stem borer infestation, green fodder and grain yield, and avoidable losses.[1]

Treatment	CSH 1					HC 260				
	Dead-hearts (%)	Green fodder yield (kg 9 m^{-2})	Avoidable losses (%)	Grain yield (g $plant^{-1}$)	Avoidable losses (%)	Dead-hearts (%)	Green fodder yield (kg 9 m^{-2})	Avoidable losses (%)	Grain yield (g $plant^{-1}$)	Avoidable losses (%)
T_1- Carbofuran 3G 15, 25, 35 & 45 DAE	8.0	20.0	0.0	0.22	0.0	12.5	40.0	0.0	0.17	0.0
T_2- Carbofuran 3G 15, 25, & 35 DAE	11.2	18.5	7.5	0.20	9.1	15.0	38.5	3.7	0.15	11.8
T_3- Carbofuran 3G 15 & 25 DAE	16.6	15.5	22.5	0.18	18.2	19.0	33.5	16.2	0.13	23.5
T_4- Carbofuran 3G 15 DAE	25.3	13.0	35.0	0.12	45.4	24.0	30.0	25.0	0.10	41.2
T_5- Control (unprotected)	65.5	10.5	47.5	0.00	100.0	42.5	25.0	37.5	0.06	65.9
LSD at 5%	4.2	2.0	-	0.025	-	3.5	2.8	-	0.021	-

1. DAE = days after emergence.

25, 35, and 45 days after seedling emergence (DAE) in different combinations to obtain variable protection levels against the spotted stem borer, as indicated in Table 1. The total number of plants and plants showing deadhearts were recorded in the central four rows of each plot at 50 DAE. Green fodder yield was recorded at 50% flowering. Grain yield was recorded at harvest.

Differences in deadhearts, fodder, and grain yield in CSH 1 and H 260 in protected and unprotected plots were significant (Table 1). Deadhearts due to *C. pattellas* damage in CSH 1 and HC 260 in plots with different protection levels varied from 8.0 to 65.5, and 12.5 to 42.5%, respectively. Fodder and grain yield of both cultivars was highest when the crop was protected at 15, 25, 35, and 45 DAE. With CSH 1 there were no productive panicles under unprotected conditions.

Fodder yield per plot in CSH 1 and HC 260 under different levels of protection ranged from 10.5 to 20.0, and 25..0 to 40.0 kg $plot^{-1}$, respectively. Thus, there were significant differences in loss in yield caused by borer damage in the two genotypes tested. Avoidable losses in fodder yield varied from 0.0 to 47.5% in CSH 1, and from 0.0 to 37.5% in HC 260. Similarly, grain yield per plot in CSH 1 and HC 260 ranged from 0.0 to 0.22 and 0.058 to 0.17 kg, respectively. Avoidable losses in grain yield were 0.0-100.0% in CSH 1, and 0.0-65.0% in HC 260. The results indicated that avoidable losses are greater in grain yield than in fodder yield. Similar results have been reported by Taneja and Nwanze (1989). They reported maximum loss in grain yield in plots infested at 15-30 days DAE.

Qualitative losses

To estimate the quality loss in forage sorghum caused by stem borer damage, two cultivars—HC 171 (sweet) and HC 260 (nonsweet)—were raised as described above. These cultivars were artificially infested by releasing 10 1st-instar larvae of *C.partellus* in each plant whorl at 15 DAE. Different levels of stem borer infestation were maintained by protecting the crop with sprays of endosulfan 0.07% at different growth stages. The stem borer intensity was recorded on randomly selected plants at 80 DAE by splitting the stalks. The total as well as the tunneled stem length of each stalk was measured. Data were converted to percentages of stem length tunneled, and categorized on a 1-9 scale (Singh 1986). Dry matter yield from each borer infestation category was recorded, and percentage loss in yield due to borer infestation was calculated.

The plant samples from each borer infestation category described above were analyzed for structural carbohydrates, namely neutral detergent fiber (NDF), acid detergent fiber (ADF), cellulose, hemicellulose, lignin, silica, and protein content (Goering and Van Soest 1970). The in vitro dry matter digestibility (IVDMD) was estimated according to Barnes et al. (1971). All the estimates were made in duplicate, and the amounts were expressed as g kg^{-1} dry matter.

There was a decrease in the dry matter yield, protein content, IVDMD, and silica content, in both sweet (HC 171) and nonsweet (HC 260) cultivars of forage sorghum, with an increase in borer infestation. The IVDMD, which takes into account all known and unknown factors affected by borer damage, decreased significantly (by about 13-16%) at 50% borer infestation. The observed decrease in IVDMD is mainly due to corresponding increase in

Table 2. Effect of spotted stem borer infestation on dry fodder yield (g plant^{-1}) and biochemical constituents (g kg DM^{-1}) of two forage sorghum cultivars.[1]

Stem borer infestation	Dry fodder yield	NDF	ADF	Hemi-cellulose	Cellulose	Lignin	Silica	Protein	IVDMD
HC 171 (Sweet)									
0 (Control)	105	680	404	276	327	380	390	700	538
10	88(-16.2)	693(+1.9)	413(+2.2)	280(+1.5)	339(+3.7)	390(+2.6)	350(-10.2)	678(-10.2)	528(-1.9)
20	80(-23.8)	708(+4.2)	418(+3.5)	290(+5.1)	342(+4.4)	450(+18.4)	310(-20.5)	656(-6.3)	508(-5.6)
30	66(-37.1)	724(+6.5)	431(+6.7)	292(+6.1)	352(+7.6)	500(+31.6)	290(-25.6)	612(-12.6)	486(-9.7)
40	56(-46.6)	725(+6.6)	437(+8.2)	288(+4.3)	353(+7.9)	570(+50.0)	270(-30.8)	612(-12.6)	464(-13.7)
50	52(-50.4)	727(+6.9)	445(+10.1)	282(+2.2)	364(+11.3)	600(+57.9)	216(-44.6)	612(-12.6)	456(-15.2)
>50	48(-54.2)	734(+7.9)	462(+14.3)	293(+6.2)	378(+15.6)	630(+65.8)	210(-46.1)	606(-13.4)	450(-16.2)
LSD at 5%	5.2	0.6	3.0	N.S.	2.8	8.1	4.2	1.6	3.6
HC 260 (Nonsweet)									
0 (Control)	95	599	337	262	275	330	290	656	542
10	77(-18.9)	612(+2.2)	358(+6.2)	264(+0.8)	296(+7.6)	350(+6.1)	270(-6.8)	612(-6.7)	538(-0.7)
20	68(-28.4)	630(+5.2)	362(+7.4)	268(+2.2)	305(+10.9)	370(+12.1)	200(-31.0)	569(-13.3)	532(-1.8)
30	59(-35.9)	643(+7.3)	368(+9.2)	275(+4.9)	310(+12.7)	390(+18.2)	190(-34.5)	569(-13.3)	520(-4.1)
40	48(-49.4)	660(+10.2)	375(+11.3)	283(+8.3)	313(+13.8)	440(+33.3)	180(+37.9)	525(-19.9)	500(-7.7)
50	42(-55.7)	671(+12.0)	388(+15.1)	285(+8.7)	386(+18.5)	450(+36.4)	170(-41.4)	525(-19.9)	488(-9.9)
>50	40(-57.9)	685(+14.4)	396(+17.5)	289(+10.3)	333(+21.1)	460(+39.4)	170(-41.4)	517(-21.2)	476(-13.2)
LSD at 5%	5.8	2.4	2.5	2.0	3.0	4.5	5.2	2.2	2.5

1. Figures in parentheses indicate % increase (+), or decrease (-) over the noninfested control plants.

fiber (NDF and ADF) and lignin contents in both cultivars. The decrease in protein content in infested plants could either be due to use of proteins by the insect, reduction in protein synthesis, or increased activity of proteolytic enzymes,

Hemicellulose and cellulose, which form the bulk of cell wall components of plants, are an important source of energy for the ruminants. The levels of these constituents in general were greater in stem borer infested plants in comparison with noninfested plants (Table 2). The NDF, which is positively correlated with ADF, increased with an increase in borer infestation. Arora et al. (1987) also observed a similar increase in NDF and ADF contents in mite-infested sorghum plants. Inerease in lignin because of spotted stem borer infestation may be the main cause for reduction in IVDMD, as observed by Arora et al. (1975). Thus, it is obvious that spotted stem borer damage results in both qualitative and quantitative losses in sorghum. There were large differences between the genotypes in parameters used to measure the quality loss in fodder caused by borer damage.

Economic Thresholds

Economic threshold levels (ETLs) for spotted stem borer on sorghum have not been computed. ETLs vary over seasons and locations and with prevailing socioeconomic conditions, and are influenced by variations in the cost of inputs, the value of the produce, and the productivity potential of the crop. Mote (1986) studied the relation between stem tunneling and reduction in grain yield. He observed a reduction of 1.0057 g plant^{-1} with an unit increase in stem tunneling for sorghum hybrid CSH 8R. Similarly, Kishore (1990) reported that a unit increase in stem tunneling and leaf injury resulted in a decrease of 0.59 and 0.002 units of grain yield, respectively. ETLs for spotted stem borer have been estimated to be 5-25% deadhearts at 20 DAE under different levels of protection. Preliminary studies conducted at Hisar indicated the ETLs for borer to be 10% deadhearts at 20 DAE.

A prerequisite for formulating any pest management program is determination of ETLs for different genotypes. Various aspects of insect/host-plant interactions for stem borers need to be investigated thoroughly in relation to host-plant resistance to this pest.

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Screening for Resistance to Sorghum Shoot Bug and Spider Mites

D u Singh

Introduction

Sorghum is cultivated under diverse agroecosystems, and its production is influenced by various abiotic factors such as extended periods of high temperatures, low humidity, and low and erratic rainfall, and these factors result in drought stress (Garrity et al. 1982; Rosenow et al. 1983; Holtzer et al. 1988). In addition, the stability of sorghum production is threatened by several insect pests. The corn planthopper, popularly known as shoot bug in India, and spider mites assume greater importance under drought stress because of increased insect establishment and rapid population build-up, thus causing considerable loss in grain and forage yields. Continuous cropping, reduced genetic variability in high-yielding varieties and hybrids, changes in cultural practices as well as reduction in the natural enemy complex, all lead to the increased severity of these pests.

Shoot Bug (*Peregrinus maidis*)

The shoot bug or corn planthopper is cosmopolitan in distribution. Its outbreaks have become frequent in many sorghum-growing states in India in the postrainy and rainy seasons. Adults and nymphs usually congregate in groups in the leaf whorls, inner leaf sheaths, panicles, and exposed roots (Chelliah and Basheer 1965), and are often found in association with ants. They suck the plant sap, resulting in reduced plant vigor, stunting, and yellowing of leaves. Severe infestations, combined with excessive oviposition in the midribs, result in gradual withering of leaves downwards from the top, or girdling by twisting of top leaves and inhibition of panicle formation (Singh and Rana 1992). However, infestation at later stages prevents either normal panicle exertion (Agarwal et al. 1978), or poor development of panicles (Rawat and Saxena 1967). It is also a vector of several viruses: maize mosaic, maize stripe, freckled yellow, and male-sterile stunt.

In general, it has been observed that two peaks of macropterous (winged) adults coincide with migratory periods at the beginning and at the end of the crop season. Brachypterous (wingless) adults appear from the 6th week onwards, with a slow growth

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in population, and decrease towards the end of the crop season. Nymphs appear by the 5th week after seedling emergence, reaching peak abundance between 8-10 weeks after plant emergence, and decline thereafter (Fernandez-Badillo and Clavijo 1989). High nymphal population determines the development of macropterous adults, and low populations result in brachypterous forms (Fernandez-Badillo and Clavijo 1990a,b).

The females prefer to lay eggs on the upper surface of the midrib of older and mature leaves at 30 days after emergence (DAE). The females of macropterous and brachypterous forms lay 18-98 and 5-64 eggs, respectively (Rawat and Saxena 1967), which hatch in 5-8 days. The nymphal stage comprises 5 instars, and development is completed in 2 days. The total life cycle ranges from 18 to 31 days, with an average of 24.5 days (Chelliah and Basheer 1965); 14-53 and 19-71 days for macropterous males and females; and 17-41 and 22-62 days for brachypterous males and females, respectively (Rawat and Saxena 1967).

Resistance-screening techniques

Techniques to screen for resistance to corn planthopper have been described by Chandra Shekar (1991) and Chandra Shekar et al. (1993a,b). The following approach can be adopted to screen for resistance to corn planthopper under field and laboratory conditions.

- Selection of hot-spot locations and adjustment of planting time so that the most susceptible stage of crop growth coincides with peak population density of the shoot bug.
- Mass-rearing of the insect on a susceptible cultivar, e.g., CSH 1, for laboratory testing and field infestation.
- Plant interlards of a susceptible cultivar (CSH 1) for screening under field conditions.
- Simulation of drought stress.

Rearing of insects, for mass multiplication and for use in laboratory and field experiments, can be carried out on the susceptible cultivar grown in pots in the greenhouse, by confining the plants with gravid females (macropterous or brachypterous) in a mylar tube.

Laboratory conditions. Plant samples of test genotypes grown in pot culture can be used at susceptible growth stages (30, 45, and 60 DAE). Excise the plant at the base and keep it in a conical flask (100 mL) filled with water, place the plants in a circular pattern in a cage at random from the center of a circular plastic trough (30-cm diameter). Release approximately 3 000 macropterous or brachypterous adult females; or 2nd-instar nymphs can be released into the plastic trough to allow free access to all the test genotypes. The number of shoot bugs are then recorded on each plant at frequent intervals (2, 6, 24, and 48 h after release) to determine the role of visual, olfactory, and gustatory stimuli in host-plant selection. Similarly, the extent of oviposition can be recorded by splitting the midribs and counting under a binocular microscope (x 50) the number of eggs laid.

Field conditions. To record insect numbers/extent of oviposition, select five plants in each replication in each genotype randomly at three growth stages (30, 45, and 60 DAE). Enclose the samples with a polythene envelope and cut the plants at the base to prevent the escape of insects during sampling. A cotton swab soaked in chloroform can be used to

immobilize the insects for counting. In addition, remove the leaves from each sample carefully, split the midribs, and count the number of eggs under a binocular microscope (x 50). Plant damage symptoms can be recorded at three crop growth stages (Chandra Shekar 1991):

45 DAE = Yellowing of leaves with stunted growth.

60 DAE = Girdling of topmost leaves without panicle development.

75 DAE = Poor panicle exertion or/development of panicle.

The extent of plant damage can be expressed as follows:

$$\text{Plant damage (\%)} = \frac{\text{Damaged plants}}{\text{Total plants}} \times 100.$$

Sources of resistance

Sources of resistance to corn planthopper have been identified. These include: Kafir Suma, Dwarf Hegari, I 753, H 109, GIB, 3677B, BP 53 (Agarwal et al. 1978), MSH 65, SPH 388, SPV nos. 475, 678, 736, 741, 756, 775, 819, 858, and CSV 10 (Rajasekhar 1989), IS 18657, IS 18677, and PJ 8K(Y) (Singh and Rana 1992), and IS 18676 and IS 19349 (Chandra Shekar et al. 1993a,b).

Mechanisms of resistance

Among the mechanisms of resistance, predominance of antixenosis for adult/nymphal colonization and oviposition (Singh and Rana 1992; Chandra Shekar et al. 1993a) and tolerance have been reported (B U Singh, unpublished).

Spider Mites (*Oligonychus indicus* and *O. pratensis*)

The spider mites are important pests of sorghum. They are usually confined along the midrib on the undersurface of the basal functional leaves. The infested area of the leaves becomes pale yellow initially, and later turns reddish or tan on the dorsal surface. As the mite population increases on the basal leaves, dense webbing is seen on the undersurface of the infested leaves. Mite densities are positively correlated with leaf area and plant maturity. Mite feeding damage is significantly lower on the late-maturing lines, demonstrating that sorghum susceptibility to mite feeding increases as the plants reach anthesis and caryopsis (Archer et al. 1986a,b).

Rahman and Sapra (1940) and Rai et al. (1989) reported that the developmental periods for egg incubation, larvae, and proto- and deuto-nymphs was 4.7, 2.2, 1.2, and 1.6 days, respectively, for females; and 1.94, 0.91, and 1.4 days for larvae, proto- and deuto-nymphs, respectively, for males. In general, the males of *O. pratensis* develop faster than the females (Tan and Ward 1977). The females reared on infested leaves produce significantly more female progeny than on uninfested leaves, suggesting that the offspring sex ratios change in response to deteriorating food sources (Stiefel et al. 1992).

On the other hand, rearing of *O. pratensis* on intact sorghum leaves at growth stages 1 to 6 have a longer life span (25.7 days) and ovipositional period (11.6 days) than on leaf disks at growth stages 6 to 8 (19.1 and 8.9 days, respectively) (Foster et al. 1977a) [Definition of plant growth stages are based on Vanderlip (1972): 1 = three leaves with fully developed sheath; 2 = five fully developed leaf sheaths; 3 =growing point differentiation; 4 = final leaf visible in whorl; 5 = boot stage; 6 = half-bloom; 7 = soft-dough; 8 = hard-dough; and 9 = physiological maturity]. However, fewer eggs (4.3) were oviposited daily by females at growth stages 1 to 6 than at 6 to 8 (Foster et al. 1977a).

Resistance-screening techniques

Techniques to screen for resistance to spider mites have been described by Dabrowski (1972), Foster et al. (1977a,b) and Sreedhar (1995). Screening under field conditions is relatively difficult because many factors affect the mite population abundance and the plant's reaction to infestation (Owens et al. 1976). The following methodology may be followed for screening under field and laboratory conditions.

- Selection of hot-spot locations and manipulation of sowing time.
- Mass-rearing of mites on a susceptible cultivar e.g., CSH 1.
- Planting border rows and interlards of a susceptible control (CSH 1).
- Simulation of drought stress.

Laboratory conditions. Screening for mite resistance under laboratory conditions can be carried out under free-choice (FC) and no-choice (NC) conditions using leaf disks and intact leaves (IL). In the FC test, the leaf disks (1.5 cm diameter) are taken from the mid-section of the larger middle leaf and arranged equidistantly in a circle on a moistened filter paper at random. A moistened sponge sheet is kept in a round plastic trough (30-cm diameter) leaving a central space. Approximately 5 000 spider mite adult females are released from the infested leaves into a petri dish (3.5-cm diameter) kept in the center of the leaf disks. The preferential response is based on orientation and colonization due to olfactory, visual, and gustatory stimuli. Similarly, oviposition can be measured by egg counts recorded at periodic intervals of 24, 48, and 72 h after adult female release.

In the IL technique, test samples of the plant are cut from the base and kept in a conical flask (100 mL), arranged randomly 15 cm apart in a cage encircling a plastic trough (30-cm diameter), and infested dry susceptible sorghum plants containing adults are kept in an empty glass jar (7.5 x 15 cm).

In the NC test, 10 gravid females are confined on leaf disks or intact leaves with tangle foot (a sticky material). The levels of preferential response is estimated by recording the mite numbers, or oviposition at 24, 48, and 72 h after infestation. In the NC test, antixenosis for feeding can also be measured.

Field conditions. Mite movement mostly depends on the direction of wind or migration, so there is considerable variation in mite abundance in research plots. Mite infestations increase rapidly and are most damaging to the crop at the reproductive stage associated with hot and dry climatic conditions. Thus, artificial infestation is the most

effective way to obtain dependable and uniform infestation. Field-collected miles can be used to infest research plots when the plants are in their late-vegetative growth stage and therefore most vulnerable to mites. Mite-infested leaves may also be collected from plants. A single sorghum leaf infested with mites can be placed across plants within a row. Mites spread rapidly between the plants (Archer 1989).

Damage evaluation

Leaf damage rating/mite counts. Leaf damage ratings estimate the total leaf area showing chlorotic stippling or death caused by mite feeding. Death of an entire leaf from mite feeding usually does not occur until the whole plant damage approaches 50%. The condition of the plant also influences the extent of damage:

- Moderately drought-stressed plants are better mite hosts than irrigated ones.
- Plant maturity may help plants escape maximum mite pressure.
- The leaf area available to the mites can influence how many mites are required to cause a given amount of damage.

Mite counts are taken at weekly intervals on 10 randomly selected plants per genotype following the procedure used by Jeppson (1951). In this system, only the number of adult female mites are recorded (Foster et al. 1977b).

In another measure of mite infestation, each leaf (from 10 randomly selected plants in each genotype) can be evaluated for susceptibility to mites (1 = no mites; 2 = few individuals above midrib only; 3 = colonies along the midrib; 4 = mites spreading away from midrib; and 5 = entire leaf covered with mites) (Foster et al. 1977b). Data presented using this technique are expressed as mean leaf ratings per plant of each test genotype.

An additional rating system can also be used to denote the damage to each test entry based on the leaf area damaged (Foster et al. 1977b) as a measure of leaf necrosis (1 = 10-20%, 2 = 21-30%, 3 = 31-40%, 4 = 41-50%, 5 = 51-60%, 6 = 61-70%, 7 = 71-80%, 8 = 81-90%, and 9 = > 91%). In order to obtain more precision, foliar damage can also be measured in comparison with uninfested leaf area with a leaf area meter.

Grain and forage yield, and 100-grain mass. Record grain and forage yield and 100-grain mass in genotypes maintained under infested and uninfested conditions. Harvest the panicles at maturity and record the panicle grain mass and 100-grain mass. Express the loss of grain/forage yield, or 100-grain mass in the infested plants, in comparison with uninfested plants of the same genotype. But note that the loss in grain and forage yields and 100-grain mass mostly depends on the time of mite infestation.

Sources of resistance

KS 30, SC 599-6, and BTx 618 are resistant to *O. pratensis* (Foster et al. 1977a). More mites have been recorded on late-maturing M 100, and fewest on early-maturing genotypes (60 M and CK 60) (Archer et al. 1986a). In respect of *O. indicus,* low foliar damage has been observed on 2219A x SB 901, 2077A x SB 905, and 168 (Kulkarni et al. 1978), CSH 5, CSH 6, SPH 890, CSV 5, and IS 3687; and SPV nos.106, 135, 192, 220, 222, 224, and 365 (Singh et al. 1981).

Mechanisms of resistance

Among the mechanisms of resistance, tolerance is a major component of resistance to *O. pratensis* (Foster et al. 1977a). Singh et al. (1981) reported antixenosis as a component of resistance to *O. indicus.* Sreedhar (1995) evaluated different components of resistance to mites and found (a) high degree of antixenosis for adult colonization in CSV 8R, CSV 14R, and IS 2146; (b) oviposition nonpreference in CSV 8R, SPV 913, and RS 29; (c) antibiosis in SPV 913, CSV 8R, ICSV 705. Sel 3, and Swati; and (d) tolerance to foliar injury in Sel 3 and ICSV 705, grain yield in IS 2146, IS 2312, IS 5613, and ICSV 705, and 100-grain mass in IS 5613, Sel 3, and SPV 913.

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Screening for Resistance to Aphids in Sorgnum

R A Balikai

Introduction

Sorghum aphids are important in several sorghum-growing areas. Information on their biology, nature of damage, and population dynamics has been published by Teetes et al. (1983), Aponte et al. (1988), and Sharma (1993).

Greenbug (*Schizaphis graminum*)

Greenbug is widely distributed in Asia, Africa, and the Americas, and is a major pest of sorghum in the latter continent. It feeds in colonies on the undersurface of the leaves and produces an abundance of honeydew. As a result of feeding, red spots appear on the leaves caused by toxins injected into the plant by the aphids. As greenbug abundance increases, the reddened areas enlarge and the leaves turn brown from the edges. The bug transmits maize dwarf mosaic virus, and may predispose sorghum to charcoal rot.

Corn leaf aphid (*Rhopalosiphum maidis*)

The corn leaf aphid often becomes abundant on sorghum and is found in almost all sorghum-growing areas of the world. It sucks the sap from the whorl leaves during the vegetative stages of crop growth. It also feeds on the panicles, produces honeydew on which molds grow, and transmits maize dwarf mosaic virus. Damage may result in yellowing, tanning, and drying up of the leaves.

Yellow sugarcane aphid (*Sipha flava*)

Yellow sugarcane aphid is an occasional pest of sorghum in North America. It prefers to feed on older leaves, secretes a-potent toxin, and causes plant mortality at the seedling stage. The damage proceeds from lower to upper leaves.

The initial symptoms of damage are leaf purpling at the seedling stage. In older plants, feeding results in severe yellowing of the plants.

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Balikai, R.A. 1997. Screening for resistance to aphids in sorghum. Pages 60-64 *in* Plant resistance to insects in sorghum (Sharma, H.C., Faujdar Singh, and Nwanze, K.F., eds.). Patancheru 502 324, Andhra Pradesh, India: International Crops Research Institute for the Semi-Arid Tropics.

Sugarcane aphid (*Melanaphis sacchan*)

Sugarcane aphid is distributed in Asia, Africa, and the Americas. It is an occasional pest worldwide. It prefers to feed on the undersurface of older leaves. The damage proceeds from the lower to upper leaves. The nymphs and adults suck sap from the lower surface of leaves, and this leads to stunted plant growth. The damage is more severe in crops under drought stress, and results in drying up of leaves and plant mortality. The aphids secrete honeydew which falls on the leaves and on the ground, on which sooty molds grow. The insect's population increases rapidly at the end of the rainy season during dry spells. Its infestation is high during the postrainy season in India. And aphid infestation spoils the crop's fodder quality. The adults and nymphs are yellowish in color. This aphid reproduces by parthenogenesis.

Resistance-Screening Techniques

Various techniques to screen for resistance to aphids have been described by Starks and Burton (1977), Kadam and Mote (1983), and Cruz and Vendramin (1988, 1989). The following approach may be adopted to screen for resistance to aphids under natural infestation and in field conditions during periods of maximum aphid density. However, because field screening is influenced by the effect of weather on aphid abundance and damage, the methods described attempt to ensure optimal efficiency.

Hot-spots

Test the material at hot-spot locations where the pest is known to occur naturally and regularly at levels that often result in severe damage to the crop. In India, Bijapur, Dharwad, Rahuri, and Hyderabad are some of the hot-spot locations that are used to screen for resistance to *M. sacchah.*

Sowing date

To screen for resistance under natural infestation, especially at hot-spot locations, adjust the sowing date of the crop so that it is at a susceptible stage, when the aphid density is at its peak. Determine the periods of maximum aphid density by monitoring aphid infestation at regular intervals. Studies conducted at Bijapur, Karnataka, India, for 2 years on cultivars M 35-1 and CSH 12R in the postrainy season have shown that heavy incidence of *M. sacchari* occurs during the 2nd fortnight of Jan. Increase in aphid abundance was observed following rains in Nov-Dec, or during continuous cloudy weather. Aphid abundance was greater, and the incidence was earlier, on CSH 12R than on M 35-1. Irrespective of date of sowing and cultivar, the peak in aphid population has been observed from the 3rd to 5th standard week (Balikai 1995). Sorghum planted in late Nov is heavily infested with *M. sacchari.* Aphid infestation can be recorded at grain-filling

stage during the 1st week of Mar. And infestations during the late stages of crop growth adversely affect the fodder quality (Narayana et al. 1982).

Augmentation of aphid density

Aphid numbers can be augmented by using the following techniques.

- Sow infester rows of a susceptible cultivar (e.g., CSH 8R, or CSH 12R) along with the test material. Sow four infester rows after every 16 rows of the test cultivars.
- Collect aphids from other fields and spread them in the infester rows to augment aphid density and to maintain uniformity in the pest load among the test cultivars.
- For better results, group the test material according to maturity, since early-maturing sorghum cultivars (which have fewer leaves) suffer more yield loss than medium- or late-maturing cultivars. The sowing date of each maturity group can be suitably adjusted so that the crop's susceptible stage coincides with the peak in aphid density.

Damage Evaluation for Resistance Screening

Aphid damage in sorghum results in plant mortality under severe infestation during the seedling stage. Signs of damage are yellowing and browning followed by drying of leaves in the older plants. Leaf injury has been found to be correlated with reduction in grain yield under severe infestation, and adverse effects on the quantity and quality of fodder. For the selection of aphid-resistant genotypes it is important to record data both on aphid density and leaf damage. To this end the aphid density:damage ratio can be evaluated by using the following criteria.

Aphid density

At peak infestation, evaluate the test genotypes for aphid resistance by recording aphid density. For this, count the number of aphids in a unit area on three leaves, and record observations on five plants selected at random from each genotype.

Damage rating

This can be evaluated in a 1 to 9 scale (where 1 = a few aphids present with no apparent damage to the leaves, and 9 = heavy aphid density on infested leaves: see Table 1). Plant injury can also be rated on a 0 to 9 scale (0 = no injury, and 9 = severe injury) (Kadam and Mote 1983). A visual damage scale of zero (no damage) to 9 (over 80% necrotic plants), and the growth differences between infested and uninfested plants, have been suggested by Cruz and Vendramin (1988, 1989).

Alternatively, the technique suggested by Starks and Burton (1977) (Table 2) can be used.

Table 1. Aphid density: injury rating based on visual scoring to screen for resistance to *Melanaphis sacchah.*

Aphid density injury rating	Aphid density/ injury (%)
1	1-10
2	11-20
3	21-30
4	31-40
5	41-50
6	51-60
7	61-70
8	71-80
9	>80

Table 2. Rating scale to screen for resistance to *Schizaphis graminum.*

Score	Remarks
1	No red spots on the leaves.
2	Red spots on the leaves.
3	Portion of the leaf killed by aphids.
4	One leaf killed by aphids.
5	Two leaves killed by aphids.
6	Four leaves killed by aphids.
7	Six leaves killed by aphids.
8	Eight leaves killed by aphids.
9	Plant killed by aphids.

Grain yield

Record grain yield of the genotypes being tested. The test material can be maintained under infested and noninfested conditions. Harvest all panicles from the middle row(s) at maturity, and record panicle and grain mass. Express the loss in grain yield in the infested plots, or panicles as a percentage of the grain yield in noninfested plots, or panicles.

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Screening for Resistance to Sorghum Head Bugs

H C Sharma

Introduction

Head bugs, *Calocoris angustatus, Creontiades pallidus, Eurystylus oldi,* and *Campylomma* spp are serious pests of grain sorghum in India and Africa. *C. angustatus* is the most important species in India and *E. oldi* in western Africa. *C. angustatus* nymphs and adults feed mainly on the developing grain, and occasionally on tender parts of the plant (Sharma and Lopez 1990). The nymphs and adults suck sap from the developing grain, which remain unfilled, and shrivel. Under severe infestation, the damaged spikelets may become completely chaffy. Damage during the early stages of grain development results in heavy yield loss; later infestations result largely in quality loss. The damaged grain shows distinct red brown feeding punctures and, after severe feeding, the grains are completely tanned. Such grains are more prone to mold incidence and show poor germination. *C. pallidus, E. oldi,* and *Campylomma* spp insert their eggs inside grains at the milk stage (Ratnadass et al. 1994). The grain tissue around the egg becomes reddish brown, and this spoils the grain quality. Other feeding symptoms are similar to those of *C. angustatus.*

Screening for Resistance to *Calocoris angustatus*

Various techniques to screen for resistance to *C. angustatus* have been described by Sharma and Lopez (1992a,b) and Sharma et al. (1992b).

Field screening

Screening for head bug resistance can be carried out under field conditions during periods of maximum bug density. Screening under field conditions is influenced by: (a) variation in flowering, (b) fluctuations in bug density, and (c) the effect of weather conditions on the bug population build-up and damage. Early- and late-flowering cultivars normally escape head bug damage, while those flowering in mid-season are exposed to very high bug abundance. The following methods can be used to increase the screening efficiency for head bug resistance under field conditions.

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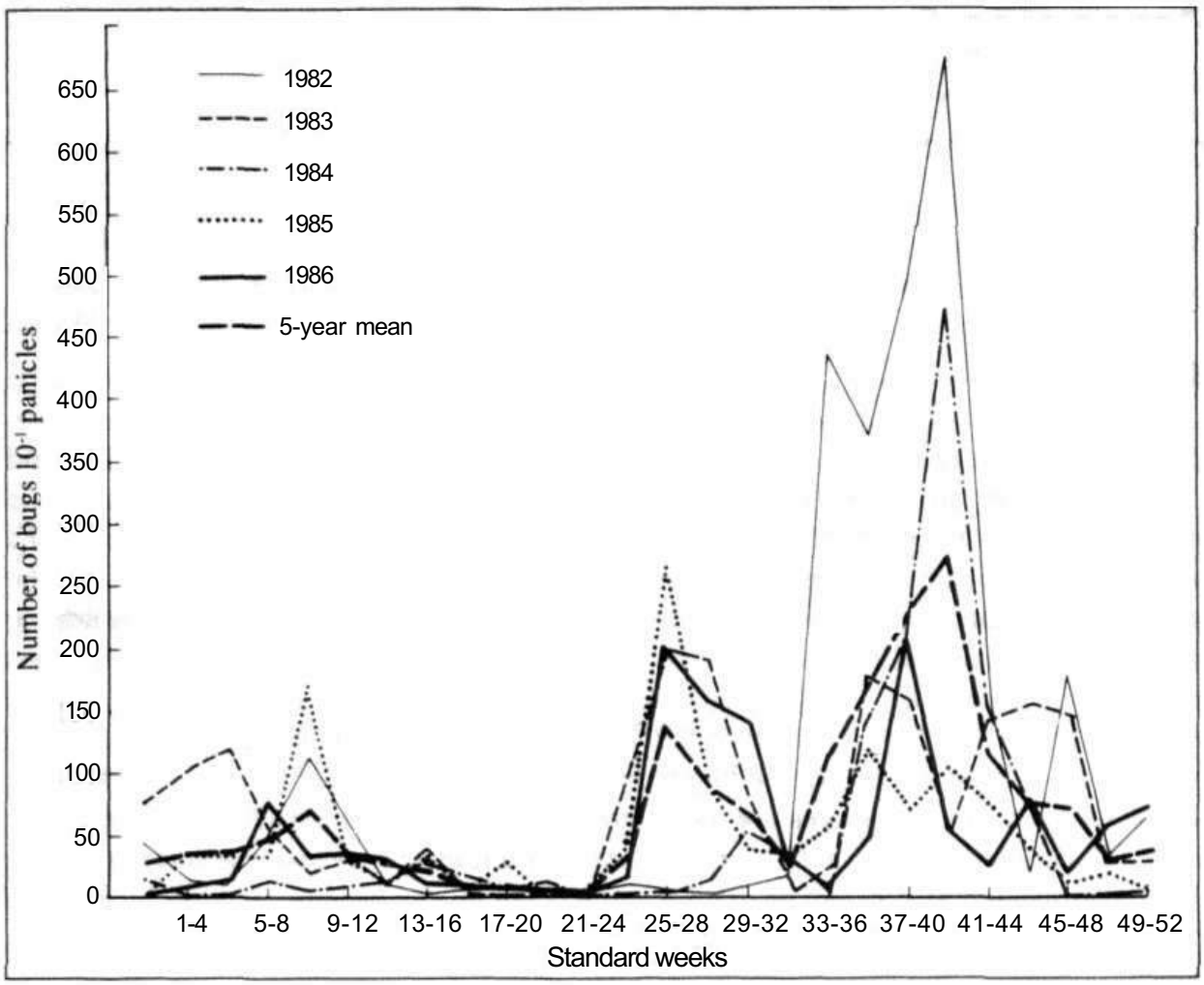

Figure 1. Population dynamics of head bug, *Calocoris angustatus* at ICRISAT, Patancheru, Andhra Pradesh, India.

Hot-spots. In India, ICRISAT-Patancheru, Bhavanisagar, Kovilpatti, Coimbatore, Palem, and Dharwad are the hot-spot locations to screen for resistance to head bugs. At ICRISAT-Patancheru, head bug density is very high during Sep-Oct, but remains quite low during the postrainy season.

Sowing date. Adjust sowing dates such that flowering coincides with maximum bug density. Determine the periods of maximum head bug density through fortnightly sowings. Maximum bug numbers at ICRISAT-Patancheru have been recorded during Sep, and a second but smaller peak has been recorded during Mar (Fig. 1). Crops sown during the 2nd week of Jul suffer the highest bug damage. At Bhavanisagar, the peak in bug density occurs during May-Jun, and the optimum time to sow for resistance screening is during the 2nd fortnight of Feb.

Infester-row technique

- Sow infester rows of mixed-maturity cultivars 20 days earlier than the test material. Alternatively, sow early-flowering (40-45 days) sorghums (IS 802, IS 13249, and IS

Figure 2. Infester-row technique to screen for resistance to head bugs under field conditions.

24439) along with the test material as infester rows. Sow four rows (Fig. 2) of a susceptible cultivar after every 16 rows of the test material.

- Collect bugs from other fields and spread them in the infester rows at panicle emergence to augment the bug abundance.
- Sow the test material in two sets, at an interval of 10-15 days between sowings, to reduce the chances of escape in the early- and late-flowering lines.
- For better results, group the test material according to maturity and height. The sowing date of each maturity group can be suitably adjusted so that flowering occurs during the peak in bug abundance.

Headcage technique

To overcome the problem of variation in flowering among the test cultivars, and fluctuations in insect abundance, the headcage technique developed for midge resistance screening has been found to be useful for head bugs also (Fig. 3). This technique also permits an increase in bug abundance and grain damage to be studied under no-choice conditions in relation to different infestation levels and stages of panicle development.

- Select 5-10 sorghum panicles at the top-anthesis stage in each plot/genotype.
- Tie the headcage around the sorghum panicle and cover it with a white muslin cloth bag.
- Collect bugs in muslin cloth bags from sorghum panicles at the milk stage.
- Separate the adult males and females (males are smaller and darker in color than the females).
- Collect 10 head bug pairs in a 200-mL plastic bottle aspirator.
- Release the head bugs in the cage and close the cloth bag.
- Examine the infested panicles after 1 week and remove panicle-feeding caterpillars, or predatory spiders if there are any.

- Remove the muslin cloth bag along with the bugs 20 days after infestation and take them to the laboratory. Kill the bugs using ethyl acetate (2 mL bag^{-1}), or keep the bags in a deep-freeze refrigerator for 30 min. Count the total number of bugs in each cage.
- Evaluate the panicles for bug damage at maturity as described under damage evaluation.

Screening for Resistance to *Eurystylus oldi*

Techniques to screen for resistance to *E. oldi* have been discussed by Sharma et al. (1992a, 1994), and Doumbia et al. (1995).

Infester-row technique

- The infester-row technique recommended to screen for resistance to *C. angustatus* can be adapted to screen for resistance to *E. oldi* as well.
- The hot-spot locations for *E. oldi* in western Africa are Sotuba and Cinzana in Mali, Kamboinse and Farako Ba in Burkina Faso, Kolo in Niger, and Samaru and Kano in Nigeria. Peak in head bug abundance has been observed during the 1st fortnight of Oct.
- For maximum bug damage, sow the crop during the 2nd fortnight of Jul.
- For efficient screening, sow the test material twice at an interval of 15 days, and group the genotypes according to maturity and height, as described for *C. angustatus.*

Headcage technique

- Select 5-10 panicles at the complete-anthesis stage (6 days after flowering) in each genotype/plot.
- Collect adult bugs from sorghum panicles at dough to hard-dough stage in muslin cloth bags.
- Separate male and female adults (males are smaller, and the females have a wedge-shaped abdomen ventrally, with a dark ovipositor), and collect 20 pairs of bugs in a 200-mL plastic bottle aspirator. Alternatively, bugs can also be picked up randomly from the field population (the sex ratio is close to 1:1), or collect 50 III—IV instar nymphs with an aspirator.
- Release the bugs inside the cage and close the cloth bag.
- Examine the cages 1 week after infestation and remove spiders and head caterpillars, if there are any.
- Count the bugs in each infested panicle as described under damage evaluation.
- At maturity, evaluate the panicles for head bug damage. In selecting resistant genotypes, it important to maintain uniformity in panicle size among the genotypes being tested, and to record data both on head bug numbers and grain damage.

Damage Evaluation for Resistance Screening

Sorghum head bugs suck the sap from developing grain that results in shriveling and tanning of grains. Some grains may remain undeveloped. Damage symptoms are normally

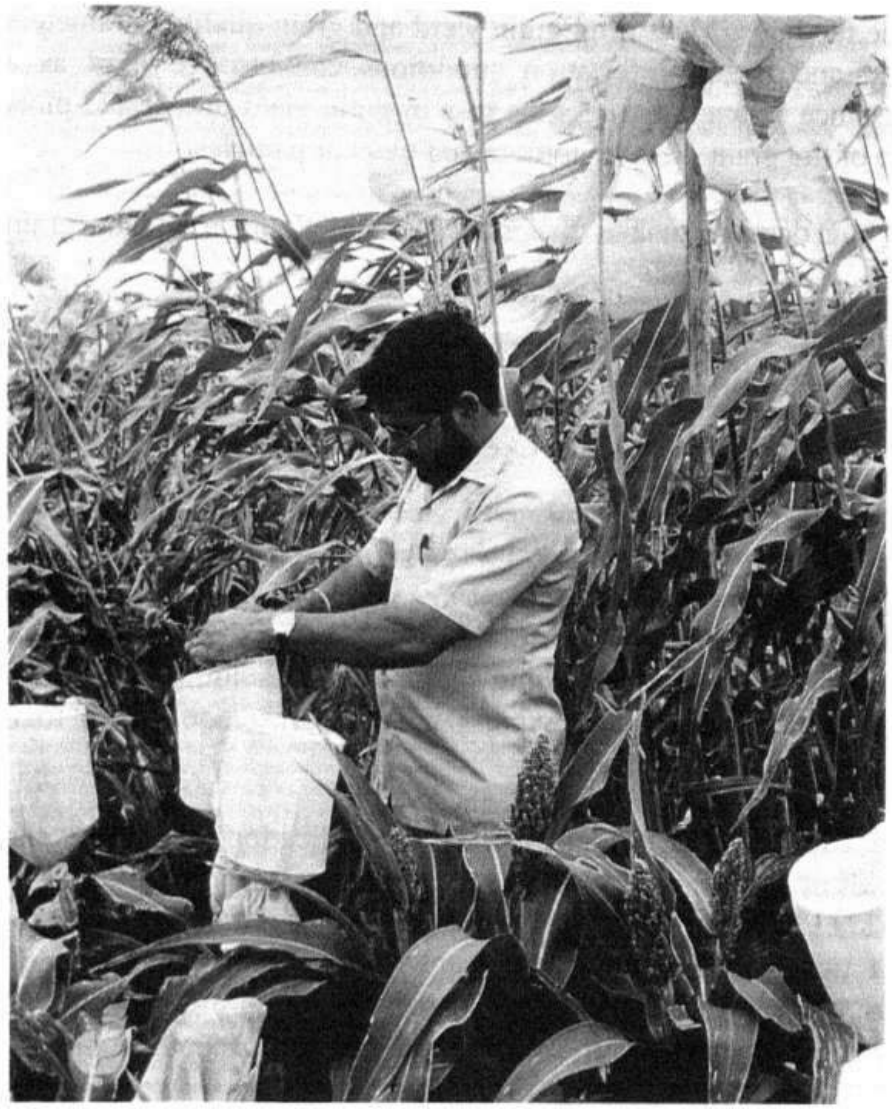

Figure 3. Headcage technique to screen for resistance to head bugs under uniform insect density.

evident on some or all of the grains. Head bug damage is generally higher inside the panicle. In some cases, a portion of the panicle may be more damaged than the rest, and some grains may be normal, while others show damage symptoms. Head bug damage can be evaluated by the following criteria.

Head bug counts. Tag five panicles at random in each genotype at the half-anthesis stage. Sample the panicles for bugs 20 days after flowering, or infestation in a polyethylene bag containing a cotton swab soaked in 2 mL of ethyl acetate or benzene. Count the total number of adults and nymphs.

Grain damage rating. Evaluate head bug damage at maturity on a 1 to 9 scale in which:

1 = all grains fully developed with a few feeding punctures;
2 = grain fully developed, with feeding punctures;
3 = grains showing slight tanning or browning;
4 = most grains with feeding punctures, and a few showing slight shriveling;
5 = grains showing slight shriveling and browning;
6 = grains showing more than 50% shriveling and turning brown or tanned;
7 = most of the grain highly shriveled and dark brown coloration;
8 = grain highly shriveled and slightly visible outside the glumes; and
9 = most of the grains highly shriveled and slightly visible outside the glumes.

Grain yield. Harvest all panicles from the middle row(s) of each plot or genotype at maturity and record panicle and grain mass in each plot or panicle. Plots, or panicles of lines being tested, can also be maintained under infested and noninfested conditions by

using cloth bags to exclude the bugs. Measuring grain yield and grain quality parameters under insecticide-protected and natural-infestation conditions can also be used as a measure of genotypic resistance to bugs. Express the loss in grain yield of infested plots or panicles as a percentage of the grain yield in noninfested plots or panicles.

Grain hardness. Head bug damage makes the grain soft and floury. Evaluate grain hardness on a 1 to 5 scale:

1 = grain completely corneous and hard;
2 = grain almost corneous;
3 = grain partly corneous;
4 = grain almost starchy and soft; and
5 = grain completely starchy and very soft.

Grain mass and floaters. Take a sample of 1 000 grains at random from each replication, or panicle. Equilibrate the moisture content overnight (12 h) at 37°C. Weigh the grain on a balance. Prepare a sodium nitrate solution of a specific density of 1.31. Keep the 1 000-grain sample in the beaker containing sodium nitrate solution. Count the number of grains floating on the surface, and express it as a percentage of the total number of grains.

Germination test. Take 100 grains at random from each replication or panicle and place them between the folds of a water-soaked filter paper in a petri dish. Keep the petri dishes in an incubator at 27±1°C, or at room temperature in the laboratory. Record the percentage of grains with radical and plumule emergence after 72 h. Data on grain hardness, 1 000-grain mass, percentage of floaters, and percentage of germination should be recorded only when the researcher intends to collect more data for in-depth studies on head bug resistance.

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Screening for Resistance to Sorghum Midge

H C Sharma

Introduction

Sorghum midge, *Stenodiplosis sorghicola* is probably the most damaging and widely distributed of all sorghum pests (Harris 1976). It occurs in all sorghum-growing regions in Africa, the Americas, Asia, Australia, and Europe. Damage is caused by the larvae, which feed on the ovary inside the glumes. This results in chaffy (empty) spikelets, and the panicles present a blasted appearance. Larvae diapause inside the glumes, and the diapause may last for 1 to several years.

Techniques to screen for midge resistance have been described by Jotwani (1978), Page (1979), Sharma (1985), and Sharma et al. (1988a,b, 1992).

The major difficulties in identifying source material with stable resistance against sorghum midge have been due to:

a) variation in the flowering of sorghum cultivars in relation to midge incidence;
b) day-to-day variation in midge populations;
c) competition with other insects, such as mirid bugs;
d) parasitization and predation by natural enemies; and
e) sensitivity of midge flies to temperature and relative humidity.

A large proportion of lines selected as less susceptible under natural conditions comprises of early- and late-flowering escapes. Because of these problems, genotypes rated as resistant under natural infestation often turn out to be susceptible in the following seasons, or at other locations. The following techniques have been standardized to screen for resistance to sorghum midge.

Field screening technique (multi-choice conditions)

Hot-spots. Hot-spot locations are useful to screen for resistance to sorghum midge. Hot-spot locations for sorghum midge are Dharwad, Bhavanisagar, and Pantnagar in India; Sotuba in Mali; Farako Ba in Burkina Faso; Alupe in Kenya; and Kano in Nigeria. Midge abundance is also high in several locations in Australia, the USA, and Latin America.

Sowing date. To screen test material for midge resistance under natural conditions, determine the periods of maximum midge density through fortnightly sowing of a susceptible

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Sharma. H.C. 1997. Screening for resistance to sorghum midge. Pages 72-78 *in* Plant resistance to insects in sorghum (Sharma, H.C., Faujdar Singh, and Nwanze, K.F., eds.). Patancheru 502 324, Andhra Pradesh, India: Internationa! Crops Research Institute for the Semi-Arid Tropics.

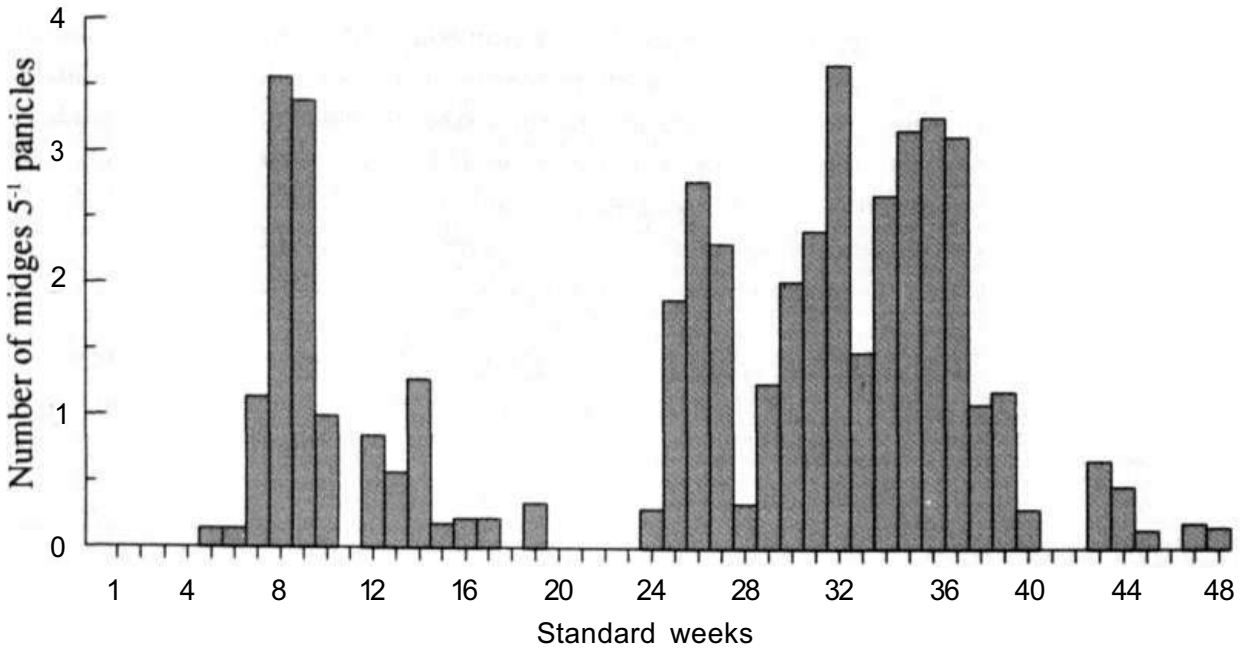

Figure 1. Population dynamics of sorghum midge at ICRISAT Center, Patancheru, Andhra Pradesh, India.

cultivar. Adjust sowing dates so that the most susceptible stage of the crop (flowering) coincides with greatest insect density. At ICRISAT-Patancheru, maximum midge density and damage have been observed in the crop planted during the 3rd week of Jul (Fig. 1). The peak in midge density occurs during Oct. A second but smaller peak has been observed during Mar in the postrainy season, for which planting is carried out during mid-Dec. At Dharwad, the peak in midge abundance has been recorded during Oct, and the optimum time for sowing test material is between 20 Jul and 5 Aug. It is necessary to determine the appropriate time for sowing the test material to screen for resistance to sorghum midge effectively at different locations.

Augmentation of midge density through infester rows and diapausing larvae.

Midge abundance can be increased through infester rows and spreading sorghum panicles containing diapausing midge larvae in the infester rows (Sharma et al. 1988a) (Fig. 2).

- Sow infester rows of cultivars CSH 1 and CSH 5(1:1 mixture) 20 days before the test material. Alternatively, early-flowering (40-45 days) lines (IS 802, IS 13249, and IS 24439) can be sown along with the test material.
- Sow four infester rows of a susceptible cultivar after every 16 rows of the test material.
- Collect midge-infested chaffy panicles containing diapausing midge larvae at the end of the cropping season. Chaffy panicles can be stored in gunny bags, or in bins until the next season.
- Spread midge-infested sorghum panicles containing diapausing midge larvae at the flag leaf stage of the infester rows. Moisten the panicles for 10-15 days to stimulate the termination of larval diapause. Adults emerging from diapausing larvae serve as a starter infestation in infester rows to supplement the natural population. Midge population multiplies for 1-2 generations on the infester rows before infesting the test material. A combination of infester rows and spreading sorghum panicles containing diapausing larvae increases midge damage 3-5 times. Infester rows alone also increase midge damage.

Sprinkler irrigation. High relative humidity is important for midge activity, adult emergence, and subsequent damage. Use overhead sprinkler irrigation to increase relative humidity in midge-screening trials during the postrainy season, or periods of low relative humidity. Operate sprinkler irrigation daily between 1500 to 1600 from panicle emergence to the grain-filling stage of the crop. Midge damage increases significantly with the use of sprinkler irrigation. Sprinkler irrigation on the crop canopy between 1500 to 1600 does not affect oviposition because peak midge abundance and oviposition occur between 0730 to 1100.

Selective use of insecticides to control other insects in a midge screening nursery. *Calocoris angustatus* and *Tetrastichus diplosidis* are the two major insects limiting midge abundance in midge resistance screening trials. Mirid bugs damage the sorghum panicles from emergence to hard-dough stage and compete for food with sorghum midge. Also, adult mirid bugs preyon ovipositing midges at flowering. *T. diplosidis* is an efficient parasite of sorghum midge at some locations.

Spray less persistent and contact insecticides such as carbaryl and malathion to control mirid bugs at the complete-anthesis to milk stage (Sharma and Leuschner 1987). The midge larvae feeding inside the glumes are not affected by the contact insecticides sprayed after flowering. Parasitism by *T. diplosidis* is also reduced in panicles sprayed at the complete-anthesis to milk stage.

Split sowing. Sow the test material twice at a 15-day interval to minimize the chances of escape from midge damage in early- and late-flowering lines. Split sowing of the test material increases the efficiency of selection for midge resistance.

Plant population affects the insect density per unit area, and in some cases influences the incidence and survival rate of insects. The level of midge damage has been observed to be higher at lower planting densities. Under field conditions, midge damage and efficiency of screening for midge resistance can be substantially increased by using a combination of timely sowing, spreading midge-damaged sorghum panicles containing diapausing larvae in the infester rows, split sowing, and selective use of contact insecticides for the control of mirid bugs and midge parasites. These techniques are useful in the initial large-scale screening of germplasm and breeding materials for resistance to sorghum midge.

Headcage technique

Caging midge flies with sorghum panicles is an important method for avoiding escape, and permits screening for midge resistance under uniform insect pressure. A headcage technique has been developed and standardized at ICRISAT-Patancheru. It consists of a cylindrical wire frame made of 1.5-mm diameter galvanized iron wire. The loop attached to the top ring rests around the tip of the panicle, and the extensions of the vertical bars at the lower ring are tied around the peduncle with a piece of G.I. wire, or electric wiring clips (Fig. 3). These prevent the cage from slipping when disturbed by wind or other external factors. Screening for resistance to midge can be carried out as follows:

- Select sorghum panicles at 25-50% anthesis stage. Remove spikelets with dried-up anthers at the top, and immature ones at the bottom of the panicle with scissors so that

Figure 2.Infester rows to increase midge abundance for resistance screening.

only the spikelets at anthesis in the middle of the panicle are exposed to the midge flies for oviposition.

- Place the wire-framed cage around the sorghum panicle and cover it with a blue cloth bag (20 cm wide and 40 cm long). The cloth bag at the top has an extension (5 cm in diameter, 10 cm long) to release the midges inside the cage.
- Collect 20 adult female midges in a plastic bottle (a 200 mL aspirator) between 0800 and 1100 from flowering sorghum panicles (only female midges visit the flowering sorghum panicles and these are collected for use in infestation).
- Release 40 midges into each cage and close the inlet. Repeat the operation the next day. Infest 5-10 panicles in each genotype, depending upon the stage of material and the resources available.
- Examine the cages 5-7 days after infestation and remove any other insects such as mirid bugs, panicle-feeding caterpillars, and predatory spiders.
- Remove the cages 15 days after infestation and evaluate the midge damage.

Spikelets with midge larvae and midge-damaged chaffy spikelets are most numerous in panicles infested with 40 midges for 2 consecutive days. There may be some variation in midge damage over seasons because of temperature, rainfall, and relative humidity, which influence both oviposition and damage by the sorghum midge. Midge damage decreases as the time of collection and release advances from 0830 to 1430. Other factors that account for decrease in midge damage over time are natural death of adults (midges die between 4 and 24 h), and reduced fecundity and oviposition because of increasing temperatures and decreasing relative humidity. Panicles infested at the top- and at half-anthesis generally suffer greater damage compared with those infested at the pre-and complete-anthesis stages. Sorghum midge behavior is influenced by different colors. Among the various colored muslin

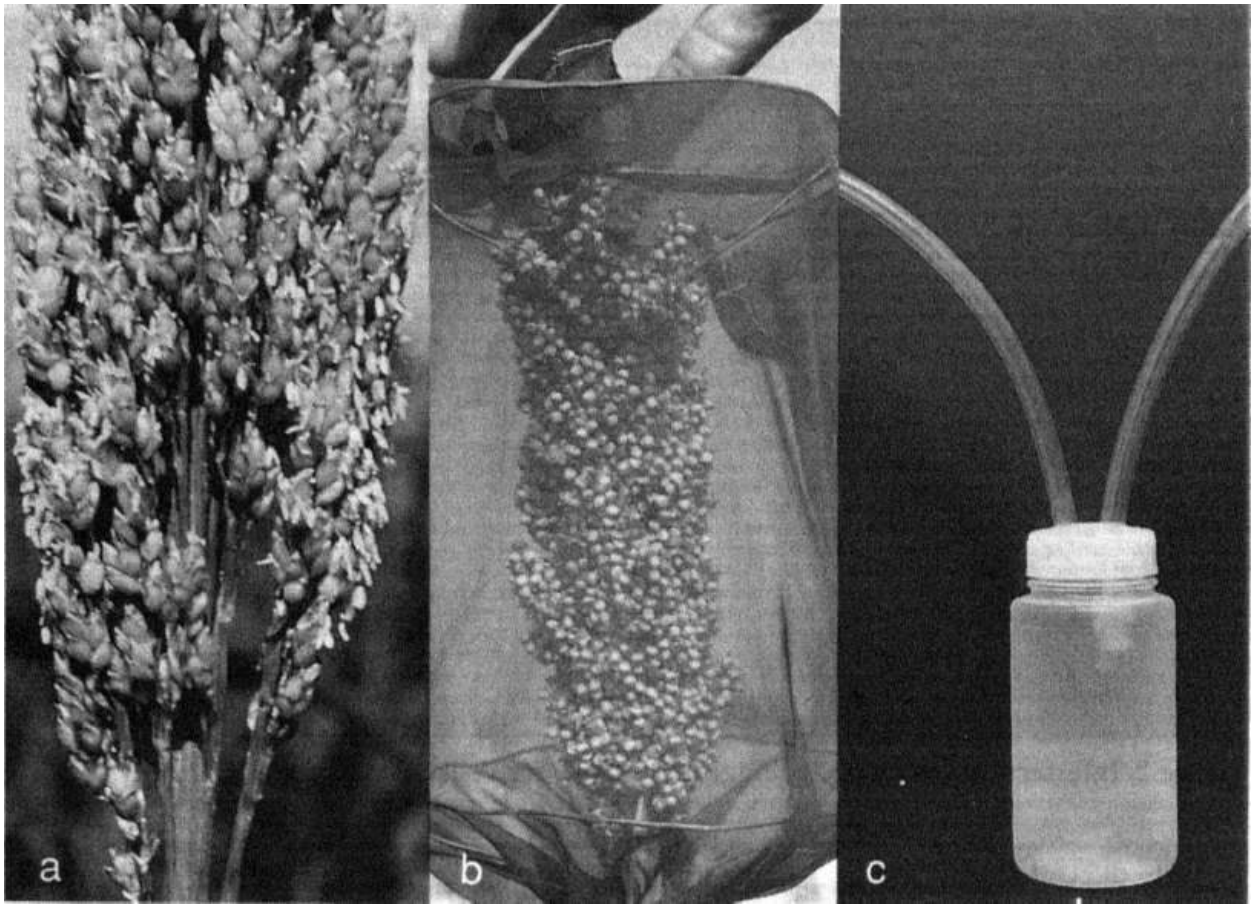

Figure 3. Headcage technique to screen for resistance to sorghum midge under uniform insect pressure: (a) panicle at anthesis trimmed with scissors for infestation; (b) headcage covered with a cloth bag: and (c) aspirator used to collect midge flies.

cloth bags tested (blue, black, red, yellow, or white), maximum midge damage has been recorded in panicles covered with blue and black bags. Blue bags are used to cover the cages because, in the semi-arid tropics, the use of black bags may create a high temperature inside the cage during the hot and dry season.

The headcage technique is quite simple, easy to operate, and can be used on a fairly large scale to confirm the field resistance of selected genotypes. Changing weather conditions influence midge activity, and can affect midge damage under the headcage. In general, it is a thorough test for use in resistance screening, and is particularly applicable in identifying stable and durable resistance. For optimal stability, test material should be screened over several environments.

Damage evaluation for resistance screening

Feeding by the midge larva on a developing grain inside the glumes leads to sterile or chaffy spikelets. However, the symptoms (chaffiness) of natural sterility and extensive grain damage by sucking insects are superficially similar to damage caused by mi,dge. The midge-infested panicles have either small white pupal cases attached to the tip of damaged spikelets,

or have small parasite exit holes in the glumes. The following methods are suggested for damage evaluation. Genotypes flowering on different dates are tagged with different-colored labels or tapes, or are marked with a paint along with panicles of resistant and susceptible checks. Selection for resistance should be based on control plants that are resistant and susceptible to midge attack and flower on the same day.

Chaffy spikelets. This is the most appropriate criterion by which to evaluate sorghum lines for midge resistance. Tag five panicles in each genotype at half-anthesis. Record the midge damage in the spikelets 15 days after flowering, as follows:

- Collect five primary branches each from the top, middle, and bottom portions of the panicle.
- Bulk the samples from all the five tagged panicles in a genotype.
- Remove secondary branches from the primary branches and mix the sample thoroughly.
- Pick up the secondary branches at random and count the number of chaffy spikelets in a sample of 500 spikelets.
- Squeeze the chaffy spikelets between the thumb and first finger, or with forceps. Record the number of spikelets producing a red ooze (this indicates midge damage). Chaffy spikelets with early-instar larvae do not produce a red ooze.
- Express the data as a percentage of chaffy or midge-damaged spikelets.
- Midge-damaged chaffy spikelets can also be recorded at harvest by adopting the procedure described above.

Visual damage rating. At crop maturity, evaluate midge damage on a 1 to 9 scale where: 1 = <10%, 2 = 11-20%, 3 = 21-30%, 4 = 31-40%, 5 = 41-50%, 6 = 51-60%, 7 = 61-70%, 8 = 71-80%, and 9 = >81 % midge-damaged spikelets.

Grain yield. Record grain yield from the genotypes being tested. The test material can be maintained under infested and noninfested conditions by using a cloth bag to calculate the percentage of midge damage, or spray insecticide to control the midge at flowering in the noninfested material. Harvest all panicles from the middle row(s) at the time of maturity and record the panicle and grain mass. Express the loss in grain yield in infested plots or panicles as a percentage of the grain yield in noninfested plots or panicles.

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Part 3

Mechanisms and Inheritance of Resistance

Mechanisms of Resistance to Insects and their Usefulness in Sorghum Improvement

H C Sharma, K F Nwanze, and V Subramanian

Introduction

Screening for resistance to insects under screenhouse/field conditions is the most effective method of developing insect-resistant cultivars (Sharma et al. 1992). However, it is not possible to rear all insect species at all locations, and the occurrence and abundance of insect populations under natural conditions are sporadic, and highly influenced by the environment. A knowledge of the mechanisms and the factors contributing to host-plant resistance to insects is useful in deciding suitable selection criteria and breeding methods for the genetic improvement of sorghum for resistance to insects (Sharma 1993). Some of the factors associated with resistance to insects can be quantified/monitored easily in plant populations, and such plant characters can be used as "marker traits" to screen and select for resistance to insects. These mechanisms are discussed in this paper for the major insects attacking sorghum.

Sorghum Shoot Fly (*Atherigona soccata*)

Shoot fly attacks sorghum from 5 to 25 days after seedling emergence. The adult fly lays white cigar-shaped eggs singly on the undersurface of the leaves. The larvae cut the growing point and feed on the decaying leaf tissues. This results in typical deadheart symptoms.

Resistance mechanisms

Nonpreference for oviposition. This is the primary mechanism of resistance to shoot fly (Soto 1974; Singh and Jotwani 1980a; Raina et al. 1984; Taneja and Leuschner 1985). Significantly higher oviposition has been recorded on the susceptible cultivar CSH 1 (66% plants with eggs) compared with resistant genotypes: IS 1034, IS 2146, IS 2265, IS 2309, IS 3962, IS 4664, IS 5566, IS 5604, IS 18369, and IS 18551 (<40% plants with

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Sharma, H.C., Nwanze, K.F., and Subramanian, V. 1997. Mechanisms of resistance to insects and their usefulness in sorghum improvement. Pages 81-100 *in* Plant resistance to insects in sorghum (Sharma, H.C., Faujdar Singh and Nwanze, K.F., eds.). Patancheru 502 324, Andhra Pradesh, India: International Crops Research Institute for the Semi-Arid Tropics.

Figure 1. Tiller production in sorghum following shoot fly damage. The tillers serve as a mechanism for recovery resistance.

eggs). However, more eggs were recorded on shoot fly-resistant cultivars, particularly IS 1082, IS 2122, IS 2195, IS 4664, IS 5484, and IS 5566 under no-choice than under multiple-choice conditions. Non-preference for oviposition to shoot fly breaks down under no-choice conditions, or under heavy shoot fly pressure in the field.

Antibiosis. Survival and development of shoot fly is adversely affected when reared on shoot fly-resistant genotypes (Jotwani and Srivastava 1970; Narayana 1975). Growth and development of the insect are retarded and the larval and pupal periods are extended by 8-15 days on resistant genotypes (Singh and Jotwani 1980b). Survival and longevity of females, and fecundity, are adversely affected when the larvae are reared on shoot fly-resistant genotypes (Raina et al. 1981).

Tolerance. Some sorghum genotypes exhibit an inherent ability to produce side tillers after the main shoot is killed by shoot fly, which in turn can produce a reasonable yield if the plant is not attacked again (Fig. 1). Tillers of resistant cultivars have been shown to be less preferred for egg-laying. Resistant cultivars have a very high rate of tiller survival compared with susceptible cultivars (Blum 1972; Doggett 1972).

Evaluation for recovery resistance (tolerance)

- Record the number of main stems (plants not damaged by the shoot fly) and shoot fly-damaged plants in the plot at harvest.
- Record the number of tillers in shoot fly-damaged plants.

- Record the number of productive tillers (tillers having panicles with grain) in shoot fly-damaged plants, and express these as a percentage of the total number of tillers in the shoot fly-damaged plants.
- Record grain yield in the shoot fly-damaged and undamaged plants. Lines showing similar yield potential under infested and noninfested conditions would have a strong tolerance mechanism of resistance.

Factors associated with resistance

Seedling vigor. Fast seedling growth may delay the 1st-instar larvae from reaching the growing point, although leaf margins may be cut without causing a deadheart. Seedling vigor is negatively associated with deadheart formation. Shoot fly-resistant lines have a rapid plant growth (Mote et al. 1986), greater seedling height and hardness (Singh and Jotwani 1980d), and longer stems and internodes, and short peduncle (Patel and Sukhani 1990a).

Evaluation for seedling vigor

- Evaluate the seedlings for vigor (height, leaf growth, and robustness) 14 days after seedling emergence in the morning or evening hours, when seedling growth can be measured properly.
- Evaluate seedling vigor on a 1 to 5 scale, where 1 = plants showing maximum height, leaf expansion, and robustness, and 5 = plants showing minimum growth, less leaf expansion, and poor adaptation.

Glossiness. The glossy trait (pale green and shiny leaves) in sorghum (Fig. 2) is associated with shoot fly resistance (Blum 1972; Maiti and Bidinger 1979). Most of the lines resistant to shoot fly exhibit the glossy leaf character during the seedling stage. Intensity of glossiness of the leaves at the seedling stage is positively associated with resistance to shoot fly.

Evaluation for leaf glossiness

- Intensity of leaf glossiness is recorded 10-12 days after seedling emergence on a 1 to 5 scale (1 = lines with light green, shining, narrow, upward-pointed leaves, and 5 = lines with dark green, dull, broad, and drooping leaves).
- Leaf glossiness is recorded in the morning hours when there is maximum reflection of light from the leaf surfaces, and clear vision of the rest of the plant.

Leaf surface wetness. Cultivars with a high transpiration rate are preferred for oviposition (Mate et al. 1988), and there are genotypic differences in surface wetness of the central shoot leaf between resistant and susceptible genotypes (Nwanze et al. 1990). Leaf moisture is important for larval movement and deadheart formation (Raina et al. 1981). Shoot fly-resistant and moderately resistant genotypes are characterized by a smooth amorphous wax layer, and sparse wax crystals. Susceptible genotypes possess a dense meshwork of crystalline epicuticular wax (Nwanze et al. 1992),

Figure 2. The glossy leaf character in sorghum seedlings associated with resistance to shoot fly.

Evaluation of leaf surface wetness

- Examine 12-day-old seedlings for leaf wetness between 0630 and 0830.
- Excise the central unfolded leaf, and spread it under a binocular microscope.
- Assess the leaf surface wetness on a 1 to 5 scale (1 = no apparent moisture, or a thin film of water on the leaf lamina, and 5 = leaf lamina densely covered with water droplets).

Trichomes. Trichomes on the undersurface of leaves (Fig. 3) are associated with shoot fly resistance (Blum 1968; Maiti et al. 1980). The wild species of sorghum that are immune to shoot fly have high trichome density on the lower surface of all leaves, which may contribute to resistance (Bapat and Mote 1982). Shoot fly-resistant germplasm lines have trichomes on the undersurface of leaves (except IS 5622, which has trichomes only on the upper surface). Trichomes are absent in shoot fly-susceptible lines.

Evaluation of trichome density on the undersurface of leaves

- Take leaf samples (1-2 cm^2) from 12-14-day-old seedlings, and place them in 20 mL of-acetic acid:alcohol (2:1) in small stoppered glass vials overnight.
- Transfer the leaf samples into 90% lactic acid in stoppered vials. The cleared leaf samples can be stored for examination.
- For examination, mount the leaf samples on a slide in a drop of lactic acid, and observe them under a microscope at 20x or 40x. Count the number of trichomes in fields selected at random, and express the trichome density as the number mm^{-2}. Trichome length may be measured using an ocular micrometer.

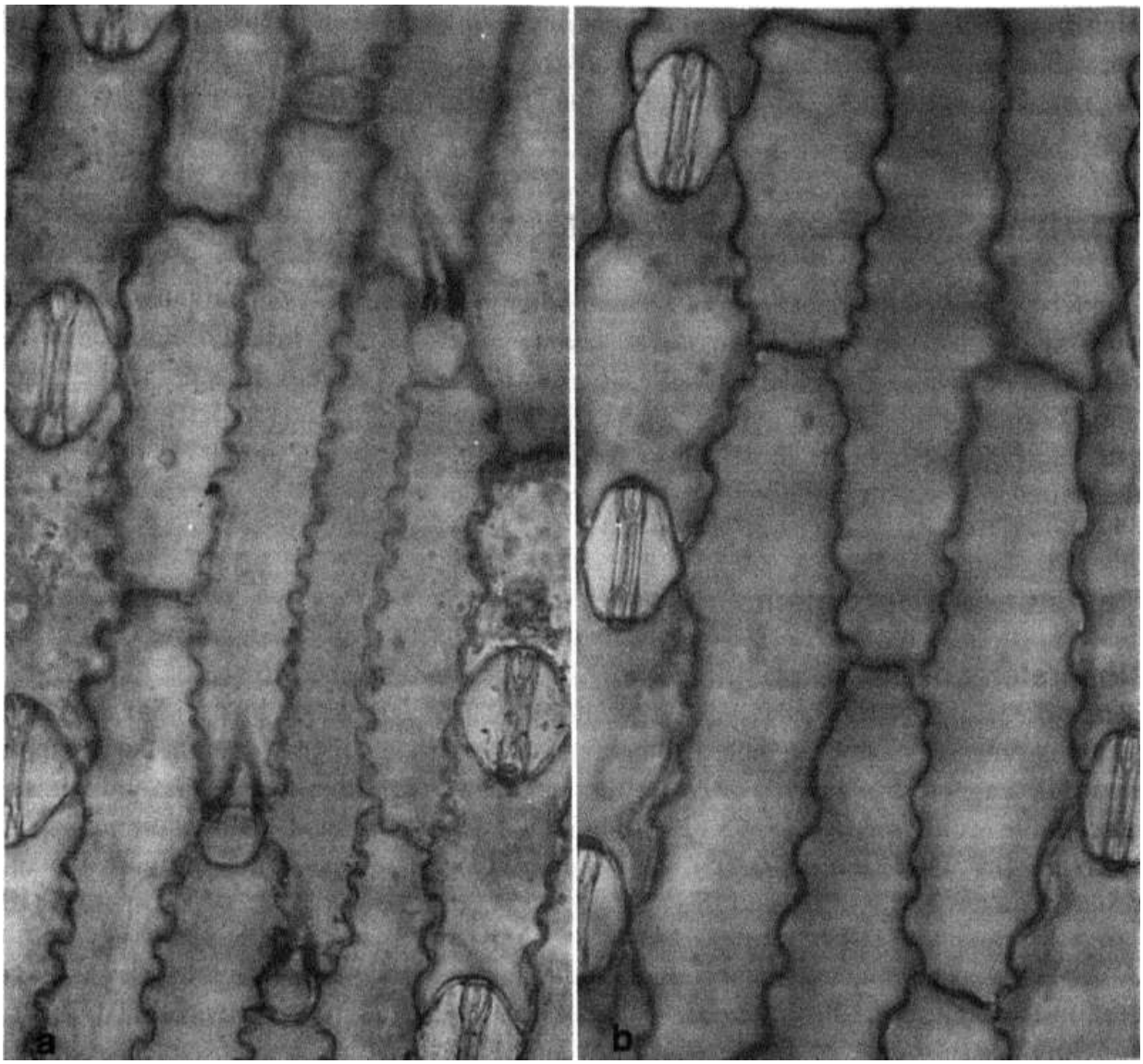

Figure* 3. *Cleared sections from* 10- *day -old sorghum seedlings showing trichomes and stomata;* (*a*)*IS 1082, with trichomes; and (b) CSH* 2, *without trichomes.

Biochemical factors. Ponnaiya (1951) reported the presence of irregular-shaped silica bodies in the plant tissue from the fourth leaf onwards in shoot fly-resistant cultivars, and from the sixth leaf onwards in susceptible ones. He suggested that the relatively late appearance of these silica bodies in the susceptible cultivars make them prone to shoot fly attack for a longer period. Blum (1968) noted distinct differences in lignification and silica deposition between resistant and susceptible cultivars; but no definite conclusions were drawn for the association between these anatomical characters and seedling resistance.

Percentage of nitrogen, reducing sugars, total sugars, moisture, and chlorophyll content of leaf in susceptible cultivars are higher than in resistant ones (Singh and Jotwani 1980c; Patel and Sukhani 1990b). Lysine is present in the leaf sheath of susceptible cultivars but absent in all the three resistant cultivars tested. Khurana and Verma (1982) observed

higher quantities of total amino acrd content in shoot fly-resistant lines than in susceptible ones. Susceptibility to shoot fly is positively correlated with phosphorus and negatively with total phenol content (Khurana and Verma 1983).

Spotted Stem Borer (*Chilo partellus*)

The first symptom of attack by spotted stem borer is leaf scarification and the presence of shot-holes caused by early-instar larval feeding in leaf whorls. Infested plants show a ragged appearance. The older larvae leave the whorl and bore into the stem at the base. Stem boring by the larvae in young plants (up to 1 month old) leads to destruction of the growing point and results in deadheart formation. In older plants the larvae feed inside the stem causing stem tunneling.

Resistance mechanisms

Nonpreference for oviposition. Ovipositional nonpreference is one of the components of resistance to *C. partellus* (Lal and Pant 1980; Dabrowski and Kidiavai 1983). More *egg* masses have been recorded on borer-susceptible genotypes than on the resistant ones. In cage tests, Saxena (1990) observed that oviposition was equally high on susceptible cultivars (IS 18363, IS 18463, and IS 2146), and moderately resistant cultivars (IS 4660 and IS 2205). However, oviposition was significantly lower on resistant cultivars (IS 18520 and IS 1044).

Antibiosis. The main mechanism of spotted stem borer resistance in sorghum is antibiosis. High mortality in the early larval stages (Jotwani 1978; Jotwani et al. 1978) and low survival rate of larvae (Lal and Pant 1980) have been reported in resistant genotypes. First-instar larval establishment, time interval between larval hatching and boring into the stem, larval mass, and survival rate are associated with resistance to stem borer (Taneja and Woodhead 1989). Saxena (1990) observed that larval establishment was 33% lower on the borer-resistant line, IS 1044, than on the susceptible control, IS 18363. Different combinations of factors are involved in conferring resistance to *C. partellus* in various genotypes, which is vital information in breeding for resistance to stem borers.

Tolerance. Jotwani (1978) reported significantly lower grain yield loss caused by stem borer in sorghum selections such as 124, 175, 177, 446, 447, 731, 780, 827 and 829 than in CSH 1, and attributed this to tolerance mechanism. Similar results were obtained in genotype IS 2205 by Dabrowski and Kidiavai (1983). In studies conducted at ICRISAT-Patancheru, lines showing resistance to deadheart formation, i.e., <20% plants with deadhearts (IS 5604, IS 5469, IS 2123, IS 5566, IS 2146, and IS 2309), also exhibited good recovery resistance (score <2). Recovery resistance to stem borers can be evaluated as described for shoot fly. Grain yield under infested and uninfested conditions can also be used as a measure of tolerance mechanism of resistance.

Factors associated with resistance

Plant morphological characters. Plant height, tassel percentage, stem thickness, number of leaves, leaf length, leaf width, leaf thickness, and leaf strength are negatively correlated with deadheart formation (Khurana and Verma 1985). Days to panicle initiation and shoot length are also associated with resistance to stem borers (Woodhead and Taneja 1987; Taneja and Woodhead 1989). Genotypes with early panicle initiation (IS 12308 and IS 13100) escape deadheart formation due to inability of the larvae to reach the growing point. Faster internode elongation is also associated with borer resistance, which is related to pushing the growing point upwards. This hampers the ability of larvae to reach the growing point, thus preventing deadheart formation.

Plant height at 10, 20, and 30 days after emergence (DAE), and seedling weight at 10 DAE, are negatively associated with leaf damage, deadheart formation, and larval survival. Shoot length at 40 DAE, ligular hairs, and leaf angle are significantly and negatively associated with deadheart formation. Moisture content of 10-day-old seedlings, and central whorl leaf at 20 DAE, are positively associated with leaf feeding and larval survival. Plant growth rate between 30 and 40 DAE and seedling vigor were negatively associated with deadheart formation. Long internodes affect the larval establishment in that the farther distance the larvae have to climb, the more they are exposed to desiccation, wash-off by rain, or attack by predators. Ligular hairs also act as a trap for the young larvae, thus reducing their success in climbing and rate of final establishment (Chapman et al. 1983).

Biochemical factors. A number of biochemical factors have been reported to be associated with stem borer resistance in sorghum. These include: low sugar content (Swarup and Chaugale 1962), amino acids, total sugars, tannins, total phenols, neutral detergent fiber (NDF), acid detergent fiber (ADF), and lignins (Khurana and Verma 1982, 1983, 1985), and high silica content (Narwal 1973). The epicuticular wax layer in sorghum plants is conspicuous and affects climbing by *Chilo* larvae (Bernays et al. 1983). On some resistant genotypes, there is a disorienting effect which has been attributed to the chemical composition of epicuticular wax (Woodhead and Chapman 1986). Concentration of 32 C marker chemical was less than half in resistant genotypes IS 2205 than in the susceptible genotypes, IS 1151 and CSH 1. Larval mortality is greater in diet impregnated with petroleum ether extract of the borer-resistant lines. Methanolic extracts from the susceptible line IS 18363 showed greater feeding stimulation than the extracts from the less susceptible cultivar, IS 2205. IS 18363 had greater phenolic and sugar contents than the less susceptible cultivar, IS 2205 (Torto et al. 1990).

Aphids (*Schizaphis graminum, Melanaphis sacchari,* and *Rhopalosiphum maidis*)

Greenbug, *S. graminum* feeds on the undersurface of leaves and secretes honeydew. The infested leaves begin to die, first turning yellow brown at the edges. Sugarcane aphid,

M. sacchari feeds on the undersurface of older leaves. The infested leaves become dry and turn brown. Under severe infestation, the plants become stunted. Infestation becomes severe by panicle initiation stage. Aphids secrete honeydew on which sooty molds grow. Corn leaf aphid, *R. maidis* feeds on young leaves in leaf whorls, and is rarely a pest.

Mechanisms of resistance

Nonpreference. This is an important component of resistance to greenbugs (Schuster and Starks 1973). Lara et al. (1981) reported that EA 266 shows nonpreference, IS 809 tolerance and antibiosis, and PI 202178 and PI 302236 show all three mechanisms of resistance.

Antibiosis. Antibiosis has been reported in PI 228928, PI 220248, IS 809, PI 302178, PI 302231, PI 2226096. PI 264953, PI 26695, KS 30, and SA 7536-1 (Schuster and Starks 1973; Teetes et al. 1974; Dixon et al. 1990). Fecundity decreases significantly with continuous rearing on resistant plants, and it is not immediately regained upon transfer to susceptible hosts (Starks and Schuster 1976). On resistant lines, greenbugs have a longer nymphal stadium, produce fewer progeny per female, and have shorter adult life span (Teetes et al. 1974).

Tolerance. This has been observed in J 242, PI 264453, and PI 229828 (Dixon et al. 1990). Nonpreference and tolerance are not influenced with continuous culturing of greenbugs on resistant plants. There is no information on mechanisms of resistance to other species of aphids infesting sorghum.

Factors associated with resistance

Imbibition of phloem sap is lower on aphid-resistant varieties (Campbell et al. 1982). Changes in nutrient supply affect the level of tolerance (Schweissing and Wilde 1979). Susceptibility to greenbug is also influenced by temperature. Differences between resistant and susceptible genotypes increase at higher temperatures (Schweissing and Wilde 1978, 1979). Genotypes with bloornless and sparse-bloom characters are nonpreferred by greenbugs (Weibel and Starks 1986; Weibel 1986a,b). These characters have an adverse effect on greenbug reproduction (Starks and Weibel 1981). Dreyer and Campbell (1984) suggested that increase in methylation of middle lamellar pectin hinders the penetration by aphids.

Spider Mites (*Oligonychus indicus* and *O. pratensis*)

Spider mites suck the sap from the undersurface of leaves. Infestation begins along the midrib of lower leaves. The infested leaves are pale yellow initially and later become red on the upper surface, and the entire leaf may turn brown. Heavily infested plants are

prone to lodging. SC 599-6 has been found to be tolerant to mites (Foster et al. 1977). Less mite damage has been reported on late-flowering lines (Perring et al. 1982), and more on senescing lines (Foster et al. 1977). But nonsenescence is not directly linked with resistance to mites (Archer et al. 1986). Leaf temperature influences the expression of resistance to spider mites in drought-resistant grain sorghum lines (Stiefel et al. 1992).

Shoot Bug (*Peregrinus maidis*)

The shoot bug sucks sap from the leaf whorls causing the leaves to become yellow red. Plants become stunted, and the top leaves begin to twist and dry first. The damage extends downwards. It is severe under dry conditions, and is a serious pest during the postrainy season. Honeydew excreted by the bugs favors the growth of sooty molds.

Mechanisms of resistance

It has been suggested that leaves tightly wrapped around the stem are associated with shoot bug resistance in sorghum (Agarwal et al. 1978). Antixenosis is a component of resistance to settling adults and nymphs in IS 18676, IS 19349, and IS 18677 (Chandra Shekar et al. 1993). Nonpreference for oviposition has been observed in IS 18676 and IS 19349.

Armyworms (*Mythimna separata* and *Spodoptera frugiperda*)

Armyworms are sporadic pests. The larvae feed on the leaves during the night, and hide in leaf whorls or in the soil/weed cover during the day.

Mechanisms of resistance

Antibiosis has been reported as a mechanism of resistance to *S. frugiperda* (Lordello et al. 1980). Tannin content of grain does not influence the development of fall army worm larvae (Wiseman et al. 1984). Chemical factors in the glumes show greater effect on survival and development of larvae (Wiseman et al. 1986).

Sorghum Midge (*Stenodiplosis sorghicola*)

Sorghum midge occurs in almost all the sorghum-growing regions of the world. The larvae feed inside the glumes on the developing ovary, which prevents normal grain formation. The damaged spikelets become chaffy.

Mechanisms of resistance

Nonpreference. Nonpreference for oviposition, or low oviposition, occurs because of closed glumes (Rossetto 1985). A short, shiny, and tight glume is the most important component of resistance to sorghum midge (Sharma 1985a; Sharma et al. 1990, 1991; Franzmann 1993). Fewer eggs are laid in the spikelets of midge-resistant genotypes (DJ 6514, AF 28, TAM 2566, and IS 15107) (<50 eggs 100^{-1} spikelets) compared with the midge-susceptible control, CSH 1 (153 eggs 100^{-1} spikelets). There are genotypic differences in their attraction to the midge females (Sharma and Vidyasagar 1994).

Antibiosis. Fewer midge flies emerge from the panicles of resistant cultivars compared with susceptible ones (Melton and Teetes 1984; Sharma et al. 1993b). The postembryonic developmental period (egg to adult) of the sorghum midge is prolonged by 5-8 days when reared on midge-resistant genotypes such as DJ 6514, IS 3461, IS 15107, IS 7005, etc. Antibiosis to midge is also expressed by the smaller size of larvae. The larvae remain smaller (<1.8 mm long and <1.0 mm in width) when reared on TAM 2566 and IS 10712, compared with those reared on CSH 11 (2.9 mm long and 1.6 mm in width). The mass of the larvae is also lower (<2.6 mg 10^{-1} larvae) on these genotypes compared with CSH 11 (5.3 mg 10^{-1} larvae). The fecundity of the midge females is also substantially reduced when reared on the midge-resistant genotypes. Low larval survival is also one of the components of resistance to sorghum midge (Melton and Teetes 1984; Rossetto 1985; Waquil et al. 1986; Sharma et al. 1993b).

Tolerance. There are conflicting reports on the compensation in grain mass due to damage by sorghum midge. Montoya (1965) reported slight compensation for midge damage. He observed that, as the mean percentage spikelet damage increased from 5 to 47%, the mass of 1000 undamaged grains increased from 30.3 to 35.1 g. Harris (1961) found no relationship between midge damage and the weight of surviving grains. Hallman et al. (1984) observed that there was a significant inverse relation between the midge damage and the mass of undamaged grains in two of the three susceptible hybrids, and three of the seven midge-resistant hybrids. However, the relations were not significant at damage levels below 40%. They suggested that, at economic threshold levels, there was no compensation for midge damage. Grain mass and volume were greater in the infested panicles than the noninfested panicles in hybrids based on midge-resistant females (PM 7061A and PM 7068A) than the hybrids based on the midge-susceptible females (ICSA 42 and 296A). Similar differences in grain mass and volume were also observed for the midge-resistant and midge-susceptible restorers. It appears that midge-resistant genotypes have a better capability for compensation in grain mass than the midge-susceptible ones.

Factors associated with resistance

Glume and grain Characters. Short, shiny, tight, and light yellow glume characters are associated with resistance to sorghum midge (Fig. 4). Rate of grain development

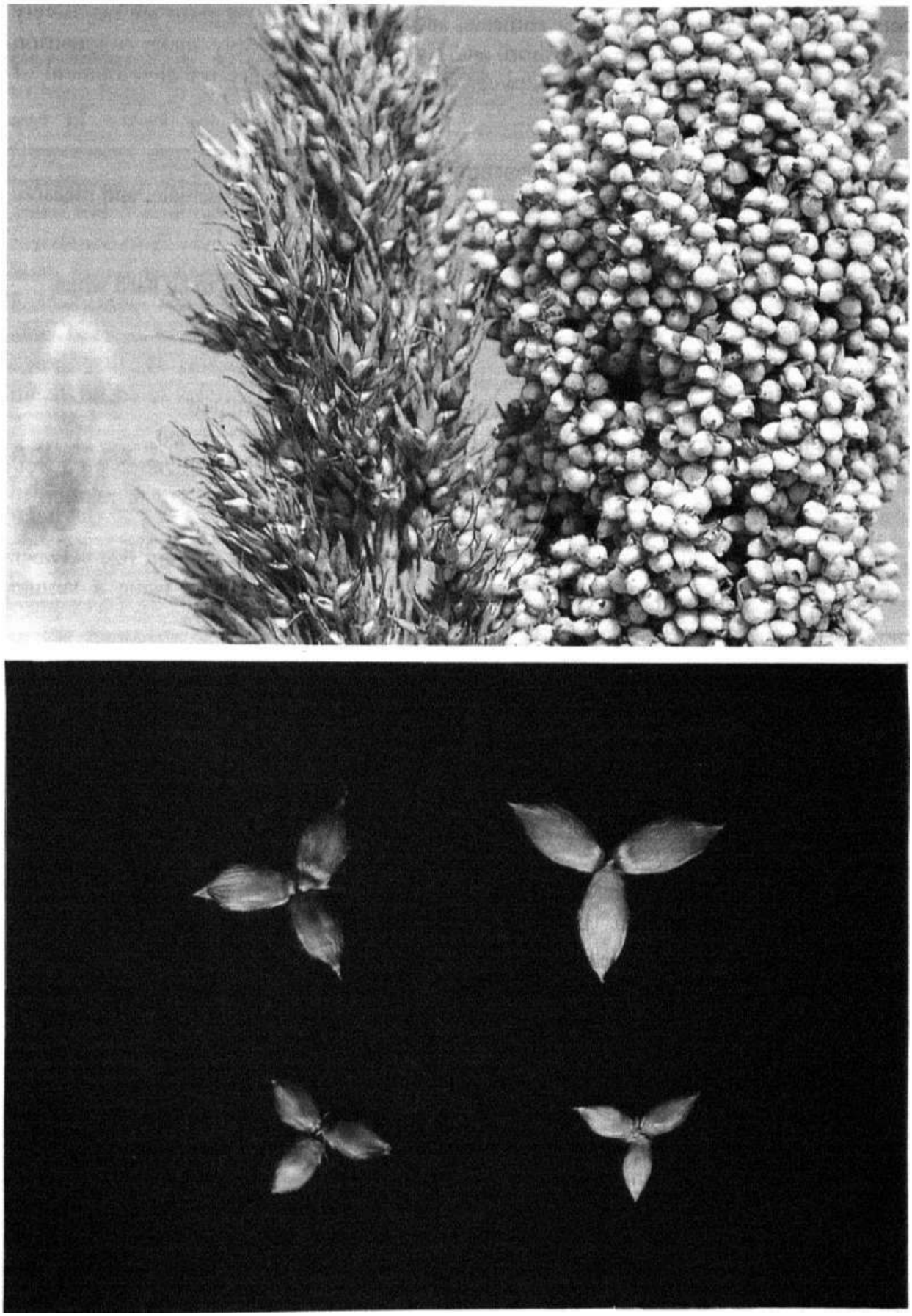

Figure 4. Glume size of midge-resistant (short) and susceptible (long) sorghum genotypes.

between the 3rd and 7th day after anthesis, and tannin content of grain are negatively associated with midge damage. Short and tight glumes possibly make oviposition difficult, and leave limited space between glumes and the ovary for development of midge larva.

Evaluation of grain and glume characters

- Collect glume samples at anthesis from the middle portion of the panicle, and measure glume length and width.
- Glume characters can be evaluated visually on a 1 to 5 scale:
  1 = glumes <2.5 mm long, light yellow, shining, tight, and appear to be hard when pressed between the thumb and fingers;
  2 = glumes 2-3 mm long, light yellow, shining, and tight;
  3 = glumes 3-4 mm long, light green, and medium-hard;
  4 = glumes 4-5 mm long, green, and slightly soft when touched; and
  5 = glumes >5 mm long, dull green, and appear to be soft when touched.
- Measure grain mass between the 3rd and 7th day after anthesis/fertilization on a wet- or dry-weight basis. Compute the rate of grain development per day in relation to mean mass of the grain during the observation period.
- Rate of grain development can be measured in terms of increase in grain size between the 3rd and 7th day after anthesis. Glume/grain size is measured using a Vernier calliper, or a specially designed grain size meter (Sharma et al. 1991).

Biochemical factors. Santos and Carmo (1974) suggested that tannin content of grain may be one of the factors imparting resistance to sorghum midge. Tannins have been found to be greater in some midge-resistant lines than in susceptible ones, while the soluble sugars are lower in midge-resistant lines (Sharma et al. 1993b). However, there are distinct exceptions, e.g., DJ 6514 is highly resistant to the midge, but has low tannin content. Tannins may act as antifeedants and/or produce antibiotic effects on the midge larvae. The tannin content of grain is negatively associated with adult emergence and fecundity. The sugar content of 10-day-old grain is positively associated with adult emergence and the fecundity of the emerging females. Chemical composition of the sorghum grain varies over seasons, and these changes have been linked with the variation in expression of resistance to midge (Sharma et al. 1993b).

Head Bugs (*Calocoris angustatus* and *Eurystylus oldi*)

Head bugs are important pests of grain sorghum in Asia and Africa. *C. angustatus* is the most important species in India, while *E. oldi* is the most damaging bug species in western Africa. Grain damaged during the early stages of development under heavy infestation becomes completely shriveled, while lower infestation levels or late infestations lead to tanning and browning of the grain. This leads to both quantitative and qualitative losses. The bug-damaged grain is infected by the grain molds, and becomes unfit for human consumption.

Mechanisms of resistance

Nonpreference. Nonpreference for feeding is one of the components of resistance to head bugs. In mulli-, double-, and no-choice tests, IS 2761, IS 17610, IS 17618,, and IS 17645 are not preferred by *C. angustatus* (Sharma and Lopez 1990). Oviposition nonpreference is another component of resistance to head bugs. The percentage of spikelets with head bug eggs were lower in IS 2761, IS 6984, IS 17645, and CSH 5 compared with Swarna. Cultivar nonpreference is also a component of resistance to *E. oldi.* IS 14332, CSM 388, Malisor 84-7, $83F_6$-16, and $83F_6$-111 have been found to harbor <5 females panicle^{-1}, compared with 11 females panicle^{-1} in E 35-1 under free-choice conditions in the field. The nonpreference of CSM 388 has also been confirmed in cage tests in the laboratory (Sharma et al. 1994). This component of resistance to head bugs can be used in conjunction with other mechanisms of resistance.

Antibiosis. Postembryonic development of *C. angustatus* is prolonged by 1-2 days on IS 17610, IS 17618, and IS 17645. Survival and establishment of 1st-instar nymphs is relatively lower on IS 17610 and IS 17645 as compared with that on the susceptible controls, CSH 5 and CSH 9 (Sharma and Lopez 1990, 1993). Growth rate and efficiency of conversion of ingested food into body matter are lower on IS 6984 and IS 2761 as compared with CSH 5 (Sharma and Lopez 1990). A marginal decrease has been observed in the fecundity of head bug females when reared on head bug-resistant genotypes (IS 2761, IS 14334, IS 16357, IS 20740, and IS 17610) compared with those reared on the susceptible control, CSH 1, over 3 generations (Sharma et al. 1993a).

Tolerance. Tolerance to head bug feeding is greater in IS 9692, CSH 1, IS 17645, and IS 17610 compared with IS 2761, IS 6984, and CSH 9 (Sharma and Lopez 1993). CSH 1 and CSH 5, although susceptible to head bugs, are more tolerant of bug feeding compared with CSH 9. The former also suffered lower loss in grain yield than CSH 9.

Factors associated with resistance

Panicle compactness. Genotypes with loose panicles are in general less susceptible to head bugs. Grain damage and bug population increase are positively associated with panicle compactness (Sharma 1985b; Shanna et al. 1994).

Glume and grain Characteristics. Cultivars less susceptible to *C. angustatus* tend to have long, hard, and less hairy glumes (Sharma 1985b). Days to glume opening (>20 days from anthesis), longer glumes (>5 mm), >50% of the grain surface covered by the glumes, hard corneous grain, and quicker grain ripening contribute towards resistance to *E. oldi* (Sharma et al. 1994). Toure et al. (1992) reported that faster rate of grain filling, low waten:carbohydrate ratio in the grain, grain hardness, glume length, and days to glume opening are the major factors contributing to the resistance of Malisor 84-7 to *E. oldi.*

Panicle-Feeding Caterpillars (*Helicoverpa,Eublemma, Cryptoblabes, and Pyroderces*)

Panicle-feeding caterpillars feed on the developing grain inside the panicle. The inside of the panicle is converted into frass consisting of fecal matter and silken webs. Genotypes with loose panicles suffer little damage by panicle-feeding caterpillars, possibly because of easy access for parasites and predators (Balasubramanian et al. 1979).

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Inheritance of Resistance to Insect Pests in Sorghum

Faujdar Singh

Introduction

This paper discusses the inheritance of resistance to insects vis-a-vis the development of appropriate strategies to breed for resistance to insect pests in sorghum.

Genetic basis of resistance

In classical genetics, resistance may be oligogenic, polygenic, or cytoplasmic.

- Oligogenic resistance is due to a major gene (monogenic), or a few genes. The gene has a distinct effect. Therefore, it is easy to detect and transfer such resistance to another genotype.
- Polygenic resistance is controlled by many genes, each with a small but cumulative effect. The effect of the individual gene is difficult to detect unless all the genes contributing to resistance are present in the genotype.
- Cytoplasmic effects are due to self-duplicating and mutable substances in the cytoplasm; it is therefore inherited through the female parent.

Expression of resistance and insect biotypes

Expression of resistance to insects can be judged at different growth stages of sorghum plants in the presence of insects under favorable conditions for their growth and multiplication. It is difficult to assess the resistance/susceptible reaction unless the three components, i.e., host, parasite (insect), and conditions favorable for insect multiplication are assured either by artificial or natural infestation.

Climatic and edaphic factors such as soil moisture, drought stress, and insect populations also contribute towards the expression of resistance/susceptibility to insect attack. High soil fertility leads to faster plant growth, thus providing apparent resistance to insects, when plants may otherwise suffer high levels of insect damage. In general, low temperatures have a negative effect on resistance to insects.

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Expression of resistance is also influenced by the presence of insect biotypes, number of genes, environment, and interaction of all these factors. Biotypes are the strains of the same insect species that differ in their ability to attack different genotypes of the same host species. The genetic basis of resistance may be vertical, or horizontal.

Vertical resistance. This is a monogenic or oligogenic resistance. Host genotypes with this type of resistance show resistance to one biotype. Therefore, it requires a series of differential cultivars (pure lines/isogenic lines) to detect the biotypes. The same cultivar exposed to different biotypes of an insect shows a differential reaction. In other words, the same cultivar could be resistant to one biotype, but susceptible to the other biotype. Therefore, such a resistance is also termed biotype-specific resistance.

Horizontal resistance. This is controlled by many genes. The same cultivar shows resistance to different biotypes of an insect, i.e., the level of resistance of a genotype is similar against all/many biotypes of the same insect. It is also called biotype-nonspecific resistance (general resistance, or field resistance, or stable resistance).

Genetic parameters as a basis of resistance

Host-plant resistance to insects is evaluated on a rating scale of 1 to 9, by percentage of damaged plants, infestation severity, or loss in yield, or other economic factors. Various genetic parameters can be computed for-experiments conducted using a suitable design. Different generations of the host are evaluated/tested for their reactions (resistance or susceptibility) to insects under favorable situations for insect infestation, i.e., either natural or artificial.

Fisher (1918) partitioned the genetic variances into additive, dominance, and their interactions, i.e., additive x additive, additive x domipance, and dominance x dominance, called epistasis. These parameters provide the basis for selection and improvement of crop plants for resistance to insects.

Additive gene effects. This is due to the average effect of alleles at the same locus. Each gene has a small but cumulative effect. It is also called the average effect of genes. If this effect is high, selection for resistance can be highly effective. Besides the direct estimation of this effect by using parents and segregating generations, it is also estimated by general combining ability (GCA) variance. High GCA variance indicates a high additive gene effect. This parameter can be effectively used for selection purposes because it indicates the degree to which progeny are likely to resemble the parents.

Dominance. This term is applied when a member of an allelic pair has the ability to manifest itself to the exclusion of the expression of the other allele. It is due to intra-allelic interaction of gene effect. This type of resistance could therefore be useful for the development of hybrids by crossing diverse parents. When dominant effects govern the expression of resistance to insects, it is not possible to go for direct selection, as in the case of additive gene effects.

Epistatic gene effects. These are due to the interaction of alleles at different loci. In broad terms, any deviation from additive and dominance gene effects is called epistasis. As noted above, there could be additive x additive, additive x dominance, and dominance x dominance interactions. Gene effects other than additive are called nonadditive. All these effects are useful for calculating genetic parameters such as heritability, genetic advance, and degree of dominance.

Heritability. This is a measure of the transmissibility of a character that provides the value of selection for the character. In a broad sense, it is expressed as a proportion of genetic variance to phenotypic variance. In a narrow sense, it is a proportion of additive genetic variance to phenotypic variance.

Genetic advance. This comprises improvement in the performance of selected lines over the original population. It is estimated by using selection differential, phenotypic standard deviation, and heritability.

Correlations and path analysis. These parameters express the positive, or negative, relation of one character with another. These are helpful in selection, e.g., when two characters are positively correlated, and both characters are desirable, selection for one character will automatically improve the other character. Such a selection can also be carried out using a selection index. It is therefore important to understand the genetic basis of resistance to insects to facilitate selection, and to achieve faster progress. Many designs are available for estimation of these parameters. Systematic planning and experimentation to estimate the genetic basis of resistance are necessary to draw valid conclusions. The following points may be helpful in conducting such experiments.

- Desirable material, i.e., pure parents and their crosses (F_1, F_2, and other generations).
- A suitable environment, artificially created, or hot-spot locations for insect development, and suitable methods for scoring/damage assessment.
- Proper design of the experiment and the recording of data.
- Analysis of data using transformation techniques, if applicable, and the correct interpretation of results.
- Experimentation over years, or over locations, in order to obtain authenticity in the results and conclusions.

Inheritance of Resistance to Insects in Sorghum

The various mechanisms that contribute to resistance to specific insects in sorghum have already been discussed. Information available on the inheritance of resistance to major insects and their use in future sorghum improvement are discussed below.

Sorghum shoot fly (*Atherigona soccata*)

Both additive and nonadditive gene effects are important for inheritance of resistance to shoot fly in sorghum. It is quantitatively inherited, predominantly by additive gene effects

(Indra et al. 1972; Nimbalkar and Bapat 1992). High broad-sense heritability (52-82%) for resistance has been reported (Sharma et al. 1977).

Resistance to shoot fly is predominantly controlled by nonadditive genes (Agrawal and Abraham 1985). Hybrids (F_1) showed an increase in resistance over the midparental value under low infestation, but the reverse was true under high infestation (Sharma and Rana 1983). Eggs per plant showed nonadditive gene effects controlled by two dominant genes (Nimbalkar and Bapat 1992).

Both additive and nonadditive gene effects have been found to be important in a line x tester analysis for shoot fly resistance. Nonadditive gene effects are important for leaf-surface wetness with low-to-moderate heritability. Glossiness exhibited high heritability and genetic advance, and strong correlation with deadheart percentage, followed by trichome density on the leaf abaxial surface (Jeewad 1993).

The percentage of eggs laid on plants on the 14th day after seedling emergence are positively correlated with the number of deadhearts. Leaf trichome density and plant height are negatively correlated with the number of deadhearts. Trichome density and seedling vigor can therefore be used to select for resistance to shoot fly (Karanjkar et al. 1992).

Path analysis has shown that selection for shoot fly resistance should aim to reduce drooping depth of leaf, and increase glossiness intensity, and early plant height in tall types. In dwarfs, increasing trichome density, glossiness intensity, and leaf length (without increasing droopiness, or reducing early plant height) would be desirable. A judicious emphasis on glossiness, early plant height (vigor) should be given for selection of genotypes for resistance to shoot fly and high yield (Vijaya Laxmi 1993).

Spotted stem borer (*Chilo partellus*)

Genotypic resistance to spotted stem borer is poorly expressed under low fertility, drought, and unfavorable weather conditions (Sharma 1993). The stage of infestation is very critical for the expression of resistance, as a progressive delay in infestation reduces the production of deadhearts (Taneja and Leuschner 1985).

Resistance is quantitatively inherited (Rana and Murty 1971). The general combining ability (GCA) effects are predominant for leaf feeding, whereas specific combining ability (SCA) effects are important for stem tunneling. Resistance to leaf feeding, deadheart formation, and stem tunneling are inherited in different ways (Singh et al. 1983). Both additive and nonadditive gene effects are important, but additive gene effects are predominant for deadheart formation and leaf injury (Singh and Verma 1988).

Leaf feeding, deadhearts, and stem tunneling are polygenic traits. Both additive and nonadditive genes are important for inheritance of stem borer resistance. However, additive gene effect is more important (Pathak 1990).

The inheritance of characters associated with resistance to stem borers, such as early panicle initiation (Taneja and Woodhead 1989), ligular hairs (Woodhead and Taneja 1987), low sugar and high amino acids and high tannin, total phenols, neutral detergent fibers, and acid detergent fibers (Khurana and Verma 1982, 1983), and high silica content (Narwal 1973) is not known.

Aphids (*Schizaphis graminum and Melanaphis sacchari*)

A single dominant gene controls resistance to sugarcane aphid, *M. sacchari* (Chang 1981; Hagio 1992). Both additive and dominance variances are significant with predominance of additive gene effects. The complementary gene effects have also been reported (Hsieh and Pi 1988). Inheritance shows incomplete dominance (Tan et al. 1985). Partial dominance exists for both antibiosis and tolerance to greenbug, *S. graminum.* GCA, SCA, maternal, and specific reciprocal effects are significant for seedling antibiosis and tolerance, but GCA effects are more important in determining tolerance (Dixon et al. 1990).

Virulence of *S. graminum* biotype F-resistant "Piper" is dominantly inherited and governed by duplicate dominant genes, and is influenced by a third modifier gene. Virulence to biotype C and E is also controlled by a duplicate dominant gene modifier (Puterka and Peters 1995). No information is available on the inheritance pattern for resistance-contributing factors, such as increased methylation in middle lamellar pectin which hinders penetration by aphids (Dreyer and Campbell 1984).

Sorghum midge (*Stenodiplosis sorghicola*)

Resistance to sorghum midge is inherited quantitatively, and is controlled by additive genes and some cytoplasmic effects (Widstrom et al. 1984; Agrawal et al. 1988). Susceptibility to midge is completely or incompletely dominant in some parents. At least two pairs of recessive genes determine the resistance in genotype AF 28, and genes with minor effects are also present. Similarly, Tift MR 88 genotype also has recessive gene control for midge resistance (Hanna et al. 1989).

Both additive and nonadditive gene effects are important for midge resistance because estimates for GCA and SCA variances are significant (Patil and Thombre 1985). Resistance to sorghum midge is governed by additive genes. The GCA effect is more important than the SCA effect. Glume length shows negative and significant GCA effects, while glume hardness and glume hairiness in genotype PM 7061B had significant and positive correlation. Similarly, glume length, glume hairiness, and glume hardness in ICSB 42 had a significant correlation. Resistant x resistant parents are needed to produce midge-resistant hybrids (Sharma et al. 1996).

Genetics of morphological traits associated with midge resistance such as the degree of apposition of glumes (Geering 1953), closed spikelets (Rossetto et al. 1984), and short and tight glumes that hinder oviposition and limit the space between glumes and ovary for the development of midge larvae (Sharma 1993), need to be determined. Other traits, such as short and thick floral parts, and faster rate of grain development, and a high grain tannin content (associated with resistance to midge) could be useful criteria by which to select for resistance to sorghum midge (Sharma et al. 1990).

Head bugs (*Calocoris angustatus* and *Eurystylus oldi*)

Information on the inheritance of resistance to head bugs is scanty. The morphological factors associated with head bug resistance/tolerance are colored grain/high tannin and

Table 1. Morphological characteristics associated with insect resistance and their inheritance.

Character	Insect	Genetics
High trichome density	Shoot fly	Presence of trichomes is recessive (*tr*) to normal (*Tr*) on abaxial leaf surface (Gibson and Maiti 1983; Maiti and Gibson 1983).
Glossy leaf	Shoot fly	Recessive (*gl*) to normal (Tarumoto 1981). High heritability and genetic advance, highly correlated with deadheart (%), and trichome density (Jeewad 1993).
Seedling height	Shoot fly	Tall dominant (*Dw1*, *Dw2, Dw3,* and *Dw4)* to short (Doggett 1988).
Internode length	Shoot fly	Governed by four major genes (*Dw1*, *Dw2, Dw3,* and *Dw4).* Long internode partially dominant and the effect of four *Dw* genes is additive (Quinby and Karper 1954).
Cleistogamous glumes	Midge	Cleistogamy is due to rolling of papery inner glumes. The rolled glume condition is dominant to unrolled. It is controlled by two genes with epistatic effects (Merwine et al. 1981).
Bloomlessness	Aphids	Presence of bloom is dominant to its absence (*bm*) on leaf sheath as well as on internodes (Ayyangar and Ponnaiya 1941).
Narrow angle between leaf and stem	Stem borer	Broad leafjunction dominant to narrow (*Jb*) (Ayyangar 1942).
Epicuticular wax	Stem borer	Waxy bloom dominant to sparse bloom (Ayyangar et al. 1937).
Loose panicles	Head bug and head caterpillars	Loose panicle dominant (*Pa*) to compact panicle (Ayyangar and Ayyar 1938)
Long glumes	Head bug	Long glume recessive to short glume (Vinall and Cron 1921). Small glume strongly but incompletely dominant with epistatic effects (Jowett 1968).
Hairless glume	Head bug, midge	Glabrous glume is recessive to hairy (Ayyangar andPonnaiya 1941).
Colored grain/ high tannin	Head bug, midge	Red (R) grain pericarp dominant to yellow (*rrY*) and white (-yy) (Graham 1916).

loose panicles (Sharma et al. 1994), less hairy glumes, days to glume opening (>20 days from anthesis), longer glumes (>5 mm), more than 50% of grain surface covered by the glumes, hard and corneous grain, and fast grain ripening (Sharma et al. 1994). The genetics of some of these traits need to be worked out.

Other pests

Genetic information on spider mites (*Oligonychus indicus* and *O. pratensis*), shoot bug (*Peregrinus maidis*), and chinch bug (*Missus leucopterus*), armyworms (*Mythimna separata* and *Spodoptera frugiperda*), and head caterpillar (*Helicoverpa armigera*) in sorghum is not available. The traits contributing to resistance to these insects have been summarized by Sharma (1993).

The genetics of morphological characters associated with resistance to insects in sorghum has been summarized in Table 1.

Genetic information on traits other than those listed in Table 1 that contribute towards resistance to shoot fly, such as stem hardness, peduncle length, leaf wetness, early appearance of silica bodies in leaf, absence of lysine, high amino acids, phosphorus, and phenolic compounds, is not available. Inheritance of resistance mechanisms to midge, such as degree of apposition of glumes, short and tight glumes, faster grain development, and tannins is not fully understood.

There is no information on the inheritance of such characters as fast internode elongation, ligular hairs, low sugar, high amino acids, tannins, and phenols that provide resistance to stem borers. Factors associated with resistance to head bugs, such as panicle compactness, days to glume opening, grain hardness, and fast grain ripening need to be studied in greater detail. Tight leaf wrapping of the stem associated with shoot bug resistance (Agrawal et al. 1988), and high tannin content provide resistance to *S.frugiperda* (Diawara et al. 1991). Inheritance of these characters needs to be worked out.

There is a lot of scope for genetic studies on the inheritance of resistance to insects in sorghum. This research will be helpful in shaping the future of breeding programs for sorghum improvement.

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Part 4

Breeding Methodologies

Breeding Sorghums for Insect Resistance

Belum V S Reddy, H F W Rattunde, and J W Stenhouse

Introduction

Sorghum is widely grown in the semi-arid tropics for food, feed, fodder, and forage. Shoot fly (*Atherigima soccata*), stem borer (*Chilo partellus*), midge (*Stenodiphsis sorghicola*), and head bugs (*Calocoris angustatus* and *Eurystylus oldi*) are important pests of sorghum. Insect pests are a major limiting factor in sustainable agriculture production in the semi-arid tropics where modern agricultural inputs are not easily accessible. Plant resistance to insect pests is an important component of integrated pest management in sorghum. Breeding for host-plant resistance involves no additional cost to farmers. It is an important aspect of the crop improvement strategy at ICRISAT-Patancheru.

Target materials

In sorghum, heterosis for grain yield is well established. It became possible to produce hybrid seeds cheaply because of availability of the cytoplasmic-genic male-sterile (cms) system (Stephens and Holland 1954). Improvement of male-sterile and restorer lines is important in producing high-yielding hybrids for commercial cultivation. In developing countries such as India, Argentina, Brazil, Mexico, and China, and in developed countries such as USA and Australia, the target materials are hybrids. In Africa, where the seed industry is not well established, varieties are most important. Breeding of restorers, or varieties, male-sterile lines, and hybrids with resistance to insects is therefore described in this paper. Also, broad-based population or gene pools comprise intermediate forms from which the target material (restorers/varieties, or male-sterile lines, and eventually hybrids) may be developed through appropriate breeding methods.

There are different breeding methods for improving various characters of sorghum, including resistance to insect pests (Allard 1960; Eberhart 1970; Gardner 1972). Each method can be modified to suit local needs. The number of genes governing a character primarily decides the choice of a breeding method. The pedigree method can be used for transferring the resistance governed by a large number of genes. The backcross method is used when resistance is under the control of a few genes (two to four). Inheritance of resistance to shoot fly, stem borer, midge, and head bugs is complex, and under the control of a large number of genes. In all these situations it is better to use the pedigree method. If resistance mechanisms

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Reddy, Belum V.S., Rattunde, H.F.W., and Stenhouse, J.W. 1997. Breeding sorghums for insect resistance. Pages 115-126 *in* Plant resistance to insects in sorghum (Sharma, H.C., Faujdar Singh, and Nwanze, K.F., eds.). Patancheru 502 324, Andhra Pradesh, India: International Crops Research Institute for the Semi-Arid Tropics.

are controlled by relatively few genes, then the backcross method of breeding can be adopted, e.g., short, tight, pale green glumes are associated with midge resistance, and loose panicles with head bug resistance, etc. These are under the control of a few genes.

On the other hand, when resistance is controlled by many genes distributed in many source lines, population improvement methods are useful to bring these genes together.

Thus, it is important to know the factors/mechanisms contributing to resistance and the genetics of resistance mechanisms in order to select the most appropriate method of breeding. The genetics of various traits associated with resistance to insects is discussed in Part 3 above.

Selection of parents

Breeding programs aimed at developing materials resistant to insects have additional objectives such as improving the yield, grain characters, and fodder quality. Lines having high levels of resistance in appropriate agronomic backgrounds should be selected as the source materials. The resistant source lines are usually restorers, while the high-yielding lines are available either in a maintainer or a restorer background. Some high-yielding restorers are: ICSRs 30, 103, 105, 107, 117, 112, 144, 146, 154, 160, 89016, 89028, 91026, 91034, 92026, 92027,93005, and 93009. High-yielding maintainers are: ICS 8,9, 11, 30,44, 2968, etc.

Resistant source lines with desirable agronomic characters that have been used as parents in resistance programs at ICRISAT-Patancheru are:

Shoot fly : ICSV 705, PS 30715-1,ICSV 708, and PS 35805.
Stem borer : IS 18432, ICSV 200, and ICSV 702.
Midge : ICSV 197, PM 17467, PM 7061, and ICSV 745.
Head bug : Malisor 84-2. Malisor 84-7 and other selections of Malisor 84-7.

A stepwise breeding process, i.e, to continue breeding for resistance to any one or two insect pests with high grain yield and agronomic desirability has been adopted at ICRISAT-Patancheru. *A guide to sorghum breeding* by House (1985) provides further details on various aspects of crop flowering and crossing procedures.

Selection criteria

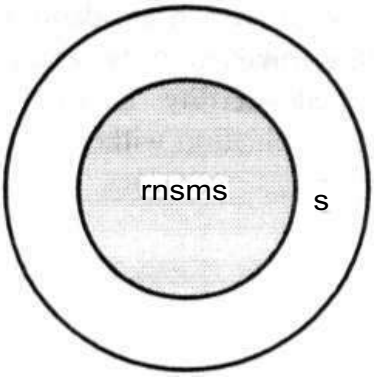
In resistance breeding programs, the following are the criteria used in addition to yield and agronomic desirability:

- shoot fly: deadheart %, leaf glossiness, and trichome density;
- stem borer: leaf feeding, and deadheart %;
- midge: chaffy spikelets %, and glume characters (small, thick, leathery);
- head bug: grain damage (shriveled and chaffy grain), and panicle compactness.

Measurements of these selection criteria, and related screening procedures, are described in Part 2 above.

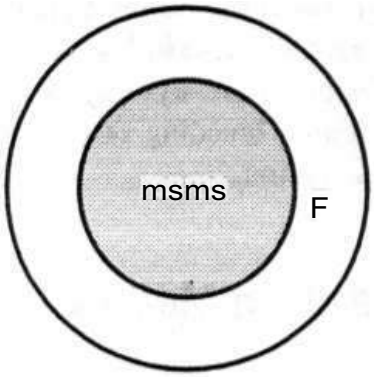
Restorers (or varieties), maintainers, and populations

Restorers are the lines that restore fertility on male-sterile lines. These are usually taller (up to 0.5 m) than male-sterile lines. They should have high pollen shedding ability, for



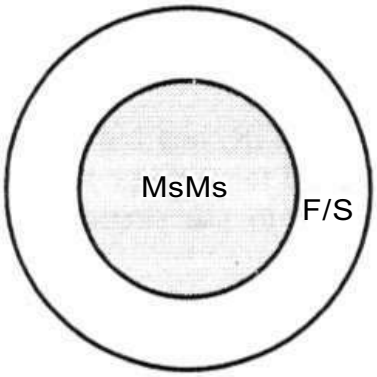
1. Male-sterile plant/line

(A-line)



2. Maintainer plant/line

(B-line)



3. Male-fertile plant/line

(R-line)

Ms: Gene for fertility restoration

ms: Gene for male sterility; an alternative form of Ms gene

S : Sterile cytoplasm

F : Fertile cytoplasm; an alternative form of 'S'

Figure 1. Interaction between cytoplasms and nuclear genes causing male-sterility in sorghum.

use as pollinators in a hybrid program. In addition, they should have high performance for grain yield and combining ability. Restorers with high yield and other desirable agronomic traits may also be released as varieties.

Maintainers are the lines that maintain male-sterility in cytoplasmic-genic male-sterile lines. These are usually short (1.0-1.5 m) and must have high grain yield and good combining ability. Male-sterile lines are called A-lines, maintainer lines B-lines, and restorer lines R-lines. The genetic constitution of these lines is represented diagrammatically in Figure 1. The *msms* nuclear genes with S-cytoplasm lead to male-sterility, and those with fertile cytoplasm make the lines male-fertile, which can then be used to maintain male-sterile lines. The *MsMs* genes (alternative forms of *ms* genes) are fertility-restorer genes that restore fertility in hybrids. This system is used in producing hybrids on a large scale for cultivation (Stephens and Holland 1954).

A population is a group of individual plants sharing a common gene pool. The sorghum flower is bisexual, self-pollinated, and some outcrossing (5-20%) is caused by

wind. However, some mutant male-sterile genes *(ms_1,ms_2 ... ms_8)* have been identified, and each of these in homozygous condition contributes to genetic male-sterility, which is different from cytoplasmic-genic male-sterility. Genetic male-sterility is used in population breeding programs to provide opportunities for recombination without hand-emasculation.

Breeding Methods

Pedigree method

This is by far the most suitable method of breeding for insect resistance. It provides an opportunity to 'fix' new gene complexes into the final selection. It is relatively easy to operate, and the method is used to develop restorers/varieties and maintainer lines.

Breeding insect-resistant restorers/varieties

The breeding methodology is outlined schematically in Figure 2. Eight major steps involved are :

1. Single or three-way crosses are made between insect-resistant (R) and high-yielding parent(s). Three-way crosses [e.g., (R_1 x S_1) x S_2] can be used for a midge-resistance breeding program, while in all other cases it is advisable to use single crosses. Advance the F_1s to F_2s.
2. Grow large F, populations (2 000 plants) with or without artificial infestation. Select for highly heritable traits such as days to flowering and plant height. If the test plants are grown under insect infestation, ensure they are exposed to low levels of infestation. Selection can be carried out among plants with low insect damage, or plants free from insect damage.
3. From F_3 to F_6, grow the families in nurseries under artificial infestation, or at hot-spot locations. Families should be interspersed with an appropriate resistant control and a susceptible control at every 10 plots.
4. Evaluate the material for desirable morphological characters and use resistance index (RI) values in selecting for insect resistance.

$$RI = \frac{X-(CI+C2)/2}{1+(C1xC2)/2} \times 100$$

where X = percentage deadhearts (or the measurement criteria) in the test plots; and C1 and C2 = percentage deadhearts (or the measurement criteria) in the two adjacent resistant control plots. The RI takes care of the variability in pest incidence in different plots (there is evidence, for example, that there are more deadhearts in the susceptible control in blocks with a large number of susceptible genotypes than in blocks with a larger number of less-susceptible genotypes). Genotypes with low RI values are considered to be more resistant. It is better to confine the selection for resistance specifically to season of adaptation, especially for shoot fly.
5. Select individual plants for agronomic desirability and high yield within the selected families.

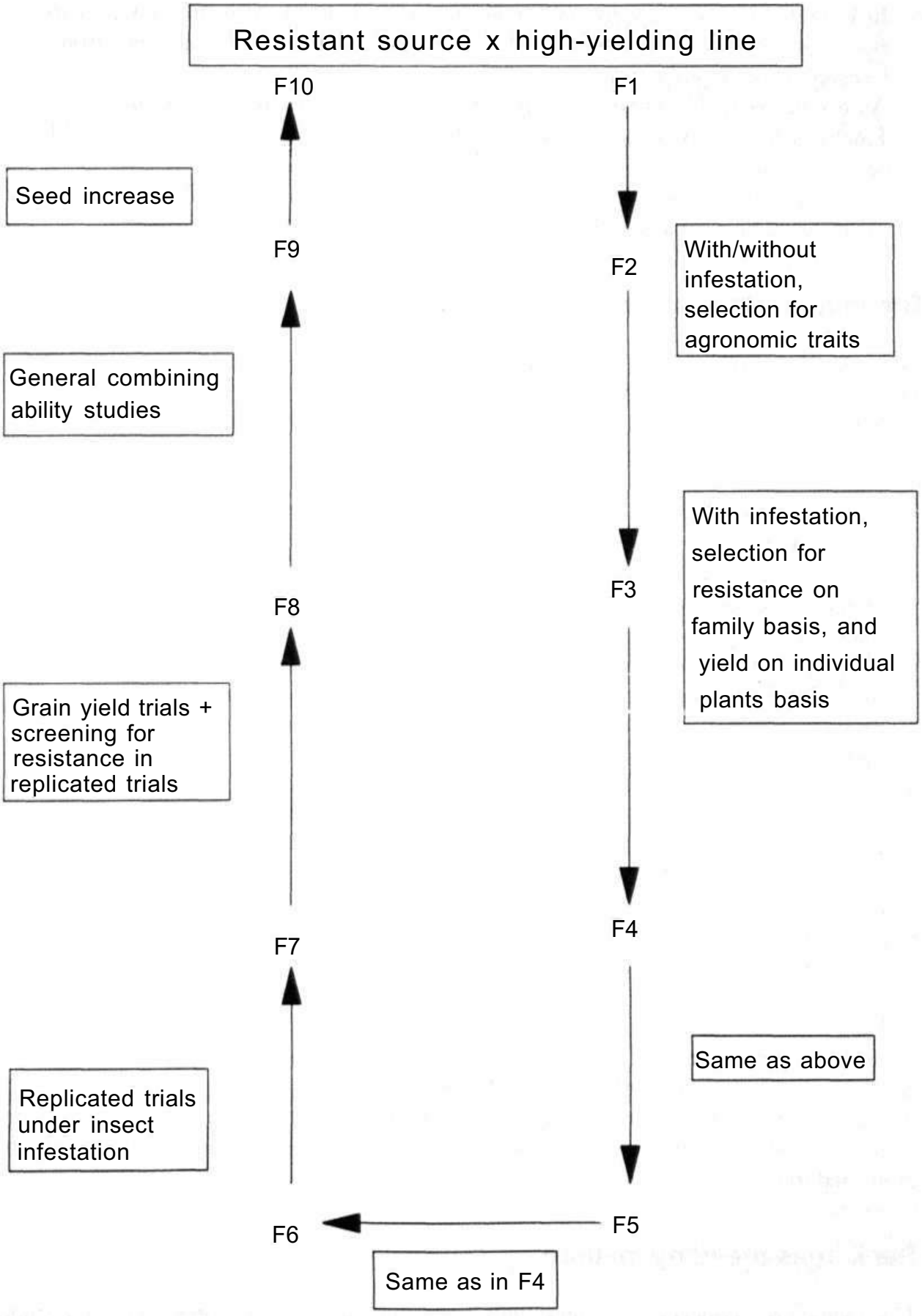


Figure 2. Development of insect-resistant restorer lines in sorghum by the pedigree method.

6. In F_6 or F_7, assess resistance and grain yield potential separately in replicated trials. Select entries based on means: first for the RI and then for grain yield from the selected resistant progenies.
7, Assess the general combining ability (GCA) of the progenies in a suitable design. Finally, select the progenies with high GCA for resistance and grain yield, and high resistance and grain yield potential.
8. Check fertility restoration in the pure lines thus developed by test-crossing these onto male-sterile lines, and also check seed-set under bagging in the test crosses.

Breeding male-steriles

The procedure followed at ICRISAT-Patancheru is given schematically in Figure 3. Various steps in this process are as follows:

a) Same as in Step 1 above, except that at least one of the parents chosen should be a male-sterile maintainer line.
b) Same as in Step 2 above.
c) Same as in Step 3 above, but, in addition, make individual plant test crosses involving a stable male-sterile line and three or four plants selected for agronomic desirability from each of the selected families in F_4 or $F_{5'}$ Assess the pollen parents' sterility maintenance ability by sowing the test crosses under protection in a separate nursery, but adjacent to the screening nursery. Backcross the test crosses with male-steriles by taking pollen from one or two individual plants from the respective pollen parents. In the following seasons, a male-sterile plant nursery should be sown separately under protection from the nearby screening nursery. Further, backcrossing should be taken up to convert the selections to male-steriles.
d) Advance the material as explained under steps 4 to 7 above.

The proportion of maintainer line genes brought into A-line cytoplasm with each backcross generation is given in Table 1.

The RI-based selection procedure may be used in breeding for low-heritable traits such as resistance to shoot fly and stem borer. Measurements taken in breeding for resistance to midge and mirid bug may be used directly without converting them into RI values, because these are often based on scores taken on a 1 to 9 scale.

Plot size can be 20-30 rows of 9-m length in F_2, 6-8 rows of 4-m length in F_3, and 2-4 rows of 4-m length in F_4 and above. Care should be taken to thin plots to a uniform stand. In each generation, about 20% of the families can be selected based on RI values, and 2-4 plants within the selected F_3 or F_4 families based on grain yield and agronomic desirability. From F_5 onwards, only one (or two) plant(s) can be selected from the selected families. Selection for resistance and grain yield is carried out based on replicated trials from F_6 or F_7 onwards.

Backcross breeding method

This method does not offer an opportunity to provide new recombinants; thus we cannot fix them. It requires considerably more time than the pedigree method. It consists of

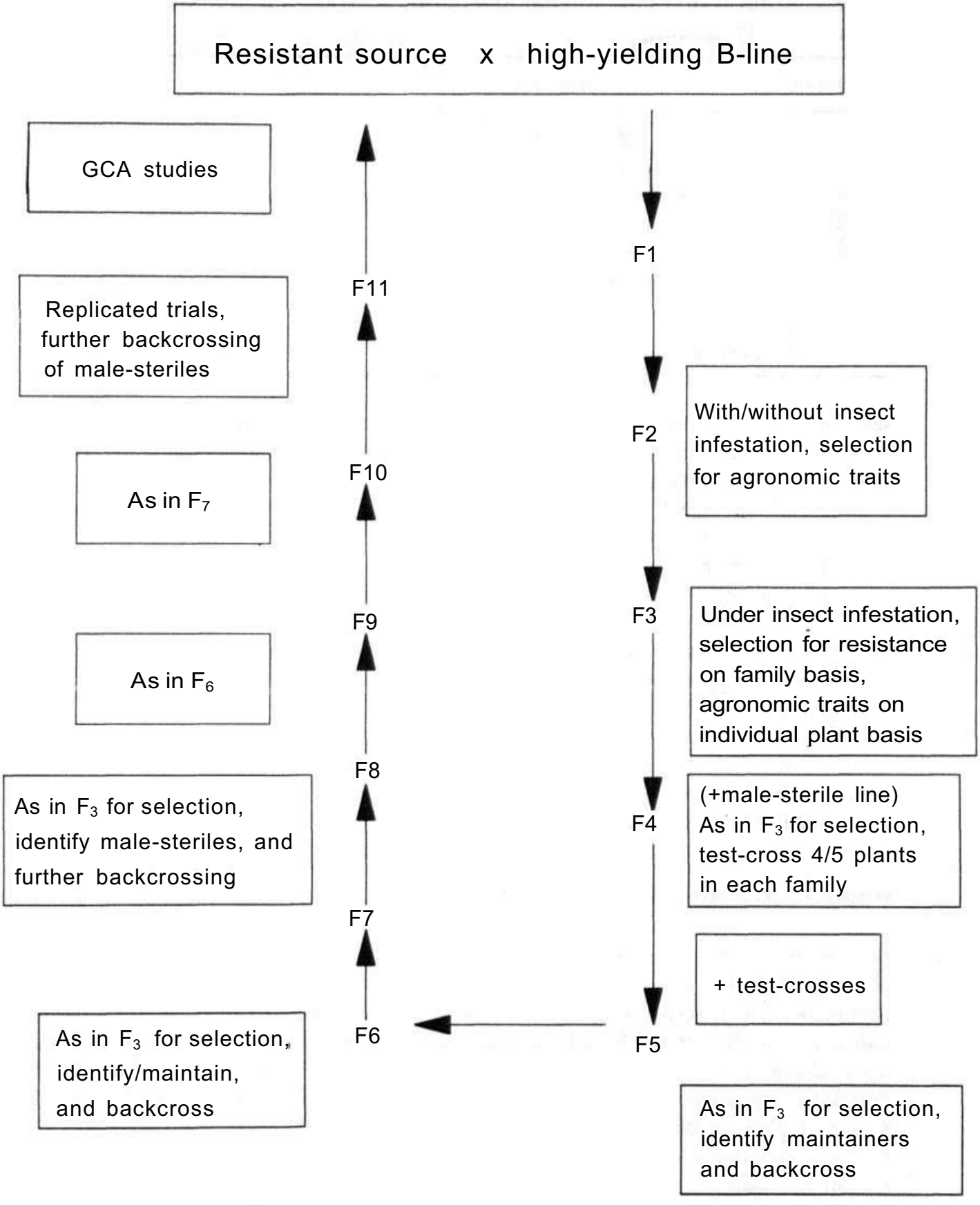


Figure 3. Development of insect-resistant male-sterile lines in sorghum by the pedigree method.

Table 1. Proportion of maintainer line genome brought into A-line cytoplasm through backcrossing.

Generation[1]	Proportion (%)
F_1	50.0
BC_1	75.0
BC_2	87.5
BC_3	93.8
BC_4	96.9
BC_5	98.5

1. F_1: 1st filial generation.
BC_1 .. BC_5 :Backcross generations 1 to 5.

selecting resistant plant(s), through screening from the F_2 generation of R x S lines, and backcrossing them to the high-yielding parent (S). Most resistances are governed by recessive genes, so that selection for resistant plant(s) is essential before backcrossing to the high-yielding parent. This method is illustrated schematically in Figure 4. Major steps involved in this method are as follows:

1. Make crosses between the resistant source (donor parent) and the selected high-yielding parent. Advance through F_1, F_2, F_3/F_4, as in the pedigree method, and select resistant plant(s).
2. Cross resistant plants to the high-yielding parent (recurrent parent), and obtain BC_1F_1. Advance to F_2 as in Step 1 above.
3. Same as in Step 2 above until BC_5F_1s are obtained.
4. From BC_5F_1, the steps are the same as described in the method for developing restorer lines, or varieties, or as in the method for developing male-sterile lines. In the latter, care should be taken that the recurrent parent chosen for improvement, or the resistant source line (donor parent) is a maintainer line.

Population improvement methods

Genetically broad-based populations are used for population improvement. These populations are gene pools possessing genes from a large number of parents. They differ from the type of populations used for pedigree selection, which are .derived from crosses between only two, or a few, parents. Improvement of broad-based populations by recurrent selection is intended to provide source populations for the extraction of inbred lines (Hallauer 1981). Population improvement involves a* selection cycle with the following three phases:

- Development of progenies to be tested.
- Evaluation and selection of these progenies.
- Recombination of the selected progenies.

All three phases may be covered in a single season, or over a period of two to four seasons, depending on the method employed.

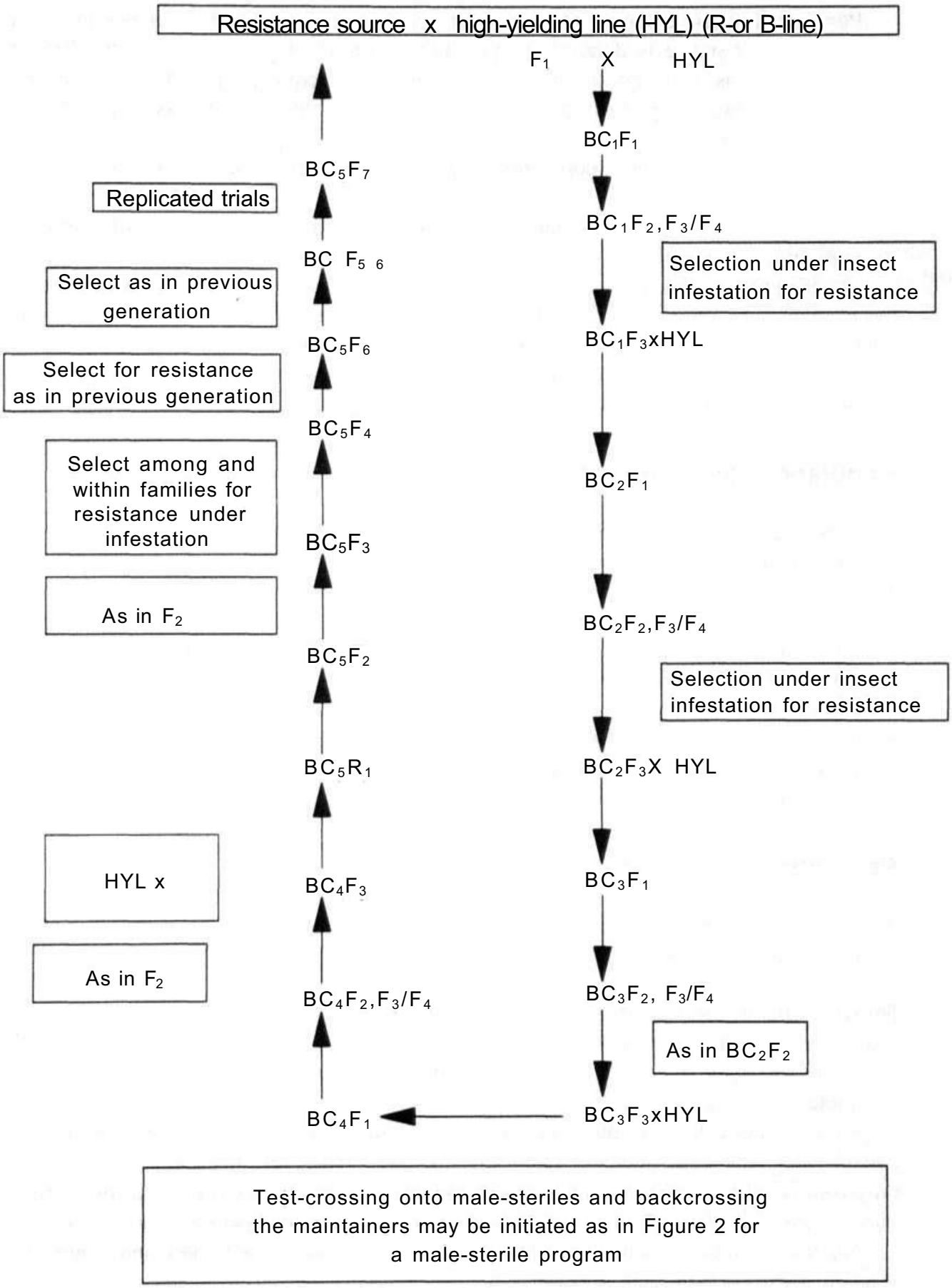


Figure 4. Transfer of resistance traits through backcrossing in sorghum.

Population improvement is an appropriate method for enhancing resistance to insect pests, where resistance is determined by a large number of genes, or even by distinct mechanisms. It is also appropriate when attempting to combine insect resistance and other traits having complex inheritance such as grain yield. The success of population improvement in these situations is based on:

a) the use of a broad-based population containing desirable genes from many distinct sources; and
b) avoiding rapid loss of resistance genes by maintaining a "gene pool" through selection and recombination of a large number (30, 50, or > 100) of lines.

Theory predicts that cyclic selection in such broad-based populations will increase the frequency of desirable genes. The consequences would be both an increase of the population mean, and occurrence of better (more resistant) lines than existed originally. Practically, such gains in the population mean and development of lines superior to those previously available can be realized.

Population formation methods

The development of a random-mating population involves the following three steps;

- Identification of insect-resistant sources, and a source of genetic male-sterility.
- Crossing the source lines with the source of genetic male-sterility, either by emasculating the source lines and/or using fertile plants from the genetic male-sterile source as the male. This method includes the cytoplasm of the original source lines in the new population, or by using male-sterile plants of genic male-sterile source to cross with the pollen from the resistant source lines.
- Recombination of the newly formed population for at least two more generations without selection. This is required to break linkages and enable new combinations of favorable genes from diverse sources.

Population improvement

A variety of recurrent selection methods is available. The appropriateness of specific methods depends on the trait(s) to be improved and the resources available.

Phenotypic selection. Mass selection is the simplest method of recurrent selection. Male-sterile plants are tagged at flowering and used for selection. A large number of selections are made, e.g., 200 or more, to maintain the broad genetic base. This method completes one cycle per season.

Advantages of the procedure are that high selection intensity may be applied and it is considerably less labor-intensive than other methods. However, this method is effective only for highly heritable traits, where identification of genetic differences among single plants is possible. The response to selection tends to be location-specific.

Population improvement for resistance to shoot fly, stem borer, and midge are one possible application of mass selection.

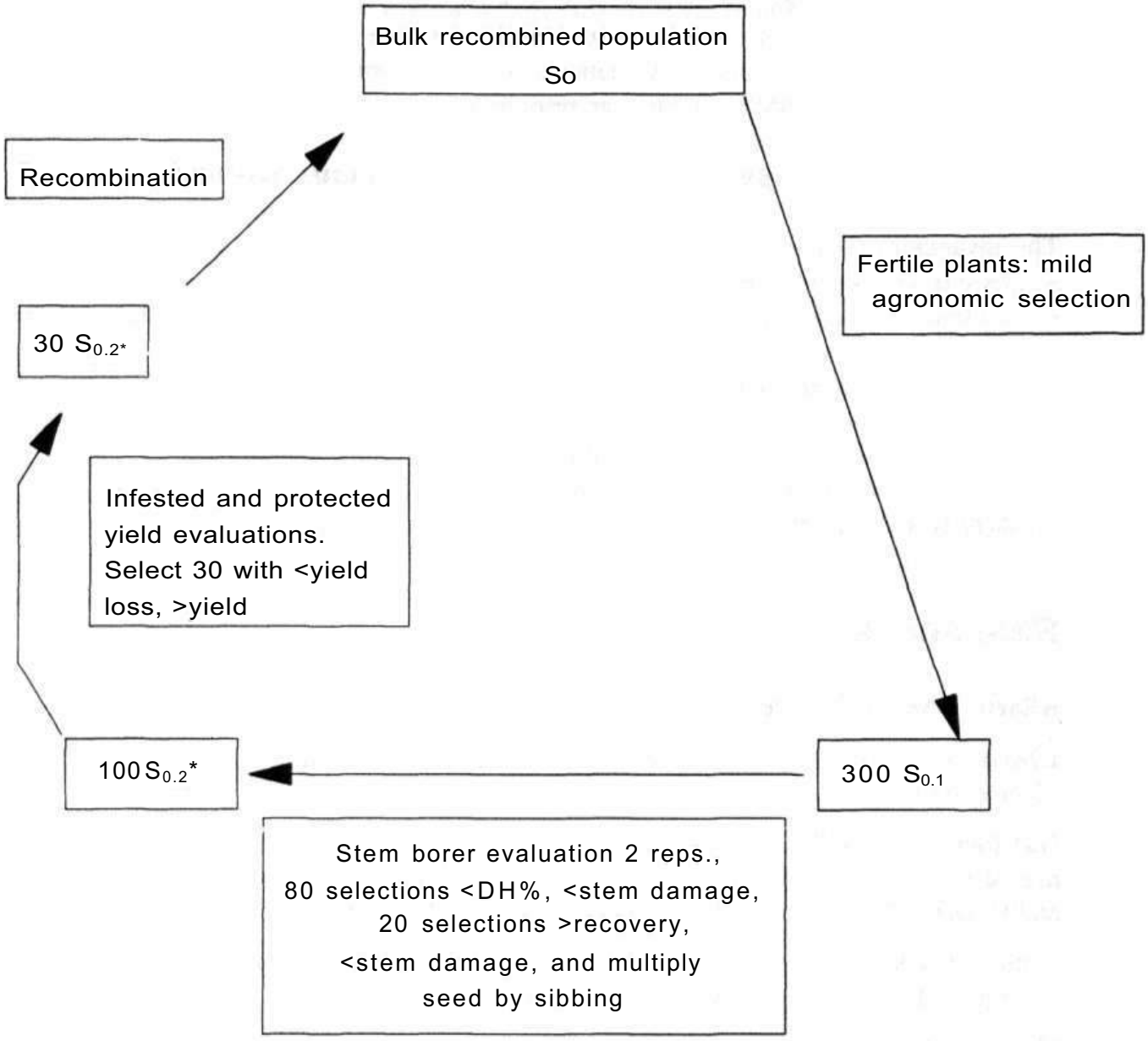


Figure 5. A recurrent selection scheme for enhancing resistance to stem borer in a broad-based population. (DH% = percentage of deadhearts.)

Genotypic selection. Whereas mass selection involves selection on the phenotype of single plants, methods based on evaluation of progenies permit replicated, multi-environment genotypic selections to be made. The three phases of each selection cycle may be distinct. For example, in the proposed selection scheme for stem borer resistance (Fig. 5), the three phases would be as follows.

- Development of S_1 progenies.
- Progeny evaluation (two-stage testing).
- Recombination of the selected progenies.

This method enables effective gains to be made for traits with lower heritability and complex inheritance. The gains realized may be stable over environments if multi-location, or multi-environment evaluations, are used. This method, however, requires heavy investment in material and human resources.

Advantages and disadvantages of population improvement

The advantages of population improvement relative to pedigree breeding methods are:

- there is less danger of loss of resistance genes;
- multiple resistance genes and mechanisms of resistance can be combined from different sources; and
- the frequency of resistance genes can be increased.

Disadvantages of population improvement relative to pedigree methods are:

- it relies more on early-generation evaluations, which may be less effective; and
- there is less opportunity to select for secondary agronomic characters.

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Biotechnology and Sorghum Improvement for Insect Resistance

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Introduction

Biotechnology offers powerful tools to all branches of biology, within which agricultural biotechnology comprises only 4% of all research and development in this area (95% in medicine). Even so, rapid increase in the popularity and acceptance of agricultural biotechnology is evident from the number of environmental release permits obtained for genetically engineered (transgenic) plants from virtually nil in the mid-1980s to over 500 in 1995. Biological applications in research are less evident (e.g., those based on linkage maps, embryo rescue, or somatic hybridization to facilitate wider hybridization), but they are equally spectacular. This discussion paper addresses the prospects for biotechnology in contributing to sorghum improvement for insect resistance.

Biotechnology *represents* a combination *of* biology, biochemistry, microbiology, genetic engineering, and applied science (field cropping in the present case) and related skills to produce new products of practical value by novel means (Mickolos and Freyer 1990; Watson et al. 1992). In sorghum improvement the products of interest are, for example, insect-resistant cultivars engineered with a bacterial gene that kills the insects. While progress continues to be made through conventional methods, it is slow, and inadequate to meet the growing demand for input-responsive high-yielding sorghum. So additional tools are needed to select sources of resistance, or progenies, and to increase both vertical and horizontal levels of resistance. Examples are identification of specific lineages of midge-resistant materials to limit the number of lines used in breeding without losing diversity, and linkage mapping of components for shoot fly resistance.

Biotechnologies Relevant to Sorghum

Tissue culture

Tissue culture is a basic tool for biotechnological research in all crops (e.g., regeneration from cells or tissues is a prerequisite for genetic transformation). In vitro selection, i.e., selecting for somaclonal variation under tissue culture, is not suitable for identifying lines

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resistant to insects. Wild species of sorghum are known to have resistance to several insects. Techniques such as embryo rescue and somatic hybridization may be useful for the introgression of wild germplasm genes into cultivated types.

Genetic transformation

Transformation is the introduction of novel genes (DNA pieces of about 1-2 kilobase) into a host sorghum cell, and later regenerating a normal plant capable of seed-set. After it has been confirmed that the introduced genes function as expected in the derivatives of such transformed plants, they can be either released as a normal variety, or used as a potential parent in a breeding program. To date, three different techniques have been used in laboratory-scale experiments for genetic transformation of sorghum:

1. By using *Agrobacterium* to carry the gene of interest as plasmids and to insert them into actively growing plant parts such as the shoot apex (either cocultivation or direct injection).
2. By bombarding gold or tungsten particles coated with the gene of interest and forcing them into cells using an air-gun (biolistics).
3. By incubating single plant cells from which walls have been digested away by special enzymes (protoplasts) with DNA fragments of interest (the gene).

In addition to the gene of interest in these three techniques one has to include a marker gene so that the transformed tissue or plants can be easily identified. In practice one uses two types of markers—selectable (e.g., transgenics that change color when incubated with some chemicals), and screenable. Introduction of a herbicide-resistant gene is an example of the latter. When all cells or tissues, after transformation with the herbicide-resistant gene are exposed to herbicide, all except those which are transformed will grow.

While the transformation method is reasonably easy to master, there is a major choice to make in selecting which genes to use to increase resistance because the technique requires isolating insect-resistance genes (DNA sequences) located within the host plants at this stage. Until now, only a few disease-resistance genes have been isolated from model plants. Also, the selected genes may not be strong enough to ensure a high level of resistance, or they may be easily overcome by the pests that develop resistance to such mechanisms. On the other hand, genes of some bacteria, such as the *Bt* (insect toxin genes isolated from *Bacillus thuringiemis)* for example, are popular. There are many strains of *Bt.* and most of them are specific to only one group of insects. Other genes, such as protease inhibitors, are also promising.

Molecular markers

Over the past four decades, entomologists have developed resistance-screening techniques, and have identified insect-resistant sources from sorghum germplasm. However, use of a wide range of available resistance sources in breeding is far from satisfactory. Some of the traits are difficult to measure, and resistance factors vary with the environmental conditions. These shortcomings can be overcome if molecular markers are

used, especially DNA markers. These DNA markers are universal in occurrence, unlimited in number, and consistent for a genotype irrespective of its growth conditions or the plant parts from which the DNA is isolated. Biochemical markers, produced from specific small (sugars), or large (protein) substances, are less reliable. Isozymes (multiple forms of the same enzyme) are useful in some cases, but they are limited in number, and may be tissue-specific (Witcombe and Duncan 1993)

Two DNA markers are relevant in sorghum: restriction fragment length polymorphism (RFLP), and random amplified polymorphic DNA (RAPD). Finding RFLP markers is an elaborate exercise, but it is more precise than using RAPDs. The latter are easy to test because only small amounts of plant DNA are needed. The RAPDs amplify parts of DNA with the help of random primers and heat-resistant DNA polymerase *(Tag.* DNA polymerase) in a polymerase chain reaction (PCR). In both cases, segregating lines of specially generated populations from crosses (usually F_2s from a resistant x susceptible cross) are evaluated for the resistant trait, and the pattern of the marker (resemblance to either of the parents [homozygous], or both [heterozygous] in the case of RFLP only). A linkage map is then drawn, and the markers close to the mapped trait are chosen as the identifiers for selection. Markers are useful for pyramiding genes for different mechanisms of resistance in a single genotype.

Complex Resistance Traits

Direct genetic intervention transformation requires a thorough understanding of the metabolic and cellular pathway responsible for any specific adaptive feature. Complete understanding is not a prerequisite for the molecular-marker based approach in which association of markers with the observed phenotypic response is being used. However, it is still useful to know the mechanisms, at least those of intermediate complexity, or structure, so that markers identified are strongly associated with the trait under study across all environmental conditions. In the case of sorghum, entomologists have made considerable progress in identifying the components of resistance to major insect pests. Thus, it is logical that biotechnologists concentrate on the few key mechanisms listed in Table 1 for each insect, so that durable resistance can be concentrated on one or a few useful genotypes (Nwanze et al. 1995).

Improvement of Insect Resistance in Sorghum Through Biotechnology

On a global scale, research on sorghum biotechnology is being carried out at the following institutions:

- Texas A&M University, College Station, USA: mapping midge resistance.
- University of Queensland, Brisbane, Australia: transformation for midge resistance.
- ICRISAT-Patancheru: mapping shoot fly resistance, and transformation for borer resistance.

Table 1. Components of interest for biotechnology research in sorghum.

Insect	Component of resistance	Comments[1]
Shoot fly	Glossy leaves, trichomes, rapid growth, volatile production, and epicuticular wax	Some are easy to score (e.g., glossy) and others are difficult (wax needs scanning EM; volatiles need GC)
Stem borer	Rapid growth, earliness, leaf angle, internode length, ligular hairs, etc.	Seasonal variation influencing plant growth should be distinguished from innate ability (genetic potential)
Midge	Small, tightly clasping glumes, faster ovary growth rate, and tannins	
Mtrid bug	Long glumes, quicker grain hardening and longer covering of grain by the glumes.	

1. EM = electron microscopy; GC = gas chromatography.

Practical advances in biotechnology are subject to the following considerations:

- The accessibility of field and laboratory screening methods.
- The high cost—which implies that biotechnology should be used only after traditional methods have been explored, or only to enhance current traditional methods.
- The effect, in the transgenic approach, of the introduced gene on the durability of resistance and on the environment. For instance, the effect on related crops in the region of introducing *Bt* is critical, such that gene rotation may be considered as an alternative option.

Conclusions

- Conventional and biotechnological approaches are complementary in nature.
- Opportunities exist for multiple trait selection, and for using novel germplasm and genes, but these should be approached with appropriate caution.

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Design and Analysis of Experiments: Basic Concepts

Subhas Chandra

The Experiment

An *experiment* is an enquiry that is undertaken to answer a *specific* question, or questions. The question(s) may be related to:

a) confirming, or disproving something doubtful;
b) discovering some unknown principle, or effect; or
c) testing, establishing, or illustrating some suggested, or known truth.

Experiments can be classified as *absolute* and *comparative.*

A *sample survey* undertaken to assess, say, the damage caused by pod borer to chickpea in a given region is an example of an *absolute experiment.* The primary aim here is to know about the *absolute value* of the damage. In a *comparative experiment,* on the other hand, *two or more* treatments are compared in their effects on a chosen characteristic of a certain reference population. Here, the absolute value of the population characteristic under any single treatment separately is of no particular interest. The primary aim is to study the differences *(comparisons)* among treatments for the chosen population characteristic(s).

This paper deals with comparative experiments (shortened below to *experiments)* which can be grouped into *single-factor* and *multi-factor* types. In a *single-factor (or unifactorial) experiment,* only a *single* factor varies while all others are kept constant. In such experiments, *the treatments are the different levels of the single variable factor.* For example, an insecticide trial, in which the single variable factor is an insecticide tested at different levels, is a unifactorial experiment. Here only the levels of the insecticide vary from plot to plot, and all other factors, such as fertilizer and water management, are applied uniformly to all plots. An experiment in which *the treatments are (all possible) combinations of selected levels of two or more factors* is a *multifactor (or factorial) experiment.* A trial having two factors each at two levels, such as two levels (11 and 12) of an insecticide and two varieties (V1 and V2), is an example of a 2 x 2, or 2^2 factorial experiment; the four treatments of this experiment are: (I1,V1), (I1,V2), (I2,V1), and (I2,V2).

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The Design

An experiment physically consists of two basic sets:

a) a set of *fixed* number, say t, of *experimental treatments;* and
b) a set *of fixed* number, say n, of *experimental units.*

The number n [(usually) a multiple of t, e.g., *n* = 2t] is referred to as the size of the experiment. An *experimental treatment* refers to *a single set of well defined conditions* whose effect is intended to be compared with other experimental treatments. The set may contain one, or more than one condition, depending on the objective(s) of the experiment. An *experimental unit* refers to *a well defined unitlportionlsubset* of the *experimental material,* the *entire* unit receiving *only a single* treatment.

For example, in an agricultural field trial, the experimental unit is a plot of land of certain size and shape in the field, the unit receiving a single treatment. The *experimental material* here is the *experimental field* which is divided into n smaller *nonoverlapping* units to test the t experimental treatments. *Note the distinction between experimental material and experimental treatments: experimental material is the medium on which experimental treatments are tested.* Depending on the problem, an experimental unit can be even a single leaf, a whole plant, or a pot in a greenhouse.

A *statistical design* is *a set of rules* that regulates the *assignment* of the n experimental units to the 't' treatments in a manner that:

a) allows *valid* comparisons to be made among the treatments; and
b) controls the principal source(s) of extraneous variation in the experimental material to ensure *efficient* treatment comparisons.

It may be noted that in so far as the *design* is concerned, it does *not* take cognizance of whether the treatments constitute a randomly or purposiveiy selected set. in *whatever manner* they have been chosen, a design treats them *as fixed and well defined* 't' *distinct* entities, and is concerned *solely* with how *these and only these* 't' should at any given occasion be assigned to the n experimental units, to ensure valid and efficient experimental conclusions. *It is at the data analysis stage that a distinction may be made as to whether the treatments be considered randomly and purposively selected.*

Basic Principles of Experimental Design

The design of an experiment is based on three basic principles: *replication, randomization,* and *local control.*

Replication

This is *the number of 'distinct' experimental units to which a treatment is assigned* out of the n units available in an experiment. The t treatments are said to be *equally replicated* when each is assigned to the same number, say r, of distinct experimental units; in this case t x r = 2n. Otherwise, the treatments are said to be *unequally replicated.* When r = 1 for each treatment, the treatments are said to be *unreplicated* with t X 1 = t = n. Correspondingly, we have an *equireplicate design,* an *unequally replicated design,* and an

unreplicated design. Note that replication is necessary to obtain:

a) an *estimate* of *experimental error variance;* and
b) a *more precise estimate* of *the mean effect of any treatment.*

To better understand these statements, a clear understanding of the term experimental error variance—generally abbreviated to experimental error—is required.

It is the *variation* among *identically treated* experimental units that constitutes *experimental error.* Or, in other words, experimental error is the variation that arises from the *failure* of experimental units receiving the *same* treatment to deliver *exactly the same* results. This may happen due to one/some/all of the following reasons:

a) inherent variability among the experimental units;
b) lack of uniformity in the conduct of the experiment, i.e., failure to standardize the experimental technique;
c) errors of observation and measurement; and
d) nonuniform behavior of all other extraneous factors, known, or unknown, that have not been/could not be accounted for in the experiment.

Note that the term experimental *error* is not synonymous with *blame/mistake.* In a statistical context, it represents the variation that is often *unavoidable.* Experimental error in any experiment is an unavoidable fact-of-life since no two experimental units receiving the same experimental treatment, despite our best efforts and care, will hardly, if ever, match exactly in all their physical manifestations.

Replication provides an estimate of experimental error because error arises from *variation* among identically treated experimental units (note the underlined "s"). This variation cannot be measured when each treatment receives only one experimental unit. The *only* way to get a measure' (estimate) of this variation, *except in some factorial experiments.* is to assign the treatments to a number of experimental units, even to an unequal number if the situation so warrants.

Note that an estimate of experimental error—the basic reason for replicating experiments—is the *basic unit of measurement* (of variation) on *a per unit basis* which serves as a yardstick for:

a) measuring the precision of the estimates of treatment effects; and
b) assessing the significance of observed treatment effects using an appropriate test of significance.

Clearly, a test of significance is possible *only when* the treatments are replicated.

Replication provides a more precise estimate of the mean effect of any treatment in the following way. Let σ^2 be the experimental error variance. The *error variance of an observed treatment mean* is then (σ/r); the reciprocal of this, i.e., (r/σ^2), is defined as the *precision of the observed treatment mean.* With $r = 2$, $(\sigma^2/r) = (\sigma^2/2)$. With $r = 3$, $(\sigma^2/r) = (\sigma^2/3)$ which is less than $(\sigma^2/2)$. The error variance of an observed treatment mean therefore decreases, and hence precision increases as the number of replications, r, is increased. But, beyond a certain number of replications, the improvement in precision may be too small to be worth the additional cost and effort.

An important point that must be clearly understood is that: **multiple observations from within any experimental unit do not represent replications; they are subsamples, and variation among them represents *sampling error, not* the experimental error.** *It is the experimental error, not the sampling error, that is valid to test the significance of observed treatment differences.*

The *number of replications* in an experiment depend on:

a) the degree of precision desired, which is generally prescribed either as the standard error of treatment means, or as the magnitude of treatment difference that can be detected; and
b) the magnitude of experimental error, generally expressed in terms of the coefficient of variation (CV) likely to be obtained in the experiment.

In any case, except for some factorial experiments, the number of replications should be *not less than two.* For a given number of treatments, and the design to be used, the number of replications should be such that the *error degrees of freedom* (df) are *at least 12.* With error df less than 12, the F-test in ANOVA will *not* be *sensitive* enough to detect treatment differences. Experimenters often confuse the terms *replication* and *block.* But they do not represent one and the same concept. *Replication is a numerical entity. A block, however, is a physical concept related to local control.* Also the number of replications is *not always* the same as the number of blocks in an experiment. Their numbers may be the same in the case of a complete block experiment. in an incomplete block, or a confounded factorial experiment, the number of blocks is more than the number of replications.

Randomization

This refers to the *random assignment* of experimental treatments to experimental units. Random assignment does *not* mean to manipulate treatments into some order that looks haphazard. Randomization should be an *objective impersonal procedure* which is devoid of personal bias and gives every treatment the same chance to receive any unit. A design in which treatments are randomly assigned to experimental units is called a *randomized design,* or alternatively a *systematic design.*

Randomization is required:

a) to provide *validity* to the estimate of experimental error, and to a statistical test of significance of observed treatment effects; and
b) to protect against any *(un)known systematic bias* that may creep into observed treatment effects due to extraneous sources of variation that have not been/could not be controlled.

The question is often asked whether or not to use randomization. To answer this question it is necessary first to understand the pros and cons of using randomization in relation to its above two functions. Any probability-based statistical tool, including any test of significance and any measure of precision, *to be valid,* deductively requires that the experimental observations constitute a *random sample* from some well defined reference population. This condition will hold good when the observations are *independent* of one another. Random allocation of treatments to experimental units allows us to proceed *as though* the observations are independent. Note that this does *not* imply that randomization *guarantees* independence of observations. This distinction is based on the premise that observations from spatially/temporally adjacent experimental units will tend to be correlated. What randomization does is only to *assure* us that the effect of this correlation on treatment effects has been made as small as possible. Some degree of

correlation may still persist since no amount of randomization can ever eliminate it completely. *True and complete independence of observations in any experiment is an ideal that can perhaps never be fully achieved, particularly in agricultural field trials.* In view of this, restraint and caution should be exercised in explaining experimental inferences.

That randomization protects against any (un)suspected systematic bias can be better understood through a simple example. Suppose that a plant breeder, in order to compare yields of two cultivars A and B, plants each to four plots in the field systematically, in this way:

PLOT 1	PLOT 2	PLOT 3	PLOT 4	PLOT 5	PLOT 6	PLOT 7	PLOT 8
A	A	B	B	A	B	A	B

If the field has a (generally unknown) fertility gradient from left to right, variety B then consistently falls on a relatively less fertile plot. The observed yield difference between the two cultivars will therefore be subject to an unknown amount of systematic bias in favor of variety A. On the other hand, had the two cultivars been assigned randomly to the eight plots, each variety would have got an equal chance of falling on any plot that happened to possess good, or poor fertility, and the bias could have been reduced.

The above arguments do *not* mean that systematic designs must never be tolerated. Provided that we possess *good* knowledge of the *form* of uncontrolled extraneous variation in our experimental material, and a systematic design is easier to work with, it may perhaps be right not to randomize. In fact, in some situations it may not be even possible to randomize. In all such circumstances, it is essential to accept the fact that any resulting inference *cannot* be supported by a meaningful probability statement, and the quoted measure of precision may be biased. So, in summary, a practical answer to the question "to randomize, or not?" is: *randomize except when there is very good reason not to; and understand that inferences from a systematic design depend heavily on the correctness of what is assumed about the underlying pattern of extraneous variation, and to report this assumption explicitly in reporting the experimental results.*

Local Control

This refers to *blocking* the experimental units. Local control will reduce the magnitude of experimental error, which improves the efficiency of the experiment and makes the tests of significance more sensitive/powerful in detecting treatment differences.

Blocking comprises the (physical) stratification of experimental units into a number of blocks in such a manner that the units within any block are relatively homogeneous. The result is that variability within each block is minimized and variability among blocks is maximized. The experimental error thus consists of only the intrablock variation. In the absence of blocking, it would consist of both the intrablock and the interblock variation.

Two decisions must be made to ensure appropriate and effective blocking. These are:

a) selection of the *source of variability* to be used as the *basis for blocking;* and
b) selection of *block size, shape, and orientation.*

An ideal source of variation for use as the basis for blocking is one that is *large and highly predictable.* For example, *soil heterogeneity* in a varietal or fertilizer trial where yield is of primary interest, *direction of insect migration* in an insecticide trial where insect infestation is of primary interest, and *slope of the field* in a study of plant reaction to drought stress. Block size, shape, and orientation should be such that they *maximize* interblock variability. Some guidelines for blocking are as follows:

- For unidirectional gradient, use long and narrow blocks; orient these blocks with their length perpendicular to the direction of gradient.
- For bidirectional gradient with one gradient much greater than the other, ignore the weaker gradient and take steps as above for unidirectional gradient.
- For bidirectional gradient with both gradients the same and perpendicular to each other, choose one of the following:
  - Use blocks that are as square as possible.
  - Use long and narrow blocks with their length perpendicular to the direction of one gradient, and use the covariance technique to account for the other gradient.
  - Use a suitable row-column design with two-way blocking—one for each gradient.
  - When gradient(s) is/are suspected to be operative, but when its/their direction(s) cannot be predicted, use blocks that are as square as possible.

Local control using blocks removes only the interblock environmental variation. The variation from plot to plot within blocks still remains, which forms the experimental error. A further reduction in experimental error may thus be achieved by measuring, in some suitable way, the varying effects of the plots within blocks, and using this plot-measure as a *covariate* to obtain corrected (adjusted) estimates of treatment effects with higher precision through *covariance analysis. A* caution to keep in mind is that the covariate values, intended to measure *environmental effects only, must not* be affected by treatments.

A third way to reduce experimental error is to *lake proper care* in the physical conduct of the experiment. Any lack of care, either at the beginning, during, or towards the end of the experiment, can be a potential source for increased experimental error. When this happens, even the most carefully and wisely chosen blocking and/or covariance analysis may not help to rescue the situation—and they might even prove counter-productive particularly when, as normally is the case, there is no way to *quantify* and *measure* the *effect* of *lack of care* in different experimental units. Since there are many ways other than local control to reduce experimental error, the principle of local control is sometimes replaced by a broader term *error control,* which includes all techniques, including blocking, to reduce the experimental error.

Increasing the number of replications is *wrongly* conceived by some as a device to reduce the experimental error. Reduction in the magnitude of experimental error may be achieved only through local/error control. Increased replication provides only a more *stable* (*consistent* in statistical terms) estimate of experimental error.

Plan your Experiment to Achieve Good Design

Design is only one facet of the whole process of planning any experiment. *The selection of an appropriate and efficient design is possible only when it is seen as an integrated*

part of The whole process of planning. The following steps, taken in sequence, can greatly increase the overall success of an experiment. Guidance is also provided in the planning proforma in the Appendix to this paper.

Step 1. Identify and state the problem *clearly* and *concisely.*

Step 2. State the objective(s) *precisely* in the form of *specific question(s)* to be answered, or *specific hypotheses* to be tested, or *specific effects* to be estimated. If you have more than one objective, list them in their order of priority. Do not be too vague, or too ambitious in stating your objective(s).

Step 3. Select the treatments *objectively* to ensure that the objective(s) can be met.

Step 4. Select the experimental material in accordance with the objectives and the population about which inferences are to be made. The experimental material should adequately represent this reference population.

Step 5. Select the experimental design considering the objective(s) and the available resources. Try to choose a simple and operationally feasible design that is likely to provide good precision.

Step 6. Select that form of experimental unit and that number of replications that are likely to provide good precision. Historical data from similar experiments can be very helpful in making these decisions.

Step 7. Decide on measures to control the effects of adjacent experimental units on each other. This can be done by treatment randomization, and other physical measures, such as using border rows.

Step8. Decide on th'e types of data, and the *manner* and *order* in which they are to be collected. Work out a *data-recording mechanism* that will later facilitate *data entry, validation, and analysis* on a computer in accordance with appropriately chosen statistical method(s) and computing software. *Give particular attention to the collection of those data that may help explain why the treatments behave as they do.*

Step 9. Prepare an outline for statistical analysis, and, if possible, a summary of expected outcomes.

Step 10. Review your plan (Steps 1-9) with a statistician and your colleagues. This work, undertaken with further guidance given in the planning proforma in Appendix, may reveal important points that have been inadvertently overlooked. Then modify your plan if necessary, in the knowledge that effective advance planning obviates avoidable problems.

Conduct the experiment as per plan (Steps 1-10), validate the collected data to check for possible errors in data and unexpected values, analyze the validated data and interpret the results, and prepare a complete, correct, and readable report.

In conducting the experiment, use experimental procedures that are free from personal bias. *Make sure* that differences among individuals, or differences associated with the order in which data have been collected do not contribute to experimental error. *Avoid* fatigue in data collection. Immediately *recheck* observations that seem dubious.

Proper data entry and validation of the entered data are important parameters to make sure that the results obtained from analyzing the data are free from incorrect/unexpected data values. *Without proper data entry and validation, the entire effort in analyzing the data may well prove to be futile.* Epi Info (version 6) is excellent software to use for data

entry and validation. It enables you to introduce many effective data-check options at the entry stage as well as later for data validation. Analyze all data using appropriate statistical tools. Interpret the results in the light of experimental conditions, hypotheses tested, effects estimated, and their relation to previously reported results.

In preparing the report, remember that there is no such thing as a *negative* result. If null hypothesis is not rejected, it is *positive* evidence that there may not be real differences among the treatments.

Statistical Data Analysis

Analysis of variance (ANOVA) is the most widely used (but not always correctly applied) statistical tool in analyzing experimental data. A formal analysis of data using ANOVA requires:

a) identification of the *treatment structure* and the *design structure* which together comprise the *data structure;* and
b) formulation of an equation (a *statistical model)* for every observation *in accordance with the data structure.* The equation, so formulated, expresses an observation as the sum of a number of components that are identified in the data structure.

The treatment structure refers to the way a treatment has been made up. In a single-factor experiment, the different levels of the factor constitute the treatments; each level stands in its own right. The different levels may be quantitative in nature; for example, the graded doses of an insecticide, or a fertilizer, or different cultivars of a crop. The levels may sometimes be classified into biologically relevant groups, each group consisting of a number of distinct levels; *this gives rise to a hierarchical structure, and not to a cross-classified treatment structure (as sometimes incorrectly interpreted).* In a factorial experiment, on the other hand, any single treatment is made up of a cross-classified combination of the different levels of the factors involved; there is a main effect for each factor as well as interaction effects of various orders among the factors, which is not the case in a single-factor experiment.

The design structure refers to the way local control is (physically) exercised in an experiment. For example, in an experiment laid out in a randomized complete block design (RCBD), local control is exercised through blocks. The blocks are structured in a way that every treatment appears in each block at least once. In a split-plot experiment, local control is exercised at two levels. The field is divided into blocks within each of which the main-plot treatments are randomly allocated. Each main plot within every block is subdivided into subplots to which subplot treatments are randomly assigned.

Consider an RCBD with t treatments and b blocks. Let the t treatments represent t different methods of cultivating groundnut. A plot-mean-observation $y_{..}$ (corresponding to the plot receiving treatment i in block j), as per the treatment and the design structures, is here the sum of the following four components:

a) A general average (μ) about which the observations are presumed to be fluctuating.
b) A component (τ_i) due to the treatment i applied to the plot.
c) A component (β_j) due to the block j in which the plot falls; it represents the effect of the environmental factor(s) which the experimenter may have been able to properly identify and which the design subsequently permits him/her to isolate.

d) A residual component (ε_{ij}), representing all other factors that influence the observation; this component constitutes the "experimental error".

The above components put together make up the statistical model for any plot-mean-observation y_{ij} in an RCBD which can be written as:

$$y_{ij} = \mu + \tau_i + \beta_j + \varepsilon_{ij} \quad (1)$$

If there are t treatments and b blocks in the experiment, then i = 1, 2. . . .t; and j = l,2,...,b; there being a total of n = tb observations corresponding to the n = tb experimental plots. The ANOVA partitions the total variation among the n observations into as many components as are *identified* and *included* in the statistical model. The statistical test commonly used in ANOVA to test the *significance/existence* of treatment differences is the *variance ratio* (also called F). This is calculated from the relevant mean squares (MS) as F = {Treatment MS/Error MS}.

To carry out a *valid* F-test, it is important to identify and use the appropriate composition of mean squares in its numerator and denominator. This identification is done on the basis of the *expected values of the mean squares.* The structure of these expected values is governed by the assumptions made regarding the nature *{random and/or fixed)* of treatment effects. *Lack of awareness/understanding of these basic concepts on the part of experimenters often leads to the use of invalid F-tests and hence to invalid inferences.*

The calculated value of F = {Treatment MS/Error MS}, say F_c, is compared with its corresponding theoretical (called by some the "tabulated") F value to decide *whether or not to reject the* (*null*) *hypothesis of no treatment differences* and, accordingly, whether or not to declare the observed treatment differences as significant. The theoretical F values for different significance levels of *P* have been tabulated and can be found, for example, in Appendix E of the book by Gomez and Gomez (1984). *A significance level is the probability of rejecting a true (null) hypothesis of no treatment differences.*

To illustrate this, assume t = 4 and b = 5 in an RCBD. There will be f_1 = (t-1) = 3 degrees of freedom for treatment MS, and f_2 = (t-1)(b-1) = 12 degrees of freedom for error MS. A reference to the above Appendix shows that for f_1 = 3 and f_2 = 12, the theoretical F-value is 3.49 at *P* = 0.05 and 5.95 at *P* = 0.01. If 3.49<F.<5.95, the observed treatment differences will be declared significant at ***P* <0.05.** A similar inference will be drawn at *P* <0.01 if F >5.95.

Most statistical software packages have obviated the need to refer to the F-tables in order to arrive at conclusions in the above manner. Instead, they compute and report the *P*-value for any calculated F-value in the ANOVA table. The *P*-value is the probability of getting an F-value as large, or larger, than the calculated F-value under the condition that the null hypothesis is correct. If the reported *P*-value, say P_c, is such that 0.01 <P_c <0.05, the observed treatment differences are declared significant at *P* <0.05. If P_C <0.01, a similar statement can be made at *P* <0.01. However, if desired, the reported value of P_c can itself be quoted to indicate the level of significance. The smaller P_c value, the stronger is the evidence that significant treatment differences exist.

The *reliability* of an estimated treatment effect is usually indicated by its *standard error* (SE). The SE can be estimated from ANOVA and may be used to set limits within which the true (unknown) value falls with *any* specified degree of confidence. These limits are called *confidence limits.* For example, if the estimated treatment mean is m and its SE is s, the 95% confidence limits for the treatment effect are (m-1.96s) and

(m+1.96s); this means that the true value of the treatment effect will fall within these limits in 95 out of 100 such experiments that could have been conducted under similar conditions.

In general, the appropriate multiple (e.g., 1.96 in the above case) of the s to be subtracted from and added to the estimated treatment effect m, depends not only on the specified degrees of confidence (e.g., 95% in the above case) but also upon how accurately the s has been estimated. Tables such as the t-table—e.g., Appendix C in Gomez and Gomez (1984)—furnishes the values of the appropriate multiple for s in determining the confidence limits. In a t-table, possible inaccuracy in the SE is indicated by its degrees of freedom, which is the effective number of observations used in the estimation of s.

Assumptions of ANOVA

The assumptions of ANOVA, which the experimental data should fulfil for the resulting analysis to be valid, are:

a) the observations can be represented by a *linear additive* model, e.g., the model (1) in case of an RCBD;
b) the errors e (see equation 1) behave like a *normal distribution;*
c) the errors e are *independent* of one another; and
d) the error e in observations should not be affected by the nature of observations, which is equivalent to saying that the errors should exhibit the *same degree of variation* for each of the experimental treatments.

Failure to meet an assumption affects the significance level as well as the sensitivity of the F- and t-tests. The true significance level, as a result, is usually larger than the apparent one. This leads to getting too many significant results. Loss of sensitivity occurs, accompanied by a loss of accuracy in the estimates of treatment effects, in the sense that a more powerful test than the ANOVA F-test, and a more accurate estimate of treatment effects, could be devised if the correct model were known. As a result, the significance levels and confidence limits *must* be considered as *approximate* rather than exact. Also, for similar reasons, the *rigid* application of 5% or 1% significance levels, to divide the treatment effects as real and unreal, appears to be hardly justified.

The nonconstancy of error variance (assumption d) can be the most serious problem. This may happen when:

a) certain treatments are erratic in their expression; and
b) the errors obey a highly skewed distribution that is very different from the normal distribution.

In situation (a), the problem may be overcome by dividing the Error SS into parts, each of which is homogeneous. In situation (b), the error variance for any treatment tends to be some function of the treatment mean which may be detected by plotting the variance against the mean on graph paper. A suitable transformation, depending on the nature of the functional relation between variance and mean, may make the error variance nearly constant.

The nonadditivity of treatment and environmental effects could be turned into additivity by a suitable transformation. Fortunately, such transformations may also bring

the distribution of errors closer to normality. In most cases, the requirement of independence of errors may normally be met by the physical act of randomization.

Usually, moderate deviations from the assumptions will not unduly affect the validity of the analysis. For example, an approximate normal distribution of errors will give comparable significance levels. Slight heterogeneity in error variation from treatment to treatment will have little effect on the confidence limits. However, where large deviations from the assumptions occur, quite misleading results may be obtained from ANOVA.

Missing Data

When data on certain experimental units are missing, either due to accident or gross error in recording, the analysis as well as its interpretation becomes involved. The correct way to analyze an experiment with missing data is to construct a model for all data that are *available* and use the *method of least squares* (LS). This may be a difficult task for the experimenter but, to overcome it, Yates introduced the *missing plot technique* which, on the face of it, provides a complete set of data by estimating the missing data values.

Suppose a single data value is missing for which a value x is substituted. If the ANOVA is now computed in the usual way for the complete data, the Error SS turns out to be of the form $ax^2-2bx+c$; where (a,b,c) are numbers that depend on the type of design and the available data values (a >0). The value of x, determined by minimizing the Error SS, is x = b/a. When this value is inserted in place of the missing data value, and if the data are analyzed *as if no* data were missing, then:

a) the estimates of treatment and block effects will be exactly the same as those obtained by the LS method;
b) the Error SS will be exactly the same as given by the LS method; and
c) the df for the Total SS and the Error SS are each reduced by 1 to get the correct partition of the df.

There are two *defects* in the missing plot technique. The Treatment SS is always slightly larger than the correct Treatment SS. This overestimation is unlikely to be large unless an appreciable number of data values are missing. It is possible to measure this overestimation and get the corrected Treatment SS and, hence, the correct F-value to test the treatment differences. The second defect is that it may not give proper t-tests because, in the analysis of so-to-say complete data, r replications are ascribed to the treatment that contains the missing data value, whereas there are only (r-1) replications. Rules are available that allow for this discrepancy and provide approximately correct t-tests.

A point that must be remembered is that substitution of any missing value by its estimate *does not in any way* recover the information that is lost through the missing data value(s). It only attempts to reproduce the results in the simple way that would have been obtained by application of the LS method to the available incomplete data. *The only complete solution of the missing data problem is not to have any missing data values.*

Selection of Design

An appropriate design for an experiment depends on:

a) the *form* and *magnitude* of extraneous variation present in the experimental material;
b) the *nature* and *number* of treatments to be tested; and
c) the *degree of precision* desired.

Normally a design should be chosen that is based on the principles of replication, randomization, and local control *unless* you have very good reasons not to.

The selection of a design should be based *not solely* on the number of treatments, as is often advocated in design textbooks. Once the nature and number of treatments are known, the *major* consideration in the selection of an appropriate design *must* be the form and magnitude of extraneous variation in the experimental material if they are fairly known to the experimenter. The reason is very simple: for a given number of treatments and their nature, the right design should be one that effectively controls the extraneous variation, so that least the possible experimental error is obtained and, consequently, the treatment effects are estimated and tested more precisely.

There are two broad categories of designs: complete block designs (CBD), and incomplete block designs (IBD). The CBDs are useful for experiments in which the experimental material stratifies itself into homogeneous blocks within each of which *all* treatments can appear *together.* The completely randomized design (CRD), randomized complete block design (RCBD), and latin square design (LSD) are CBDs. Some features of CBDs are:

a) all treatments appear together in each block;
b) analysis of data is simple;
c) missing data are easier to cope with: and
d) they can be used for single- as well as multifactor experiments.

The IBDs are useful in experiments wherein the experimental material stratifies itself into homogeneous blocks within each of which all treatments *cannot* be tested together. Lattice designs and confounded factorials are examples of IBDs. Some features of IBDs are:

a) all treatments do not appear together in each block;
b) analysis of data is more complicated than for CBDs;
c) missing data are not easy to handle; and
d) they can also be used for single- as well as multifactor experiments.

Some general guidelines/features for a few commonly used designs are given below where: t = number of treatments, r = number of replications, b = number of blocks, k = number of experimental units per block (i.e., block size).

Complete block designs

Completely randomized design (CRD)

- All experimental units are homogeneous.
- Treatments can be (un)equally replicated.
- Main effects and interactions in case of factorial experiments are studied with equal precision.

Randomized complete block design (RCBD)

- Unidirectional gradient in the experimental material.
- Homogeneous blocks each of size k = t are available.
- r = b.
- Main effects and interactions in case of factorial experiments are studied with equal precision.

Latin square design (LSD)

- Bidirectional gradient in the experimental material.
- Perpendicular blocks of t homogeneous rows and t homogeneous columns are available such that, within each row and within each column, the treatments can appear together.
- k = r = t.
- Main effects and interactions in case of factorial experiments are studied with equal precision.

Incomplete block designs

Balanced lattice design

- t is such that $t = p^2$, e.g., $t = 36 = 6^2$, which gives p = 6.
- $r = p + 1$.
- b = p.
- Equal precision for all comparisons between pairs of treatments.

Partially balanced lattice design

- t is such that $t = p^2$.
- $r \geq 2$.
- b = p.
- Some pairs of treatments will have higher precision than others.

Confounded factorials

- More than two factors involved.
- Accuracy on certain higher-order interactions sacrificed.

In addition to the above, there is a special class of designs—split-plot designs—which are frequently misused or overused by researchers. These designs should be used *only when* there is no other alternative. General guidelines/features for their use are as follows:

Split-plot design

- At least two groups of factors under study: this includes the situation where each group has only one factor.
- One group requires larger plots and the other(s) can be tested on smaller plots, the large plots being called main plots and the small plots subplots.
- The group(s) tested on subplots, and their interaction with the group tested on main plots, are studied more precisely.

Strip-plot design

- Two groups of factors under study: this includes the situation where the groups have only one factor each.
- Both groups require large plots.
- The interaction between the two groups is studied more precisely.

A word of caution

There goes a saying that it is possible to prove anything by statistics. This may appear to be true for *bad* statistics; but exactly the *converse* is valid for *good* statistics. Use of a statistical design, followed by a proper analysis of data, does *not* provide absolute proof of the effectiveness of experimental treatments, it *only* enables you to *estimate* the *likelihood or reliability* of their continued effectiveness *at the level indicated by the experiment. This is all statistics can do.* Remember that *there is always a probability, even if small, of the results being wrong.* Therefore, avoid jumping to conclusions on the basis of your statistically significant results if they do not make sense. *Statistical significance may not necessarily mean biological significance. Similarly, a statistically nonsignificant result should not be rigidly interpreted as biologically unimportant.*

Reference

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APPENDIX: Proforma for Planning an Experiment

(The term "plot" is synonymous with "experimental unit" in items 09, 10, 16.)

01. Experiment title:

02. Objective(s): (If > 1, classify as **major** and **minor,** and avoid vagueness)

03. Details of experimental treatments

Experimental factor(s):

Levels of factor(s):

Total number of treatments/treatment-combinations: t =

Relevant information on (nature of) treatments/factors:

(e.g., checks, require larger plots, interplot interference, etc.)

04. Observations (within units) to be recorded:

(Indicate their order and manner)

05. Treatment effects/differences to be estimated:

06. Hypotheses to be tested:

07. Details of experimental site/material

(If >1 site, give information by site)

Site/field label:

Dimensions:

Any (suspected) gradient(s)?:

Direction of gradient(s):

Other relevant information about experimental site(s)/material:

08. Extraneous factors likely to distort treatment effects:

09. Plot size and shape required to facilitate experimental operations:

Plot size	:	Gross :	Net:
Plot shape	:		
Total number of plots	:	n =	
Number of plots in a row	:	n_{row} =	
Number of plots in a column	:	n_{col} =	

10. **Plot orientation:**

11. Block size: k = **Block shape:**

12. Block orientation:

13. Number of blocks: b =

14. Number of replications: r =

15. Experimental design:

(Fully randomized/partially randomized/nonrandomized)

16. Plot sampling plan:

17. Outline of statistical analysis:

Part 5

Insect Pest Management Strategies

Role of Plant Resistance to Insects in Sorghum Integrated Pest Management

H C Sharma

Introduction

The role of host-plant resistance (HPR) in pest management in sorghum has been discussed earlier by Jotwani (1978), Teetes (1985), and Sharma (1985, 1993). In this paper, information on HPR to insects in sorghum is reviewed to assess the role of insect-resistant cultivars in integrated pest management (IPM).

Sorghum shoot fly (*Atherigona soccata*)

Several workers have screened sorghum germplasm for resistance to shoot fly (Singh et al. 1968; Doggett et al. 1970; Singh and Rana 1986; Taneja and Leuschner 1985a; Jotwani 1978; Patel and Sukhani 1990; Sharma et al. 1992). Most of the sources resistant to shoot fly originate from postrainy-season sorghums grown in India under stored soil moisture. Cultivars M 35-1 (IS 1054), IS 1057, IS 2123, IS 2146, IS 4664, IS 2205, IS 5604, and IS 18551 have been widely tested, and possess moderate levels of resistance. Wild species of sorghum (*Sorghum purpweosericeum* and S. *versicolor)* possess very high levels of resistance to shoot fly (Mote 1984).

Efforts to breed for resistance to shoot fly have been made in the All India Coordinated Sorghum Improvement Project, and at ICRISAT. M 35-1, grown widely during the postrainy season in India, is a selection from landraces grown in the postrainy season. Improved varieties CSV 5, CSV 6, CSV 7R, Swati (SPV 504), and CSV 8R have been developed using landraces, and possess moderate levels of resistance to shoot fly (Singh and Rana 1986). Some of the improved lines such as ICSV 700, ICSV 705, and ICSV 717 developed at ICRISAT-Patancheru have better yield potential than the landraces (Agrawal and Abraham 1985).

Stem borers (*Chilo partellus* and *Busseola fusca*)

Sources of resistance to stem borers have been identified by several workers (Jotwani 1978; Jotwani et al. 1979; Singh et al. 1983; Singh and Rana 1984, 1989; Taneja and

ICRISAT Conference Paper no. CP 1167.

Sharma, H.C. 1997. Role of plant resistance to insects in sorghum integrated pest management. Pages 151-160 *in* Plant resistance to insects in sorghum (Sharma, H.C., Faujdar Singh, and Nwanze, K.F., eds.). Patancheru 502 324, Andhra Pradesh, India: International Crops Research Institute for the Semi-Arid Tropics.

Leuschner 1985b, Alghali 1985, Patel and Sukhani 1989, Agrawal and Taneja 1989; Reddy 1985; Sharma et al. 1992). IS 1055 (BP 53), IS 1044, IS 2123, IS 2195, IS 2205, IS 2146, IS 5469, and IS 18551 show moderate levels of resistance to spotted stem borer. Genotypic resistance is poorly expressed under low fertility, drought, and unfavorable weather conditions. The stage of infestation is critical for expression of resistance because a progressive delay in infestation reduces the production of deadhearts (Taneja and Leuschner 1985b). ICSV 705, SPV 135, CSV 8R, SPV 104, SPV 238, and SPV 842 are improved genotypes with moderate levels of resistance to spotted stem borer with a better yield potential than the original sources of resistance. Several improved sorghum varieties released in India (CSV 2, CSV 3, CSV 5, CSV 6, CSV 7R, CSV 8R, and Swati) have been developed by using landraces resistant to spotted stem borer (IS 1122, IS 1055, IS 1151, and IS 1054) (Singh and Rana 1989).

Aphids (*Schizaphis graminum, Melanaphis sacchari* and *Rhopalosiphum maidis*)

Extensive screening for resistance to *S. graminum* has been carried out in the USA. IS 809, KS 30, PI 264456, SA 7536-1, Tx 2567, and DK 46 are good sources of resistance against biotype C (Teetes et al. 1974; Schuster and Starks 1973; Starks and Schuster 1976; Lara et al. 1981; Baletka and Castellano 1983; Kofoid et al. 1991). *R. maidis* is a sporadic pest with no apparent effect on grain yield. Four biotypes of the aphid have been identified. TAM 428 (SC 110-9) exhibits high levels of resistance. Genotypes PI 954177, and IS 8100C are resistant to *M. sacchari* (Chang 1981; Chang and Fang 1984; Hagio and Ono 1986; Hagio et al. 1985). Midge-resistant lines ICSV 197 and ICSV 743 are relatively less damaged by this aphid (H.C. Sharma, unpublished).

Spider mites (*Oligonychus indicus* and *O. pratensis*)

SC 599-6 has been found to be tolerant of mites (Foster et al. 1977a,b). Mite damage has been reported to be less on late-flowering lines (Perring et al. 1982), and more on senescing lines (Foster et al. 1977a,b), but nonsenescence is not directly linked with resistance to mites (Archer et al. 1986).

Shoot bug *(Peregrinus maidis)*

Lines 1753, H 109, GIB 3677B, BP 53, IS 8884, IS 8887, IS 8891, and IS 8918 are less damaged by the shoot bug (Agarwal et al. 1978; H.C. Sharma, unpublished). IS 18657, IS 18677, and PJ 8K(R) are resistant to shoot bug under field conditions (Singh and Rana 1992).

Armyworms (*Mythimna separata* and *Spodoptera frugiperda*)

Armyworms are sporadic pests, and their feeding results in extensive damage on most cereals during outbreaks. In such situations, plant resistance is unlikely to be very helpful.

However, available levels of resistance may be useful under normal conditions to restrict population build-up. Lines E 302 and E 709 (resistant to borers) have been reported to be less damaged by *M. separata* (Kulkarni et al. 1978). SC 109-12, 1821 CM, and NK Savana 5 are resistant to *S.frugiperda* (Lordello et al. 1980, Wiseman et al. 1984, 1986).

Sorghum midge (*Stenodiplosis sorghicola*)

Substantial progress has been made in the identification and use of resistance to sorghum midge (Johnson et al. 1973; Rossetto et al. 1975; Jotwani 1978; Page 1979; Peterson et al. 1985; Teetes 1985; Singh 1987; Agrawal et al. 1987; Wiseman et al. 1988; Sharma et al. 1993). Sorghum midge resistance is also being used in breeding programs in Africa, Argentina, and El Salvador. Lines IS 2579C, IS 12666C, TAM 2566, AF 28, DJ 6514, IS 10712, IS 7005, IS 8891, and IS 8721 are diverse sources of resistance.

Major progress in breeding for midge resistance has been made in the USA, Australia, and at ICRISAT-Patancheru. ICSV 197, ICSV 735, ICSV 745 (DSV 3), ICSV 88013, and ICSV 88032 (SPV 1010) have high levels of midge resistance. Their yield potential is comparable to commercial cultivars. Sorghum midge resistance is being transferred to hybrid parents with improved agronomic backgrounds, and experimental hybrids are being tested. Sorghum hybrids with different levels of midge resistance are widely cultivated in Australia. The use of host-plant resistance in the management of sorghum midge is therefore most promising, because the levels of resistance to midge are quite high. Midge-resistant sorghum cultivars will provide greater flexibility in sowing times in the search for maximum yields and the appropriate use of available rainfall without risking midge damage.

Head bugs (*Calocoris angustatus* and *Eurystylus oldi*)

A major effort in identifying resistance to head bugs has been made in India (Sharma and Lopez 1990, 1991, 1992a,b), and in western Africa (Sharma et al. 1994; Ratnadass et al. 1995; Doumbia et al. 1985). IS 17610, IS 17645, IS 21443, and IS 17618 have moderate levels of resistance to *C. angustatus.* CSM 388, S 29, IS 14332, Malisor 84-7, and Sakoika are sources of resistance to *E. oldi.* Most of the sources of resistance have either colored grain/high tannin content, or are *guinea* sorghums from western Africa. Malisor 84-7, a line derived from *guinea* sorghums, has a moderate yield potential, is medium-dwarf in height, and has good grain quality. It can be cultivated in areas endemic to bugs in western Africa, and also used in resistance-breeding programs.

Panicle-feeding caterpillars (*Helicoverpa, Eublemma, Cryptoblabes, Pyroderces,* etc.)

Panicle-feeding caterpillars feed on the developing grain inside the panicle. The interior of the panicle is converted into frass consisting of fecal matter and silken webs. Resistance to these caterpillars has not been studied specifically, although some lines suffering less damage have been identified. Chencholam, SPV 130, SPV 69, SPV 9, RS

160, and K-Tall are resistant to head caterpillars (Balasubramanian et al. 1979; Wilson 1976; Natarajan and Sundara Babu 1988). Genotypes with loose panicles suffer little damage by head caterpillars, possibly because of easy access for parasites and predators and the adverse effect of abiotic factors (Balasubramanian et al. 1979).

The role of insect-resistant cultivars in integrated pest management (IPM)

Adequate levels of resistance are present against a few sorghum pests, e.g., sorghum midge and greenbug. However, varieties with low to moderate levels of resistance have been identified against shoot fly, stem borers, mirid bugs, and armyworms. These lines can be very useful for pest suppression over a period of time. The adverse effects of resistant genotypes on pest populations are continuous, cumulative, and without cost to the farmers. Pesticide application requires money, their effect on pest populations declines over time, and they need to be re-applied. Reduction in pest density through plant resistance also makes control operations easier through natural enemies, and reduces the number of pesticide sprays required.

Resistant varieties slow down the rate of pest population increase. This also delays the time required in which insect infestations reach economic threshold levels (ETLs). This is especially true if the resistance mechanisms increase the mortality of immature stages and prolong the developmental period of survivors. This delay may also result in the insect population reaching ETL after the most susceptible stage of the crop is over, thus eliminating the need for insecticide use. Use of HPR in IPM conserves natural enemies, preserves environmental quality, and slows down the rate of development of insecticide-resistant insect populations.

Effect of insect-resistant cultivars on insect population dynamics

The impact of insect-resistant cultivars on insect populations can be explained by using the simple insect population models of Knipling (1967) as adapted by Sharma (1985). In one season, insect population in an area planted to a midge-resistant cultivar would be 1000 times lower than in areas planted to a susceptible cultivar. By the end of 1 year, insect population would be rare in areas planted to a resistant cultivar, and remain constant in areas planted to a moderately resistant cultivar.

The most common form of IPM involves the use of moderately resistant cultivars and insecticides. Assuming that one insecticide application reduces the insect population by 90%, the combined action of an insect-resistant cultivar and insecticide would produce a 24-fold difference in population between an area sown to a susceptible cultivar, compared with an area cropped to a moderately resistant cultivar (Sharma 1985). Further, it would reduce the population carryover by 31 times, and thus result in a substantial reduction in insect numbers in the following year.

HPR based on unbalanced nutrition or toxic substances may also increase the susceptibility of insects to insecticides. Insect-resistant cultivars are also compatible with biological/cultural control. The advantage of using insect-resistant cultivars is that they can help to preserve natural enemies through reducing the need to use pesticides. The

combined use of insect-resistant cultivars and biological/cultural control would be the same as that achieved by using insect-resistant cultivars and insecticides. However, biocontrol is density-dependent, whereas insecticides are not.

Effect of plant resistance on economic threshold levels (ETLs)

Insect-resistant cultivars may increase the time required for insect infestation to reach threshold levels, or increase the threshold levels according to the nature of resistance and the criterion on which the threshold is based (Teetes 1985). If the ETL is based on the amount of damage (e.g., % deadhearts for shoot fly and stem borer, number of leaves damaged by aphids, or % leaf area consumed by army worms), and the major component of resistance is tolerance, nonpreference, and antibiosis (e.g., shoot fly, stem borers, aphids, armyworms, etc.), then a susceptible cultivar will suffer economic damage in Jul, a moderately resistant cultivar will suffer in Aug, and a resistant cultivar by the end of the season (Fig. 1 A,B,C). In cases where insect damage is limited to a particular stage and a short span of time (e.g., deadheart formation due to shoot fly and stem borer), then, based on the level of resistance, sowing can be undertaken until the expected insect

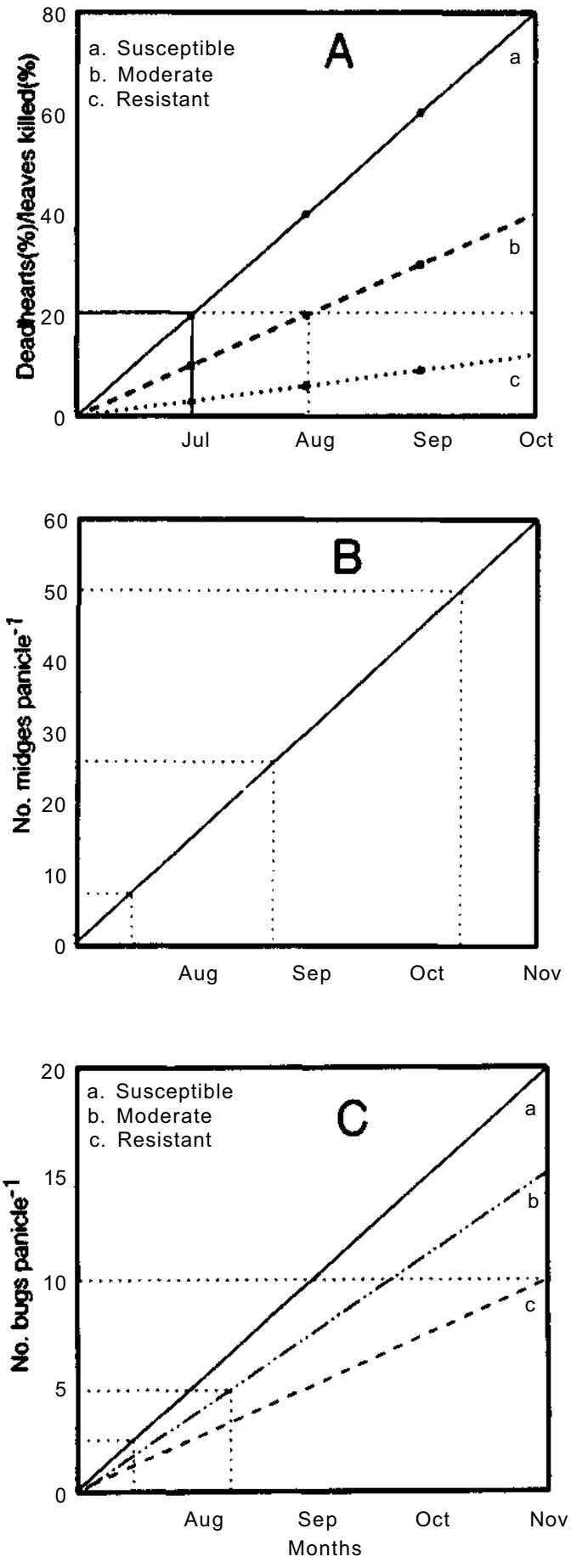

Figure 1. Effect of host-plant resistance on the economic threshold levels (ETLs) where the ETL is based on damage (1A), the nondamaging adult stage (1B), and damaging adults, on genotypes with nonpreference and antibiosis mechanism of resistance (1C).

density is below the ETL for a particular cultivar.

If the ETL is based on a nondamaging stage of insect attack (e.g., midge adults on panicles, or the number of moths of borers and army worms caught in pheromone or light traps), the ETL will increase with an increase in the level of insect resistance. In the case of sorghum midge, the ETL may be 5 adults panicle^{-1} for a susceptible cultivar, 25 adults panicle^{-1} for a moderately resistant cultivar, and >50 adults panicle^{-1} for a highly resistant cultivar (Fig. 1B). If the ETL is based on adults that also cause the damage (e.g., head bugs), and the mechanism of resistance is nonpreference and antibiosis (which will decrease the rate of population increase), then the resistant varieties will also increase and delay the ETL (Fig. 1C). ETLs for *C. angustatus* have been estimated to be 0.06-0.12 adults at the half-anthesis stage, and 5.4-10.5 adults or 7.9-15.0 nymphs at milk stage. For *E. oldi* ETLs have been estimated to be 0.97-2.52 bugs panicle^{-1} at the milk stage (O. Ajayi, pers. comm.). ETLs for head caterpillars have been worked out for a range of production levels and control costs for corn earworm in sorghum (Fuchs et al. 1993). For example, when the value of the crop is US$ 650 ha^{-1}, the ETL is 1 larva panicle^{-1}.

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Effect of Plant Resistance to Insects on the Effectiveness of Natural Enemies

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Introduction

The most effective method of reducing losses caused by insect pests involves such cultural practices as early and uniform regional planting. The crop is thus able to escape shoot fly incidence, and late-season build-up of midge populations. Additionally, resistance of plants to insects enables them to avoid, tolerate, or recover from injury that would otherwise cause greater damage to other plants of the same species under similar pest density and environmental conditions. Host-plant resistance can effectively keep pest populations below economic threshold levels, especially under low-input smallholder farming in the semi-arid tropics.

One of the consequences of pest control that combines more than one method is interactions between the different components (Emden 1981). These interactions are fundamentally important to the concept of integrated pest management (IPM).

The use of plant varieties resistant to insects is a proven, effective, economical, and safe method of pest control ideally suited to IPM. Genotypes with reasonable levels of resistance against shoot fly, stem borer, midge, and mirid bugs have been identified (Sharma 1993; Sharma and Lopez 1992; Sharma et al. 1992, 1993, 1994). A distinct advantage of using resistant plants is their compatibility with other methods of insect control. Insects feeding on resistant plants may be less vigorous and more easily killed by weather, or more easily handled, if necessary, with reduced amounts of insecticides.

Predation and parasitism have been shown to be greater on some resistant genotypes and, as a rule, natural enemies have a relative advantage if the rate of increase of their prey is slowed down, as is the case on moderately resistant plants. Thus, even a slight increase in host resistance could lead to an increase from partial to complete effectiveness of biological control. However, the secondary plant substances such as flavonoids, terpenoids, and alkaloids used as a resource by a herbivore not only affect the physiology and behavior of the herbivore, but also affect the quality of the herbivore as a resource for the beneficial insect. Changes in the host suitability due to host diet can influence the developmental rate, size, percentage emergence, success of parasitization, sex ratio, fecundity, and life span of parasitoids.

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Resistant plants may limit the availability of nutrients to parasitoids, both directly by making nutrients in the host inaccessible to a parasitoid, and indirectly by limiting the availability of nutrients to the host which would in turn result in nutrient limitation for the parasitoid. Pests stressed by antibiosis may be more susceptible to certain natural enemies, but, on the other hand, antibiotic chemicals acquired from the plant may adversely affect the natural enemies (Boethel and Eikenbary 1986). A well known example is in tomato, where tomatine absorbed by the endoparasitoid, *Hyposoter exiguae,* from its host, *Heliothis zea,* prolongs the parasitoid larval period, reduces pupal eclosion and adult size, and shortens its longevity and fecundity (Campbell and Duffey 1979). However, an antagonistic relationship between a host and the natural enemy of an insect pest does not always apply. Feeding of the fall armyworm (*Spodoptera frugiperda)* on resistant corn does not affect its parasitoid, *Campoletis sonorensis* (Isenhour and Wiseman 1989).

A number of predators and parasitoids have been reported on shoot fly, mirid bugs, stem borers and midge, especially the last two. Successful use of natural enemies for insect control depends on understanding the biology and the ecology of both the pest and the beneficial organisms operating on it. Certain mechanisms of resistance to stem borers in sorghum may increase the efficacy of some natural enemies by prolonging the exposure of the larvae/pupae to natural enemies. An increase in time required to bore into harder stems may also expose the larvae for longer periods to parasitoids and predators.

Although several studies have clearly indicated the prospects for biological control of sorghum midge, the extent to which parasitoid activity is affected by sorghum genotype is clearly not understood. In order to optimize the benefits from integrating the breeding for resistance to midge with biological control, it is desirable that these management options are either complementary, or synergistic, and not antagonistic. The effect of resistance in sorghum on midge development should be exhibited in the next trophic level of association, i.e., on midge parasitoids.

Genotype-Host-Parasitoid Interactions

Sorghum midge (*Stenodiplosis sorghicola*)

Field studies were conducted at ICRISAT-Patancheru, India, over six cropping seasons (postrainy and rainy seasons 1992-95). Three midge-resistant (ICSV 745, ICSV 89058, and IS 10712) and three susceptible (Swarna, CSH 9, and ICSV 112) sorghum genotypes were used in these studies. Experiments were laid out in a randomized complete block design. Plant spacing was 75 and 10 cm between and within rows, respectively. There were three replications. Experiments were sown on three dates in each season. Staggered multiple sowing dates were used to facilitate monitoring of sorghum midge and parasitoid populations throughout the season. Standard agronomic practices were followed for raising the crop. No insecticide was applied during the reproductive phase of the crop.

In each season, three sorghum panicles from each genotype at the half-anthesis stage in each replication (and in each sowing) were artificially infested with 40 female midges

on two successive days using the headcage technique (Sharma et al. 1988). Five days after infestation the panicles were exposed to natural parasitization for 10 days, and thereafter re-caged for parasitoid development and emergence. Similarly, another set of naturally infested panicles, but not exposed for parasitization, was used to determine the level of natural midge infestation. Emerging insects were collected using the modified headcage as described by Kausalya (1994). Natural enemies were sent to the British Museum for identification/confirmation. The level of parasitization (LP) was calculated on the basis of total emerging midge flies (m) and parasitoids (p) in the artificially infested panicles by using the formula:

$$LP = \frac{P}{m+p} \times 100.$$

Adult midge flies emerged 2-3 weeks prior to the parasitoid emergence. Midge infestation in resistant genotypes ICSV 745 and ICSV 89058 was very low. Levels of parasitization were generally higher in second and third plantings than in the first planting. There were no significant differences between genotypes *(P* 0.05) for levels of parasitization. There was, however, a clear interaction between sowing date and level of parasitization. Midge-resistant genotypes were generally less favorable for parasitoid development at all stages of host development.

An increase in midge activity was followed by an increase in *Aprostocetus* activity, and this agrees with the earlier report of Mote and Ghule (1986). The emergence of *Aprostocetus* adults occurred 1-2 weeks after the start of midge emergence. The delay in parasitoid emergence favors the build-up of midge populations because the life cycle of midge is completed in 17-20 days, and each female produces an average of 100 eggs (Passlow 1973; Murthy and Subramaniam 1975). In contrast, the life cycle of *Aprostocetus* is completed in 21-25 days, and fecundity of the parasite (50 eggs female^{-1}) is much lower (Taley et al. 1978; Garg 1979; Thontadarya et al. 1983). Such a disparity in the developmental period and fecundity between the pest and the parasitoid results in considerable midge damage in susceptible sorghum genotypes.

Parasitization levels by *Aprostocetus* were greater during the postrainy season than in the rainy season. This was observed in all the genotypes in spite of a low damage level in midge-resistant genotypes. Parasitoids were always associated with sorghum midge irrespective of midge densities. However, midge parasitization was low in the midge-resistant genotype ICSV 745. Low numbers of host larvae (midge) in the resistant genotype have been attributed to nonpreference for oviposition (Sharma 1985) and a faster rate of grain development (Sharma et al. 1990).

The role of rainfall on *Aprostocetus* populations was not clear. Mote and Ghule (1986) reported a positive correlation between rainfall and sorghum midge populations. However, rainfall distribution during the period of midge activity was not associated with either midge or parasitoid abundance in the present investigations. Peak midge activity was recorded in Oct when maximum temperature and relative humidity (RH) ranged from 27 to 31°C and 82 to 96%, respectively. Sharma (1985) and Garg and Taley (1978) reported maximum midge abundance in Sep/Oct, when temperatures ranged from 25 to 27°C and RH from 75 to 80%. Parasitism by *Aprostocetus* adults was recorded at the slightly higher temperatures of 29-36°C and at the lower RH levels of 61-76%. Perhaps

this difference between parasitoid and host insect in environmental requirements may account for the delay in parasitoid build-up.

Spotted Stem Borer (*Chilo partellus*)

Two stem borer-resistant (IS 2205 and ICSV 700) and two susceptible (ICSV 1 and CSH 9) sorghum genotypes were sown in three replications in 3 x 4-m plots. The rows were spaced at 75 cm, and plants were thinned to a spacing of 10 cm within a row. There were two main treatments of artificially and naturally infested plots, and three subtreatments of three infestations at 20, 30, and 40 days after emergence (DAE). Artificial infestation was carried out on individual plants in the center of four rows with laboratory-reared neonate larvae.

Five days after infestation, destructive sampling was undertaken at weekly intervals. Five plants were dissected per plot and examined for larvae which were collected and reared in the laboratory on fresh sorghum stem pieces in glass vials for possible parasitoid emergence and identification. Stem pieces were changed at regular intervals until successful pupation in the case of unparasitized larvae. Sampling continued until crop harvest.

Five parasitoid species were associated with spotted stem borer larvae at ICRISAT-Patancheru, namely: *Cotesia flavipes, C. ruficrus, Sturmiopsis inferens, Temelucha* spp, and *Glyptomorpha deesae. C. ruficrus* was the predominant parasitoid species (45%), followed by *S. inferens* (32%), and *C. flavipes* (13%). In general, parasitoid activity increased with crop age, and was highest at 40 DAE. Higher levels of parasitization were recorded on stem borer-resistant genotypes than on susceptible ones, irrespective of the time and method of infestation. The level of parasitization was generally higher under natural than under artificial infestation, due in part to the design of the experiment, and also due to the cumulative effect of parasitoid attack in the former.

Changes in stem borer parasitoid activity and species predominance with crop age indicates possible host plant-parasitoid interactions. The predominance of *S. inferens* in early crop growth stages may be associated with the crop's micro-environment. Similarly, the predominance of *C. flavipes* at a later stage of crop growth is perhaps related to the searching ability of this species, which is limited to large-stemmed grasses and the physiological suitability of host stem borers (Overholt et al. 1994).

On-farm Surveys

The incidence of damage by spotted stem borer and midge in farmers' fields has been monitored in order to assess the extent of parasitoid activity in various sorghum-growing areas in India. The study involved frequent on-farm surveys at intervals of 3-6 weeks in the major sorghum-growing districts of Andhra Pradesh, Maharashtra, Karnataka, and Tamil Nadu. Depending on field size, planting density, and on the frequency of sorghum fields in an area being sampled, samples of 40-200 plants were randomly selected and examined for pest incidence and parasitoid activity. Farmers, or farm laborers when present, were interviewed concerning their recognition of pests and pest damage, their perceptions of their importance relative to other production constraints, and control inputs used.

Midge infestation was relatively low in Andhra Pradesh (14%) compared with that in Maharashtra (30%). *Aprostocetus gala* was by far the most predominant parasitoid species, making up to 85-90% of the species complex.

Borer larval parasitization was significantly higher in Andhra Pradesh (>50%) than in Maharashtra (<15%), while pupal parasitization was greater in Maharashtra (34%) than in Andhra Pradesh (18%). The parasitoid species' complex and predominance also varied between regions. The tachinid *Sturmiopsis inferens* was the predominant larval parasitoid in Andhra Pradesh whereas, in Maharashtra, it was the brachonid *Cotesia flavipes. Xanthopimpla stemmator* was the predominant pupal parasitoid in both States, and was the most active stem borer parasitoid in Maharashtra.

The survey findings reported indicate that natural enemies are closely associated with sorghum insect pests in farmers' fields. The species complex and predominance varies considerably across regions. Farmers in Maharashtra predominantly (90%) cultivate high-yielding sorghum hybrids. In Andhra Pradesh, the situation is the reverse where >75% of the fields are sown to varieties and landraces which usually are more tolerant of or moderately resistant to these pests than are hybrids. Although stem borer incidence was similar in both States, it can be inferred that differences in parasitoid composition and predominance may be associated, at least in part, with differences in sorghum genotypes cultivated in these States.

Response of Spotted Stem Borer Parasitoids to Odors from Pupae and Frass

An experiment was conducted to investigate interactions between the insect, the host plant, and the parasitoid, with respect to *Pediobius furvus* response to chemical cues emanating from the host habitat and/or the host itself. A cylindrical glass olfactometer measuring 20 x 20 x 20 cm was placed 3 cm above an overhead projector. Light from the projector was passed through the glass box and through a prism where the image was reflected from a mirror onto the surface of a table. The apparatus used was two cylindrical glass tubes of 20 x 1.80 cm, and a third ruled cylindrical glass tube of 20 x 20 cm that had an outflow port of 2 x 1.2 cm. From the image reproduced on the table, a tracing was made of the parasitoid's movements without disturbing the insect (Duale 1993). The presence of an observer near the apparatus did not seem to disturb the insects, and the relay of diffusion gradients of particular odors in a given environment. Parasitoids to be tested were introduced individually in the chamber through the outflow port on the floor, and allowed a maximum of 30 min to respond to the odors emanating from pupae and frass. The position of the pupae and the frass to be tested was alternated from one arm to the other at frequent intervals during each set of experiments. In this way any errors due to imperfections in the apparatus were cancelled out.

The test using cylindrical tubes to study the behavior of *P. furvus* indicated that no detectable bias was present in the system because the mean time spent per field by the insect did not differ significantly between the fields. *P. furvus* female parasitoids showed a significant attraction to a combination of host and host frass. In all the trials undertaken,

the number of *P. furvus* females responding to a combination of host pupae with frass was twice as large as those responding to either frass or pupae alone.

Larval frass and odor from the host pupa are known to have an olfactory stimulant effect. The results clearly demonstrate that *P. furvus* females can use olfactory cues to locate their hosts. Female parasitoids were attracted by a combination of odors from the frass and the host itself. Such attraction should increase the parasitoids' searching efficiency in the field since females would move towards plants or plant parts on which stem borers are present.

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Pesticide Application and Plant Protection for Sorghum

S K Pal and V S R Das

Introduction

Plant protection and surveillance is an integral part of the insect pest management for sorghum. Three levels of insecticide protection are required: intensive, research, and economic.

Intensive protection

This involves extensive treatments and observations because the smallest crop damage can jeopardize experimental results, e.g., growth analysis studies, leaf area measurements, and other physiological and pathological trials. But the area involved is usually small. This level of protection requires surveillance twice a week, and heavy application of pesticides. Considerable commitment of resources is therefore necessary to achieve the desired degree of protection. The threshold levels on which applications are based are very low (Table 1).

Research protection

Fields under research protection include those allocated for experiments that can tolerate some pest damage without detriment to experimental data, e.g., yield trials, seed multiplication, etc. This level of protection requires surveillance once a week, and the threshold levels are higher than those required for intensive protection.

Economic protection

Experiments in this category are protected against insect pests only when the infestation threatens to cause economic damage, or loss of research material. Surveillance at this level is also done weekly, and threshold levels are higher than those for research protection.

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Table 1. Economic thresholds for major insect pests of sorghum.

Insect	Growth stage	Thresholds under different degrees of protection			Control measure[2]	Dosage	Remarks
		Intensive	Research	Economic			
Shoot fly	After 1 Jul 7 DAE[1]	>5% plants with eggs and DH[1]	20% DH before thinning	30% DH before thinning	Carbofuran 3G, or phorate 10G granules Endosulfan spray	40 kg ha^{-1} 1 L 100^{-1} L water	Apply carbofuran/ phorate at sowing. Foliar spray.
Stem borer	20 DAE	Leaf damage, or >5% DH	Leaf damage, or >10% DH	Leaf damage, or >25% DH	Endosulfan 35 EC Carbofuran 3G	1 L 100^{-1} L water 2–3 g whorl^{-1}	
Armyworm	45 DAE	One larva $plant^{-1}$, or 10% leaf damage	Leaf damage on 20% plants	Leaf damage on 40% plants before boot stage	Endosulfan 35 EC	1 L 100^{-1} L water	
Mites		Mite colonies and leaf yellowing on 5% plants	10% plants infested	25% plants infested	Dimethoate 30 EC Dicofol 18.5 EC	0.5 L 100^{-1} L water 1 L 100^{-1} L water	
Shoot bugs	Before 60 DAE	Shoot bugs, or damage in 1% plants	Damage, or infestation in >20% plants	Damage, or infestation exceeds 30%	Endosulfan 35 EC	1 L 100^{-1} L water	
Aphids	60 DAE	>5% plants with aphids at 28 DAE, or 20% plants with aphids at 60 DAE	>30% plants infested at 28 DAE or >50% plants infested at 50 DAE	>50% aphid infestation at 50 DAE. Do not spray if predators are present	Dimethoate 30 EC Malathion 50 EC	0.5 L 100^{-1} L water 0.5 L 100^{-1} L water	
Mirid bugs	60 DAE	1 adult or 10 $nymphs\ panicle^{-1}$	2 adults or 20 $nymphs\ panicle^{-1}$	5 adults or 50 $nymphs\ panicle^{-1}$	Carbaryl 50 WP Endosulfan 35 EC	1 kg 100^{-1} L water 1 L 100^{-1} L water	
Helicoverpa	90 DAE	1 medium- to large-sized larva $panicle^{-1}$	5 medium- to large-sized larvae $panicle^{-1}$	No spraying	Endosulfan 35 EC	1 L 100^{-1} L water	
Midge	60 DAE	1 midge fly $panicle^{-1}$	2–3 midge flies $panicle^{-1}$	5–6 midge flies $panicle^{-1}$	Carbaryl 50 WP Endosulfan 35 EC	1 kg 100^{-1} L water 1 L 100^{-1} L water	

1. DH = deadhearts; DAE = days after seedling emergence.
2. 3G = 3% granules; 10G = 10% granules.

Insect Control in Sorghum

Wireworms (*Gonocephalum, Eleodes, Conoderus,* and *Aeolus*)

Several species of Elateridae (click beetles) are present at ICRISAT-Patancheru research farm, the larvae of these beetles are called wireworms. The adults deposit eggs in the soil. Young larvae are creamy white, but change to shiny yellow as they grow older. Fully grown larvae are nearly 25 mm long. Wireworms hollow out the seed, and this prevents germination. The symptoms of wireworm damage in fields are bare patches of variable dimensions, and a general thinning of the crop stand. Seed treatment with insecticides (heptachlor and BHC), and soil drenching with endosulfan 35 EC (0.17%) are effective for wireworm control.

White grubs (*Holotrichia* and *Phyllophaga)*

Adults, commonly referred to as May or June beetles, are brownish black. The larvae feed on plant roots. They are C-shaped, with brown heads and white bodies. Substantial damage to sorghum occurs after seedling emergence. Seedlings begin to die and stand loss can occur within 7-10 days in severely infested fields. Infested plants that are not killed at the seedling stage are stunted and do not produce grain. A basic application of carbofuran at sowing, or soil drenching with endosulfan 35 EC (0.17%), achieves effective control.

Cutworms (*Agrotis* spp)

Cutworm larvae have a curled-up appearance and are grayish white, black, or brown. The larvae remain underground during the day and feed at night. Larvae cut off sorghum plants at, or slightly below, the soil surface. Soil drenching with endosulfan 35 EC (0.17%), or spraying with endosulfan 35 EC (0.35%), provides effective control.

Sorghum shoot fly (*Atherigona soccata*)

Shoot fly is an important pest of sorghum. It attacks the crop from 5 to 25 days after seedling emergence. The adult is a small gray fly that deposits small, white, cigar-shaped eggs singly on the undersurface of the leaf. The eggs hatch in 2-3 days and the maggots enter the plant through the whorl and destroy the growing point, resulting in deadheart formation and tiller initiation. As a result of shoot fly attack, the plant stand can be greatly reduced. Late sowing during the rainy season increases the likelihood of shoot fly damage. Adjustment of sowing dates (early sowings), a high seeding rate, use of high-yielding shoot fly-resistant cultivars, application of cypermethrin (0.05 kg ai ha^{-1}) at 6 and 12 DAE provides adequate control of this pest.

Spotted stem borer (*Chilo partellus)*

Spotted stem borer is an important pest of sorghum. It attacks the crop 2 weeks after seedling emergence until crop harvest, and affects all plant parts except the roots. Eggs are laid in masses of 10-100 on the undersurface of leaves. The first symptom of attack is elongated windows in young whorl leaves, where the larvae eat the upper lamina and leave the lower lamina intact. The grown-up larvae leave the whorl and bore into the stem at the base. Stem boring by the larvae in young plants damages the growing point, and results in deadheart formation. Both stem and peduncle damage lead to the production of completely or partially chaffy panicles. Plowing after crop harvest, destroying plant stubbles, and removal of affected plants reduces borer damage. Observation of a closed season from 15 Apr to 15 Jun (during which period no sorghum crop is allowed to grow) has been found to achieve low incidence of shoot fly and stem borer during the rainy season in India. Application of endosulfan 35 EC (0.70 kg ai ha^{-1}) gives adequate control of this pest. In experiments under intensive protection, patch application of carbofuran granules in the whorls of infested plants provides good control.

Aphids (*Rhopalosiphum maidis* and *Melanaphis sacchari)*

The corn leaf aphid, *R. maidis* is commonly found in the whorl leaves, but also on the underside of leaves, on stems, or in panicles. The sugarcane aphid, *M. sacchari,* infests the older leaves, and the infestation progresses upwards. The young and adults suck the plant sap, leading to a yellowish mottling of the leaves. The aphid produces honeydew on which molds grow.

Shoot bug *(Peregrinus maidis)*

Nymphs and adults of the shoot bug suck plant sap and cause stunted growth and death of the leaves, and sometimes of the plants. They secrete honeydew, which favors the growth of sooty mold. Demeton-methyl (0.2 kg ai ha^{-1}) and dimethoate (0.3 kg ai ha^{-1}) are recommended for aphid and shoot bug control.

Leaf defoliators (*Amsacta, Hieroglyphus, Myllocerus, and Mythimna)*

Oriental armyworm, *M. separata,* is a sporadic pest and, under heavy infestations, the entire crop may be lost. The larvae are dirty, pale brown to dark brown, with three dark brown dorsal lines. They feed on the leaves, leaving only the midrib. The red hairy caterpillar (*Amsacta albistriga*), the flea beetles, grasshoppers *(Hieroglyphus* spp), leaf weevils *(Myllocerus* spp), etc., also feed on the leaves. A number of insecticides give effective control: e.g., endosulfan 0.7 kg, quinalphos 0.5 kg, or carbaryl 1.0 kg ai ha^{-1}.

Spider mites *(Oligonychus indicus* and *O. pratensis)*

Spider mites suck sap from the underside of the leaves. The infested leaf areas are pale yellow initially, and later become reddish on the upper surface of the leaves. The entire leaf may turn brown. In case of severe infestation, the mites may invade and web sorghum panicles. Hot and dry weather usually increases mite infestation. Dimethoate (0.3 kg ai ha^{-1}) and dicofol (0.35 kg ai ha^{-1}), sprayed on the undersurface of leaves, are effective for mite control.

Sorghum midge *(Stenodiplosis sorghicola)*

Sorghum midge is 1.6 mm long and has a yellow head, brown antennae and legs, an orange-red thorax and abdomen, and grayish hyaline wings. The females lay eggs in flowering spikelets. They live for less than 24 h. Eggs are cylindrical and hatch in 2-3 days. Damage to the crop is caused by the larvae which feed on the ovary inside the glumes. This results in chaffy spikelets, and the panicles present a blasted appearance. Chemical control includes the application of dimethoate (0.3 kg ai ha^{-1}), endosulfan (0.7 kg ai ha^{-1}), or carbaryl (1.0 kg ai ha^{-1}).

Head bugs *(Calocoris angustatus* and *Eurystyius oldi)*

Head bugs are serious pests of sorghum. The adult female is about 5 mm long and yellowish green. It lays eggs inside the glumes. The nymphs are yellow to orange red, and complete their development in 17-18 days. Both nymphs and adults infest the panicles and suck the sap from developing grain, which becomes discolored, remains unfilled, and becomes chaffy. Spray carbaryl (1.0 kg ai ha^{-1}), or dimethoate (0.3 kg ai ha^{-1}) to control these head bugs.

Panicle-feeding caterpillars *(Helicoverpa, Eublemma, Cryptoblabes,* and *Pyroderces)*

American bollworm, *H. armigera,* is an important panicle-feeding caterpillar. Eggs are laid on the floral parts. The larvae feed on the developing grain. Endosulfan (0.7 kg ai ha^{-1}) and carbaryl (1.0 kg ai ha^{-1}) are effective for controlling the larvae.

When the crop is sown after 1 Jul, carbofuran should be applied in all categories of protection. Other chemicals, such as fenvalerate and cypermethrin, can also be used for insect control on sorghum. Use of dimethoate against shoot bugs is recommended in the postrainy season.

Integrated Pest Management (IPM) in Sorghum

K F Nwanze

Introduction

Brader (1979) defined integrated pest control as a pest management system that, in the context of the associated environment and the population dynamics of the pest species, uses all suitable techniques and methods such as cultural practices, host-plant resistance, insecticides, biological control, and legislation, in as compatible a manner as possible, and maintains the pest populations at levels below those causing economic injury.

Wightman (1993) described IPM as "management activities that are carried out by farmers...". Both definitions merit consideration in that they describe both ends of a continuum in research and development (R&D). In essence, the former emphasizes the process of developing IPM strategies, and the latter its implementation. However, Brader's definition, as in the early years of IPM, was a concept in which all possible control options were implied, and single option-based IPM strategies were not accommodated within the IPM framework. Today, IPM is synonymous with environmental safety and sustainability, and any nonchemical control option can therefore readily find a place in this framework. The effective management of crop pests, which sustains rather than destroys basic ecological relations in the environment, is embodied in the concept of IPM. Thus the IPM R&D continuum can be subdivided into four main phases:

- research into individual IPM components or options;
- on-station evaluation of a combination of options;
- on-farm evaluation and validation studies; and
- farmer implementation.

Existing publications on IPM of sorghum and pearl millet insect pests suggest that, in the past 20 years, our efforts have mostly been directed at the first two phases. Only recently has attention been given to the implementation phase. Apart from work done at the International Center for Insect Physiology and Ecology (ICIPE) in western Kenya (Saxena et al. 1989, 1990), there has not been a concerted effort to develop well focused IPM strategies for farmers. Individual components have been tested, and the results have been put together in publications as parts of an IPM strategy (Ajayi 1990; Nwanze 1985; 1991; Reddy 1984; Ndoye and Gahukar 1987; Sharma 1985, 1993; Gebre-Amlak 1988; Gahukar 1988, 1989; Omolo et al. 1993; Minja 1990; Sukhani 1986; Saxena et al. 1989; 1990; Sagnia 1983).

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A major gap in these attempts is the absence of diagnostic research on the characterization of the physical and biological environment, assessment of on-farm losses, socioeconomic analysis of farmers' perceptions of insects as pests, and their attitudes towards human and capital investment in pest management. These are pivotal elements in any research agenda that is targeted at the development of viable management options.

K M Harris (1959), addressing the inaugural meeting of the Science Association of Nigeria, concluded that "the first consideration, over and above technical considerations, is whether the farmer is interested in reducing attacks on his crops. There is a lack of interest, and since without interest no progress can be made, it seems that any approach towards stem borer control must be based on the education of the farmer" (Stem borers of cereals in Northern Nigeria, unpublished report). Thirty years later, Nwanze and Mueller (1989) again concluded that "most recommendations are impractical as they do not take sufficient account of the situations, resources, and needs of farmers." These conclusions are applicable not only to stem borers but also to all major insect pests of sorghum.

Components of Pest Management in Sorghum

A rapid online search of databases, 1975-94, revealed that varietal resistance, cultural methods (especially intercropping), and biological control are the three major elements in the IPM of food crops of the semi-arid tropics. It is an accepted fact that prospects for chemical control in sorghum and pearl millet production in Africa and Asia will remain a low priority as long as these crops continue to fetch low market prices compared with those for maize and rice. Other control methods, such as use of pheromones and novel bio-insecticides, are also cited.

Cultural practices

There are good scientific data to support the potential contributions of several cultural practices in IPM (sowing date, crop-residue destruction, tillage, and soil water and fertilizer management). These data are usually classified as impractical because they conflict with socioeconomic values, traditional uses of crop residues, labor-intensive cropping, and the lack of adequate financial resources. The effects of intercropping on pest populations and crop damage are well documented and are believed, among other factors, to be due to increased diversity in the agroecosystem, increased fertilization, and nonhost effects.

Water management as a cultural control tactic in rice has been shown to have potential as a substitute for the use of insecticides to control the rice weevil, *Lissorhoptrus oryzophilus,* in Louisiana, USA (Quisenberry et al. 1992). This study involved the removal of water to alter the habitat necessary for larval survival. Water management resulted in higher grain yield and graimstraw ratio than other treatments, which also involved insecticide applications. Similarly in Colombia, water drainage in rice fields (a weekly flush as against permanent flooding) resulted in fewer eggs, mines, shoot fly and pupae of the rice leafminer, *Hydrellia wirthi.*

The sorghum shoot fly, which is a major seedling pest of late-sown sorghum in Africa and India, is the major production constraint in the postrainy season crop in India where over 500 000 ha are grown under irrigation. Studies at ICRISAT-Patancheru have shown that reduced irrigation of postrainy-season sorghum during the first 4 weeks after sowing resulted in a 4-fold reduction in shoot fly oviposition and seedling deadheart formation. Plant biomass and grain yield increased by 75 and 80%, respectively (K F Nwanze, unpublished). The environmental impact of water management in these examples underscores the savings in water resources.

Other mechanical and physical practices, such as "catch and kill" or "uproot and burn", are often practiced by farmers. For example, the systematic removal of weeds and shoot fly deadhearts as soon as they appear, and their destruction by burning or dipping in an insecticide, is a common practice in Buldana district of Maharashtra, India.

Intercropping

The effects of intercropping on pest populations and crop damage are well documented. Crop combinations comprise sorghum/legume, sorghum/cereal, and a range of food and nonfood crops, involving cropping pattern and crop density combinations. The effects of intercropping are believed to be due to increased diversity in the agroecosystem, increased fertilization and crop growth, and nonhost effects of the associated crop. Intercropping is an age-old farming practice, and research at the farm level is necessary because on-station experiments are often too small, too well managed, and too often unrealistic to adequately represent on-farm conditions. On-station research in this area should therefore be limited to the initial testing of intercropping configurations.

Biological control

Published lists of the natural enemies of crops, which include parasitoids, predators, and insect pathogens are impressive, but no lasting successes have been reported in sorghum. However, recent work at ICIPE, the International Institute of Tropical Agriculture (IITA), and the CABI Institute of Biological Control indicates that there are prospects in the near future for using insect pathogens in the control of locusts and grasshoppers (IITA 1991; ICIPE 1993).

Several lists of the natural enemies of *Chilo partellus, Busseola fusca,* and *Coniesta ignefusalis,* have been published. These provide an excellent reference on species composition, distribution, and, if available, information on their efficiencies as natural control agents. The most recent of these are by Sharma and Davies (1988), Mohyuddin (1990), Harris and Nwanze (1992), and Nwanze and Harris (1992). These lists are not exhaustive, but they are comprehensive and include parasitoids, predators, and insect pathogens. However, it is known that existing natural enemies do not appear to be effective in regulating the abundance of insects (Youm 1990; Nwanze and Harris 1992). Because of the range of natural enemies, it has been suggested that applied biological control should be explored. This should be considered with caution since we really need to know more about indigenous natural enemy complexes before planning wider implementation of classical biological control by the introduction of exotic parasitoids.

Based on the large number of hymenopterous parasitoids of stem borers in Asia, Mohyuddin and Greathead (1970) recommended the introduction of five species into eastern Africa. Gilstrap (1985) also indicated that the prospects were excellent for biological control of seven sorghum stem borers, including *C. partellus* and *B. fusca,* and that both importation and conservation tactics are most likely to be useful. Information on natural enemies has been summarized by Appert et al. (1969), Alam et al. (1972), Ingram (1983), and Skoroszewski and Van Hamburg (1987). Records show that the only effective case so far was against stem borers on sugarcane in the Caribbean, and no lasting success has been reported on sorghum. However, efforts are continuing, and prospects are encouraging in the work being carried out at ICIPE. Studies of an exotic parasitoid, *Cotesia flavipes,* indicated higher searching ability and acceptability than the indigenous species. Similarly, research on insect pathogens has shown compatibility between resistant sorghum genotypes and *Bacillus thuringiensis, Nosema marucae,* and *Beauveria bassiana,* which alone are reported to effectively reduce *C. partellus* damage (ICIPE 1993).

Chemical control

There are strong proponents for the "safe use" of insecticides. Even with so-called environmentally safer chemicals, there is little convincing evidence of the economic soundness of some of the recommendations made for insecticide use on major food crops, given the current yield levels in sorghum-based subsistence agriculture. Unfortunately, as greater importance is still attached to cash crops, insecticides continue to be a major component of international aid, governments continue to subsidize insecticides, and, more often than not, developing countries have sadly become the dumping ground for products long-since banned in the developed world.

Apart from problems in procurement and proper application, known cases of insecticide mismanagement continue to be a threat to animal and human health, and a major factor in environmental pollution. Where insecticides must be used, their application should be based on economic threshold levels (ETLs). ETL estimates are available for shoot fly, sorghum midge, and head bugs, but much research remains to be done on stem borers, aphids, shoot bugs, and spider mites.

Biopesticides

Insect control involving pheromones, bacteria, viruses, chemosterilants, genetic sterility, irradiation, antifeedants, and repellents have been tried on some crops/insects with varying degrees of success. Sex pheromones can be used as male attractants for monitoring abundances of *C. partellus, Stenodiplosis sorghicola,* and *Helicoverpa armigera.* Research on the pearl millet stem borer sex pheromone is well advanced in western Africa. Field dispensers impregnated with the synthetic formulation and an efficient locally made water-based pheromone trap have been developed, and can be used for mass trapping or mating disruption (Youm and Beevor 1995).

Extracts from neem *(Azadirachta indica)* and custard apple *(Annona squamosa)* seeds reduce damage by spotted stem borer, oriental army worm, shoot bug, and head bug. This

can result in a yield increase of 25-30%. Several neem-based formulations are now available in the market (Sharma et al. 1996). Toxins from *Bacillus thuringiensis* are also effective against the stem borer, armyworm, and some species of head caterpillars. More research is needed on the efficacy and usefulness of these control measures against the insect pests of sorghum.

Host-plant resistance

Insect management through host-plant resistance is recognized as a long-term control measure. Its success is highly dependent on access to world germplasm reserves for systematic screening using insect bioassays that permit easy identification of resistant material, and which guarantee reliable and consistent results. Screening techniques and resistance identification parameters have been developed and standardized at 1CRISAT-Patancheru for the major pests (Sharma et al. 1992). These methods have been used to screen over 30 000 accessions from the world sorghum collection to identify sorghum genotypes resistant to shoot fly (60), stem borer (72), midge (30), and head bug (18).

Sharma (1993) has provided information on the use of resistant sorghum cultivars in IPM in different ecosystems. There has been a remarkable success in India, Australia, and the USA in developing high-yielding midge-resistant sorghums, and stem borer-tolerant selections such as Maldandi and Serena, which are widely cultivated by farmers in India and eastern Africa, respectively. But, in general, sorghum insect pests have not yielded to successful conventional resistance-breeding approaches. Over 99% of the genotypes listed by Sharma (1993) are described as "highly promising", having "good potential", or are "superior to susceptible controls", but they have not gone beyond research stations and onto farmers' fields. Basically, resistance levels are either too low to result in significant genetic improvement when transferred into agronomically improved cultivars, or conventional breeding techniques have not yielded agronomically desirable products.

Our knowledge of resistance mechanisms and factors, and the bases of gene action and inheritance, is not lacking. The range of morphological, physiological, and chemical factors, or traits associated with resistance to insects clearly indicates an area that has been extensively studied. In spite of this, these traits present problems for traditional breeding approaches. Apart from the fact that cultivated sorghums lack sufficient levels of resistance to major insect pests, resistance traits are quantitatively inherited and have been difficult to manipulate (Stenhouse 1991). An immediate question therefore is: can existing knowledge and material be exploited in ways other than traditional breeding methods? This question needs a critical examination.

Farmer participation in research

Applying methods and principles of entomology in subject-matter research on crop pests in Africa and Asia is necessary. However, the direct transfer of approaches to problem-solving in crop pest research from developed to developing countries is not advisable. It could easily bias problem identification and encourage the acceptance of recommendations without critical appraisal. As an example, at the very outset of any problem-

solving pest management research, we should provide evidence on whether the insect is merely a pest, or a pest problem for farmers. This distinction between pests and pest problems is important, because farmers must be able to associate the pest with economic damage in relation to the perceived losses caused by other biotic and abiotic yield-reducing factors. One does not need to emphasize the contrast between farmers and their environment in the developing and the developed countries from where research approaches are borrowed. These differences often prevent successful solutions from being directly transferred from developed to developing countries.

Evaluating new technologies in on-station experiments is the mainstay of traditional pest management research. However, a much broader set of criteria is required for evaluating the results of similar experiments under on-farm conditions. The most crucial test of any new technology is its adoption by farmers. Such studies currently rely on surveys to document the success of the research. But, if such surveys are to provide unbiased input in the design of the next generation of technologies, as far as possible the technology-developers should be excluded from evaluation and appraisal work, except as resource persons. Information from impact assessment studies can become an important component in a rigorous and relentless canvassing of support from farmers—the primary beneficiaries of new and improved agricultural technologies—and from governments.

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Part 6

International Insect Resistance Testing Program

International Insect Resistance Testing Program

H C Sharma and K F Nwanze

Introduction

The prime objective of the ICRISAT Sorghum Improvement Program is to increase and stabilize sorghum production in the semi-arid tropics. To achieve this goal, it is necessary to provide national agricultural research systems with genotypes that have higher and stable yield potential than those currently grown by the farmers. To obtain yield stability it is essential to have genotypes with some level of resistance to insect pests. Thus, one of the important objectives of our work is to identify resistant sources for various insect pests from world germplasm resources and use them in pest-resistance breeding research. To ensure that such resistance is broad-based, it is necessary to test the material across a range of environments and under differing pest situations.

The International Sorghum Insect Pest Resistance Testing Program was therefore established as a cooperative effort for:

a) identifying broad-spectrum and stable sources resistant to insect pests;
b) distributing sources of resistance and improved breeding lines to interested workers;
c) providing information on variability in insect populations at different locations; and
d) acting as a communication link between entomologists and breeders in different regions of the semi-arid tropics.

International Sorghum Shoot Pest Nursery (ISSPN)

Shoot fly (*Atherigona soccata*) is a widespread pest of sorghum in Asia, Africa, and Mediterranean Europe. It causes damage to young sorghum seedlings, usually 10-30 days after sowing. White elongate eggs are laid singly on the underside of leaves parallel to the midrib on the third to seventh leaf. Damage is characterized by wilting and subsequent drying of the central shoot, thus causing a deadheart to develop. Deadhearts can easily be pulled out, following which a white or yellowish legless maggot may often be seen. Wilting of the main shoot often results in tillering, and these tillers may also be attacked.

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Sharma, H.C., and Nwanze, K.F. 1997. International insect resistance testing program. Pages 185-194 *in* Plant resistance to insects in sorghum (Sharma, H.C., Faujdar Singh, and Nwanze, K.F., eds.). Patancheru 502 324, Andhra Pradesh, India: International Crops Research Institute for the Semi-Arid Tropics.

There are different species of stem borers that attack sorghum, and they vary considerably from area to area. In Asia and the lowland areas of eastern Africa, the spotted stem borer (*Chilo partellus*) is usually the most important species. Corn stalk borer (*Busseola fusca*) is predominant in western Africa and the highland areas of eastern Africa. Other borers attacking sorghum are *Sesamia* spp, *Diatraea* spp, and *Eldana saccharina,* causing significant losses in some areas. *C. partellus* damage is most commonly characterized initially by chewing by the insect of the young leaves in the central whorl, which leads to 'windowing'. The insect then attacks the growing tip and kills the central shoot causing the characteristic 'deadheart' symptoms. The stem may be extensively tunneled by the larvae. In late attacks the peduncle (last internode of stem bearing the panicles) may also be tunneled, resulting in a withered panicle with no seed (chaffy) or in breakage of the stem below the panicle.

Some germplasm lines and breeders' material have been identified as resistant to shoot fly and stem borer at ICRISAT-Patancheru. It is proposed to test this material under different environmental conditions to find out whether the resistance is stable across locations.

Sowing

To ensure adequate insect attack, the trial should be sown 3-4 weeks later than the normal sowing time, or when the shoot fly and stem borer abundance is maximum. Thinning should be done 1 week after germination, keeping a plant-to-plant distance of 10 cm. Otherwise, recommended agronomic practices should be followed. Normally, no insecticides are applied in this trial, but plant protection measures may be adopted, if necessary, to control panicle-feeding pests (midge and head bug).

Observations

Researchers participating in the ISSPN program are recommended to use the sample proforma Data Recording Sheet, with self-explanatory columns for data entry, given on the following page.

Four weeks after crop emergence, count the total number of plants, plants with shoot fly deadhearts (SFDH), plants showing stem borer leaf-feeding symptoms (PSBLF), leaf-feeding score (LFS), and stem borer deadhearts (SBDH). The leaf-feeding score should be recorded on a 1 to 9 scale (where 1 = <10% leaf area eaten by the larvae, and 9 = >80% leaf area eaten). Six weeks after crop emergence, take a second count for stem borer infestation by recording plants with leaf-feeding symptoms and deadhearts.

At harvest, count the number of plants with peduncle damage, the number of tillers, and tillers with harvestable panicles, and chaffy/broken panicles. Also record the grain yield. Additional information, such as days to 50% flowering, could usefully be given in the Remarks column of the sheet.

Data Recording Sheet (ISSPN)

Location_____Date of sowing_____Date of emergence_____

Plot no.	Entry no.	4 WAE[1]					6 WAE[1]			Plants with peduncle damage	No. of harvestable panicles	No. of chaffy/ broken panicles	Grain yield	Remarks
		Total plants	SFDF	PSBLF	LFS	SBDH	PSBLF	LFS	SBDH					
101	14													
102	25													
103	21													
104	12													
105	2													
106	4													
107	3													
108	1													
109	20													
110	13													
111	5													
112	9													
113	10													
114	22													
115	16													
116	11													
117	6													
118	23													
119	18													
120	7													
121	17													
122	19													
123	15													
124	8													
125	24													

1. SFDH = Shoot fly deadhearts; PSBLF = Plants showing borer leaf feeding; LFS = Leaf feeding score; SBDH = borer deadhearts; WAE = Weeks after emergence.

International Sorghum Midge Nursery (ISMN)

Sorghum midge (*Stenodiplosis sorghicola*) is the most destructive pest of grain sorghum and is a major problem in all the sorghum-growing areas of the world. Yield losses are frequently very severe. There is evidence that sources of resistance do occur, but resistant materials are unstable at different locations, i.e., lines showing resistance at one place can become susceptible at another. The primary aim of the International Sorghum Midge Nursery is to identify the broad-spectrum stable resistance sources, and to distribute high-yielding midge-resistant lines to national agricultural research systems.

Sowing

To maximize the opportunity of midge attack, trials should be sown about 3 weeks later than the normal sowing date of the main crop at each location. Infester rows of a susceptible cultivar are sown 20 days in advance to encourage midge attack. Infester rows of early-flowering lines (<40 days to flowering after seedling emergence: e.g., IS 802, IS 13249, and IS 24439) could be sown along with the test material. Four rows of infester material should be sown after every 16 test rows. Each entry should be sown in two rows and two replications. To ensure reasonable crop uniformity, a basal dose of 40 kg nitrogen (N_2) and 20 kg phosphorus (P_2O_5) are given. A side dressing of 40 kg N_2 can be given 30-40 days aftef germination. Normally, no insecticide should be used in this trial, but it is recognized that plant protection measures could be necessary in some situations to control seedling pests. No insecticide should be applied before 2 weeks of panicle emergence or after panicle emergence.

Observations

One of the major difficulties encountered in midge resistance evaluation is the effectiveness of a criterion or technique adopted to screen for resistance to sorghum midge. Variable day-to-day midge populations and differences in flowering of different germplasm lines pose a major problem in attempting to obtain uniform midge population pressure on all test entries. In addition, the symptoms (chaffiness) of natural sterility and extensive grain damage by sucking insects are superficially similar to midge damage. Midge-infested panicles have either small white pupal cases attached to the tip of damaged spikelets, or small parasite exit holes in the glumes. The following methods are suggested for resistance evaluation.

Midge damage

This is the most appropriate criterion by which to evaluate sorghum lines for midge resistance (see on the following page the sample ISMN Data Recording Sheet). Tag 5 panicles in each genotype at half-anthesis. Record midge incidence in the spikelets 15 days after flowering, as follows:

Collect 5 primary branches each from the top, middle, and bottom portion of the panicle. Bulk the samples from all five tagged panicles in a genotype. Remove secondary branches from the primary branches and mix the sample thoroughly. Pick up the secondary branches at random and separate the developed grains and undeveloped chaffy spikelets in a sample of 500 spikelets. Enter the number of chaffy spikelets (per 500 spikelets collected) on the data sheet. Squeeze the chaffy spikelets between the thumb and forefinger or with forceps. Record the number of spikelets producing red ooze (midge-damaged spikelets produce a red ooze on squeezing), and enter the number of midge-damaged spikelets per 500 spikelets.

Midge-damaged chaffy spikelets can also be recorded at harvest by adopting the procedure described above.

Visual scoring. When the crop matures, visual scores should be given on a 1 to 9 rating scale as follows:

Damage rating	Midge-damaged spikelets (%)
1	<10
2	11-20
3	21-30
4	31-40
5	41-50
6	51-60
7	61-70
8	71-80
9	>80

Grain yield. Grain yield under protected and unprotected conditions can also be recorded as a measure of resistance to sorghum midge. Midge damage in the protected panicles/plots can be avoided either by covering the panicles with muslin cloth bags before flowering, or through insecticide application at flowering. Harvest all panicles from the middle row(s) of each plot at maturity and record the panicle and grain mass of the protected and unprotected panicles/plots.

International Sorghum Head Bug Nursery (ISHBN)

Head bugs (*Calocoris angustatus,* and *Eurystylus oldi)* are major pests of sorghum in Asia and western Africa. Under heavy infestation, the grains are totally shriveled or remain undeveloped, and are unfit for human consumption.

Information on the species involved and their relative importance is incomplete. Cooperators should therefore indicate the predominant species, and the other head bug species involved. A number of sorghum lines that are moderately resistant to the head bug *C. angustatus* have been identified. It is proposed to test these lines under different environmental and pest situations to identify broad-spectrum and stable sources of resistance to head bugs.

ISMN Data Recording Sheet

Location_____Replication_____Date of sowing_____

Plot no.	Entry	Days to 50% flowering	DR[1]	Midge-damaged spikelets (%)	Grain yield (tha^{-1})	Agronomic score[2]
101						
102						
103						
104						
105						
106						
107						
108						
109						
110						
111						
112						
113						
114						
115						
116						
117						
118						
119						
120						
121						
122						
123						
124						
125						

1. DR = Damage rating (1 = 10%, and 9 = >80% midge-damaged spikelets).
2. Agronomic score (1 = Good, and 5 = Poor).

Sowing

Trials should be sown about 3 weeks later than the normal sowing date of the main crop at each location to augment bug infestation. Infester rows of susceptible cultivars can be sown 20 days in advance to encourage head bug attack. Two rows of infester material should be sown after every eight test rows. To ensure reasonable crop uniformity, a basal dose of 40 kg N_2 and 20 kg P_2O_5 should be given. A side dressing of 40 kg N is also useful 30-40 days after germination. Normally, no insecticide should be used in this trial, but it is recognized that plant protection measures may be necessary in some situations to control seedling pests. No insecticide should be applied 2 weeks before panicle emergence, or 2 weeks after panicle emergence.

Observations

Head bugs suck the sap from developing grains, which leads to the shriveling and tanning of grains. Some grains remain undeveloped. The damage symptoms are normally evident on some or all the grains. In some cases, a portion of the panicle may be more damaged than the rest, and some grains remain normal while others show damage symptoms.

Visual scoring. Sorghum lines should be evaluated visually by looking inside the panicles at maturity. The panicles are rated for bug damage on a 1 to 9 scale, as follows:

1 = all grains fully developed with a few feeding punctures.
2 = grain fully developed, with feeding punctures.
3 = grains showing damage symptoms with slight tanning/browning.
4 = most grains with feeding punctures, and a few showing slight shriveling.
5 = grains showing slight shriveling and browning.
6 = grains showing more than 50% shriveling and turning brown or tanned.
7 = most grains highly shriveled with dark brown coloration.
8 = grains highly shriveled and slightly visible outside the glumes.
9 = most of the grains remaining undeveloped and invisible outside the glumes.

Head bug counts. Tag five panicles at random in each geqotype at half anthesis. Sample the panicles for bugs 20 days after flowering in a polyethylene bag containing a cotton swab soaked in 2 mL of ethyl acetate or benzene. Count the total number of adults and nymphs.

Grain yield. Harvest all panicles from the middle row(s) of each plot at the time of maturity and record the panicle and grain mass in each plot.

Grain hardness.[1] Evaluate grain hardness on a 1 to 5 scale (1 = very hard, and 5 = soft grain).

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1. Note that Data on grain hardness, 1 000-grain mass, floaters (%), and germination (%) should be collected only in properly conducted trials, when the researchers concerned intend to collect additional data for in-depth assessment of resistance to head bugs. The sample General Information Data Sheet is provided for use by researchers when the recording of further background information relating to the Testing Program is considered to be necessary.

ISHBN Data Recording Sheet

Location_____Replication_____Date of sowing_____

Plot no.	Genotype	Days to 50% flowering	DR[1]	No. of head bugs panicle[1]	Grain yield (kg ha^{-1})	Agronomic score[2]
101						
102						
103						
104						
105						
106						
107						
108						
109						
110						
111						
112						
113						
114						
115						
116						
117						
118						
119						
120						

1. DR = Damage rating (1 = Grains with a few feeding punctures, and 9 = Grains showing >60% shriveling).
2. Agronomic score (1 = Good, and 5 = Poor).

Grain mass and percentage floaters. Take a sample of 1000 grains at random from each replication. Equilibrate the samples for moisture content overnight (12 h) at 37°C. Record the grain mass on an electronic balance. Prepare a sodium nitrate solution of 1.31 specific density. Put the 1 000-grain sample in a beaker containing the sodium nitrate solution. Count the number of grains floating on the surface and express it as a percentage of the total number of grains.

Germination test. Take 100-grains at random from each replication and place them between the folds of a water-soaked filter paper in a petri dish. Keep the petri dishes in an incubator at 27±1°C or at room temperature in the laboratory. Record the percentage of grains with radical and plumule emergence after 72 h.

General Information Data Sheet

1. Trial _____

2. Institute _____ Location _____

3. Cooperator(s) _____

4. Latitude _____ Longitude _____ Altitude _____

5. A. Date of sowing _____ Date of emergence _____

   B. Date of thinning _____ Distance between rows _____

   C. Distance between plants _____ Date of harvesting _____

6. Weather data

Parameter	J	F	M	A	M	J	J	A	S	0	N	D
A. Rainfall (mm)												
B. No. of rainy days												
C. Temperature (max)												
Temperature (min)												
D. Relative humidity (max)												
Relative humidity (min)												

7. Irrigation _____
   (quantity and dates)

8. Fertilizer applied _____
   (amount, type, and time)

9. Pesticide _____
   (insecticide, date of application, and dosage)

10. *Striga* (% incidence) _____

11. Other relevant notes _____

Role of Networks in Collaborative Research and Technology Exchange

C L L Gowda and A Ramakrishna

Introduction

Networking is being widely used to avoid duplication of effort, and to engage, at relatively low cost, a critical mass of research and development staff to address and solve specific problems. Networks enhance interaction and exchange of information, knowledge, and material among the members.

Networks have been defined in different ways according to their purpose, form, and method of operation. An agricultural research network is a group of individuals, or institutions linked together because of commitment to collaborate in solving or addressing a common agricultural problem, or set of problems, and to use existing resources more effectively (Faris 1991).

Network types

Networks can be formal or informal. If a group of scientists meet to discuss and review past research results and plan future research on a topic, that meeting can be regarded as a form of 'network' if the participants interact and exchange information and material. A classification of networks has been proposed by Plucknett et al. (1990), as follows:

- Information exchange network: Disseminates available information, methodologies, and research results to members [e.g., the Semi-Arid Tropical Crops Information Service (SATCRIS) at ICRISAT].
- Material exchange network: Exchanges germplasm and breeding materials (e.g., the International Nurseries and Trials Network), or machinery (e.g., ARNAM) among cooperating scientists.
- Scientific consultation network: Allows individuals or groups to conduct independent research, slightly modifying on-going research to serve the goals of the network, and share the results (e.g., the International Soybean Program, INTSOY).
- Collaborative research network: Jointly plans and conducts research to address common research interests (e.g., the Cereals and Legumes Asia Network, CLAN).

ICRISAT Conference Paper no. CP 1184.

Gowda, C.L.L., and Ramakrishna, A. 1997. Role of networks in collaborative research and technology exchange. Pages 195-200 *in* Plant resistance to insects in sorghum (Sharma, H.C., Faujdar Singh, and Nwanze, K.F., eds.). Patancheru 502 324, Andhra Pradesh, India: International Crops Research Institute for the Semi-Arid Tropics.

Network objectives

Sharing of research responsibilities, resources, information, and technology are the main goals of most networks. The following is a generalized list of objectives for agricultural research networks (modified from Faris and Ker 1988).

- To strengthen (basic and applied) research capability of members to identify, address, and solve common problems.
- To generate appropriate technology by using existing research staff, facilities, and other resources more effectively.
- To ensure stability of agricultural production through a responsive research capability.
- To provide technical and financial support to facilitate coordination of activities.

Network components

The following five major components characterize a network (Faris 1991):

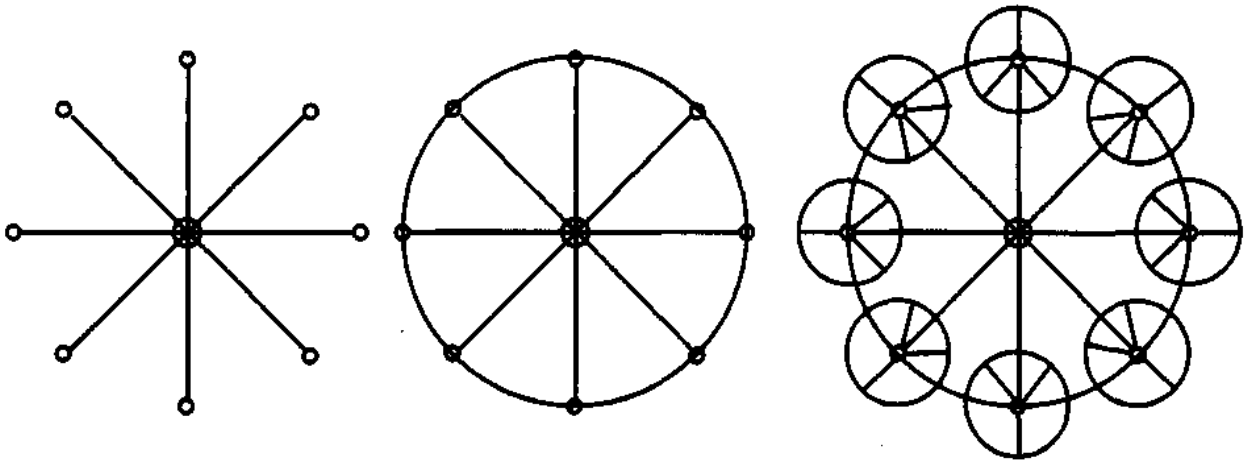
- *Membership:* comprising interested scientists and administrators who form the body of a network.
- *Assets:* include the members and the facilities and resources made available with them.
- *Coordination:* which is required to effectively organize and harmonize network activities.
- *Communication:* which enables interchange of information, material, and technology.
- *Research:* the major component around which a network is organized, including all research-related components such as information, literature, genetic material, technology, and methods.

Network structure

The structure of a network, depicting how its components are linked and how they interact, also indicates a network's nature and function. The key element is the linkages among members which encourage exchange. The most commonly used structure is the wheel-model, where the 'hub' represents the coordination unit which is connected to the 'nodes' (the members) through the 'spokes' (coordination). The nodes are themselves linked to form the 'rim' which represents direct communication among members—an important consideration for a successful network (Fig. 1).

Network costs and benefits

There are costs involved, and there can be problems associated with networks. They demand commitment of staff and resources, and may alter research priorities of individual members. Although networks are likely to benefit the members by strengthening their research capabilities, they do not build facilities or employ many permanent staff. The research costs are funded by the operational costs of the members'



(a) (b) (c)

Figure 1. A wheel-like description of networks showing the coordinating hub in the center, the spokes linking the nodes (a), the rim joining the nodes (b), and the nodes forming the research network units or subnetworks (c).

research projects. However, additional funds may be provided from other sources to undertake special projects, activities, and for coordination.

The Cereals and Legumes Asia Network (CLAN)

CLAN was established in 1992 to serve as a research and technology exchange network for Asia involving sorghum, pearl millet, chickpea, pigeonpea, and groundnut. CLAN amalgamated the activities of two earlier networks, the Asian Grain Legumes Network (AGLN) and the Asian component of the Cooperative Cereals Research Network (CCRN). CLAN consists of scientists and administrators in Asian countries who have indicated their interest and willingness to commit resources to undertake collaborative research, participate in network activities, and share research results and technology. Membership includes staff from more than 15 Asian countries, regional and international institutions primarily in Asia and elsewhere, and ICRISAT scientists. Currently the Coordination Unit is located at, and supported by, ICRISAT-Patancheru.

The overall objective of CLAN is to support, coordinate, and enhance technology exchange involving CLAN priority crops and their resource management among Asian scientists. The ultimate goal is to improve the wellbeing of the Asian farmers by improving the production and productivity of crops in a sustainable manner. The specific objectives are to:

- Strengthen linkages and enhance exchange of germplasm, breeding material, information, and technology options among members.
- Facilitate collaborative research among members to address and solve high-priority production constraints, giving attention to poverty and equity issues as per needs and priorities of member countries.
- Assist in improving the research and extension capability of member countries through human resource development.
- Enhance coordination of regional research on sorghum, pearl millet, chickpea, pigeonpea, and groundnut.

- Contribute to the development of stable and sustainable production systems through a responsive research capability in member countries.

Network activities

CLAN supports diverse activities in the region and member countries, based on expressed needs.

A bilateral work plan between ICRISAT and a member country becomes a part of the Memorandum of Understanding (MOU). These work plans are prepared at Review and Work Plan Meetings held in each country. At these meetings, scientists from the national agricultural research system (NARS) present a review of previous research on CLAN mandate crops and outline future plans. The work plan for each country is prepared on the basis of need, interest, and capabilities of the national programs. In addition to collaborative research plans and a list of experiments, work plans contain details of germplasm exchange, monitoring tours, meetings, training, and administrative and protocol procedures. They also identify a commitment and responsibility to carry out the agreed plan of work. Some major activities are listed below.

Germplasm and breeding material exchange. Because national programs have contributed local landraces to the ICRISAT gene bank for storage, multiplication, and sharing with other NARS, sorghum, pearl millet, chickpea, pigeonpea, and groundnut germplasm is available at the ICRISAT gene bank, and genetically improved breeding material of these crops is shared with network members. The network facilitates these exchanges, and assists NARS in testing, evaluation, and use of these materials in the national programs.

Training. The Coordination Unit assists the Training and Fellowships Program and CLAN members by:

- identifying training needs and financially supporting the participants;
- organizing special training courses, and tailored individual programs, on specific topics to develop and update research skills; and
- arranging in-country training programs to share the latest knowledge on research techniques, results, and technology related to the network's mandate.

Information exchange. CLAN members are given access to the services provided by ICRISAT's Information Management and Exchange Program (IMEP). These include the following:

- Research and information bulletins; proceedings of workshops, conferences, and meetings; field and laboratory handbooks and manuals; international newsletters; and other ICRISAT publications.
- Selective dissemination of information, literature searches, and other documents/ reprints as requested.

  In addition, the Coordination Unit collects reports, books, unpublished manuscripts, etc., from member countries for sharing with other members.

Collaborative research. Agreed joint research between NARS and ICRISAT, and with mentor institutions and ICRISAT, forms an integral part of network activities.

Working groups. These are formed by a group of committed scientists having a common interest in addressing and finding solutions to a high-priority regional problem. Their work as a group helps to avoid duplication and engages a critical mass of scientists in solving research problems. Group membership comprises interested scientists with relevant expertise from NARS and regional, mentor, and international research institutions who are committed to working together, and to sharing resources and data. Each working group nominates a Technical Coordinator (who is an expert in the subject) to liaise, coordinate, and harmonize research efforts. The Technical Coordinator is normally supported by a network or institution which provides logistic and administrative support.

Several specialized working groups have been established under CLAN auspices to carry out research on specific high-priority regional constraints to production. Examples are:

Asia-Pacific groundnut viruses, bacterial wilt of groundnut and drought, *Botrytis* gray mold of chickpea, and tolerance in sorghum.

Special projects. Based on the requirements of national programs and on donor interests, CLAN executes special projects in member countries, either on a bilateral or a multilateral basis. Examples are the Sri Lanka Pigeonpea Production Project in which CLAN executes the project that has been funded by the Asian Development Bank to enhance the production and use of pigeonpea to reduce imports of lentil dhal; and the FAO project on Asian Grain Legumes On-farm Research (AGLOR) in Indonesia, Nepal, Sri Lanka, and Vietnam concerning on-farm adaptive research that leads to sustainable increase in the production of legume crops.

Funding

Most of the funding support for network activities comes from member countries. The NARS use existing staff, facilities, and resources in their institutions to carry out collaborative research. ICRISAT provides support for the Coordination Unit, and partially supports the costs of scientists' travel, training, and workshops/meetings, in addition to supporting ICRISAT-based research programs aimed at developing intermediary or end-use technologies for member NARS in the region. CLAN provides additional support funds, solely on a basis of need, to support quality research, special-topic research, and working group research; and for organizing meetings, workshops, study tours, training courses, and information dissemination. Additional funding for network activities comes from the Asian Development Bank, FAO, and other donors.

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Appendix 2. Abbreviations and Acronyms

ADF	acid detergent fiber
CBD	complete block design
CLAN	Cereals and Legumes Asia Network
CRD	completely randomized design
DAE	days after emergence
df	degrees of freedom
EM	electron microscopy
ETL	economic threshold level
FC	free choice
GC	gas chromatography
GCA	general combining ability
HPR	host-plant resistance
IBD	incomplete block design
ISHBN	International Sorghum Head Bug Nursery
ISMN	International Sorghum Midge Nursery
ISSPN	International Sorghum Shoot Pest Nursery
IPM	integrated pest management
IVDMD	in vitro dry matter digestibility
LP	level of parasitization
LS	least square
LSD	latin square design
MS	mean square
NARS	national agricultural research system
NC	no-choice
NDF	neutral detergent fiber

PCR	polymerase chain reaction
RAPD	random amplified polymorphic DNA
RCBD	randomized complete block design
R&D	research and development
RFLP	restriction fragment length polymorphism
RH	relative humidity
RI	resistance index
SCA	specific combining ability
SE	standard error

About ICRISAT

The semi-arid tropics (SAT) encompasses parts of 48 developing countries including most of India, parts of southeast Asia, a swathe across sub-Saharan Africa, much of southern and eastern Africa, and parts of Latin America. Many of these countries are among the poorest in the world. Approximately one-sixth of the world's population lives in the SAT, which is typified by unpredictable weather, limited and erratic rainfall, and nutrient-poor soils.

ICRISAT's mandate crops are sorghum, pearl millet, finger millet, chickpea, pigeonpea, and groundnut; these six crops are vital to life for the ever-increasing populations of the semi-arid tropics. ICRISAT's mission is to conduct research which can lead to enhanced sustainable production of these crops and to improved management of the limited natural resources of the SAT. ICRISAT communicates information on technologies as they are developed through workshops, networks, training, library services, and publishing.

ICRISAT was established in 1972. It is one of 16 nonprofit, research and training centers funded through the Consultative Group on International Agricultural Research (CGIAR). The CGIAR is an informal association of approximately 50 public and private sector donors; it is co-sponsored by the Food and Agriculture Organization of the United Nations (FAO), the United Nations Development Programme (UNDP), the United Nations Environment Programme (UNEP), and the World Bank.

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