Techniques to Screen Sorghums for Resistance to Insect Pests

Information Bulletin no. 32

International Crops Research Institute for the Semi-Arid Tropics

Abstract

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Sorghum (*Sorghum bicolor* (L.) Moench] is one of the most important cereal crops in the semi-arid tropics. Grain yields on peasant farms are generally low, insect pests being one of the major factors limiting production. There are over 150 species which damage sorghum crops, of which sorghum shoot fly (*Atherigona soccata*), spotted stem borer (*Chilopartellus*), sorghum midge (*Contarinia sorghicola),* and head bugs *(Calocoris angustatus* and *Eurtystylus immaculatus)* are the major pests worldwide.

This bulletin describes techniques to screen for resistance under choice (field) and no-choice (cage) conditions, methods of evaluating insect damage, and the sources of resistance to the major pests.

Résumé

Référence : Sharma, H.C., Taneja, S.L., Leuschner, K., et Nwanze, K.F. 1992. **Techniques de criblage des sorghos pour la résistance aux insectes ravageurs**. Bulletin d'information n° 32. Patancheru, A.P. 502 324, India : International Crops Research Institute for the Semi-Arid Tropics. 48 p. 28 références. Mots-clés : *Sorghum bicolor*, techniques de criblage, insectes ravageurs, mouche des pousses, foreur des tiges, cécidomyie, punaises des panicules, résistance. ISBN 92-9066-213-1.

Le sorgho [*Sorghum bicolor* (L.) Moench] est une des plus importantes cultures céréalières des régions tropicales semi-arides. Les rendements en grains de cette culture sont généralement peu élevés en milieu paysan, car les insectes ravageurs se présentent comme un des facteurs importants qui limitent la production. Il existe plus de 150 espèces d'insectes qui dévastent les cultures du sorgho, dont la mouche des pousses (*Atherigona soccata*), le foreur des tiges (*Chilo partellus*), la cécidomyie (*Contarinia sorghicola*), et les punaises des panicules (*Calocoris angustatus* et *Eurystylus immaculatus*) constituent les principaux ennemis dans diverses régions du monde.

Ce bulletin présente les techniques de criblage pour la résistance dans des conditions de choix multiple (au champs) et de choix unique (en cage), les méthodes d'évaluation des dégâts causés par l'insecte, et les sources de résistance aux ravageurs importants.

Resumen

Citación: Sharma, H.C., Taneja, S.L., Leuschner, K., y Nwanze, K.F. 1992. **Técnicas de selección en sorgo para resistencia a insectos dañinos.** Boletín de Información no. 32. Patancheru, A.P. 502 324, India: International Crops Research Institute for the Semi-Arid Tropics. 48 p. 28 referencias. Palabras claves: *Sorghum bicolor*, técnicas de selección, insectos dañinos, mosca del vástago, barrenador del tallo, mosca midge, chinches de la panoja, resistencia. ISBN 92-9066-213-1.

El Sorgo (*Sorghum biclor* (L.) Moench) es uno de los más importantes cereales en los trópicos semi-áridos. Los rendimientos en conditiones de campo son generalmente bajos, siendo el ataque de insectos uno de los factores principales que restringen la producción. Existen más de 150 especies que dañan el sorgo, entre las cuales, la mosca del vástago (*Atherigona soccata*), el barrenador del tallo (*Chilo partellus*), la mosca midge (*Contarinia sorghicola*), y las chinches de la panoja (*Calocoris angustatus* y *Eurystylus immaculatus*) son las plagas de más amplia distribución mundial.

Este boletín describe las técnicas de selección para resistencia bajo condiciones no fijas (en el campo) y fijas (en cajas insectarias) así como los métodos para evaluar el daño hecho por insectos y las fuentes de resistencia a las plagas más dañinas.

Front cover: *Headcage covered with a cloth bag: a technique used to screen for resistance to sorghum midge.*

Back cover: *Field infestation with larvae using the bazooka applicator.*

Techniques to Screen Sorghums for Resistance to Insect Pests

ICRISAT

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About the Authors

H.C. Sharma and *S.L. Taneja:* Cereals Entomologists, Cereals Program, ICRISAT.

K. Leuschner: Principal Cereals Entomologist, SADCC Regional Sorghum and Millet Improvement Program, Zimbabwe.

K.F. Nwanze: Principal Cereals Entomologist, Cereals Program, ICRISAT.

Editing: V. Sadhana
Design: G.K. Guglani
Cover: A.A. Majid
Page makeup: T.R. Kapoor and K.C.S. Rao

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Contents

Foreword

Sorghum is one of the most important cereals in the semi-arid tropics. It is grown as human food or as animal feed and fodder on about 50 million ha annually. Insect pests are one of the major factors limiting sorghum production. It is accepted that the most appropriate long-term strategy for insect control is one based on insect-resistant cultivars, associated with cultural practices to reduce infestation, natural enemies, and need-based application of insecticides. Chemical control is expensive and often beyond the means of farmers in the semi-arid tropics. Growing insect-resistant cultivars is therefore essential in keeping pest population below economic threshold levels.

Development of appropriate screening techniques is essential in identifying stable sources of resistance, and in transferring genes conferring resistance into high-yielding cultivars. This Information Bulletin describes the resistance screening techniques developed at ICRISAT Center. It also identifies sources of resistance among germplasm lines, and lists breeding lines developed at ICRISAT Center with resistance to the major sorghum insect pests.

This information is certain to prove useful to sorghum scientists throughout the world. I am sure that this bulletin will assist breeders in developing insect-resistant cultivars with high and stable yield across a range of agroecosystems of the semi-arid tropics.

J.M.J. de Wet
Director, Cereals
ICRISAT

Introduction

Sorghum (*Sorghum bicolor* Moench (L.)] is one of the most important cereal crops in the semi-arid tropics (SAT). Grain yields on peasant farms are generally low (500-800 kg ha^{-1}), insect pests being one of the major factors limiting sorghum production. In India, nearly 32.1% of the actual produce is lost due to insect pests (Borad and Mittal 1983). The losses due to panicle-feeding insects alone have been estimated to be over $ 100 million annually (Leuschner and Sharma 1983).

There are over 150 insect species which damage sorghum pianis from sowing to crop harvest (Seshu Reddy and Davies 1979a). In most of the sorghum-growing areas, the important pests found are the sorghum midge (*Contarinia sorghicola* Coq.), stem borers (*Chilo partellus* Swinhoe and *Busseola fusca* Fuller), head bugs (*Calocoris angustatus* Leth., *Eufystylus immaculatus Odh., Creontiades pallidus Ramb.* and *Campylomma* spp.), green bug (*Schizaphis graminum* Rondani), and shoot fly (*Atherigona soccata* Rondani). Others like the spider mite (*Oligonychus* spp.), shoot bug (*Peregrinus maidis Ashm.*), corn leaf aphid (*Rhopalosiphom maidis* Fitch.), and head caterpillars (*Helicoverpa armigera* Hb; *Eublemma* spp., *Cryptoblabes* spp., and *Pyroderces simplex* Wsm.) can be regarded as occasional pests. The locusts (*Schistocerca gregaria* Forsk. and *Locusta migratoria migratorioides* Linn.) and armyworms (*Mythimna separata* Walker, *Spodoptera exempta* Walker, and *Spodoptera frugiperda* J.E. Smith) also cause sporadic defoliation of the crop (Sharma 1985a).

The major components of pest management in agroecosystems are cultural practices, natural enemies, insecticides, and host-plant resistance. Cultural practices are effective against certain pests, some having become an integral component of crop husbandry and farming systems. However, some of the cultural practices are only partially effective and are difficult to implement under rainfed conditions. It is difficult for all farmers to plant at times when pest incidence can be avoided so that population buildup of insects can be minimized. Chemical control is expensive and numerous applications may be required. This is often beyond the reach of most farmers in the SAT. In many areas, insecticides and spraying equipment are either not available or farmers lack the proper knowledge of their use. Under such circumstances, use of resistant or less-susceptible cultivars is one of the most important methods of keeping insect populations below economic threshold levels. Host-plant resistance does not involve any extra costs or application skills in pest control techniques. However, host-plant resistance is not a panacea for all pest problems. It is most useful when carefully utilized with other components of pest management (Painter 1951).

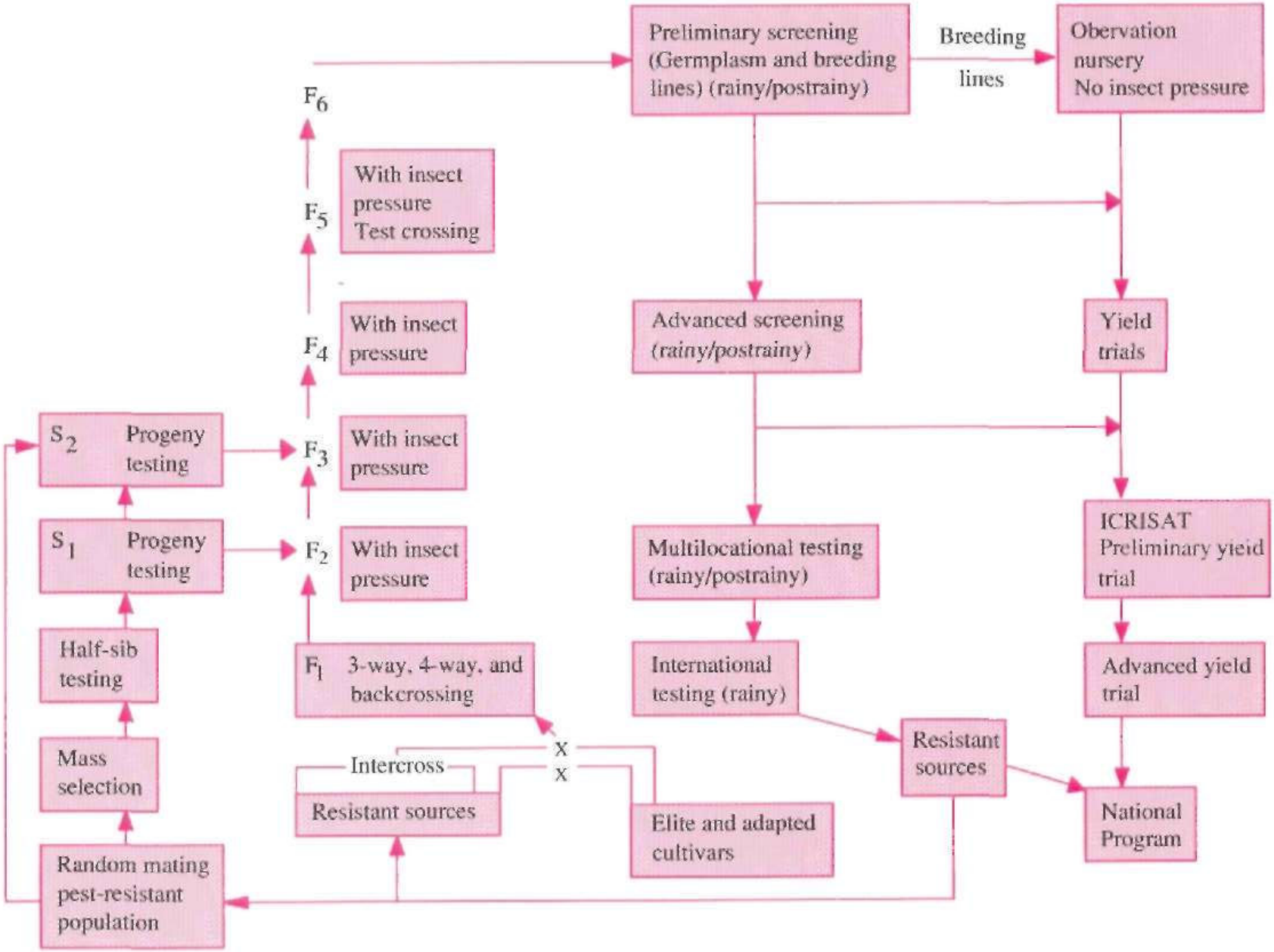


Figure 1. Screening and breeding for insect-pest resistance in sorghum at ICRISAT Center.

Plate 1. Shoot fly (inset): egg laying and deadheart symptoms.

In the Cereals Program at ICRISAT Center, we have placed major emphasis on developing insect-resistant cultivars through a multidisciplinary approach. A general scheme for identification and utilization of resistance to insects is presented in Figure 1. This information bulletin presents an overview of the techniques developed at ICRISAT Center, to screen for resistance to key pests of sorghum in the SAT.

Shoot Fly (*Atherigona soccata*)

Shoot fly is an important pest of sorghum in Asia, mediterranean Europe, and Africa. It attacks sorghum from 5 to 25 days after seedling emergence. The adult fly lays white, elongated, cigar-shaped eggs singly on the undersurface of the leaves, parallel to the midrib (Plate 1). The eggs hatch in 1-2 days, and the larvae crawl *to the plant whorl and then* move downward between the folds of the young leaves till they reach the growing point. They cut the growing point and feed on the decaying leaf tissues, resulting in deadheart formation (Plate 1). As a result of shoot fly attack, plant stand is greatly reduced. The death of the main shoot often results in the production of tillers, which often serve as a mechanism *of* recovery resistance and produce productive panicles. However, the tillers are also attacked under high shoot fly pressure. Larval stage lasts for 8-10 days. Pupation takes place in the soil or in the stem; this stage lasts for 8-10 days. In general, the shoot fly completes its life cycle in 17-21 days.

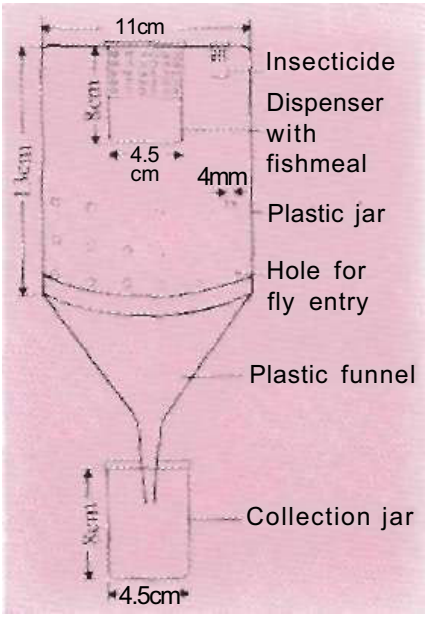


Figure 2. A diagrammatic representation of the plastic jar trap, showing the various parts.

Resistance Screening Techniques

Various techniques to screen for resistance to shoot fly have been discussed by Pradhan (1971), Jotwani (1978), and Taneja and Leuschner (1985a).

Field Screening

Sowing Date. Adjust the sowing date so that the test material is exposed to optimum insect pressure. Use fishmeal-baited trap (Figure 2) (Plate 2) to study the population dynamics of the shoot fly (Taneja and Leuschner 1986) to determine periods of greatest insect density (Figure 3). In India, to screen for resistance to shoot fly, the best time to sow the test material is the second fortnight of July for the rainy season and October for the postrainy season (Taneja et al. 1986).

Interlard-Fishmeal Technique. To ensure high and uniform

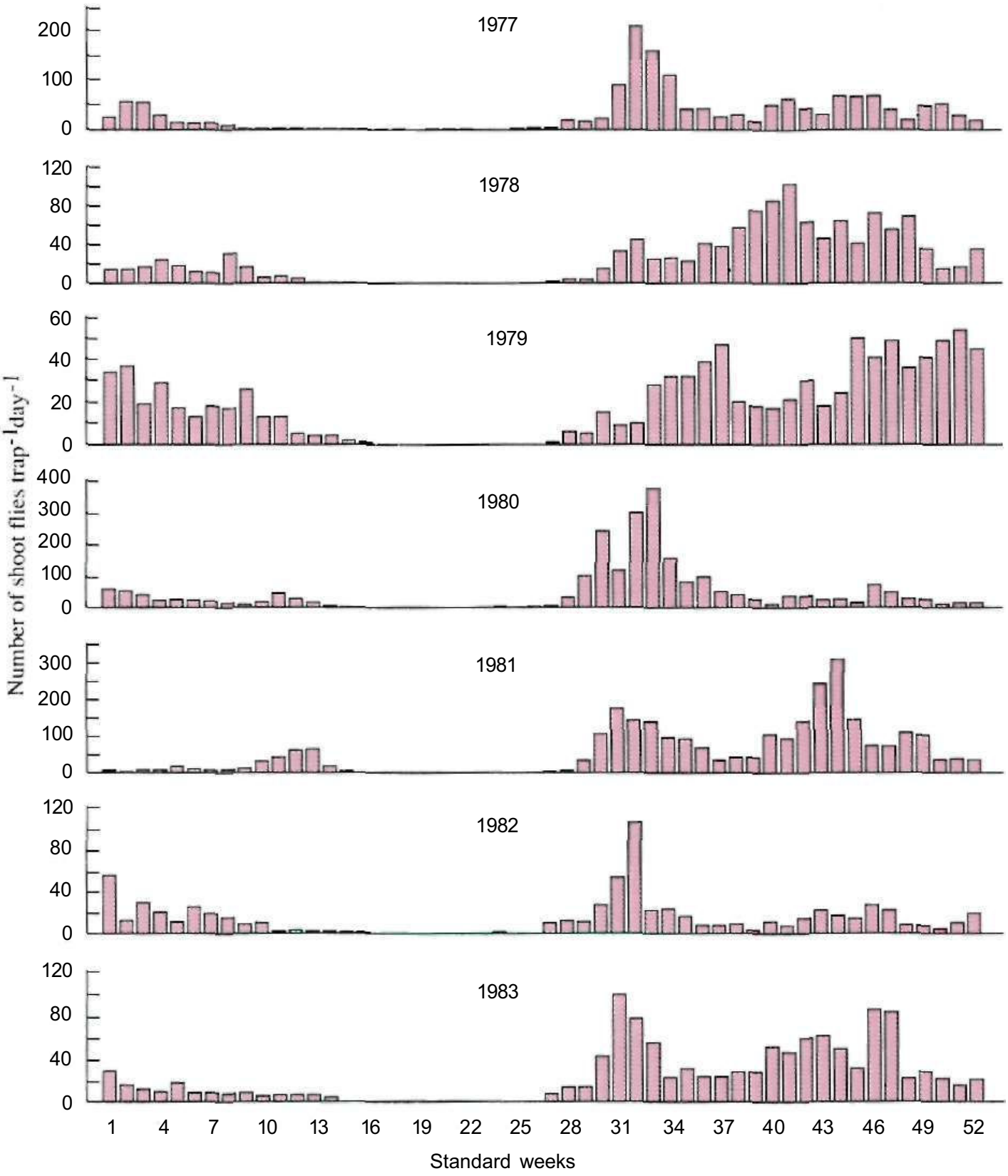


*Figure 3. Shoot fly (*Atherigona *soccata) catches in fishmeal-baited traps at ICR1SAT Center, India, 1977-83*

Plate 2. Plastic jar trap used to monitor shoot fly populations.

Plate 4. Fishmeal-baited trap to collect live shoot flies for resistance screening.

shoot fly pressure under field conditions, use the interlard-fishmeal technique:

- Sow four rows of interlards of a susceptible cultivar (usually CSH 1) after every 24 rows, 20 days before sowing the test material.
- One week after seedling emergence, spread moistened fishmeal uniformly or keep it in plastic bags (Plate 3) in the interlards to attract the shoot flies. One generation of the shoot fly is completed on the interlards and the emerging flies infest the test material. Fishmeal can also be spread or kept in plastic bags in the test material. This method provides adequate and uniform shoot fly pressure for resistance screening (Taneja and Leuschner 1985a).

Plate 3. Interlard fishmeal technique to screen for shoot fly resistance..

Cage Screening

To confirm resistance observed under field conditions, and to study various resistance mechanisms, a cage-screening method can be used. The cage-screening technique described by Soto (1972) has been modified at ICRISAT Center to closely simulate field conditions.

- Catch shoot flies in the fishmeal-baited trap (Plate 4), and collect them from the trap in the mornings and/or evenings.
- Separate A *soccata* from other species.
- Confine the flies with the sorghum seedlings for 1-2 days in a 30 x 30 x 30-cm cage.
- For every 100 plants, release 40 flies for 1 day or 20 flies for 2 days.

The cage-screening technique can also be used for multiple- as well as no-choice conditions. For a multiple-choice test, several genotypes can be sown in the field in 3.4 x 2-m beds with a row spacing of 15 cm. Ten days after seedling emergence, cover the plants with a 3.4 x 2 x 1-m screened cage (Plate 5a), and introduce flies in the cage. After 1 week, count the eggs and deadhearts after removing the cage. For a no-choice test, sow only one genotype in 1 * 1-m beds and cover six beds with a 2 x 3 x 0.5-m cage having six compartments (Plate 5b). Ten days after seedling emergence, release 20 flies in each compartment and record observations as described earlier.

Rapid screening can also be carried out using plastic tray cages (Plate 5c). This system consists of two plastic trays (40 x 30 x 14 cm), one for sowing test material and the other (fitted with fine wire-mesh as shown in Plate 5c) clamped over the first tray thus serving as a cage. Ten days after seedling emergence, the second tray is placed over the first one upside down using clamps and 20 flies are released in each cage and observations recorded as described earlier.

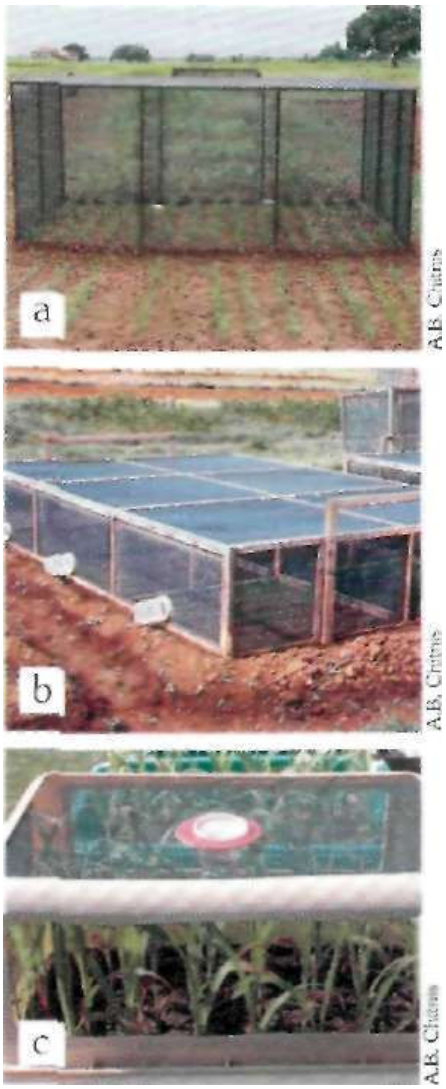


Plate 5. Cage technique to screen for shoot fly resistance under (a) Single cage, multi-choice conditions, (b) multi-compartment, no-choice conditions, and (c) single compartment, no-choice conditions.

Damage Evaluation for Resistance Screening

In the preliminary evaluation of the material (where a large number of lines are to be tested), count shoot fly deadhearts 28 days after crop emergence. Record the total number of plants and those showing deadhearts separately, and calculate the percentage of plants with deadhearts. Shoot fly damage can also be rated visually on a 1-9 scale (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 = >80% plants with deadhearts). Select lines with <50% deadhearts or a damage score of < 5 for further testing.

For advanced evaluation, sow the test material in 2-3 replications. Record the number and percentage of plants with eggs and deadhearts 21 and 28 days after crop emergence. Genotypes selected in the advanced evaluation can be further screened in cages under multi-choice and no-choice conditions.

Sources of Resistance

At ICRISAT Center, over 25 000 sorghum germplasm accessions have been screened for resistance to shoot fly. Forty germplasm accessions and 11 breeding lines have been identified as sources of resistance (Table 1). Stability analysis of 42 germplasm lines tested over five seasons indicated that IS 1054, IS 1071, IS 2394, IS 5484, and IS 18368 were quite stable for shoot fly resistance across locations. IS 2123, IS

Table 1. Sources of resistance to sorghum shoot fly identified/ developed at ICRISAT Center.

Genotype	Plant height (cm)	Time to 50% flowering (days)	Deadhearts (%)
IS 923	325	75	42
IS 1034	315	73	27
IS 1096	265	66	37
IS 2122	305	80	33
IS 2146	280	80	23
IS 2195	260	75	44
IS 2205	300	89	33
IS 2265	430	112	43
IS 2269	270	69	20
IS 2291	255	79	18
IS 2309	285	89	34
IS 2312	*290*	75	26
IS 2394	265	71	42
IS 4646	450	98	32
IS 4663	295	73	38
IS 4664	300	82	31
IS 5210	315	75	38
IS 5470	310	77	32
IS 5480	290	82	17
IS 5484	305	70	28
IS 5511	390	98	26
IS 5538	365	98	29
IS 5566	310	87	46
IS 5604	355	*86*	*38*
IS 5613	325	80	27
IS 5622	350	87	38
IS 5636	305	71	29
IS 5648	270	69	28
IS 6566	300	81	39
IS 18366	305	72	20
IS 18368	300	67	47
IS 18369	305	72	24
IS 18371	305	69	37
IS 22114	370	84	37
IS 22121	380	73	19
IS 22144	350	74	29
IS 22145	350	73	39
IS 22148	345	79	34
IS 22149	390	89	28
IS 221%	300	70	31
ICSV 705	110	71	19

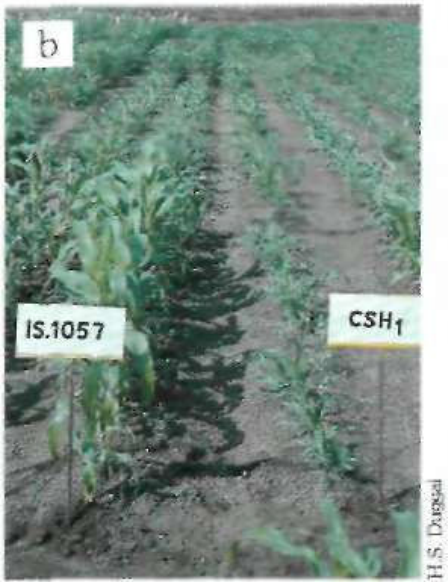
Continued

Table 1. *Continued.*

Genotype	Plant height (cm)	Time to 50% flowering (days)	Deadhearts (%)
ICSV 707	180	72	25
ICSV 708	180	70	27
ICSV 711	170	77	29
ICSV 712	185	79	26
ICSV 713	170	80	19
ICSV 714	135	82	11
ICSV 717	240	78	40
PS 35805	150	87	22
PS 35832-1	200	76	31
PS 35832-2-2	220	84	37
Resistant control			
IS 18551	330	71	28
Susceptible control			
CSH 1	155	58	72

2195, IS 4664, and IS 18551 showed low incidence (<35%) and moderate stability. ICSV 700, ICSV 701, ICSV 705, ICSV 714, and ICSV 717 are the improved breeding lines with resistance levels comparable to the original sources of resistance. Differences in susceptibility to shoot fly under field conditions are shown in Plate 6a and b.

Plate 6. Differences in cultivar susceptibility to shoot fly under field conditions: (a) IS 18551—resistant (left) and CSH 1—susceptible (right); (b) IS 1057—resistant (left), and CSH 1—susceptible (right).



Spotted Stem Borer (*Chilo partellus*)

The spotted stem borer is an important and common pest of sorghum in Asia and in eastern and southern Africa. It 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 (Plate 7a). 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 (Plate 7b). In older seedlings where internode elongation has started and the growing point has moved upwards, the larva feeds inside the stem causing stem tunneling (Plate 7c). Later infestations also result in peduncle tunneling and breakage (Plate 7d). Both stem and peduncle damage sometimes lead to the production of complete or partially chaffy panicles (Plate 7e).

The female lays 400-500 flattened, overlapping, yellowish eggs in masses of 10-100 on the undersurface of leaves, usually close to the midrib. Eggs hatch in 4-6 days. Larvae complete development in 19-27 days and pupation occurs inside the stem, and lasts for 7-10 days. During winter and/or summer, larvae may enter into hibernation/or aestivation in stubbles and stalks. With the onset of the rainy season, the diapausing larvae pupate, giving rise to first-generation moths.

Resistance-Screening Techniques

Several techniques have been developed to screen for resistance to the spotted stem borer (Pradhan 1971; Jotwani 1978; Taneja and Leuschner 1985b; and Taneja 1987).

Screening under Natural Infestation

Hot Spots. Test the material at hot-spot locations where the pest populations are known to occur naturally and regularly at levels that often result in severe damage to the crop. Hot-spot locations for *Chilo partellus* are Hisar in northern India; Afgoi and Baidoa in Somalia; Panmure and Mezarbani in Zimbabwe; Kiboko in Kenya; and Golden Valley in Zambia.

Sowing Date. To screen for resistance under natural pest 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 density of the stem borer population is at its peak. Determine the periods of maxi-

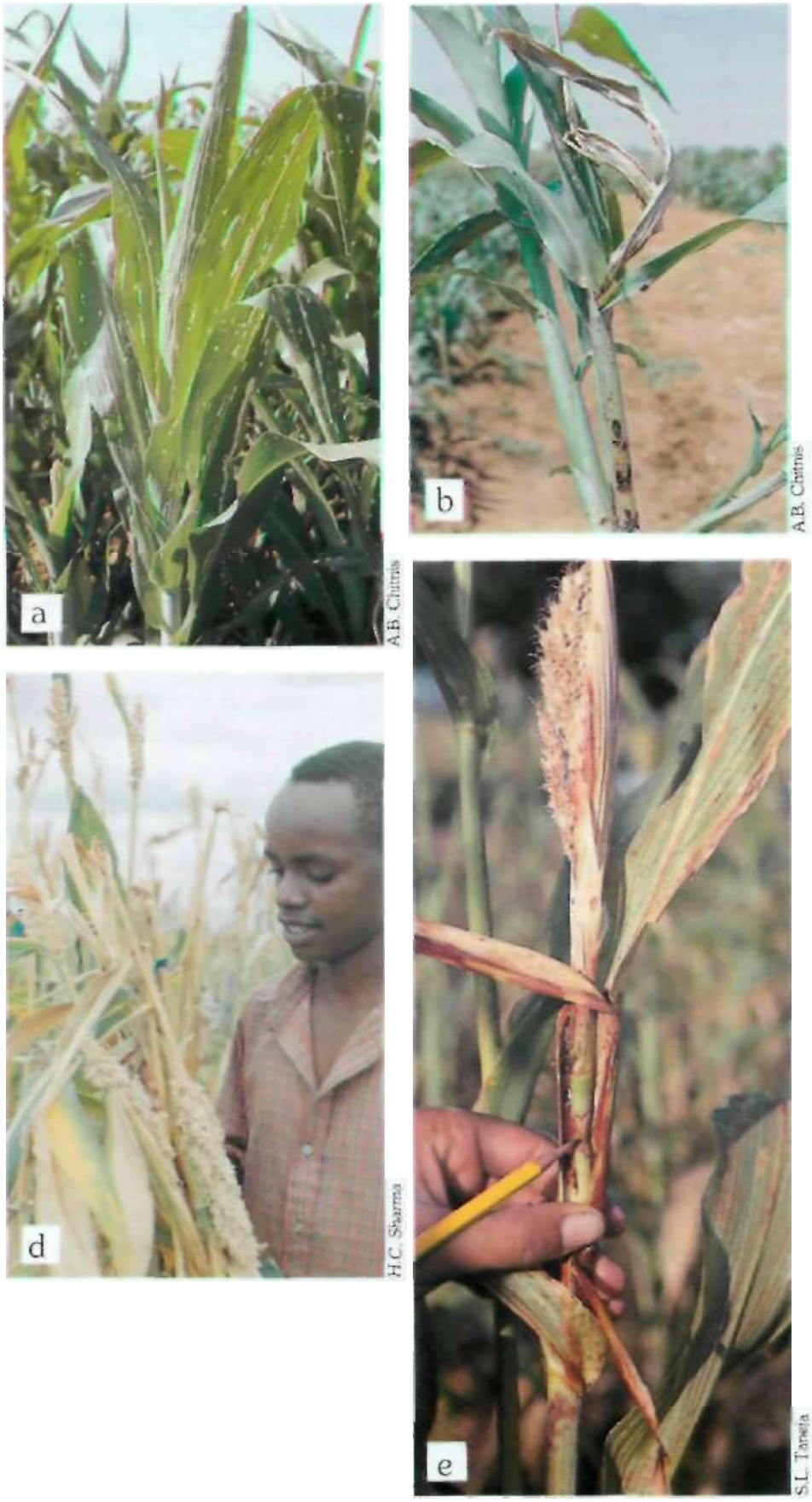


Plate 7. Nature of damage by the spotted stem borer, Chilo partellus: *(a) Leaf feeding, (b) deadheart formation, (c)stem tunneling, (d) peduncle breakage, and (e) chaffy panicle.*

mum borer density through pheromone traps, light traps, or by monitoring pest infestation at regular intervals. At Hisar, *C. partellus* is most active in August-September (Figure 4). Sorghum crop sown between the first and third week of July suffers maximum stem borer damage.

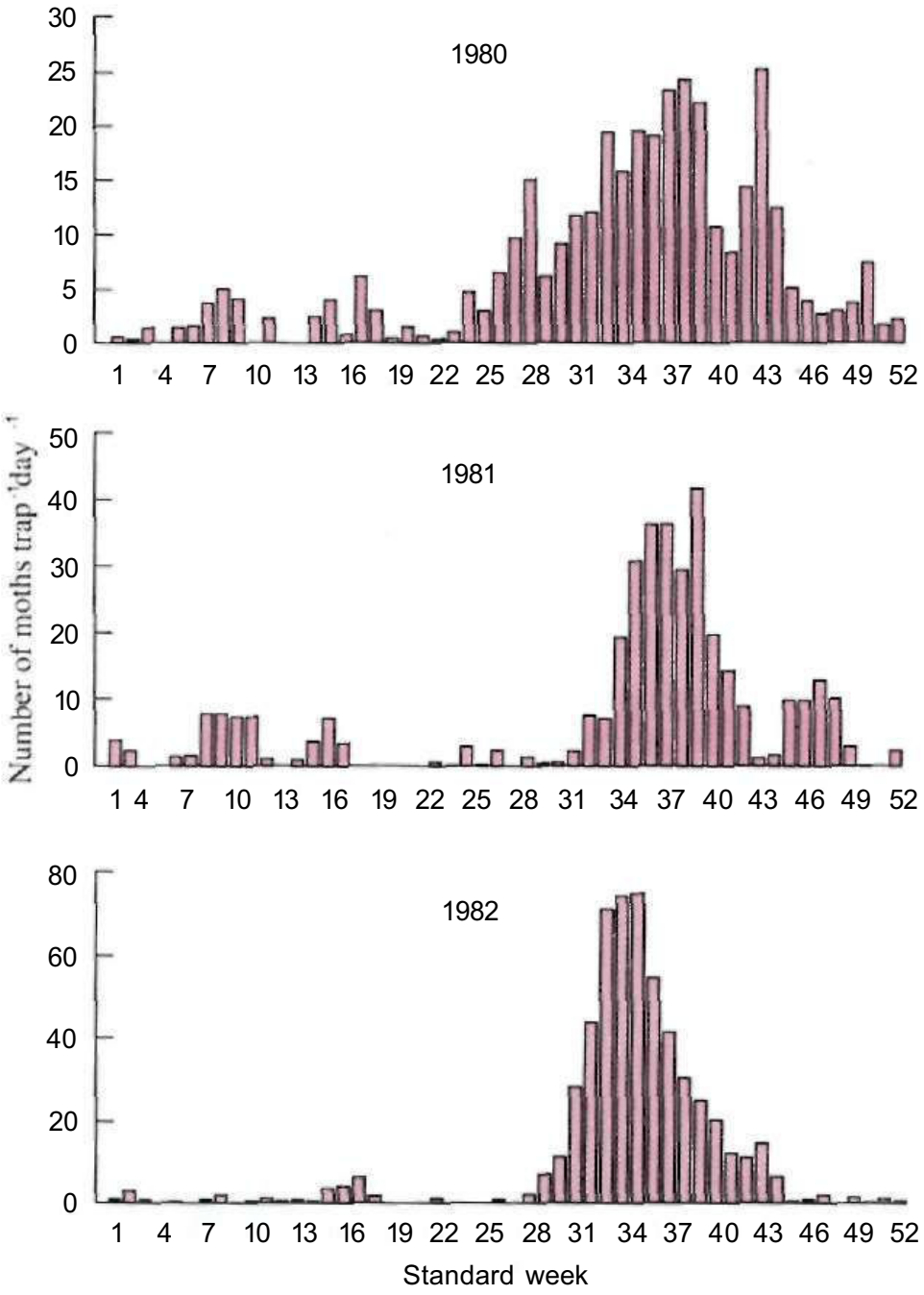


Figure 4. Seasonal activity of spotted stem borer,* Chilo partellus, *based on light trap catches of moths at Hisar, Haryana, 1980-82.

At ICRISAT Center, maximum moths in the light traps have been recorded during September, followed by smaller peaks during November and February-April.

Mass Rearing and Artificial Infestation

The efficiency of any resistance-screening program depends on the uniform and timely infestation of 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 1979b). An artificial diet to rear *C. partellus* has been developed at ICRISAT Center (Taneja and Leuschner 1985b). Most of the ingredients of this diet (Table 2) are available in the local market. For sorghum leaf powder, collect CSH 1 leaves from a 35-40 days-old crop. Wash, dry, and grind the leaves to a

Table 2. Artificial diet used for mass rearing spotted stem borer, *Chilo partellus,* at ICRISAT Center, 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	160.0 g
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.

fine powder, and autoclave for 15 min at 120° C at 5 kg cm^{-1} pressure for use in the artificial diet.

Diet Preparation.

- Blend the ingredients of fraction A (Table 2) (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 each of 1 L plastic jar (Plate 8a).
- Allow the diet in the jar to cool to room temperature.
- Place about 100 eggs, which are at the blackhead stage, in each jar (Plate 8b) and keep the jars in a dark room for 2 days. This discourages the photopositive behavior of first 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 (Plate 8c).

The larval period lasts on the artificial diet for 22-28 days and the pupal period for 5-6 days. Moth emergence begins 30 days after the larval inoculation and continues up to the 40th day (Plate 8d). Females emerge 2-3 days later than the males (Figure 5). 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.

Plate 8. Mass rearing of spotted stem borer in the laboratory. (a) Diet preparation, (b) inoculation with eggs, (c) rearing in the laboratory, and (d) pupation and adult emergence.

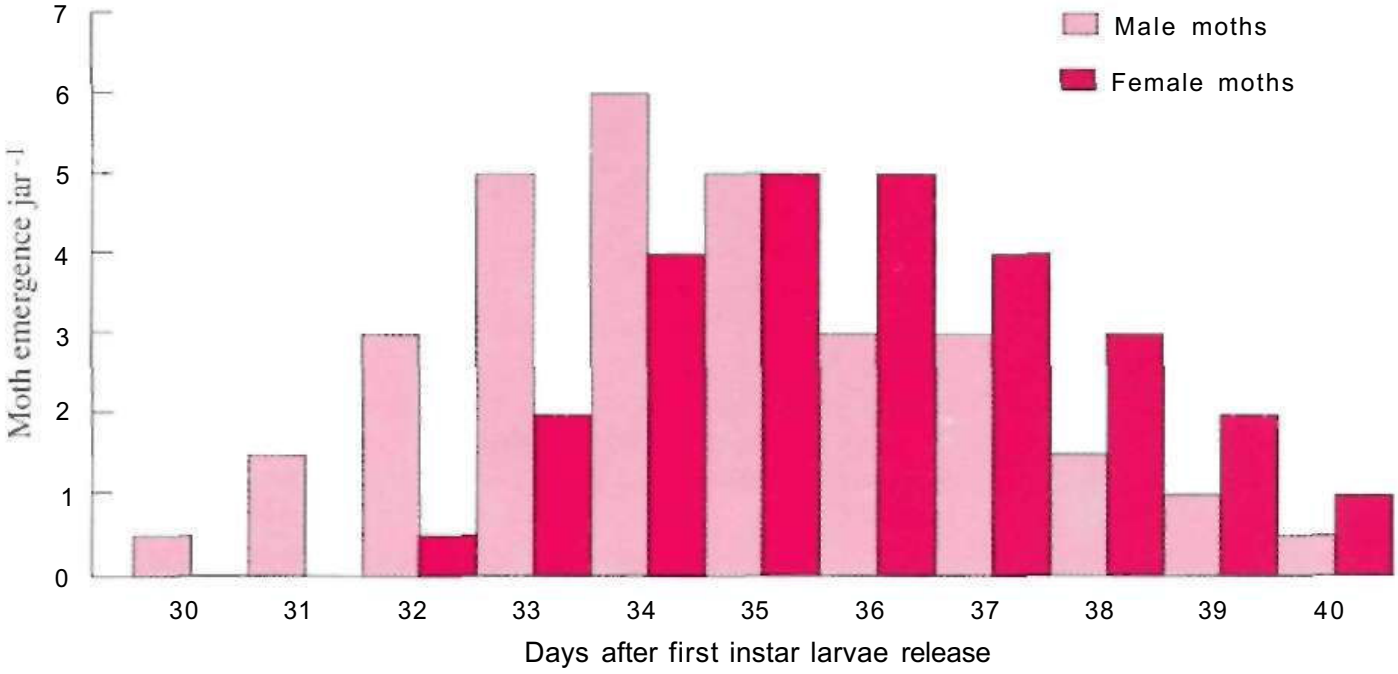


Figure 5. Rates of emergence of spotted stem borer moths from the larvae reared on an artificial diet, ICRISAT Center, India, 1990/91.

Moth Collection. Collect the moths with the help of a vacuum cleaner attached to a suction pipe (Figure 6) (a bifurcated tube is fixed

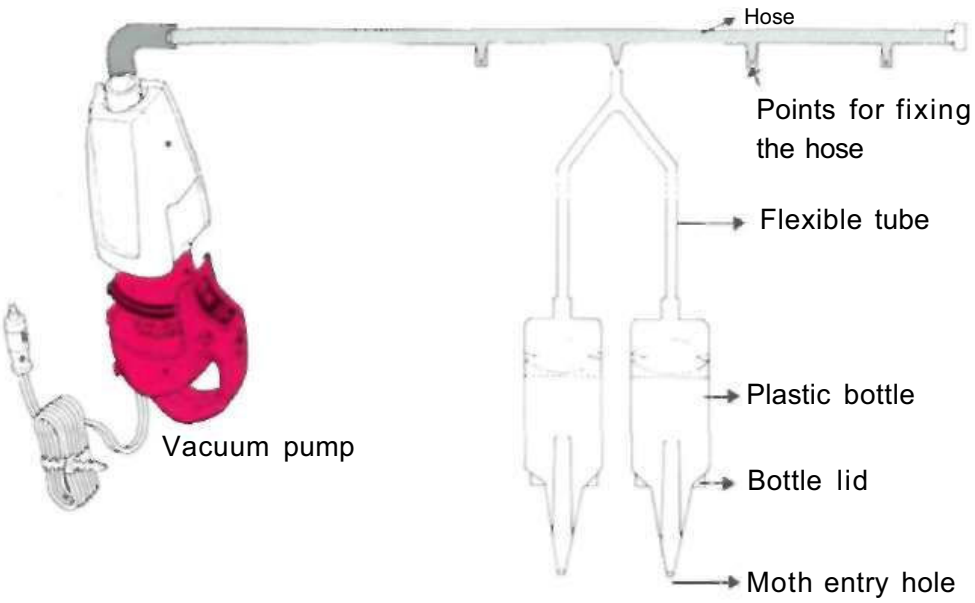


Figure 6. Stem borer moth-collection device used at ICRISAT Center.

Plate 9. Oviposition cage. (a) Releasing 50 pairs of spotted stem borer moths in an egg-laying cage, and (b) eggs laid on the glycine paper.

to this pipe, which terminates in the collection bottles) or with the help of hand-held aspirators. Collect the male and female moths separately (males are smaller in size with dark forewings and smaller 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 (Plate 9a). A fine georgette cloth with 6 * 6 mm holes at regular intervals is fitted around the outer side of the cylinder, around which a sheet of white glycine paper (25 ^{x} 80 cm) is wrapped 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 (Plate 9a). A female lays an average of 10-12 egg masses (500-600 eggs) over a period of 4 days, the maximum being laid on the second and third day. The eggs are laid in batches on the glycine paper through the holes in the wire-cage (Plate 9b). Replace the glycine paper daily. Feed the moths with water using a cotton swab.

Egg Storage. High humidity (80-90%) is needed for normal embryonic development, and hatching is drastically reduced when relative humidity falls below 50%. To obtain high humidity, place the glycine papers containing egg masses on a rod in a plastic bucket containing water (Plate 10). Cover the plastic bucket with a lid. Store the eggs at

Plate 10. Glycine paper containing spotted stem borer eggs hung on a rod to keep them under high humidity in a bucket (containing water at the bottom) for uniform hatching.

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 Center, screening for stem borer resistance is carried out during the rainy and the postrainy seasons. The rainy season sowing is generally done in mid-June and the postrainy season sowing at the end of September. A schedule for diet preparation, crop sowing, and infestation is given in Figure 7. This schedule may be adapted in different locations with modifications as required.

Preparation of "Bazooka". For field infestation, the "bazooka applicator" developed at the Centro Internacional de Mejoramiento de Maiz y Trigo (CIMMYT) in 1976 (CIMMYT 1977) to infest maize with corn earworm was modified to suit our requirements (Plate 11a). Keep 500 black-head stage egg masses along with 85 g of poppy seeds *{Papaver* sp.) (locally known as *Khas Khas)* overnight in a plastic jar with a tightly fitting lid. In the morning, mix the first instar larvae gently with the carrier and transfer them into the plastic bottle of the bazooka.

Field Infestation.

- Take the bazooka to the field, and infest the plants 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 (Plate 11b). Generally 5-7 larvae per plant are sufficient to cause appreciable leaf feeding and deadhearts (> 90% damage in susceptible genotypes).
- Infest 15-20-days-old plants. Deadheart formation decreases progressively as the infestation is delayed (Figure 8).
- 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 higher temperatures. However, on cloudy days, infestations can be carried out at any time of the day.
- Agitate 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 of 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 the first infestation.

Piste 11. Field infestation with larvae. (a) Bazooka applicator for releasing uniform number of larvae in each plant, and (b) field infestation using the bazooka applicator.

Control of Shoot Fly. Shoot fly infestation interferes with the screening for resistance to stem borer. A selective insecticide may be used to control shoot fly without leaving any residual effect on stem

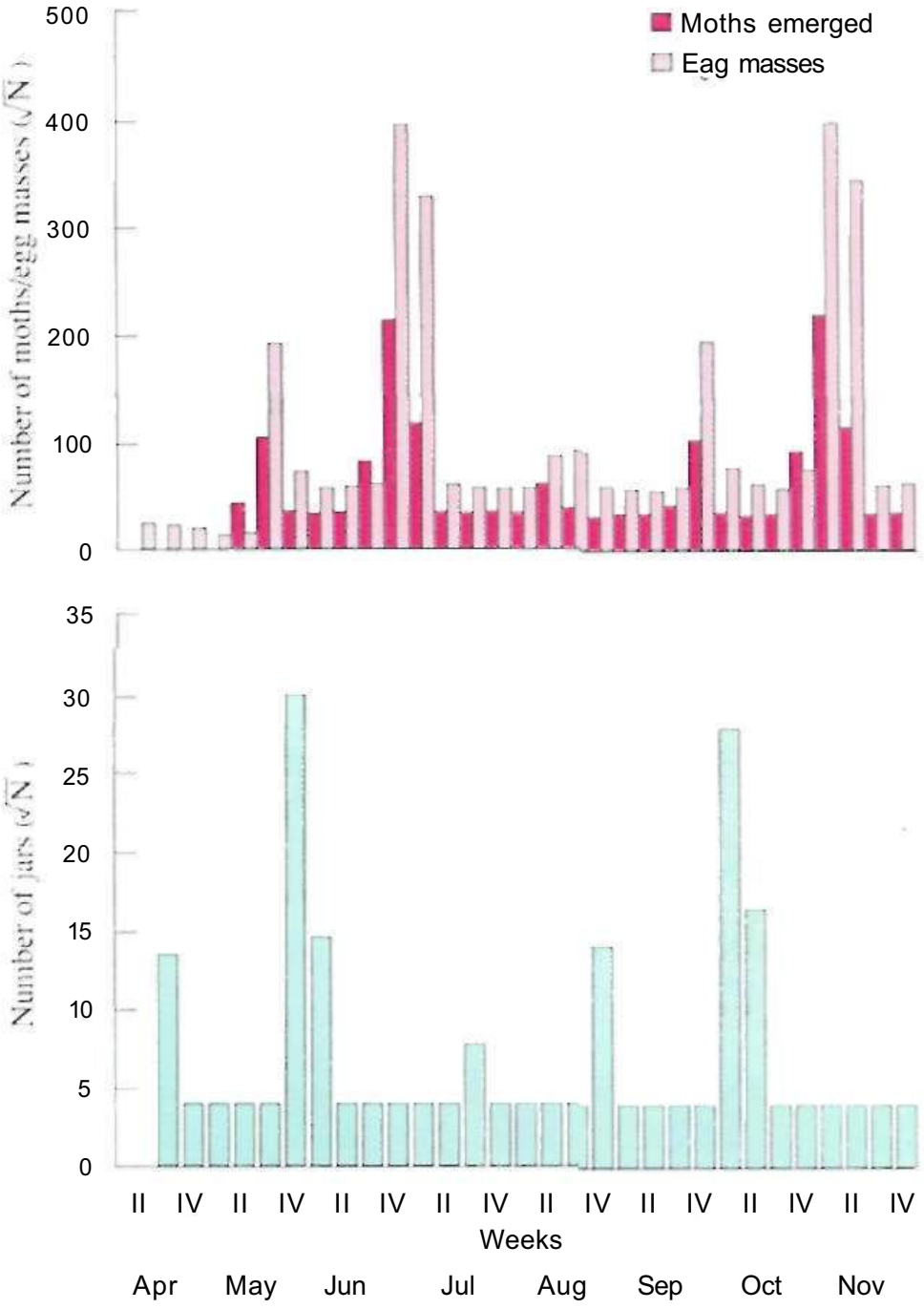


Figure 7. Schedule for diet preparation, moth emergence, and oviposition for artificial infestation of Chilo partellusa at *ICRISAT Center.*

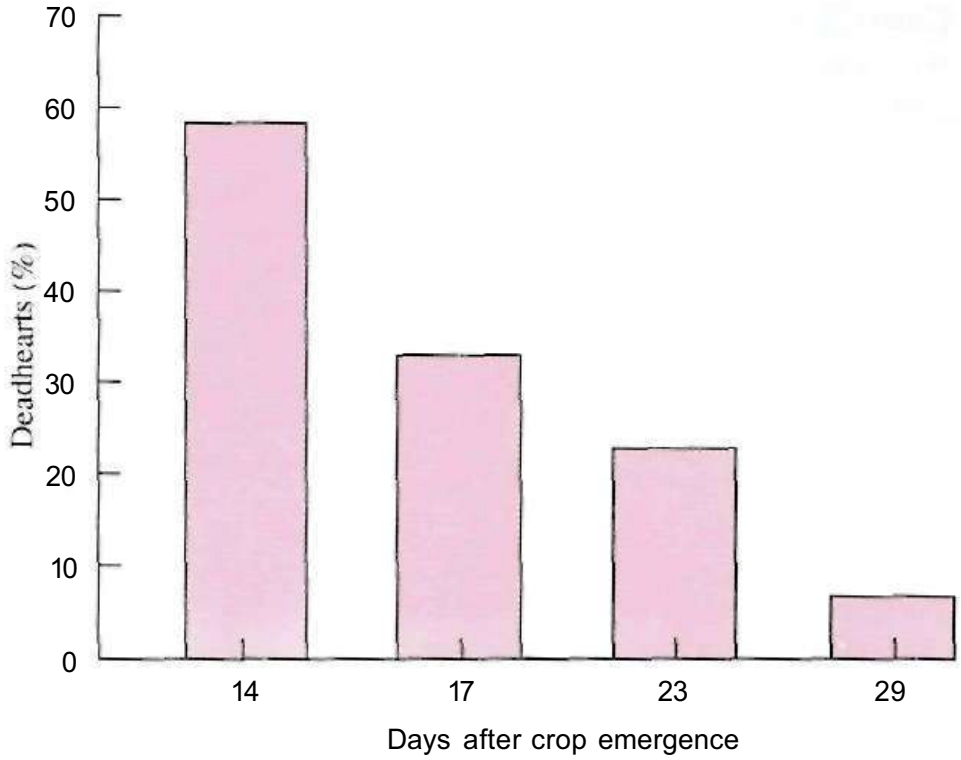


Figure 8. Deadheart formation in susceptible sorghum CSH 1 infested with spotted stem borer larvae at 14,17, 23, and 29 days after seedling emergence.

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 Electrodyne® sprayer 1 week before the borer infestation controls the shoot fly effectively without any detrimental effect on borer establishment. Also, plant 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/peduncle tunneling, and production of chaffy panicles. All these symptoms are not necessarily related to yield loss. Leaf injury, which is the first larval feeding symptom, has been 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 in situations of high wind velocities, which would break the peduncle. Deadheart

formation causes the most critical damage. This parameter is therefore, the most important criterion for differentiating degrees of resistance. The second important criterion is the production of chaffy panicles. The following observations are recorded for the evaluation of damage.

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

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

Chaffy Panicles. At crop harvest, record observations on the number of partial and complete chaffy panicles, number of broken

Figure 9. Leaf damage rating scale for spotted stem borer.

Table 3. Visual damage rating scale for leaf feeding, deadheart formation, and chaffy and broken panicles due *to C. partellus.*

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%***

panicles, and visual score (1-9) for chaffy/broken panicles and grain mass.

Sources of Resistance

Screening for spotted stem borer resistance by artificial infestation at ICRISAT Center started in 1979 (Seshu Reddy and Davies 1979b). Later on, testing of the material was initiated at Hisar, where the natural stem borer infestation was found *to* be quite high and regular. Out *of* nearly 20 000 germplasm lines tested over three seasons, 77 have been reported as resistant (Table 4). Differences in susceptibility to stem borer are illustrated in Plate 12a and b. Stability analysis of 62 germplasm lines over six seasons indicated that IS 5470, IS 5604, IS 8320, and IS 18573 are the most stable for resistance to spotted stem borer. ICSV 443, ICSV 700, and PB 12779-1 are improved sources of resistance.

Sorghum Midge (*Contarinia sorghicola*)

Sorghum midge is probably the most damaging and widely distributed of all sorghum pests (Sharma 1985b). It occurs in all sorghum-growing

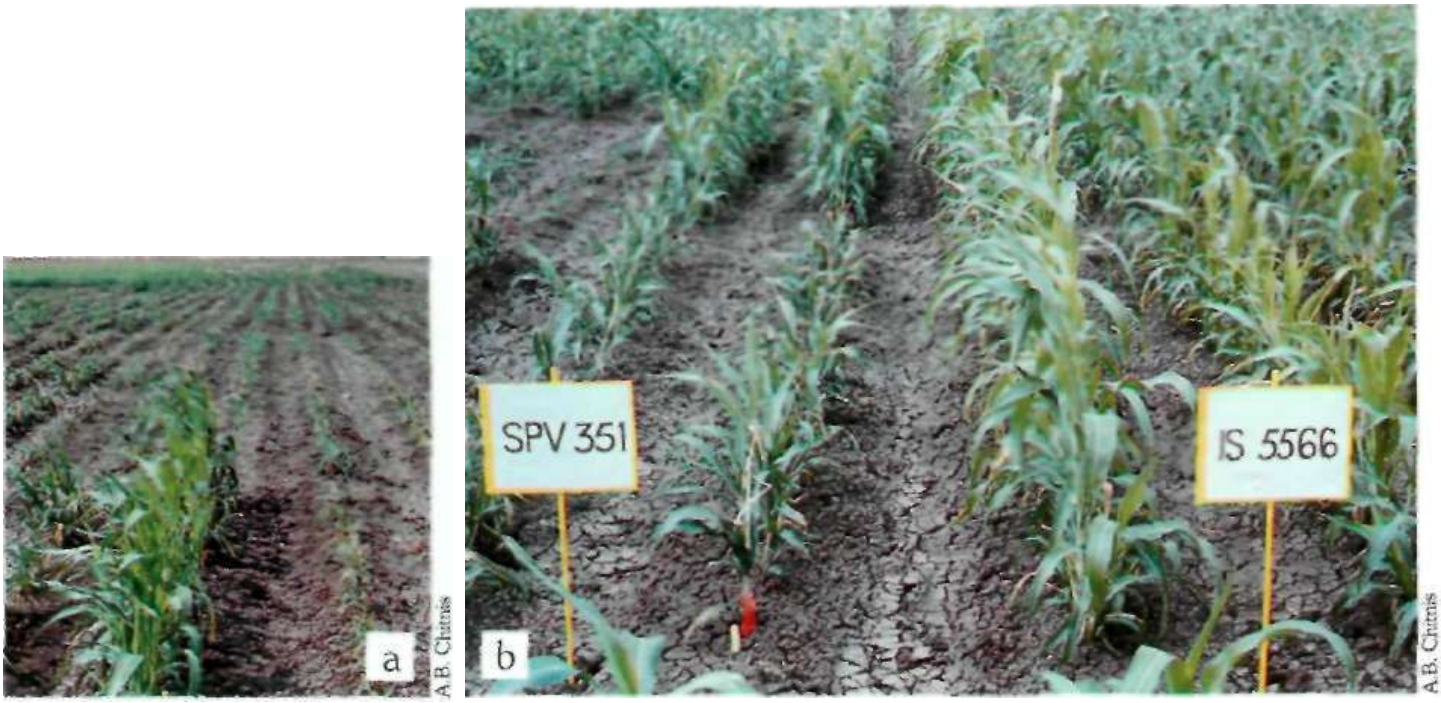


Plate 12. Cultivar differences in susceptibility to spotted stem borer under artificial infestation: (a) IS 2205—resistant (left), and CSH1—susceptible (right), and (b) IS 5566—resistant (right), and SPV 351—susceptible (left).

Table 4. Sources of spotted stem borer resistance identified/ developed at ICRISAT Center.

Genotype	Plant height (cm)	Time to 50% flowering (days)	Deadhearts (%)	
			Artificial infestation	Natural infestation
IS 923	325	75	11.1	24.6
IS 1044	375	93	3.4	23.3
IS 1057	340	71	37.0	31.0
IS 1082	260	82	16.2	25.1
IS 1096	265	66	6.7	30.2
IS 1104	315	73	16.2	25.3
IS 2122	305	80	4.9	12.1
IS 2123	300	80	15.4	14.1
IS 2195	260	75	13.7	11.5
IS 2263	305	80	13.1	23.7
IS 2265	430	112	16.4	18.2
IS 2269	270	69	27.7	21.8
IS 2291	255	79	16.8	34.6

Continued

Table 4. *Continued*

Genotype	Plant height (cm)	Time to 50% flowering (days)	Deadhearts (%)	
			Artificial infestation	Natural infestation
IS 2312	290	75	11.2	10.1
IS 2375	180	53	16.8	22.8
IS 2376	180	61	8.2	9.7
IS 3962	400	100	1.0	13.6
IS 4546	295	79	13.7	34.5
IS 4637	290	66	22.3	39.1
IS 4646	450	98	21.7	23.6
IS 4663	295	73	16.6	18.9
IS 4756	345	82	4.5	12.7
IS 4757	275	71	15.7	14.4
IS 4776	325	84	7.4	20.6
IS 4995	420	108	2.3	21.1
IS 5072	285	89	4.8	16.0
IS 5210	315	75	23.0	39.5
IS 5268	300	91	7.5	25.7
IS 5469	295	71	13.1	13.7
IS 5470	310	77	5.9	11.6
IS 5480	290	82	6.1	11.3
IS 5484	305	70	7.5	15.7
IS 5490	290	67	1.1	7.4
IS 5511	390	98	45.7	17.1
IS 5571	370	96	9.8	14.4
IS 5579	360	82	3.2	23.3
IS 5585	295	66	17.1	25.7
IS 5604	355	86	24.6	24.2
IS 5613	325	80	7.6	16.8
IS 5619	360	73	29.6	13.5
IS 5648	270	69	7.6	17.1
IS 5658	335	89	8.7	11.2
IS 6566	300	81	11.4	18.3
IS 7224	465	125	4.0	23.1
IS 8549	280	131	7.7	21.0
IS 8811	240	68	35.7	28.6
IS 12308	180	50	4.8	23.6
IS 13100	240	58	11.1	20.5
IS 17742	320	89	16.2	28.6
IS 17745	390	98	6.9	22.3
IS 17948	340	88	9.0	14.0
IS 18551	330	71	8.4	24.5
IS 18573	400	87	9.5	13.3
IS 18577	400	89	5.6	14.3
IS 18578	395	89	23.7	24.7

Continued

Table 4. *Continued*

Genotype	Plant height (cm)	Time to 50% flowering (days)	Deadhearts (%)	
			Artificial infestation	Natural infestation
IS 18579	290	75	10.1	9.4
IS 18581	330	135	8.4	10.5
IS 18584	310	72	17.7	17.0
IS 18585	305	72	20.5	10.4
IS 18662	230	64	19.7	27.5
IS 18677	210	58	32.6	32.5
IS 22039	340	71	8.7	26.0
IS 22091	305	70	43.4	25.9
IS 22113	365	77	17.5	36.5
IS 22114	370	84	13.5	28.8
IS 22121	380	73	20.2	28.2
IS 22129	380	92	13.4	16.6
IS 22144	350	74	21.4	21.7
IS 22148	345	79	16.8	14.1
IS 22196	300	70	24.0	16.7
IS 23962	390	50	7.7	31.8
PB 10306	300	83	15.2	16.1
PB 12779-2	285	81	8.8	12.1
PB 12891-1	250	83	13.1	36.6
PS 14413	300	88	9.0	24.7
PS 30715-2	200	75	4.4	37.9
ICSH 90127	270	71	12.5	26.9
Resistant control				
IS 2205	300	89	14.9	18.7
Susceptible control				
ICSV 1	155	58	62.2	70.0

Plate 13. Midge-damaged panicle showing chaffy spikeiets, and inset, midge fly ovipositing in a flowering spikelet.

regions in Africa, Americas, Asia, Australia, and Europe. Damage is caused by the larvae, which feed on the ovary inside the glumes. This results in chaffy (empty) florets, and the panicles present a blasted appearance (Plate 13). Egg laying occurs in the morning and a female lays 75-100 eggs in florets at anthesis. Eggs hatch in 2-3 days. Larvae are orange-red and feed on the developing grain inside the glumes. Larval development is completed in 9-12 days, and pupation occurs beneath the glumes. The pupal period lasts for 3-4 days after which the pupa wriggles its way to the tip of glumes, and the adult emerges from

the pupal case leaving the characteristic white pupal skin attached to the glumes. Larvae may also diapause inside the glumes, and the diapause may last for one to several years.

Resistance Screening Techniques

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

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 head 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 consist of early and late 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 (Multichoice Conditions)

Hot Spots. Use hot-spot areas to screen effectively for midge resistance. Dharwad, Bhavanisagar, and Pantnagar in India, Sotuba in Mali, Farako Ba in Burkina Faso, Alupe in Kenya, and Kano in Nigeria are the hot-spot locations for sorghum midge.

Sowing Date. For successful screening of test material for midge resistance under natural conditions, determine the periods of maximum midge density through fortnightly sowings of a susceptible cultivar. Adjust sowing dates so that the most susceptible stage of the crop (flowering) coincides with greatest insect density. At ICRISAT Center, maximum midge density and damage have been observed in the crop planted during the 3rd week of July. The peak in midge density occurs during October. A second but smaller peak has been observed during March in the postrainy season, for which the optimum planting date is mid-December (Figure 10). At Dharwad, the peak in midge numbers has been recorded during October, and the optimum time for sowing test material is between 20 July and 5 August.

Augmentation of Midge Density. Midge populations can be

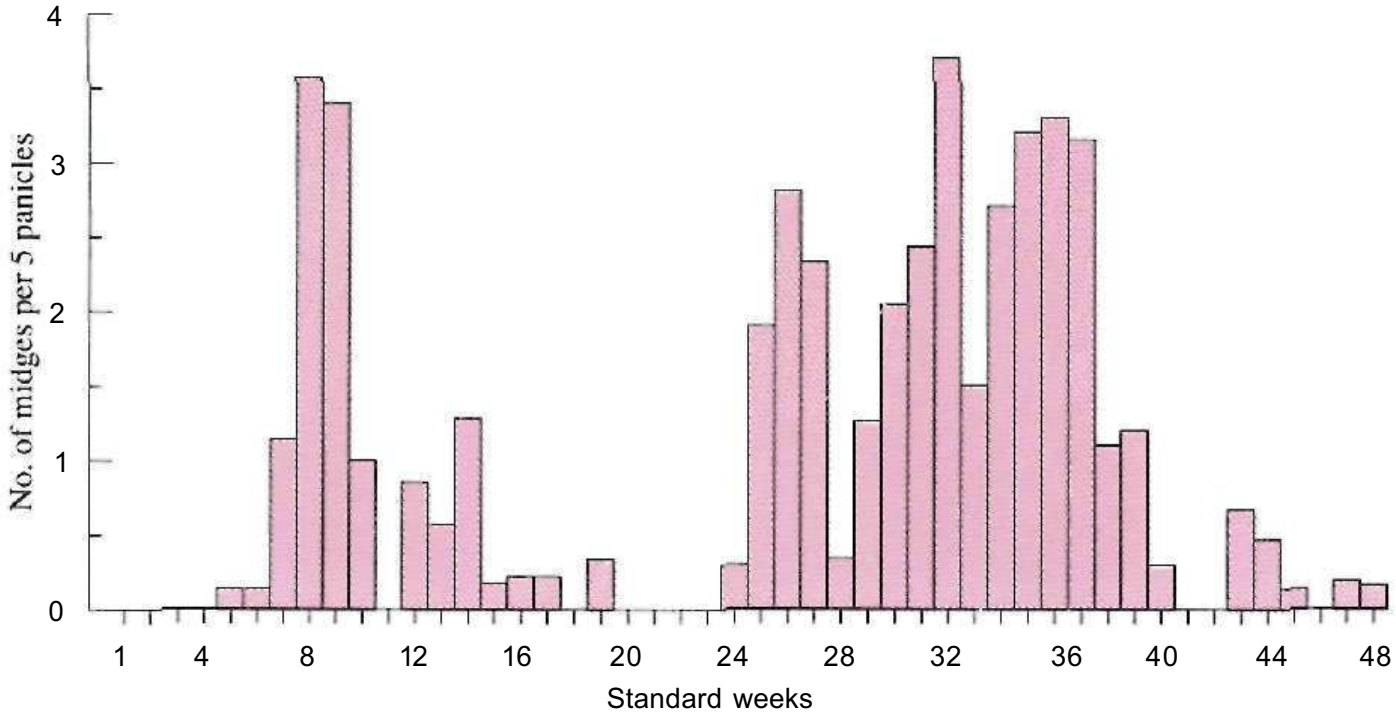


Figure 10. Population dynamics of sorghum midge (Contarinia sorghicola) *at ICRISAT Center, India, 1980/81.*

Plate 14. Infester row technique to screen for midge resistance. Four rows of a susceptible cultivar are planted after 16 rows of the test material.

augmented through infester rows and sorghum panicles containing diapausing midge larvae (Sharma et al. 1988a).

- Sow infester rows of CSH 1 and CSH 5(1:1 mixture) 20 days before the test material (Plate 14). Alternatively, early-flowering (40-45 days) lines (IS 802, IS 13249, and IS 24439) can be sown along with the test material to avoid problems in field management.
- Sow four infester rows after every 16 rows of the test cultivars.
- Spread midge-infested sorghum panicles containing diapausing larvae at the flag leaf stage of the infester rows. Moisten the panicles for 10-15 days to stimulate the termination of larval diapause for pupation and adult emergence. Adults emerging from diapausing larvae serve as a starter infestation in infester rows to supplement the natural population. Midge population multiplies for one to two 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 (Figure 11). Infester rows alone also increase midge damage.

Sprinkler Irrigation. High relative humidity is important for

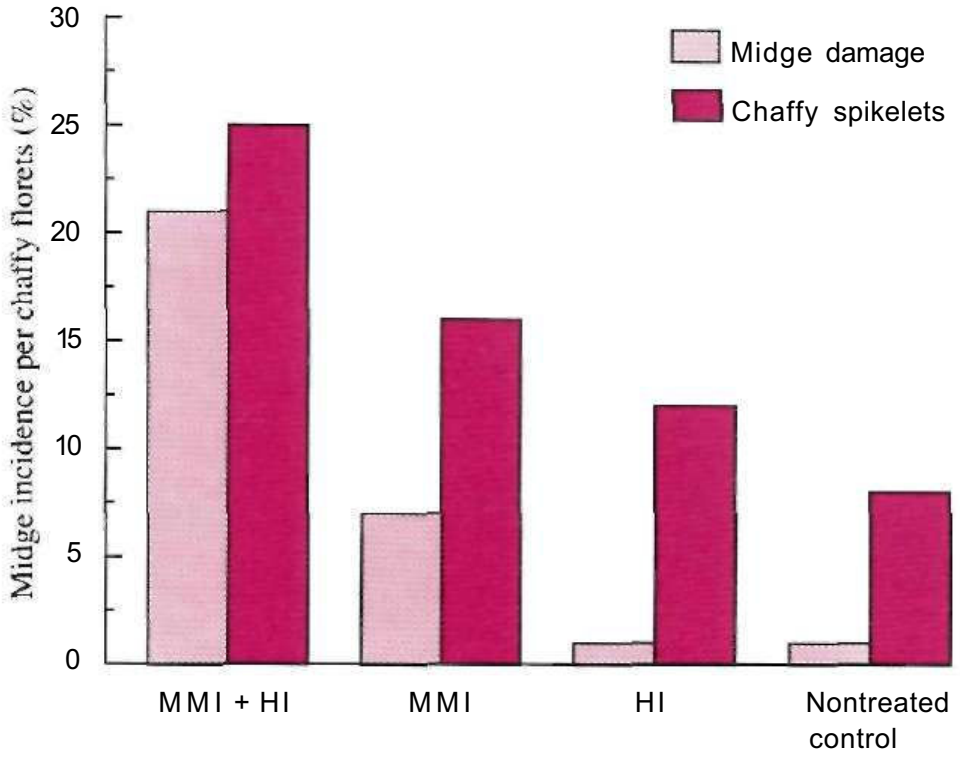


Figure 11. Midge population management for the resistance screening of sorghum using (1) mixed maturity infester rows (MMI) plus head inoculation (HI), (2) MMI only, and (3) HI only, on sorghum hybrid CSH 1, ICRISAT Center, India, postrainy season 1981/82.

midge activity, adult emergence, and subsequent damage. Use overhead sprinkler irrigation to increase relative humidity (RH) in the midge-screening trials during the postrainy season or periods of low relative humidity (Plate 15). 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. Use of sprinkler irrigation over the crop canopy between 1500 to 1600 does not affect oviposition because the peak midge abundance and oviposition occurs between 0730 to 1100 (Sharma et al. 1990).

Selective Use of Insecticides to Control *Calocoris angustatus* and *Tetrastich us diplosidis.* *C. angustatus* and *T. diplosidisare* the two major biotic factors limiting midge abundance in trials of screening for midge resistance. Head bugs damage the sorghum panicles from emergence to hard-dough stage and compete for food with sorghum midge. Also, adult head bugs prey on ovipositing midges at

Plate 15. Overhead sprinklers used to increase relative humidify in midge resistance screening.

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 head 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. 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 material increases the efficiency of selection for midge resistance.

Plant Density. Plant population affects the insect density/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 a lower planting density.

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 sowings, *and* selective use of contact insecticides for the control of head bugs and midge parasites. These techniques are useful in the initial large-scale screening of germplasm and breeding materials for resistance to sorghum midge.

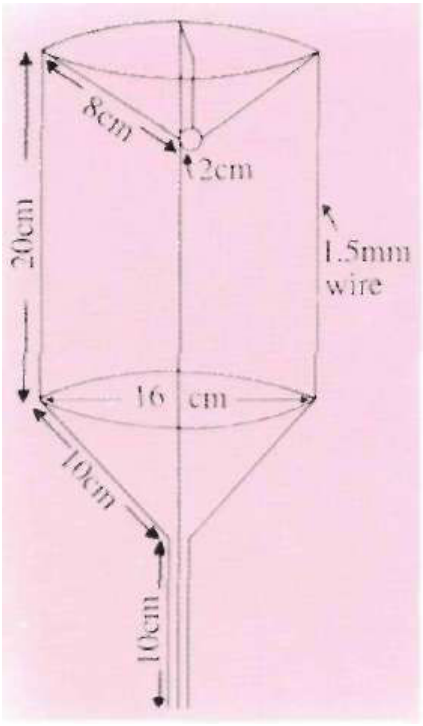


Figure 12. Measurements of headcage to screen for midge resistance under no-choice conditions.

Headcage Technique

Caging midge flies with sorghum panicles is an important method of avoiding escape, and allows screening for midge resistance under uniform insect pressure. A headcage technique developed and standardized at ICRISAT Center 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. These prevent the cage from slipping when disturbed by wind or other factors (Figure 12). Screening for resistance to midge can be carried out as follows:

- Select sorghum panicles at 25-50% anthesis stage. Remove florets with dried anthers at the top, and immature ones at bottom of the panicle with scissors (Plate 16a) so that only the florets at anthesis in the middle of the panicle are exposed to the midge flies.
- Place the wire-framed cage around the sorghum panicle and cover it with a muslin or any similar thin blue cloth bag (20 cm in diameter, 30 cm long) (Plate 16b). The cloth bag at the top has an extension (5 cm in diameter, 10 cm long) to introduce the midges.
- Collect 20 adult female midges in a plastic bottle (200 mL) aspirator (Plate 16c) between 0800 and 1100 from flowering sorghum panicles (only female midges visit the flowering sorghum panicles and these are collected for infestation).
- Release 40 midges into each cage and tie up the opening. 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 head bugs, head caterpillars, and predatory spiders.
- Remove the cages 15 days after infestation and evaluate midge damage as described in the following pages.

Florets with midge larvae and midge-damaged chaffy florets are greatest in panicles infested with 40 midges for two consecutive days. There is some variation in midge damage over seasons because of temperature, rainfall, and relative humidity which influence both ovi-

position and damage by the sorghum midge. Midge damage decreases as the time of collection and release advances from 0830 to 1430 (Table 5). Other factors which account for decrease in midge damage over

Plate 16. Headcage technique to screen for resistance to sorghum midge; (a) panicle trimmed with scissors, (b) headcage covered with cloth bag, and (c) aspirator used to collect midge flies.

Table 5. Effect of time of midge collection on midge damage under headcage conditions (40 midges/panicle).

Time of collection	Florets with midge larvae (%)		Chaffy florets (%)	
	Rainy season 1982	Postrainy season 1982/83	Rainy season 1982	Postrainy season 1982/83
0830	47.8(43.67)[1]	81.6(64.61)	67.0(55.16)	87.8(69.67)
1030	36.2(36.94)	44.0(41.54)	58.4(49.94)	53.2(46.86)
1230	37.2(37.39)	10.0(18.01)	74.0(59.80)	27.6(31.69)
1430	17.4(23.86)	7.4(15.36)	66.4(55.24)	39.4(38.86)
SE	±(2.89)	±(1.67)	±(3.67)	±(1.14)

1. Figures in parentheses are angular transformations.

time are natural death of adults (midges die between 4 and 24 h), 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 (Figure 13). Sorghum midge behavior is influenced by different colors (Sharma et al 1990). Among the various colored (blue, black, red, yellow, or white) muslin cloth bags tested, maximum midge damage has been recorded in panicles covered with blue and black bags (Table 6). Blue bags are used to cover the cages because the black bags may cause very high temperatures inside the cage during the hot and dry season in the semi-arid tropics.

The headcage technique is quite simple, easy to operate, and can be used on a fairly large scale to confirm the resistance of field-selected cultivars. Changing weather conditions influence midge activity, and

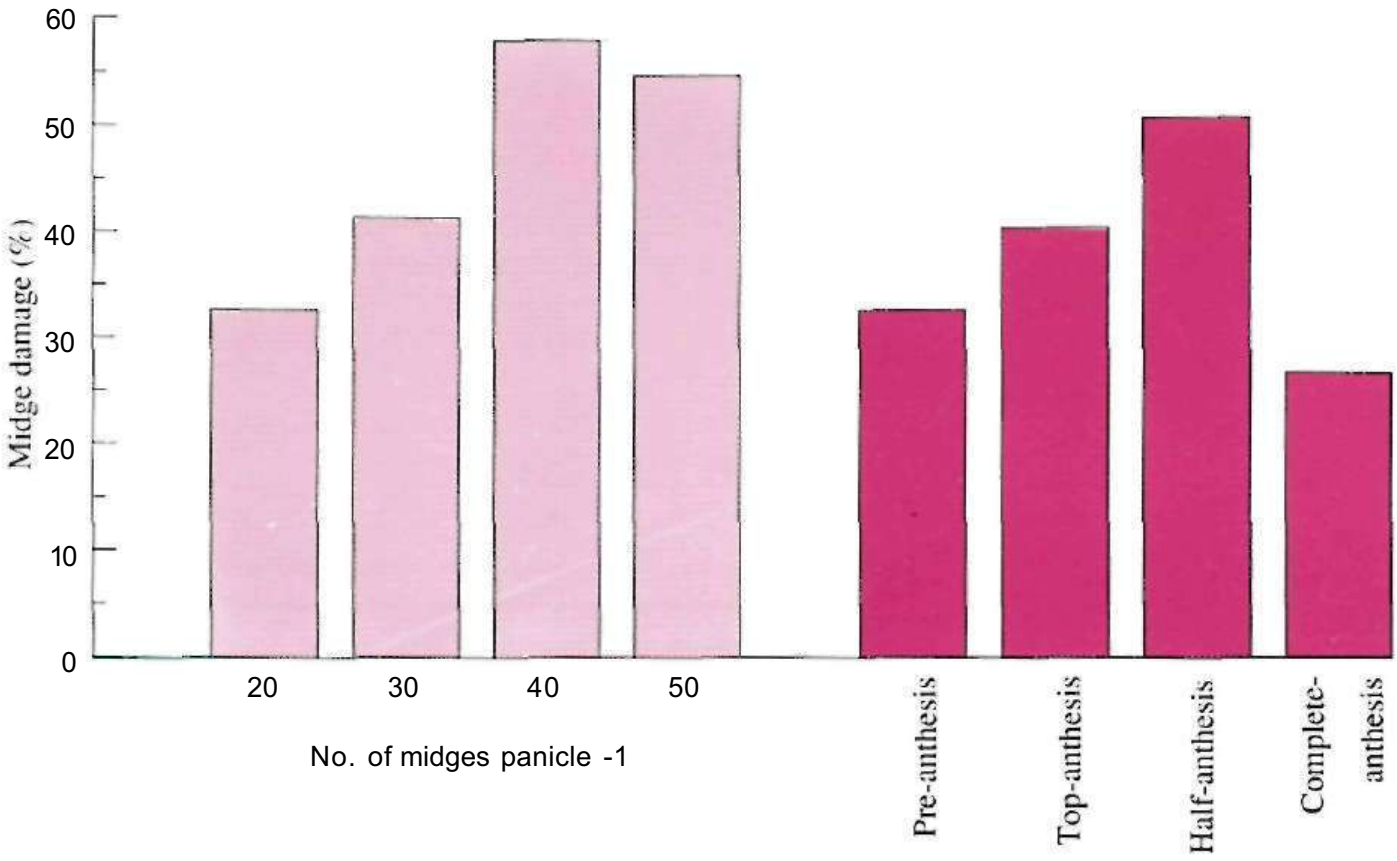


Figure 13. Midge damage in sorghum panicles (cv CSH 1) at different levels of midge pressure and stages of panicle development under headcage conditions, ICRISAT Center,1981/82.

Table 6. Effect of bag color on midge damage.

Bag color	Florets with midge larvae (%) Postrainy season 1981/82	Rainy season 1982
Blue	71.8(58.2)[1]	77(61.4)
Black	70.3(57.2)	76(60.1)
Red	66.9(55.1)	61(51.6)
Yellow	66.0(54.4)	63(52.7)
White	69.8(56.7)	66(54.7)
SE	±(1.84)	±(2.11)

1. Figures in parentheses are angular transformations.

can affect midge damage under the headcage. But in general, it is a thorough test for resistance screening, and is most useful for identification of stable and durable resistance. Test material should be screened under the headcage and over several testing environments to identify lines with stable resistance (Sharma et al. 1988b).

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 midge. However, the midge-infested panicles have either small white pupal cases hanging to the tip of spikelets or have small parasite exit holes in the glumes. The following methods are suggested for damage evaluation.

Chaffy Spikelets. This is the most appropriate criterion to evaluate sorghum lines for midge resistance. Tag five panicles in each genotype at half-anthesis. Record midge incidence in the florets 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-damaged florets).
- 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 1-9 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 = >80% midge-damaged spikelets (Plate 17).

Grain Yield. Record grain yield in genotypes being tested. The test material can be maintained under infested and noninfested conditions. Harvest all panicles from the middle row(s) at the time of maturity and record 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.

Sources of Resistance

We screened over 15 000 germplasm accessions for resistance to sorghum midge. The cultivars selected under natural conditions were tested under no-choice conditions in the headcage over many seasons and locations. Cultivars selected as midge resistant were tested at several locations through the International Sorghum Midge Nursery.

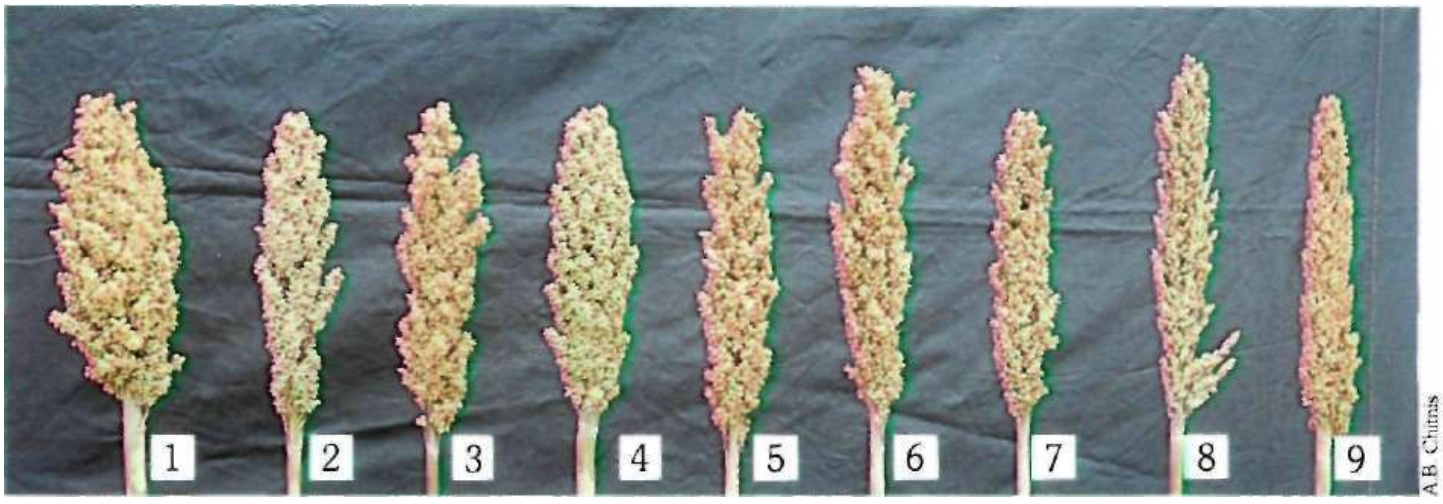


Plate 17. Visual damage rating scale for midge incidence.

Table 7 lists the cultivars showing resistance to sorghum midge. DJ 6514, TAM 2566, AF 28, IS 10712, IS 8918, and IS 7005 are stable and diverse sources of resistance to sorghum midge. ICSV 197, ICSV 745, ICSV 843, ICSV 88013, and ICSV 88032 are the improved cultivars with high levels *of* midge resistance with yield potential comparable *to the* commercially released cultivars. The differences in susceptibility to sorghum midge are illustrated in Plate 18a, b, c, and d.

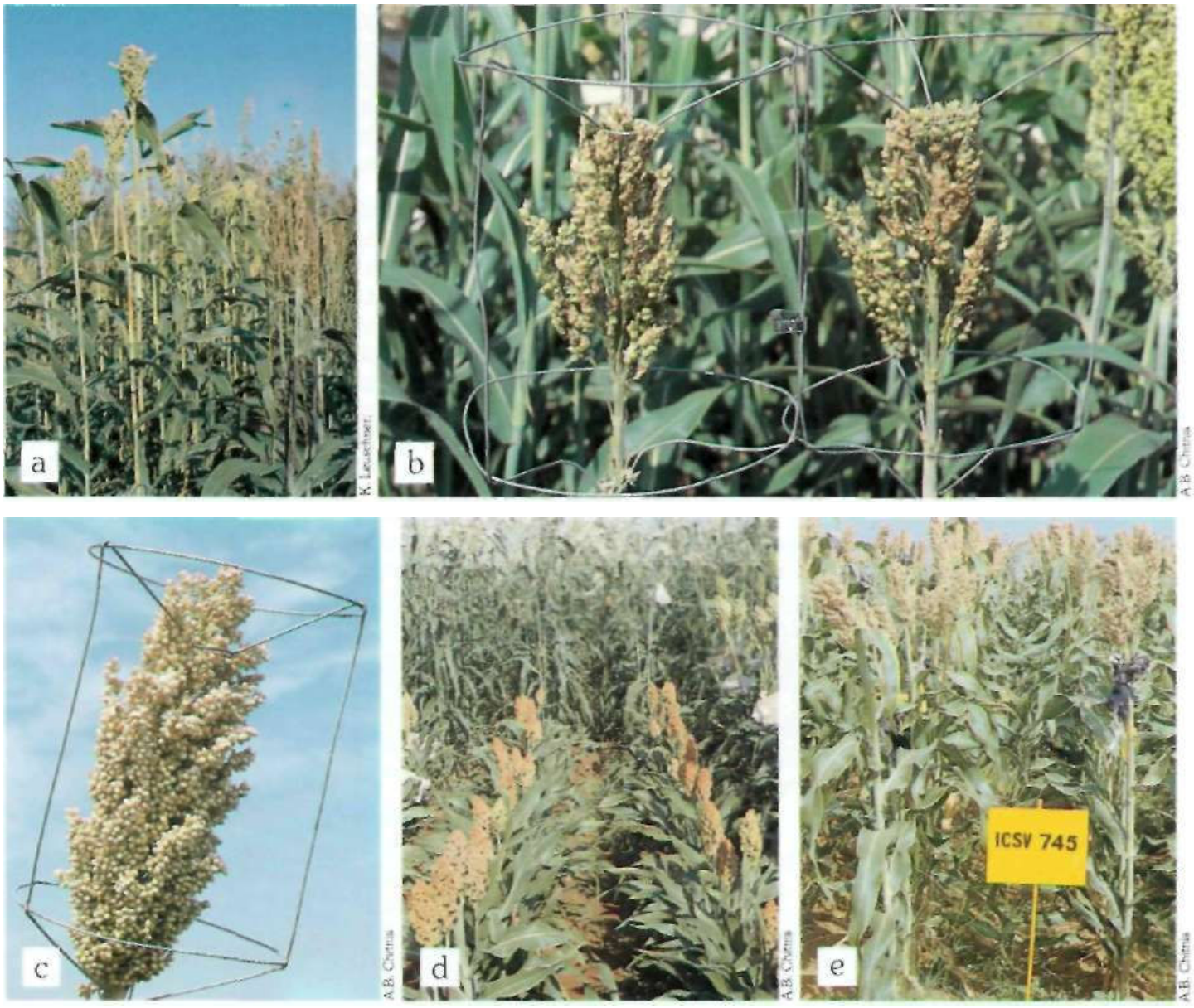


Plate 18. Cultivar differences in susceptibility to sorghum midge. (a) Differences in midge damage under natural infestation: ICSV197(left) showing no damage and CSH1 (right) showing cent percent damage. (b) Midge susceptibility under headcage: CSH1 susceptible, and (c) ICSV 745—resistant. (d) Sources of resistance to sorghum midge. (e) ICSV 745, an improved midge-resistant variety.

Table 7. Midge damage ratings of resistant sources under natural and headcage conditions, ICRISAT Center, India.

Genotype	Plant height (cm)	Time to 50% flowering (days)	Damage rating[1] ± SE		Midge damage (%)	
			Natural infestation	Headcage conditions	Natural infestation	Headcage conditions
IS 3461	385	71	2.0±0.15	2.0±0.00	21	19
IS 7005	300	75	2.3±0.15	2.4±0.24	24	18
IS 8671	185	75	2.6±0.69	4.0±0.00	24	16
IS 8751	390	60	2.4±0.16	2.8±0.37	26	22
IS 8884	275	112	2.0±0.24	2.6±0.37	18	26
IS 8887	290	112	2.4±0.20	2.6±0.24	20	28
IS 8891	320	109	1.7±0.19	4.0±0.50	24	11
IS 8918	290	111	2.0±0.00	2.0±0.00	26	18
IS 9807	370	75	2.5+0.17	2.6±0.24	23	26
IS 10712	195	78	2.5±0.43	3.0±0.36	31	31
IS 15107	260	84	3.0±0.22	3.4±0.40	33	32
IS 18563	240	74	3.3±0.33	2.5±0.50	23	28
IS 18695	75	65	3.6±0.26	3.4±0.51	18	14
IS 18698	315	70	2.2±0.39	2.8±0.48	20	23
IS 19474	365	76	1.9±0.29	1.910.52	22	24
IS 19476	370	72	2.3±0.13	2.0+0.00	16	15
IS 21871	90	71	2.0±0.28	1.4+0.38	26	46
IS 21873	95	71	4.3±0.64	5.010.00	22	48
IS 21879	100	70	2.5±0.34	3.8+0.75	21	21
IS 21881	90	68	3.1±0.43	3.9±0.70	28	28
IS 21883	110	69	3.0±0.26	4.0±0.76	25	27
IS 22806	330	71	1.9±0.26	1.610.29	13	12
IS 26789	230	69	2.9±0.22	3.2±0.44	39	23
IS 27103	195	71	1.6±0.21	1.610.37	22	17
ICSV 197	278	80	1.4±0.18	1.410.19	15	18
ICSV 386	141	80	2.0±0.30	2.8±1.44	22	26
ICSV 387	168	65	2.9±0.35	3.0±0.52	24	22
ICSV 388	291	62	2.4±0.51	1.8±0.25	17	19
ICSV 389	126	68	2.9±0.32	3.2±0.73	30	20
ICSV 391	145	73	3.7±0.36	3.8±0.41	18	15
ICSV 393	156	60	3.3±0.44	4.5±0.96	26	31
ICSV 397	253	84	3.0±0.00	2.0±0.00	28	31
ICSV 563	149	59	2.8±0.23	2.5±0.00	28	35
ICSV 564	191	60	3.4±0.26	3.6±0.47	10	22
ICSV 690	152	57	4.3±1.36	2.3±0.75	14	28
ICSV 692	199	59	2.9±0.40	2.7±0.60	21	18
ICSV 729	74	66	2.3±0.36	2.4±0.43	21	16
ICSV 730	130	78	3.1±0.46	3.2±0.73	18	15
ICSV 731	140	72	3.1+0.44	4.3±0.60	22	27
ICSV 736	239	76	3.9±0.47	3.3±0.33	18	13

Continued

Table 7. *Continued*

Genotype	Plant height (cm)	Time to 50% flowering (days)	Damage rating[1] ± SE		Midge damage (%)	
			Natural infestation	Headcage conditions	Natural infestation	Headcage conditions
ICSV 737	285	76	2.8±0.33	3.2H.03	24	27
ICSV 739	241	70	3.0±0.82	2.5±0.00	22	28
ICSV 744	282	77	3.8±0.52	2.2±0.33	22	8
ICSV 745	215	71	2.0±0.50	2.5±0.20	18	22
ICSV 746	259	77	2.4±0.26	2.4±0.24	17	11
ICSV 748	279	81	2.8±0.18	4.0±0.00	12	15
ICSV 752	166	71	3.1±0.52	3.5±0.50	26	19
ICSV 753	147	72	2.9±0.52	2.5±0.50	40	32
ICSV 757	221	77	2.5±0.54	3.0±0.00	16	9
ICSV 843	260	78	4.0±1.00	3.5±0.00	14	28
ICSV 88006	250	68	3.8+1.25	2.510.00	14	32
ICSV 88013	217	70	4.1±0.69	2.8±0.32	21	15
ICSV 88014	267	69	3.4±0.37	3.5±0.00	17	10
ICSV 88028	149	70	3.4±0.42	3.0±0.29	28	19
ICSV 88032	201	61	3.4±0.76	2.1±0.13	14	12
ICSV 88035	200	69	3.0±0.34	4.8H.25	10	39
ICSV 88036	145	66	2.9±0.41	2.6±0.24	17	30
ICSV 88041	123	66	2.6±0.49	2.3±0.43	18	11
ICSV 89049	129	67	3.0±0.55	3.1±0.51	16	18
ICSV 89051	302	83	3.1*0.38	2.7±0.30	17	11
ICSV 89052	302	84	2.5±0.38	2.710.30	22	8
ICSV 89053	160	74	3.1±0.39	3.010.29	20	28
ICSV 89054	246	68	3.1±0.23	3.5±0.50	19	18
ICSV 90001	160	65	3.0±0.61	3.8±0.52	21	17
ICSV 90002	165	63	3.0±0.50	3.3±0.80	14	32
ICSV 90003	155	71	3.5±0.61	3.4±0.58	24	27
ICSV 90004	200	71	3.3±0.31	3.911.20	19	18
ICSV 90005	180	65	2.510.32	3.410.48	22	27
Resistant controls						
DJ 6514	230	71	1.3±0.14	1.8±0.43	21	20
TAM 2566	85	64	2.210.40	3.310.63	22	17
AF 28	320	71	1.7±0.29	1.0±0.00	25	18
Susceptible controls						
CSH 1	155	58	8.4±0.28	9.0+0.16	92	90
CSH 5	200	67	8.3±0.25	8.8±1.03	77	82
CSH 9	210	68	7.4±0.55	8.5±0.00	72	85
CSH 11	210	64	6.3H.02	7.2+1.11	84	89
Swarna	155	65	8.2+0.40	8.2±1.01	88	95
SE					±6.6	±4.7
CV (%)					25	21

I. Damage rating : 1 = < 10% midge damage, and 9 = > 80% midge damage.

Sorghum Head Buss

Mirid head bugs (*Calocorisangustatus, Creontiades pallidus, Eurystylus immaculatus,* and *Campylomma* spp) are very serious pests of grain sorghum in India and Africa, of which *C. angustatus is* the most important species in India and *E. immaculatus* in West Africa.

C. angustatus nymphs and adults feed mainly on the developing grain, and occasionally on other tender parts of the plant (Plate 19). The nymphs and adults suck sap from the developing grain, which remain unfilled, shrivel, and under severe infestation, become completely chaffy. Damage during the early stages of grain development results in heavy yield loss; later infestation results largely in loss of quality. The damaged grains show distinct red-brown feeding punctures, and in cases of severe feeding, become completely tanned (Plate 19). However, such grains are more prone to mold incidence and show poor seed germination.

C. pallidus, E. immaculatus (Plate 20), and *Campylomma* spp insert their eggs inside the grain at the milk stage. 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.*

Females of *C. angustatus,* after a pre-oviposition period of 2-4 days, lay cigar-shaped eggs inside the glumes before anthesis. Eggs hatch in 7-8 days, and the five nymphal instars complete development in 8-12 days. A female lays 182±21 eggs during the rainy, and 113±12 eggs during the postrainy season (Sharma and Lopez 1990). The off-season carryover of this bug is not known, except that the bugs are known to feed on sorghum fodder grown during summer.

Plate 19. Nature of damage caused by head bug (C. angustatus,), *and inset, adult head bug.*

Plate 20. Eurystyius immaculatus *damage on sorghum panicle, and inset, nymphs and adults.*

Resistance-Screening Techniques

Various techniques to screen for resistance to head bugs have been described by Sharma (1985c), Sharma and Lopez (in press), and Sharma et al. (in press).

Field Screening

Screening for head bug resistance can be carried out under field conditions during periods of maximum bug density. However, screening under field conditions is influenced by: (a) variation in flowering of sorghum cultivars; (b) fluctuations in bug population, and (c) the effect of weather conditions on the head bug population buildup and damage. Early- and late-flowering cultivars normally escape head bug damage, while those flowering during midseason are exposed to very high

populations. The following methods can be used to increase the screening efficiency for head bug resistance under field conditions.

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

Sowing Date. Adjust sowing dates such that flowering coincides with maximum head bug density. Determine the periods of maximum

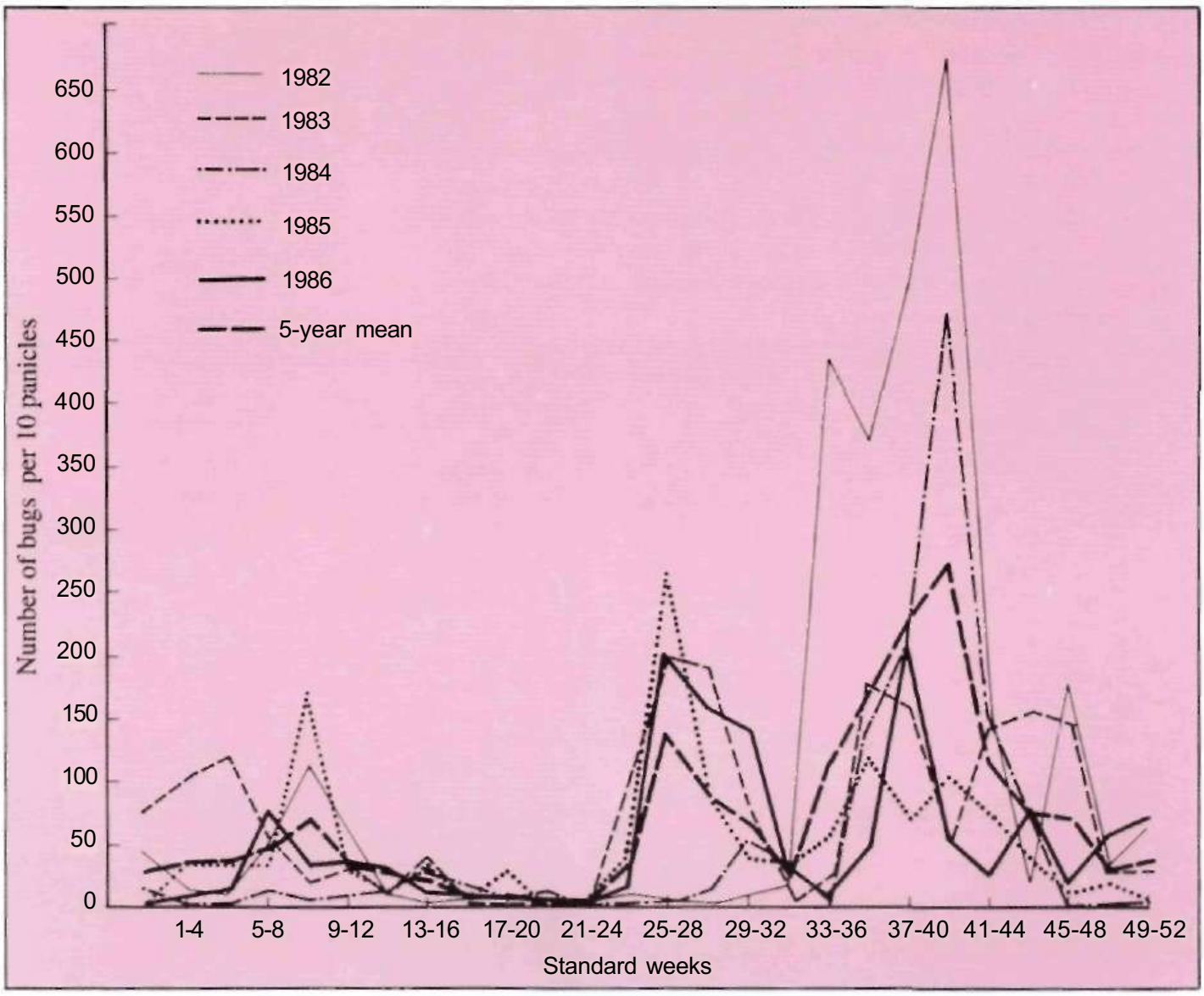


Figure 14. Seasonal abundance of Calocoris angustatus *at ICRISAT Center, India, 1980-86.*

head bug density through fortnightly sowings. Maximum head bug numbers at ICRISAT Center have been recorded during September, and a second but smaller peak has been recorded during March (Figure 14). Crops sown during the second week of July suffer the greatest head bug damage. At Bhavanisagar, the peak in head bug density occurs during May-June, and the optimum time to sow for resistance screening is during the second fortnight of February.

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 24439) along with the test material as infester rows. Sow four rows of a susceptible cultivar after 16 test rows (Plate 21).
- Collect bugs from other fields and spread them in the infester rows at panicle emergence to augment the bug population.
- Use sprinkler irrigation during the postrainy season to build up the bug population.

Plate 21. Infester row technique to screen for resistance to head bugs.

- 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 population.

Plate 22. Headcage technique to screen for resistance to head bugs.

Headcage Technique

To overcome the problem of variation in flowering among test cultivars and fluctuating insect populations, the headcage technique developed for midge resistance screening has been found to be useful for head bugs also (Plate 22). The headcage technique allows bug population buildup 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 pre-anthesis to top-anthesis stage in each plot/genotype.
- Tie the headcage around the sorghum panicle and cover it with a white muslin cloth bag as described for sorghum midge.
- Collect head bugs in muslin cloth bags from sorghum panicles at the milk stage.
- Separate the adult males and females (males are smaller and have a darker color on the back).
- 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 head caterpillars or predatory spiders if there are any.
- Remove the head bugs 20 days after infestation in the muslin cloth bag, and take them to the laboratory. Kill the bugs using ethyl acetate (2 mL per bag) or keep the bags under deep freeze for 30 min. Count the total number of bugs in each cage.
- Evaluate the panicles for head bug damage at maturity as described under damage evaluation.

Greatest head bug population buildup occurs in panicles infested with 10 pairs of bugs per panicle across all stages of panicle development and in panicles infested at the half-anthesis stage (Table 8). Head bug population buildup decreases linearly with an advance in the stage of panicle development at the time of infestation.

Screening for Resistance to *Eurystylus immaculatus*

- Infester row technique described to screen for resistance to

Table S. Population buildup and grain damage by *Caloeoris angustatua* under the headcage at four levels of infestation and four stages of panicle development (cv. CSH 1), ICRISAT Center, postrainy season, 19S2/83.

No. of pairs released	No. of head bugs/panicle				Damage rating[2]			
	Pre-anthesis	Half-anthesis	Complete-anthesis	Milk stage	Pre-anthesis	Half-anthesiis	Complete-anthesis	Milk stage
5	200 (13.9)[1]	338 (18.2)	285 (16.8)	220 (14.4)	9.0	7.5	7.2	4.5
10	468 (21.5)	503 (22.4)	516 (22.7)	157 (12.0)	9.0	9.0	8.0	6.0
15	328 (18.1)	481 (21.8)	456 (21.2)	265 (15.7)	9.0	9.0	8.0	6.0
20	151 (12.3)	412 (20.1)	321 (17.7)	170 (12.8)	9.0	9.0	9.0	6.0
Mean	151 (16.5)	434 (20.6)	395 (19.6)	203 (13.7)	9.0	8.6	8.0	5.6
SE to compare head bug pairs		No. of bugs (±0.81)				Damage rating ±0.07		
Stages of panicle development		(±1.34)				±0.12		

1. Figures in parentheses are $\sqrt{N}$ transformed values.
2. Damage rating: 1 = grain with a few feeding punctures; 9 = grain showing > 75% shriveling; slightly visible outside the glumes; and highly tanned appearance.

C. angustatus can be adapted to screen for resistance to *Eurystylus* also.

- The hot-spot locations for *Eurystylus* 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 head bug incidence has been observed during the first fortnight of October.
- For maximum head bug damage, sow the crop during the second fortnight of July.
- 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 in the case of *C. angustatus.*

The head cage technique described for *C. angustatus* has been standardized to screen for resistance to *Eurystylus immaculatus.*

- Select 5-10 panicles at the complete-anthesis stage (6 days after flowering) in each genotype/plot.
- Collect adult bugs from sorghum panicles at the 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 (sex ratio is closer 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 head bugs in each infested panicle as described under damage evaluation.
- At maturity, evaluate the panicles for head bug damage.

For better results, it is important to maintain uniformity in panicle size amongst the genotypes being tested, and to record data both on head bug numbers and grain damage to select resistant genotypes.

Damage Evaluation for Resistance Screening

Sorghum head bugs suck the sap from developing grain which results in shriveling and tanning of grains. Some of the grains may remain undeveloped. The damage symptoms are normally evident on some or all 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 half-anthesis. Sample the panicles for head 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 1-9 scale (1 = all grains fully developed with a few feeding punctures; 2 = grain fully developed, with feeding punctures; 3 = grains showing 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 of the grain highly shriveled with a dark-

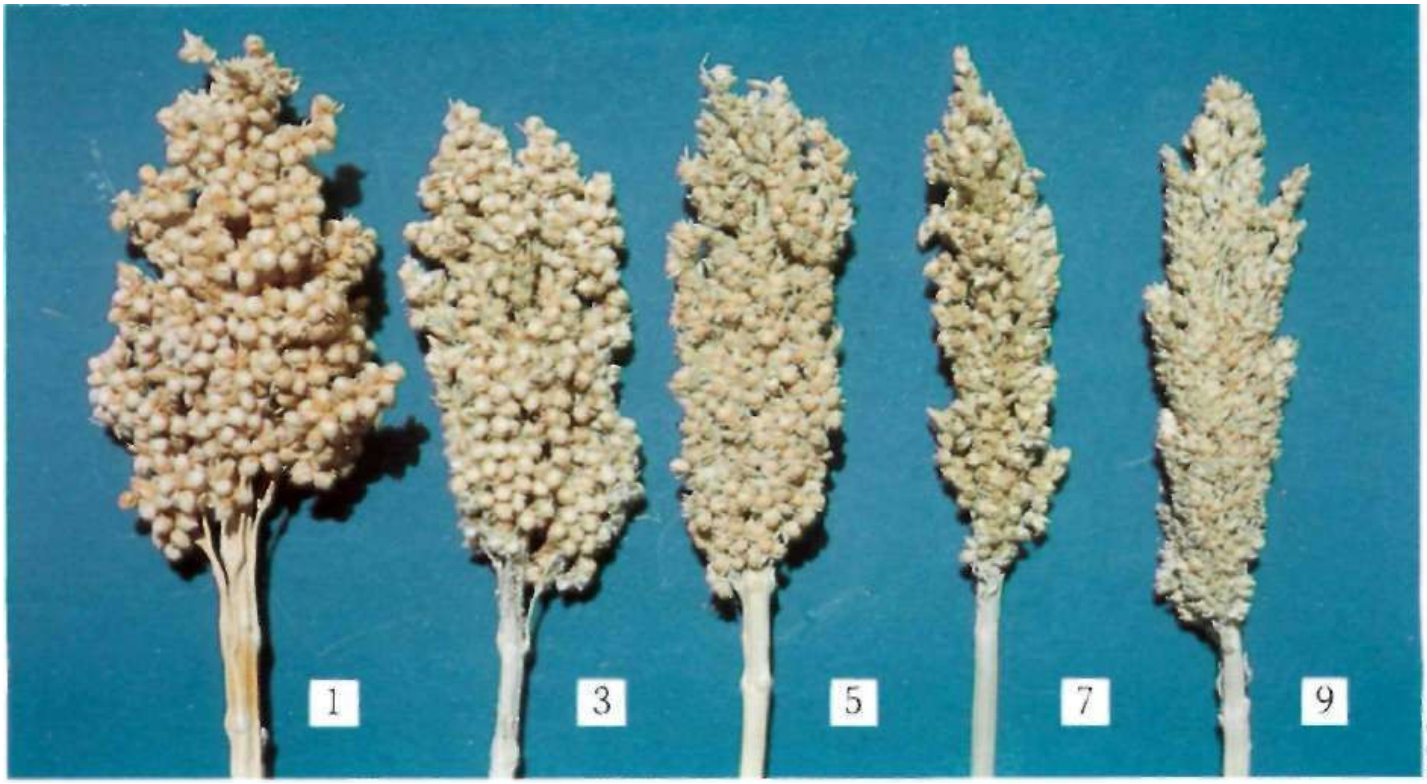


Plate 23. Visual damage rating scale (or head bugs.

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 (Plate 23).

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. Express the loss in grain yield of infested plots or panicles as a percentage of the grain yield in non-infested plots or panicles.

Grain Hardness. Head bug damage makes the grain soft, and floury. Evaluate grain hardness on a 1-5 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 1000 grains at random from each replication or panicle. Equilibriate 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

1000 grain sample in the 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 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, 1000 grain mass, percentage of floaters, and percentage of germination should only be collected when the scientists intend to collect more data for in-depth studies on head bug resistance.

Sources of Resistance

Over 15 000 sorghum germplasm accessions have been screened for resistance to *C. angustatus* at 1CRISAT Center under field conditions. Selected lines have been tested for several seasons using the headcage technique (Table 9). IS 17610, IS 17618, IS 17645, IS 20740, and IS 20664 are moderately resistant to *C. angustatus.* Differences in susceptibility to head bugs are shown in Plate 24a and b.

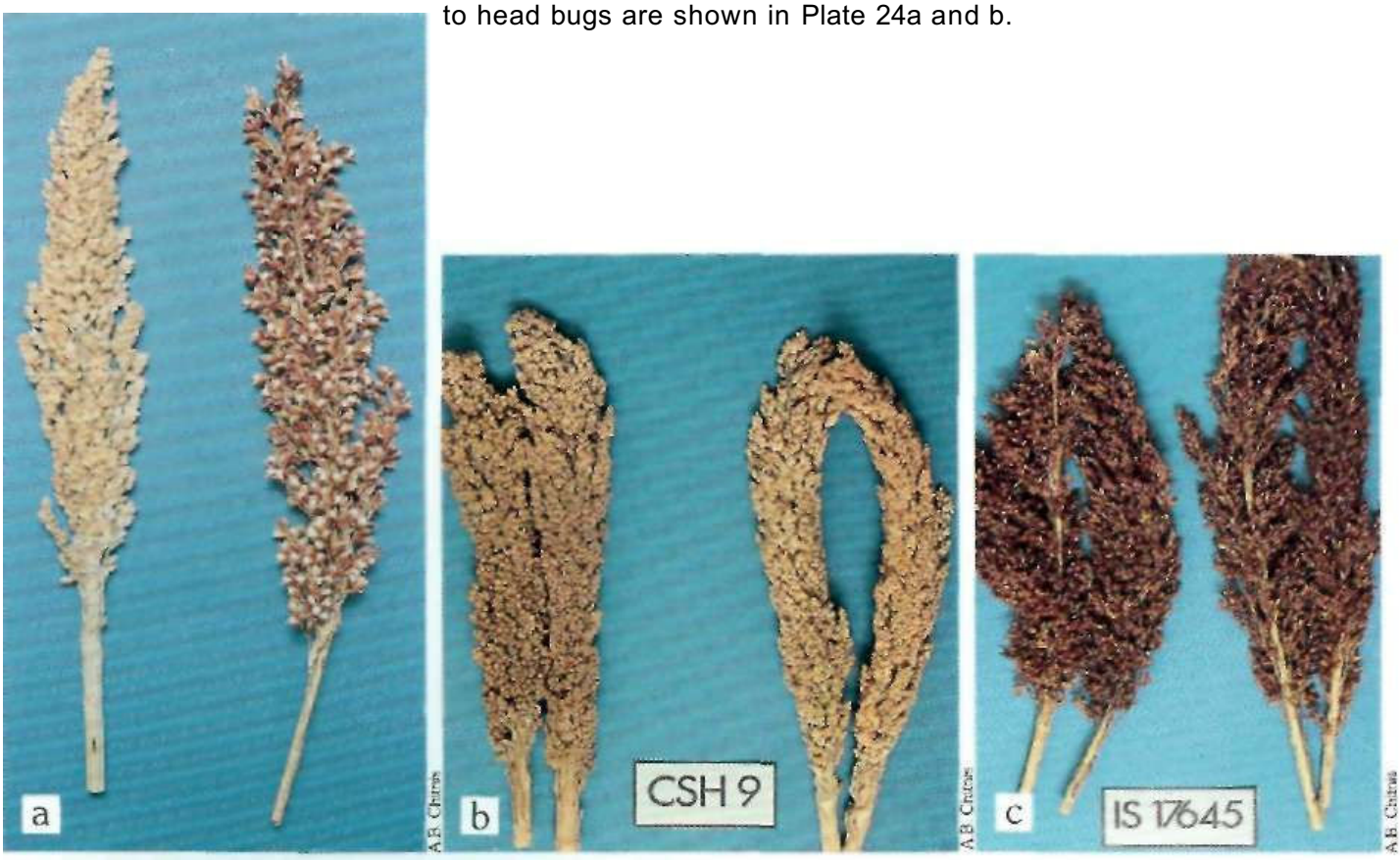


Plate 24. Differences in cultivar susceptibility to C. angustatus; *(a) CSH9—susceptible (left), versus IS 17610—resistant (right), and (b) CSH 9—susceptible versus (c) IS 17645—resistant.*

Table 9. Response of 26 sorghum genotypes to the head bug, *C. angustatus,* under natural and headcage conditions, rainy season 1989.

Genotype	Plant height (cm)	Time to 50% flowering (days)	Damage rating[1]		
			Headcage conditions with		Natural conditions
			5 pairs	10 pairs	
IS 14108	218	54	4.8	5.0	1.9
IS 14317	308	74	7.7	6.5	1.4
IS 16357	214	68	3.8	5.8	2.2
IS 17618	392	110	2.3	5.0	2.2
IS 17645	425	110	5.2	.[2]	1.7
IS 19455	267	71	6.3	6.5	2.8
IS 19948	305	76	5.9	5.8	2.2
IS 19949	285	81	4.5	5.2	2.2
IS 19950	329	78	3.6	5.0	3.1
IS 19957	308	78	4.5	4.0	2.8
IS 20059	348	72	5.2	5.9	3.1
IS 20068	329	73	5.6	5.6	1.9
IS 20664	300	77	5.0	6.7	2.5
IS 20740	255	75	4.7	6.8	2.5
IS 21443	268	72	5.9	7.0	3.1
IS 21444	258	71	6.5	5.9	3.3
IS 21574	384	75	5.2	6.5	2.8
IS 22284	252	88	5.7	6.1	1.9
IS 25760	296	72	4.7	4.7	2.8
IS 27329	326	74	5.2	5.0	1.9
IS 27452	332	85	3.9	4.5	2.8
IS 27477	332	82	3.2	3.8	2.5
Resistant control					
IS 17610	425	110	1.1	2.7	1.4
Susceptible control					
CSH 1	120	66	8.8	9.0	4.7
CSH 5	165	74	7.9	9.0	4.2
CSH 9	129	76	7.9	9.0	4.2
Mean			5.2	5.9	2.6
SE			±0.56	±0.50	±0.35

1. Damage rating: 1 = grain with a few feeding punctures, and 9 = grain showing >75% shriveling, slightly visible outside the glumes and highly tanned.
2. = not studied.

For *Eurystylus immaculatus,* over 1000 lines have been evaluated for resistance under field conditions. IS 14332, CSM 388, Malisor 84-7, Sakoika, IS 2474, IS 907, IS 22227, SK 86, E 1140, SK 140, Kamboinse Local, and S 29 have been identified to be resistant. Some of the field-selected lines have been tested under no-choice conditions in the headcage (Table 10). CSM 388 (Plate 25a and b), Malisor 84-7, IS 14332, and Sakoika have also been found to be resistant to head bugs under the headcage screening.

Table 10. Head bug (*E. immaculatus*) numbers and grain damage in 11 sorghum cultivars under natural and headcage conditions, Sotuba, Mali, rainy season 1985.

	No. of head bugs/panicle		Damage rating[1]	
Genotype	Natural infestation	Headcage conditions	Natural infestation	Headcage conditions
IS 14332	5± 0.8	56± 4.7	1.1±0.11	1.0±0.01
CSM 388	9± 3.8	133±16.7	1.0+0.11	1.0±0.29
Malisor 84-7	20± 6.9	147+10.1	1.4+0.25	2.0±0.14
83F6-87	26± 6.3	101+14.5	2.2±0.37	3.0±0.37
A 13120	74±13.7	182±72.5	2.4±0.25	2.0±0.25
83F6-111	45± 8.3	119±49.0	2.6±0.25	3.010.36
ICSV 197	49±16.9	101± 7.0	2.8±0.20	3.0±0.65
E 35-1	100±19.3	175±35.9	3.0±0.32	3.010.48
83F6-16	27±14.7	187±37.3	3.0±0.45	4.0±0.52
83F6-42	37±13.2	262±65.8	4.4±0.25	3.510.53
83F6-148	39± 5.4	170±45.1	4.4±0.25	4.0±0.65

1. Damage rating : 1 = grains with a few feeding punctures, and 5 = most of the grains become highly shriveled, and slightly visible outside the glumes.

Plate 25. Differences in cultiwar susceptibility to Eurystylus immaculatus: *(a) E 35-1—moderately susceptible (left), Malisor 84-7—resistant (center), and ICSV 1063 BF—susceptible (right); (b) CSM 388—resistant (left), and S 35—susceptible (right).*

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International Crops Research Institute for the Semi-Arid Tropics
Patancheru, Andhra Pradesh 502 324, India

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