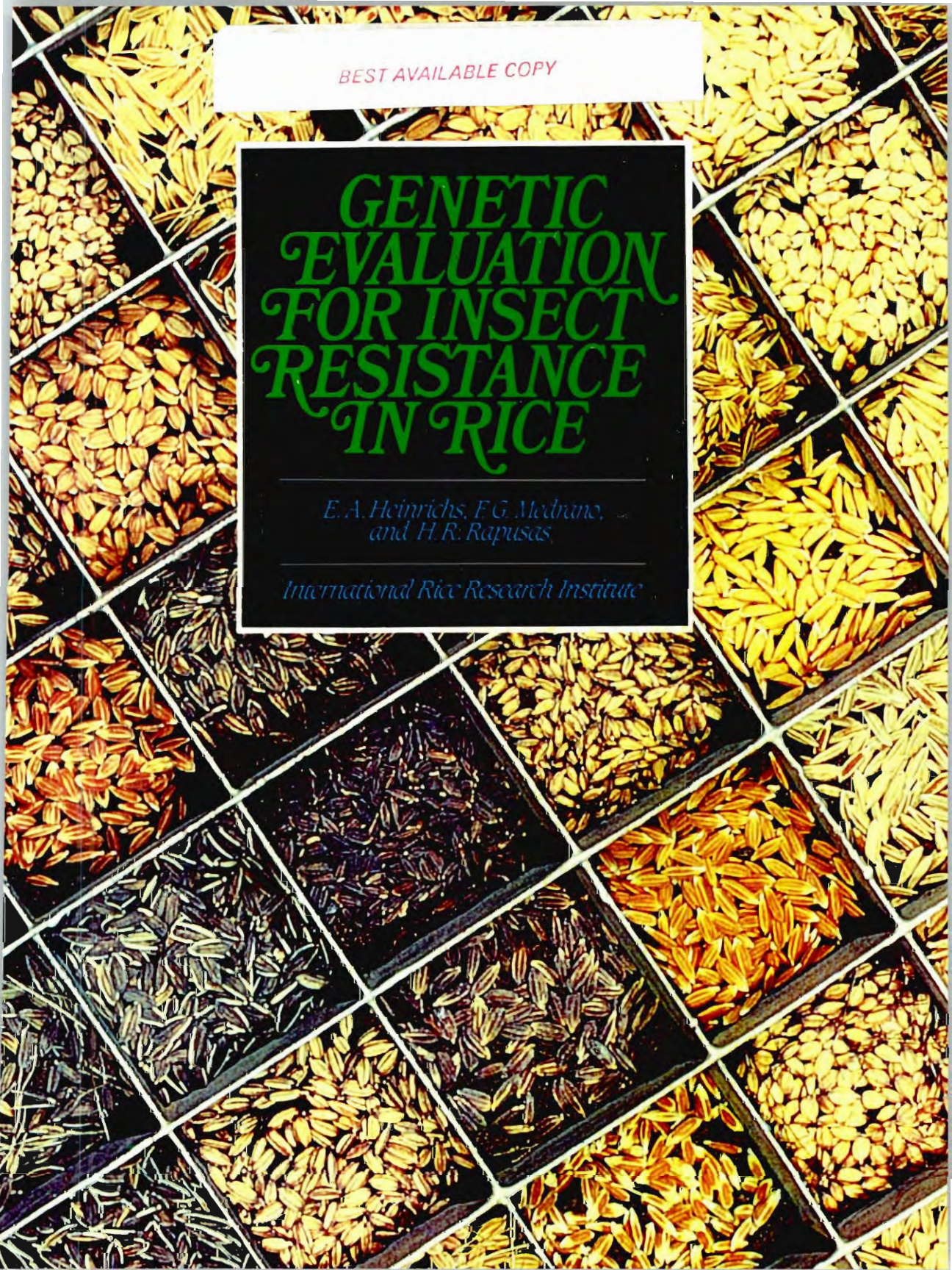

GENETIC EVALUATION FOR INSECT RESISTANCE IN RICE

E. A. Heinrichs, F. G. Medrano, and H. R. Rapusas

International Rice Research Institute

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International Rice Research Institute
Los Baños, Laguna, Philippines
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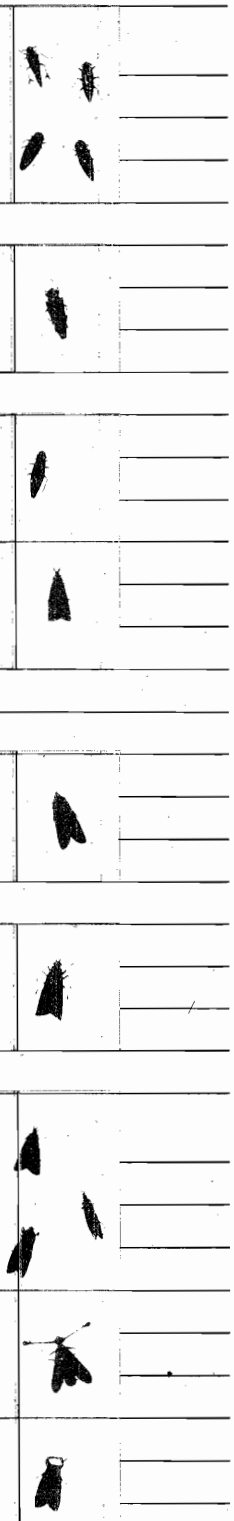

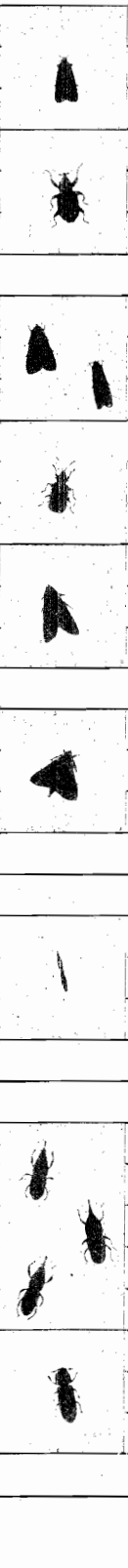

PREFACE

The growing of insect-resistant rice varieties is a major tactic in the integrated control of rice insect pests. However, the area planted to these varieties is only a fraction of the total area planted to rice. Insect resistance in commercial varieties is limited to a few insects: several hoppers, the stem borers, and the gall midge. To take full advantage of resistant varieties, we need to expand breeding programs for insect resistance. The purpose of this book is to encourage this expansion.

The introductory chapter describes the principles of insect resistance and the role of varietal resistance in integrated rice insect pest management. The second chapter gives general information on developing an insect resistance program and describes procedures for insect resistance screening. In the succeeding chapters, procedures for individual species or groups of closely related species are described. Subjects covered are

- insect rearing,
- screening for insect resistance, and
- determining the mechanisms of resistance.

Where information is available, a list of the sources of resistance is included. Each chapter is written so that it can stand alone. (There is some repetition of figures among the chapters and the references.) Appendices list sources of rice seed for resistance studies, general references on insect resistance, and an explanation of the codes used to name breeding lines in the various national and international programs. A glossary of terms used is included.

We are indebted to several scientists instrumental in the preparation of this book. Dr. R. B. Mills of Kansas State University and Dr. R. R. Cogburn, USDA, Beaumont, Texas, reviewed the chapters on stored grain insects; Dr. C. M. Smith, Louisiana State University, reviewed the chapter on the rice water weevil; and Dr. M. B. Kalode, AICRIP, Hyderabad, India, and Arifin Kartohardjono, BORIF, Bogor, reviewed the chapter on gall midge. Dr. S. Chelliah and Dr. R. Velusamy of Tamil Nadu Agricultural University, Coimbatore, India, provided unpublished material on determining levels of brown planthopper and leaffolder resistance. Dr. M. S. Alam of the International Institute of Tropical Agriculture, Nigeria, provided unpublished information on procedures used to screen for resistance to the stalk-eyed fly.

The manuscript was completed while E. A. Heinrichs was on sabbatical leave at the Entomology Department, University of Nebraska, Lincoln. The facilities provided by Dr. E. A. Dickason, department chairman there, and the typing of the edited manuscript by Mrs. C. Faust are greatly appreciated. Ms. D. C. Capili, secretary in the Department of Entomology at IRRI, typed the first two drafts. Ms. Kristin N. Heinrichs and Shawn M. Heinrichs assisted in the preparation of the glossary.

Many of the procedures described in this book have been developed over 20 years by IRRI research assistants and scholars from many countries. Special appreciation for helping with this book is due A. Marciano Romena, I. Domingo, C. R. Vega, L. Malabuyoc Sunio, V. Viajante, E. Baldos Medina, E. Camañag, and P. Paguia Schuler.

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FOREWORD

Rice varieties with multiple resistance to insects and diseases are grown on more than 20 million hectares in Asia and Central and South America. In Indonesia and the Philippines, insect-resistant rice varieties are grown on virtually all of the irrigated area. Progress in the breeding and distribution of resistant rice varieties has been phenomenal. Screening for brown planthopper (BPH) *Nilaparvata lugens* began in 1967. By 1973 the first BPH-resistant variety was released, and now BPH-resistant varieties are grown on more than 15 million hectares in Asia.

Insect-resistant varieties are a major element in the integrated control of rice insect pests, and their use is compatible with other control tactics. Resistant varieties provide pest control at essentially no cost to the farmer. This is extremely important as today's rising input costs severely erode the farmer's profit margin. In the International Rice Research Institute's overall program for improving technology to help farmers increase world rice production, the development of pest-resistant rices has been an important objective.

Great progress has been made in the development of resistant rice varieties, but more needs to be done. For many important insects, resistant varieties are not yet commercially available. Furthermore, the insect resistance breeding program has been primarily restricted to irrigated rice. We urgently need to develop varieties resistant to other insect pests, especially to those that attack upland and deepwater rice. Because yield increases in these areas will be less dramatic than those in irrigated areas, the economic advantage of insecticide use is limited; the value of varietal resistance is therefore even greater here.

Since 1962, when Dr. M. D. Pathak established the IRRI insect resistance screening program, the Institute, working with various national rice research programs, has developed procedures for rearing insects, screening rice varieties for insect resistance, and studying the nature of resistance. These procedures are described in this book. In addition, a list of varieties resistant to each insect is given. Most of the information on the sources of resistance has not been previously published.

In preparing this publication, the authors have drawn on their experience in IRRI's Genetic Evaluation and Utilization Program and on their communication with rice scientists in national and international research programs. Dr. E. A. Heinrichs, entomologist and head of the Department of Entomology, is responsible for the insect resistance screening program. F. G. Medrano, assistant scientist, supervises the brown planthopper resistance studies, and H. R. Rapusas, senior research assistant, conducts studies on the nature of resistance to leafhoppers and planthoppers.

R. C. Cabrera, assistant manager, and F. E. Manto, graphics supervisor, did the design. M. P. Dayrit and M. C. Quilloy, senior graphics assistants; P. O. Mamon and C. C. Amutan, graphics assistants; and O. M. Figuracion, Jr., senior illustrator, did the layout and artwork. W. G. Rockwood, editor, assisted by E. P. Cervantes, editorial assistant, edited this publication. G. S. Argosino, assistant editor, helped in the later stages of the book.

M. S. Swaminathan
Director-General
International Rice Research Institute

Chapter 1
INTRODUCTION

Insects are a serious constraint to rice production, and the highest losses due to insects occur in the tropics. In Asia alone, yield loss due to insects has been estimated at about 25% (Cramer 1967).

Insects not only damage the plant by feeding on its tissue, some are also vectors of devastating rice viruses. All portions of the plant — from root to panicle — are attacked by various insects. All growth stages of the rice plant — from the seedling in the nursery to the mature plant — are vulnerable. Even after harvest, the grain which is stored is attacked. Of about 1,000 insect species known to attack rice, about 30 cause sufficient damage to require control.

Control tactics used to minimize losses to insect pests are varietal resistance, insecticides, biological control agents, and cultural controls. In the integrated approach to the management of rice insect pests, varietal resistance is the major tactic. Another important tactic is the limited use of insecticides; fields are monitored and insecticides with low toxicity to the natural enemies are applied only when threshold levels are reached.

The incorporation of insect resistance into modern varieties is a major objective of most rice breeding programs in the tropics. Insect-resistant rice varieties are important for several reasons:

- Income per hectare in rice production is relatively low; any amount spent for such controls as insecticides greatly lowers the farmer's profit. Varietal resistance is essentially free to the farmer.
- The cost of insecticides is increasing faster than the price of rice in most countries.
- Landholdings of most rice farmers are small. Maintaining insecticide application equipment is not economical on many small farms.
- Farmers often do not know how to apply insecticides according to economic threshold populations: prophylactic applications are wasted.
- Insecticides may cause poisoning accidents.
- Insecticides contaminate the environment and kill fish and livestock.
- Varietal resistance is compatible with other control tactics — chemical control, biological control, and cultural control.
- Some predators are more effective against insect pests which are on resistant varieties (Kartohardjono and Heinrichs 1984).
- Resistant varieties increase the susceptibility of insect pests to insecticides (Heinrichs et al 1984).

THE ROLE OF VARIETAL RESISTANCE IN AN INTEGRATED PEST MANAGEMENT PROGRAM

With the spread of high-yielding varieties and the change in cultural practices which accompanied the green revolution, there was a shift away

from traditional cultural controls. Traditional varieties which had some tolerance for pests have in some cases been replaced by higher yielding but more susceptible varieties. Insecticides have replaced the traditional pest controls and have in many cases increased yields. Yield stability, however, has not been achieved with the repeated use of insecticides alone. Because of their cost to the farmer, the selection of insecticide-resistant strains, and the development of secondary pests into major pests when, through excessive insecticide use, their natural enemies are destroyed, insecticides will never be the farmer's panacea. Ironically, there has been greater dependence on insecticides because they destroy natural enemies. More poisoning accidents among farmers have occurred and environmental contamination has increased.

Because control programs based on one method are short-lived, scientists are developing insect pest management systems that integrate the various tactics. The integration of chemical, biological, and cultural control, and varietal resistance is designed to maintain populations at subeconomic levels with minimal cost to the farmer. Because of the many advantages of varietal resistance it should be a component of all rice insect management systems. The use of varietal resistance may be the principal control method or an adjunct to other control measures (Painter 1951).

Varieties having even only moderate levels of resistance fit well into an integrated control program. A common form of such control is to apply insecticides only when the insect population on moderately resistant varieties reaches the economic threshold. Insecticides applied to a moderately resistant variety are more effective because the insect is more susceptible on a resistant plant (Heinrichs et al 1984). Moderately resistant varieties will suppress the insect population and decrease the number of insecticide applications needed.

Resistant varieties are compatible with biological control agents (predators, parasites, and pathogens) on which they have no direct adverse effect. Moderately resistant varieties allow a subeconomic pest population to remain on the plants and serve as hosts for natural enemies. The reduction in the brown planthopper (BPH) population on resistant and moderately resistant varieties improves the natural enemy-to-pest ratio in favor of biological control (Kartohardjono and Heinrichs 1984). In addition, spiders as predators are more efficient when preying on BPH on a moderately resistant variety than on a susceptible variety. This increased predator efficiency could be due to the restless behavior of the pest. Restlessness increases the pest's exposure to natural enemies (Pathak 1975) and reduces its vigor.

Insect resistance and short duration can be incorporated into a variety. And these two qualities together exert greater control than either alone. Very early maturing (VEM) rice lines which have yields equal to those of intermediate-maturing varieties can escape the third generation of BPH, usually the one causing hopperburn.

Types of insect resistance

Insect resistance comprises the heritable qualities of a plant that enable it to reduce the degree of insect damage it suffers (Painter 1951). Cultivars differ in their degree of resistance: there may be a gradation from extreme resistance to extreme susceptibility (Russell 1978). Resistance is classified as low, moderate or intermediate, and high.

Resistance at different rice growth stages has different names. Seedling resistance is detected and measured in the greenhouse seedbox screening of seedlings. It may persist as the plants mature. Adult or mature plant resistance is manifested in older plants whose seedling stage was susceptible.

There are types of apparent resistance, not heritable, which should not be confused with true resistance. Painter (1951) used "pseudoresistance" to describe resistance due to transitory characters in potentially susceptible plants. The types he listed are:

- *Host evasion.* The host plant passes through the susceptible stage quickly or when insect populations are low. A crop which is in the field when insect populations are low or matures before the insect population reaches damaging levels shows host evasion.
- *Escape.* A particular host plant is neither infested nor injured despite the local presence of the pest insect.
- *Induced resistance.* Some environmental conditions, such as soil fertility, temporarily increase the level of resistance.

Mechanisms of resistance have been grouped into three categories by Painter (1951). Nonpreference and antibiosis refer to the response of the insect to the plant; tolerance refers to the response of the plant to the insect.

- *Nonpreference.* Nonpreferred plants lack the characteristics of hosts for insect feeding, oviposition, or shelter. Because nonpreference pertains to the insect rather than the plant host and is not parallel to antibiosis, Kogan and Ortman (1978) proposed the term *antixenosis* for nonpreference. Antixenosis (against the guest) means the plant is considered a bad host.
- *Antibiosis.* Antibiosis is an adverse effect on the biology (survival, development, or reproduction) of the insect.
- *Tolerance.* A tolerant plant can produce good yield even while it supports an insect population that would severely damage and decrease the yield of a nontolerant plant.

Much genetic variability occurs within rice insect populations. When insect-resistant varieties are grown, forms of the insect which can attack resistant varieties may be naturally selected. Although the term is controversial, biotype is generally used to describe populations within a species which can survive on and destroy varieties that have genes for resistance. The stability of insect resistance in rice varieties depends on the occurrence of these biotypes. An objective of breeding programs is to incorporate stable resistance into high-yielding varieties. Varieties usually have one of two types of insect resistance:

- horizontal (or nonspecific) resistance, effective against all known biotypes of an insect, or
- vertical (or specific) resistance, effective against certain biotypes but not to others (Fig. 1).

Horizontal resistance is quantitative: the degree of resistance depends on the number of minor genes, each of which contributes a small effect. Vertical resistance is qualitative in that the frequency distribution of resistant and susceptible plants in a population is discontinuous.

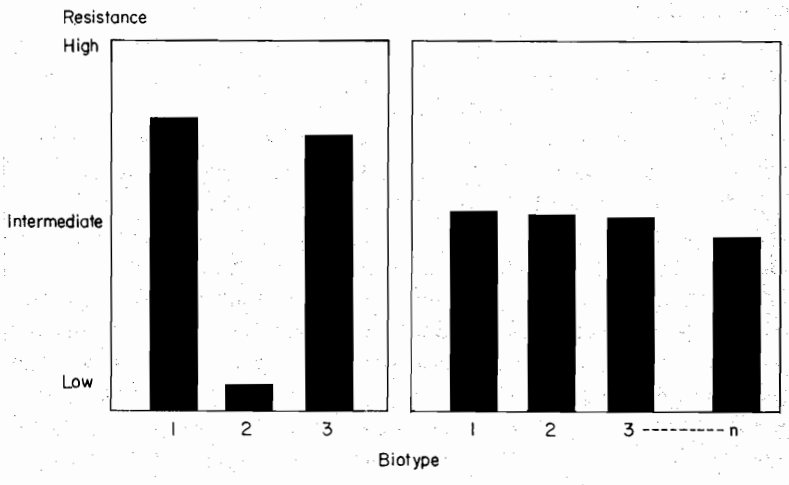
Resistant and susceptible plants are easy to classify because they fall into distinct classes. In horizontal resistance there is a continuous gradation between resistant and susceptible plants: the difference between resistance and susceptibility is not distinct. Vertical resistance is under the control of one (monogenic) or a few (oligogenic) major genes. Major genes have a strong effect and are easily identified.

Breeding for insect resistance is most rapid when single vertical resistance genes are used. Because of the biotype selection problem in monogenic resistant varieties, two breeding strategies are followed.

- *Sequential release.* A variety with a new major gene for resistance is released when, because of selection for a new biotype, a variety with another major gene becomes susceptible.
- *Gene pyramiding.* Two or more major genes for vertical resistance are incorporated into a variety to provide resistance to more biotypes and increase the resistance stability.

Additional breeding strategies (involving minor and major genes) to prevent or slow biotype selection are being considered:

- Increasing the number of minor genes to increase the level of horizontal resistance and its stability.
- Combining minor genes with major genes.



1. Comparison of a variety having vertical resistance (left) with a variety having horizontal resistance (right) to biotypes of a given insect species.

- Rotating major genes by seasonally alternating varietal groups having different major genes. This should reduce selective pressure of the resistant varieties on the insect population.
- Breeding tolerant varieties that exert no selection pressure on the insect population. Tolerance by itself not only is of value in decreasing yield loss caused by insects, but can also be combined with major genes which decrease the insect population and provide a high level of resistance. When the insect population increases on the resistant variety, monitoring will detect a shift to the new virulent biotype. A new variety with another major gene(s) can then be released. Before the new variety is released (and after the old major gene has become ineffective), the tolerance in the old variety will prevent drastic yield losses.

Current status of breeding for insect resistance

Breeding for insect resistance in rice has been a focus of rice research programs only during the last two decades. Before 1962, scientists believed there was not sufficient insect resistance in rice to be of practical value to farmers (Jennings et al 1979). In the early screening studies at IRRI, germplasm that had stem borer resistance and high leafhopper and planthopper resistance was identified. This discovery of high resistance elicited much interest, and insect resistance became an important part of many rice breeding programs. Screening methods for more than 30 rice insects throughout the world have been developed, and sources of resistance to most, identified (Table 1). Much of the IRRI germplasm collection has been screened against the BPH and green leafhopper (GLH) (Table 2). Of the 47,944 varieties screened against the GLH, a large number — 1,196 — are resistant.

Over the past two decades, several countries have established programs on varietal resistance to insects. Major emphasis has been on hoppers, stem borers, and the gall midge. All the major rice-producing countries in South and Southeast Asia have breeding programs for BPH and GLH resistance. BPH-resistant varieties occupy about 25% of the irrigated lowland rice area in Southeast Asia. In China, hybrid rice grown on about 5 million hectares has BPH resistance. Strong breeding programs for gall midge resistance in India, Sri Lanka, and Thailand have yielded gall midge-resistant varieties. Five have been released by the Indian Program (Heinrichs and Pathak 1981). Many varieties with low to moderate levels of stem borer resistance have been released in Asia.

In addition to the stem borer, many rice insects remain for which the levels of resistance are too low for breeding purposes. High levels of resistance have been found only for leafhoppers and planthoppers and the gall midge. Only a small portion of the world germplasm collection has been screened against most rice insect pests. Screening of the entire germplasm collection may identify varieties with higher levels of resistance to some of these insects. When resistance to a particular insect species is low, special breeding techniques must be used to increase the levels of resistance.

Table 1. Status of screening and breeding for varietal resistance to rice insect pests.

Insect		Status				
Common name	Scientific name	Screening methods developed	Resistance sources identified	Resistant breeding lines available	Resistant varieties released	Genes for resistance identified
Brown planthopper	*Nilaparvata lugens*	+	+	+	+	+
Whitebacked planthopper	*Sogatella furcifera*	+	+	+	–	+
Smaller brown planthopper	*Laodelphax striatellus*	+	+	+	+	–
Rice delphacid	*Sogatodes orizicola*	+	+	+	+	–
Green leafhopper	*Nephotettix virescens*	+	+	+	+	+
Zigzag leafhopper	*Recilia dorsalis*	+	+	+	–	+
White leafhopper	*Cofana spectra*	+	+	–	–	–
Blue leafhopper	*Empoascanara maculifrons*	+	+	–	–	–
Striped stem borer	*Chilo suppressalis*	+	+	+	+	–
Yellow stem borer	*Scirpophaga incertulas*	+	+	+	+	–
Small moth borer	*Diatraea saccharalis*	+	+	+	–	–
African striped stem borer	*Chilo zacconius*	+	+	–	–	–
African white stem borer	*Maliarpha separatella*	+	+	–	–	–
Lesser cornstalk borer	*Elasmopalpus lignosellus*	+	+	–	–	–
African pink stem borer	*Sesamia calamistis*	+	+	–	–	–
S. A. white stem borer	*Rupela albinella*	+	+	–	–	–
Stalk-eyed fly	*Diopsis macrophthalma*	+	+	+	–	–
Rice stem maggot	*Atherigona oryzae*	+	+	–	–	–
Whorl maggot	*Hydrellia philippina*	+	+	+	+	–
Gall midge	*Orseolia oryzae*	+	+	+	+	+
Rice seedling fly	*Atherigona exigua*	+	+	–	–	–
Armyworm	*Mythimna separata*	+	–	–	–	–
Thrips	*Stenchaetothrips biformis*	+	+	–	–	–
Rice bug	*Leptocorisa oratorius*	+	+	–	–	–
Caseworm	*Nymphula depunctalis*	+	+	–	–	–
Leaffolder	*Cnaphalocrocis medinalis*	+	+	+	–	–
Rice water weevil	*Lissorhoptrus oryzophilus*	+	+	–	–	–
Hispa	*Dicladispa armigera*	+	+	–	–	–
Bloodworm	*Chironomus tepperi*	+	+	–	–	–
Rice weevil	*Sitophilus oryzae*	+	+	–	–	–
Maize weevil	*Sitophilus zeamais*	+	+	–	–	–
Lesser grain borer	*Rhyzopertha dominica*	+	+	–	–	–
Angoumois grain moth	*Sitotroga cerealella*	+	+	–	–	–

Table 2. Status of breeding for varietal resistance to insects at IRRI, January 1983.

Insect	Year screening started	Accessions tested	Resistant accessions		Resistant breeding lines	Resistant varieties released
			No.	%		
Brown planthopper Biotype 1	1967	30,790	281	0.91	+	+
2	1975	8,922	126	1.41	+	+
3	1975	10,711	148	1.38	+	+
Whitebacked planthopper	1970	46,488	391	0.84	+	–
Green leafhopper	1967	47,944	1,196	2.50	+	+
Zigzag leafhopper	1973	2,370	36	1.52	+	–
Yellow stem borer	1962	19,961	25[a]	0.13	+	+
Striped stem borer	1962	ca 15,000	23	0.15	+	+
Whorl maggot	1965	15,787	1[a]	0.01	+	+
Leaffolder	1979	17,914	35	0.20	+	–
Caseworm	1982	4,768	–[b]	–	–	–
Thrips	1983	79	11	13.92	–	–
Rice bug	1983	–[c]	–	–	–	–

[a]Moderately resistant. [b]Entries were selected in varietal testing, and retesting is needed to confirm the resistance. [c]Initial screening is still in progress.

Resistance may exist in the wild rices *Oryza* spp; resistance to grassy stunt virus was found in the wild rice *O. nivara.* The IRRI program has evaluated more than 300 of the 1,100 accessions in the IRRI germplasm collection. Many of these are resistant to the hoppers (Table 3). Screening of the wild rice collection has identified varieties with high yellow stem borer and whorl maggot resistance not found in *O. sativa.* However, the genomes of many of the wild rice species differ from those of *O. sativa* and it is difficult to incorporate the resistance from wild rice into *O. sativa.* Tissue culture and genetic engineering may make it possible.

INTERDISCIPLINARY APPROACH TO DEVELOPING INSECT-RESISTANT RICE VARIETIES

The most successful insect resistance breeding programs have been those in which scientists of several key disciplines worked as a team. This is because the degree to which a new variety is a success depends on factors involving many disciplines. Thus, entomologists, plant pathologists, physiologists, cereal chemists, agronomists, and soil scientists must work with the plant breeder. At the beginning, a plant breeder and an entomologist may be sufficient. The initial step is for the entomologist to identify sources of resistance and then provide these sources to the breeder for use in the hybridization program. As the program develops, scientists from the other disciplines should become involved. The objective of the team must be to develop a high-yielding variety with acceptable grain quality and resistance to biological and physical stresses. The role of the entomologist is to

- develop efficient screening methods,
- identify resistant donors,
- screen breeding lines developed by the breeder,
- determine the nature and causes of resistance,

Table 3. Screening for insect resistance of the IRRI wild rice germplasm collection, 1983.

Insect	Accessions screened	Resistant accessions[a]	
		No.	%
Brown planthopper			
Biotype 1	248	119	48.0
2	248	97	39.1
3	248	98	39.5
Whitebacked planthopper	228	86	37.7
Green leafhopper	228	112	49.1
Zigzag leafhopper	208	115	55.2
Striped stem borer	167	7	4.2
Yellow stem borer	181	24	13.3
Leaffolder	264	8	3.03
Whorl maggot	328	7	2.13
Caseworm	204	0	0.0

[a]Damage ratings of 0-3.

- select for biotypes to be used in the screening program,
- determine the rate of biotype selection, and
- collaborate with the breeder in identifying genes for insect resistance.

SETTING PRIORITIES FOR THE INSECT RESISTANCE BREEDING PROGRAM

Several criteria must be considered when deciding which insect pests are to be included and what resources are to be spent on each. Manpower, facilities, and financing must be allotted.

- The extent of economic losses caused by the various insects should be known. The potential of the insects to damage plants and to cause yield losses, their distribution within the geographic area concerned, and the periodicity of outbreaks should be considered. Insects which may cause only moderate yield losses but are widely distributed and have regular outbreaks may cause more financial loss than an insect which has a higher damage potential but seldom reaches economic proportions, or occurs only in isolated areas. Furthermore, an insect which causes direct damage by feeding and, in addition, transmits a virus would have higher priority than one which causes only feeding damage.
- Screening should be undertaken against insects for which there is a good possibility of success in selecting resistance sources and transferring the resistance into good agronomic types. This selection requires familiarity with the insects' behavior. If resistance to a given species exists in one location, it is likely to exist in another. Resistance to one insect species may mean resistance to another. Screening varieties which have resistance to several insect species may identify some with resistance to other related species. High resistance to stem borers or other insects with chewing mouthparts is unlikely, but low levels of resistance can be combined with other methods such as biological, chemical, and cultural controls.
- The availability of low-cost alternative controls must be considered. Populations of some insects can be maintained at subeconomic levels by biocontrol agents, cultural controls, or minimal insecticide application.
- Researchers must study the facilities, land, and manpower required. For some insects (such as hoppers) greenhouse screening is the best method. Field screening can be done but often fails because of low populations or a species mix. Zigzag leafhopper screening has not been successful in the field because of low populations. Screening for yellow stem borer requires a screenhouse (and adequate manpower for manual plant infestation).
- The availability of insects for screening is important. If greenhouse screening is conducted, an efficient rearing program is required. If field screening is done, a high natural population is necessary. The accessibility of locations with high natural populations (hot spots) must be considered. Screening far from the research station is expensive. If

someone cannot continuously be at the site during screening, plot maintenance (weed and rodent control, irrigation, etc) may be poor and will adversely affect the screening program.

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Chapter 2
GENERAL PROCEDURES FOR THE SCREENING PROGRAM

The movement of seeds in the IRRI insect resistance breeding program is illustrated in Figure 1. Seeds to be screened are obtained from the IRRI germplasm collection (about 60,000 accessions) or directly from national programs. Seed of genetically improved cultivars is obtained from national and IRRI breeding programs. In the initial screening, entries are non-replicated. In retesting, entries are replicated from 3 to 10 times depending on the insect species. If reactions are still not distinct after retesting, a second retest is conducted. Resistant entries are then given to the breeder for use in hybridization.

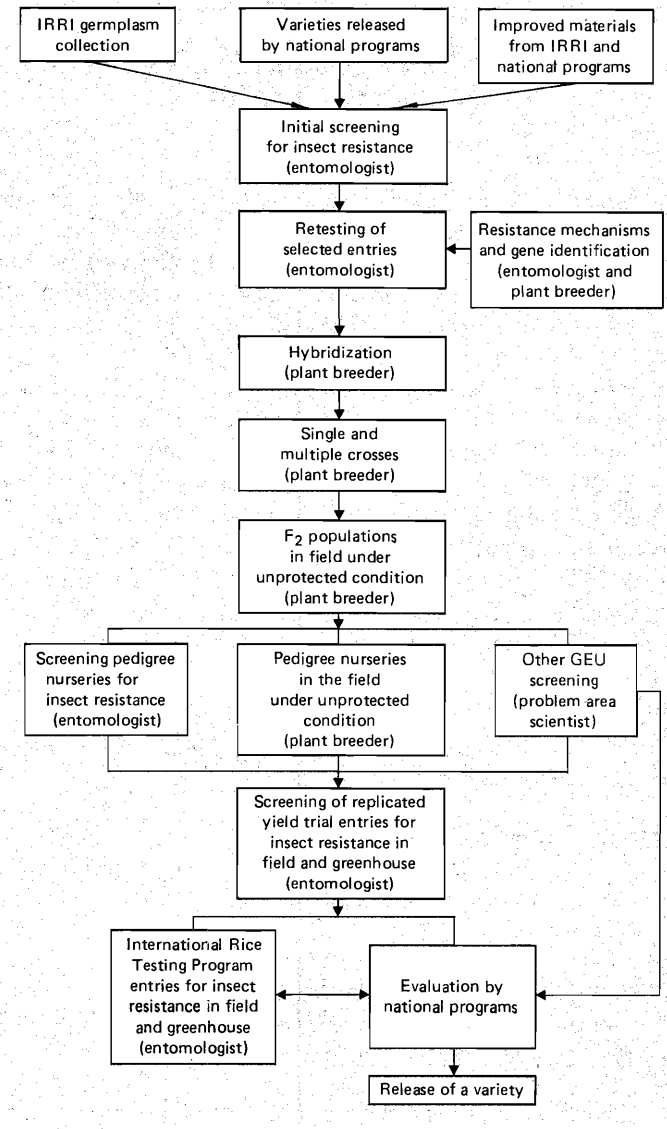
Next, single and multiple crosses are made and the F_2 populations are grown without insecticide in the field. The F_3 to F_7 are screened for insect resistance and evaluated by other scientists in the Genetic Evaluation and Utilization (GEU) Program. Entries in the replicated yield trial are screened in either the field or the greenhouse depending on the insect. The most promising entries from IRRI and national programs are included in the International Rice Testing Program (IRTP) insect nurseries and evaluated in the field or greenhouse. Where possible, a field test is preferred. Insect-resistant breeding lines are then named as varieties and released by the national programs.

Development of screening methods

Requirements for an insect resistance screening program are

- an adequate amount of test seed,
- test insects, and
- a method to evaluate the levels of resistance among test entries. To establish a screening method, one needs to understand the biology of the insect and its potential plant damage. Development of a method for one species may be simple but for another, difficult.

Developing an efficient rating method is extremely important. Ratings used in rice insect resistance screening programs are based mostly on degree of plant damage, but in some cases, insect numbers are also used. To compare entries a numerical rating system should be used. The Standard Evaluation System for Rice (SES) (IRRI 1980) was devised for this purpose and includes most of the major rice insects. All rating scales are on a 0-9 basis (0 = no damage, 9 = severe damage). Although rarely available, an understanding of the relationship between a given damage level and economic loss is useful. Resistance ratings of 0-3 include no economic losses



1. Movement of seeds in the insect resistance breeding program at IRRI.

and susceptibility ratings of 7-9, severe economic losses. Although current rating scales are not often based on degree of economic loss, the following information may be useful as a guide for improving those scales and for developing new rating scales:

Damage rating scale	Level of resistance	Economic losses
0	Immune	None
1	Highly resistant	None
3	Resistant	None
5	Moderately resistant	Moderate
7	Moderately susceptible	Severe
9	Susceptible	Severe

The screening method selected should give distinctly different reactions between susceptible and resistant entries. When these reactions are distinct, moderate resistance can also be detected. Plant reaction and subsequent damage rating depend on number of insects per plant, plant vigor, plant age, and environmental factors such as temperature. The insect population selected should also give distinctly different reactions for the susceptible and resistant entries. When insect populations are too high, all entries may appear susceptible, when too low, all entries may appear resistant. Plants that lack vigor because of soil deficiencies or plants that are extremely young may also be rated as susceptible although under normal conditions they would be resistant.

Sources of seeds for screening

Genetic diversity in rice exceeds that in most other crops. Having been cultivated for thousands of years, rice strains adapted to the many different agroclimatic regions of the world have been selected by farmers. It is estimated that there are about 100,000 rice strains in the world. The great diversity among these rices includes genetic resistance to many rice insects.

To identify insect resistance, it is important to have many genetically diverse rice strains available. Seed can be obtained from the various germplasm collections, which contain the traditional centuries-tested varieties.

National rice germplasm collections are maintained in many countries (Appendix 1). The size of the various collections may vary from a few hundred to several thousand accessions. IRRI maintains the largest collection of world rices, and currently has about 60,000 accessions of *O. sativa,* 2,614 strains of *O. glaberrima,* and 1,100 wild rices. Through collaboration with national programs, the IRRI collection should reach 100,000 accessions. About 14,000 of the IRRI accessions are currently maintained at Fort Collins, Colorado, USA. CIAT and IITA also maintain limited accessions. Country of origin, morphological and agronomic characteristics, grain quality, and known reactions to various physical and biological stresses of the IRRI accessions are computerized.

Selection of seeds to begin the screening program

The germplasm collections consist primarily of traditional varieties, a few

modern varieties, and possibly some wild rices (*Oryza* spp.). When beginning a screening program for a particular insect, it is best to first evaluate advanced breeding lines and released varieties developed by your research station. Although the chances of finding resistance in the breeding lines and varieties is limited, these plants make good donors if resistance is found. Because of the limited number of released varieties and breeding lines available, screening of this material will not take long. Seed of released varieties and of resistant lines bred at other stations should then be obtained and tested against the local population of the particular insect. This is often done for hoppers, as many hopper-resistant varieties have already been developed all over Asia. The IRTP nurseries can provide seed of these resistant cultivars.

Next, obtain seeds of traditional varieties from the germplasm collection at your research station. If unavailable there, write to other national programs or to IRRI. Breeding codes used by various programs are given in Appendix 2. Several approaches can then be followed:

- Systematically screen the entire germplasm collection.
- Select varieties from countries where the specific insect is a pest and resistant sources have probably been naturally selected.
- Select varieties resistant to the same or closely related insect in another country from a collection in that country.

India and Sri Lanka are rich sources of germplasm with resistance to BPH and to several other insects, maybe because of the area's natural selection pressure. Resistance can, however, be found where the pest does not occur. This has been the case with resistance to GLH *Nephotettix virescens* in African rices.

Where insect resistance cannot be found in *O. sativa,* wild rices should be screened. Wild rice is useful as a source of disease resistance: the only known resistance to grassy stunt virus 1 was obtained from the wild rice *O. nivara*. The IRRI germplasm collection contains about 1,100 wild rices of several species (Table 1). Species having the same genome and chromosome number as *O. sativa* ($2n = 24$) are easy to cross with it. Without the use of special techniques, species with a genome different from that of *O. sativa* have limited use in breeding programs.

Multiplication of seeds for screening

About 5 g of seed is required for screening against one insect species. If the seed is insufficient it must first be multiplied, a process during which special care must always be taken. (Small amounts of seed can easily be lost.)

Cleaning of seeds and breaking dormancy. Once removed from cold storage, seeds for screening should be quickly sown, especially in the hot, humid tropics. Remove extraneous matter such as weed seeds and broken seeds. To break dormancy, place the clean seeds in coin envelopes in an oven for 3 days at 45° C, and for 5 days (7 for wild rice) at 50° C. The lemma and palea of wild rices should be removed by hand if germination is poor.

Table 1. Chromosome numbers, genome symbols, and geographical distribution of *Oryza* species in the IRRI germplasm collection (adapted from Chang 1976a, b).

Species name, synonym	(x = 12) 2n =	Genome group	Distribution
O. alta	48	CCDD	Central and South America
O. australiensis	24	EE	Australia
O. barthii (O. breviligulata)	24	A^gA^g	West Africa
O. brachyantha	24	FF	West and central Africa
O. eichingeri	24,48	CC, BBCC	East and central Africa
O. glaberrima	24	A^gA^g	West Africa
O. grandiglumis	48	CCDD	South America
O. granulata	24	–	South and Southeast Asia
O. latifolia	48	CCDD	Central and South America
O. longiglumis	48	–	New Guinea (Irian Jaya)
O. longistaminata (O. barthii)	24	A^1A^1	Africa
O. meyeriana	24	–	Southeast Asia, southern China
O. minuta	48	BBCC	Southeast Asia, New Guinea
O. nivara (O. fatua, O. rufipogon)	24	AA	South and Southeast Asia, southern China, Australia
O. officinalis	24	CC	South and Southeast Asia, southern China, New Guinea
O. punctata	48, 24	BBCC, BB (?)	Africa
O. ridleyi	48	–	Southeast Asia
O. rufipogon (O. perennis, O. fatua, O. perennis subsp. *balunga)*	24	AA	South and Southeast Asia, southern China, New Guinea
(O. perennis subsp. *cubensis)*		$A^{cu}A^{cu}$	South America
O. sativa	24	AA	Asia
O. schlechteri	–	–	New Guinea (Papua New Guinea)

Germination and sowing. When more than 10 g of seed are available, the seed can be sown directly in seedboxes; seedlings can then be transplanted in soil beds in a screenhouse or in the field. When only 10 g or less of seed are available, extra care is needed. In this case, germinate the seeds by placing them in a petri dish containing filter paper moistened with an antifungal solution. Keep the paper moist and the petri dish in a room at 30° C. Incubate the seeds for 24 h or until hypocotyls are exserted. Then transplant in moist soil in seedboxes. Label each accession. Keep the seedboxes in full sunlight in a greenhouse. When seedlings are 25 days old, transplant them by twos in fertile, puddled soil in clay pots. Label the pots and, to avoid sun damage, leave them in the shade for 2 days.

Care of plants. For maximum seed production, the plants must have good growing conditions and must be protected from pests and adverse weather. Potted plants should be kept in a screenhouse to protect them from storms, insects, and rats. Most accessions will be susceptible to rice viruses and should be protected from the leafhopper and planthopper vectors. If hoppers can enter the screenhouse, a systemic insecticide should be

2. Panicles of wild rices are enclosed in a glassine bag to catch dropping grains. The bag is attached to a wire. The plant is held erect with a bamboo stake.

incorporated into the soil before transplanting, or the plants should be caged in fine nylon or fiberglass screen.

To prevent lodging, tall plants, ie, some of the wild rices, should be attached to bamboo stakes. Seeds of wild rices drop from the panicle as soon as they mature; the panicle should therefore be covered with a glassine bag on a wire a few days after flowering (Fig. 2).

Rats, insects, insect-vectored viruses, and typhoons are major threats in the field. Rat control through baiting with a chronic anticoagulant rodenticide should start 2 weeks after transplanting and continue until 2 weeks before harvest or until bait consumption stops. Small plots can also be protected by installing a rat fence (Fig. 3). To control insect-vectored viruses incorporate a systemic insecticide in the soil at the last harrowing and make foliar applications beginning at 30 days after transplanting.

Seed storage

Adequate short-term seed storage facilities are essential to an efficient varietal resistance program. Seed must often be held several weeks or longer before it can be screened. Even after it is screened, resistant seed is held for possible retesting and studies on resistance mechanisms. When screening breeding lines for more than one insect, it may not be possible to screen against all the insects simultaneously. Seeds are then kept in the storage room until the lines can be screened. Storage facilities must be temperature-controlled and protected from rats and storage insects. At IRRI the Entomology Department maintains seed in a room with a window air conditioner which maintains the temperature at 20° C and about 60-75% RH. Seeds are kept in coin envelopes or paper bags inside glass jars or plastic

3. Seed production plot is protected with a rat fence.

boxes where they are protected from rats and storage insects and in which humidity can be regulated (Fig. 4). Silica gel can be added to the containers to reduce moisture.

Selecting a screening site

Screening can be conducted in a greenhouse, a screenhouse, or the field, depending on available facilities and the insect species. Greenhouse screening is generally the most productive and greenhouse screening methods are available for most insects. With an efficient insect-rearing program, greenhouse screening can be conducted throughout the year. Screening against stem borers, however, is conducted in a screenhouse or in the field. Problems encountered in field screening are:

- insufficient insects,
- the presence of insect species other than the target pest, and
- the destruction of test plants by disease, rats, and adverse weather. Field screening is the best method to screen for whorl maggot *Hydrellia philippina* in the Philippines. Populations are sufficiently high for valid screening throughout the year and whorl maggot is usually the only pest of any consequence up to 35 DT when observations are made. Rat

damage and later damage by other pests do not interfere with the screening program. On the other hand, field screening for stem borer is difficult. To ensure high stem borer populations, screening is conducted when surrounding farms have no rice crop. Because of the island effect, however, the plots, especially in the later stages near flowering, are subject to invasion by rats.

Where greenhouse screening of breeding lines is conducted, field screening should be done before a breeding line is released as a variety. Where field insect populations are insufficient, methods of increasing them have been developed.

4. Rice seeds are protected from rats and storage insects in a plastic box or glass jar placed on shelves in an air-conditioned room.

Sources of insects for screening

For greenhouse and screenhouse screening, insects are usually reared and plants artificially infested. Some difficult-to-rear insects can be collected in the field or at lights at night. These insects themselves, or their progeny, can serve for screening studies. Moths of the yellow stem borer are collected live in light traps in the field and at lights around buildings. When placed in an oviposition cage containing rice plants, the moths lay egg masses on the plants. The emerging larvae are used to infest entries in the screenhouse. Techniques for obtaining sufficient insects for field screening follow:

- *Select a hot spot and proper planting date.* Certain locations have an abundance of a particular insect in certain seasons. The field screening should be conducted when the pest is abundant at such locations. If there is no experiment station at such locations, arrangements are made to use farmers' land. What was a hot spot one year may not be one the next, so several years may be required to obtain reliable field results.
- *Plant border rows of a susceptible variety.* A major portion of the screening plot can be planted to a highly susceptible variety: this will build up the population. To increase the chance of insect buildup, susceptible plants are infested with artificially reared or field-collected insects.
- *Follow cultural practices which favor pest buildup.* Spacing affects certain insects. Wide spacing causes higher whorl maggot damage while narrow spacing results in BPH buildup. High N rates favor hoppers and leaffolders. High fertilizer rates generally favor pests. Knowledge of ecological conditions affecting the target insect is necessary.

  Most rice insects are sensitive to water management. Water in the field is necessary for adequate whorl maggot and caseworm populations; it also encourages BPH populations.
- *Apply resurgence-inducing insecticides.* Insecticides can be a powerful tool in the manipulation of field insect populations. Application of resurgence-inducing insecticides to susceptible border rows increases BPH and WBPH populations. This method is used at IRRI for the field screening of BPH. Application of the right insecticide at the right time can also increase leaffolder populations.

- *Lights.* Lights are commonly used to attract gall midge adults to fields for screening. If other, unwanted insects are abundant these may also be attracted and may cause damage. For example, lights used to attract yellow stem borer moths at IRRI also attract BPH, which results in hopperburn in the areas under the lights.
- *Ratooning.* Ratooning provides a second chance if the crop escapes infestation because insect populations are low. It is a screening technique against insects which attack only at the tillering stage. Gall midge develops only in growing plants. If the gall midge does not appear in sufficient numbers during the tillering stage, plants can be cut and the ratooned plants will still be susceptible should the gall midge population increase.

Sowing seed and maintaining plants

In greenhouse screening, seeds are sown in seedboxes or in pots. Where 1- to 2-wk-old seedlings will be used for screening, eg, against hoppers, seeds are sown in seedboxes. To prevent rat or bird damage, the seedboxes are placed in a screen cage. They are watered only with a fine spray. Before the seedlings are infested with insects, the seedboxes are placed in trays containing water. When older plants are screened in the greenhouse, eg, against leaffolder, seeds are sown either directly in pots and thinned to a specific number/pot or in seedboxes and later transplanted in pots.

For field screening tests, seedlings are usually grown by the wet seedbed method in a field nursery. Make 1-m-wide raised seedbeds with a smooth, firm surface. Allow the beds to settle for 2 days. Then, using a wooden template, make rows at 10-cm intervals in the muddy surface (Fig. 5). Sow seed in the rows and cover with fine soil. Label at 10-row intervals (Fig. 6).

5. Rows for seed sowing are made with a wooden template on the muddy surface of a seedbed.

6. Seeds are sown in a seedbed. To identify the entry, a labeled stake is placed every 10th row.

7. Seedlings are removed from a flooded seedbed. Bundles of each entry are held together by a wire labeled with the accession number.

8. Labels on bamboo stakes indicate the entry in each row. Excess seedlings are held at the base of the stakes and used to replace missing hills.

Irrigate by flooding the depressions between seedbeds and splashing water on the seedbeds. Check the seedbeds daily. Protect from rats and birds until several days after seedling emergence. If susceptible to viruses, seedlings should be protected from insect vectors, either with netting or insecticides.

Before transplanting (21 days after sowing) prepare wood labels (with variety name or code number) for each row or group of rows comprising one entry. Attach a 20-cm-long wire to each label. Before pulling the seedlings, place the labeled tags at the end of the appropriate row. Flood the seedbed (to soften the mud), remove the seedlings, and tie the bundle of each entry with the corresponding label (Fig. 7). Lay out the field (already fertilized and prepared for planting) with stakes of bamboo or other material to mark each entry (Fig. 8). Place the seedling bundles at the base of the stakes and transfer each label to the top of the stake (Fig. 8). If wood labels are not available, paper labels numbered with India ink and dipped in wax may be used (Fig. 9). Using a guide wire, transplant the seedlings.

Management of field plots

Normal plant growth is important in screening for insect resistance. Plants that are not growing properly because of such stress as weed competition,

9. Paper labels marked with India ink and dipped in wax identify each entry.

drought, or low fertility will be abnormally susceptible to feeding by insects. If stress is not evenly distributed, insect damage ratings may not be reliable.

Weeds and rats are major pests. Application of herbicide against grassy weeds must be followed by hand weeding. When tests are conducted between crop seasons, the island effect results in rat immigration into test plots, so rat control is essential.

To maintain normal plant growth, fertilizer should be applied in split doses. The type and rate of fertilizer depend on the soil test, but nitrogen sufficient for a dark green plant color and vigorous growth should be applied.

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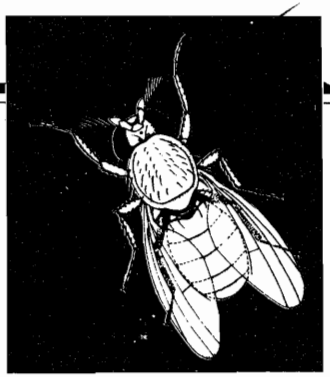


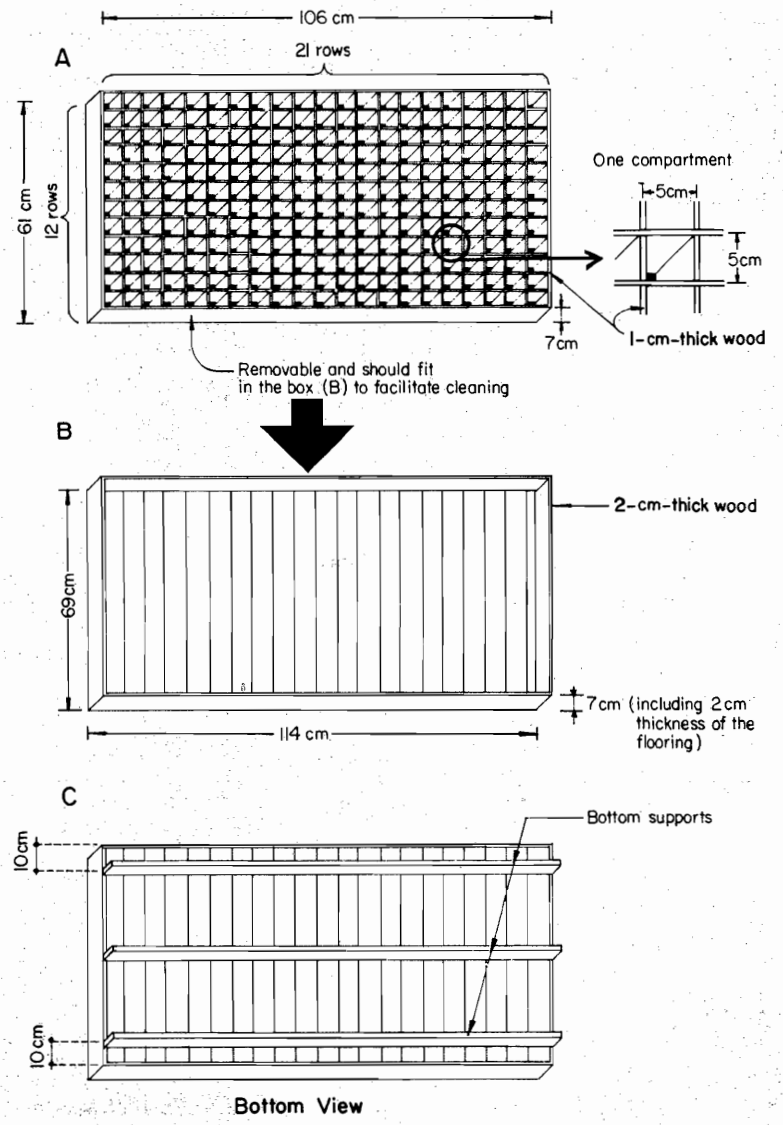
Chapter 3
WHORL MAGGOT

The whorl maggot *Hydrellia philippina* is widely distributed throughout South and Southeast Asia and in China, Japan, and Korea. The fly is hydrophilic and is a pest only in fields that have standing water. Flies oviposit on seedlings shortly after transplanting and damage occurs up to about 40 days after transplanting (DT). Maggots feeding on the leaves within the whorl cause stunting of the plants and delayed maturity. Greenhouse and field screening methods developed at IRRI are described. Only one variety with moderate resistance and several wild rices with resistance have been identified.

GREENHOUSE SCREENING

Greenhouse screening is done in seedboxes. It is more laborious than field screening because flies are collected in the field to infest the seedlings.

Steps	Key points
1. Sowing seed in seedboxes	• Use compartment seedboxes 114 × 69 × 7 cm (Fig. 1). • Place soil containing fertilizer (40 kg N/ha) in the seedbox. • Place a sowing guide consisting of 252 compartments over the soil in the seedbox and sow 5 seeds/entry in each compartment. • Sow a susceptible check variety (TN1, IR8, or any local susceptible variety) and a resistant check (IR40) into the compartments (in 1 compartment on each of the 4 sides and 1 compartment in the middle). • Cover the seed with fine soil and place the seedbox in a water pan tray with 5-cm-deep water.
2. Thinning seedlings and removing the sowing guide	• At 10 days after sowing (DAS), thin the seedlings to 3 per compartment. • Remove the sowing guide taking care not to uproot the seedlings.
3. Covering the seedbox with a cage	• Cover the seedbox with a nylon mesh cage 123 × 76 × 40 cm.
4. Infesting with whorl maggot flies	• Collect flies in the field early in the morning. Use a cylindrical mylar film cage (40 cm high and 5 cm in diam) (Fig. 2). Place the cage over the flies on rice



1. Compartment seedbox. A) compartment structure consisting of 252 compartments measuring 5- × 5-cm, B) top view of the seedbox, and C) bottom view of the seedbox.

Steps	Key points
	seedlings in the field. Flies that fly upward into the cage are trapped and carried to the greenhouse. • Release the field-collected flies into the caged seedbox at the rate of 5 flies per compartment (3 seedlings). • Allow the flies to oviposit on the plants for 48 h then remove the cage and release the flies.

Steps	Key points
5. Recording oviposition preference	• After the cage is removed, record the number of eggs on each plant to determine oviposition preference.
6. Rating damage	• Take damage ratings 15 days after infesting the plants with the flies. • Grade the leaves individually and average the damage rating for all leaves of each plant. Disregard the first two leaves because they are already developed at the time of infestation and thus escape damage. • Leaf damage ratings are based on the following scale:

Scale	*Damage description*
0	No feeding lesion
1	Feeding lesions are the size of a pinhead.
3	Feeding lesions up to 1 cm long
5	Feeding lesions longer than 1 cm but occupy less than half the total leaf area
7	Feeding lesions occupy about one-half of the total leaf area
9	Feeding lesions are large, occupying more than half the total leaf area, and leaf is broken.

2. Mylar cage for collecting whorl maggot flies in the field.

FIELD SCREENING

Steps	Key points
1. Infesting plants	• To ensure sufficient insect pressure for valid screening, choose a time of the year when the whorl maggot population is at its peak. At IRRI incidence is highest in February-March in the dry season crop and September-October in the wet season crop. These dates coincide with the time that farmers in the surrounding area have recently planted their crops. Transplant seedlings a few weeks after the farmers in the vicinity have transplanted theirs.
2. Sowing entries in the nursery	• Sow seed of test entries in seedboxes in the greenhouse or in a wetbed nursery in a screenhouse or in the field. • For the wetbed nursery sow about 5 g of seed in furrows spaced at 5 cm. • Label each entry. • Protect the nursery from insect damage and insect-transmitted viruses by incorporating a granular systemic insecticide (1 kg ai/ha) into the wetbed soil before sowing, by broadcasting insecticide granules, or by spraying insecticide at 10 and 15 DAS.
3. Preparing the field	• Prepare the field for planting and incorporate NPK fertilizer as needed based on local recommendations.
4. Transplanting the seedlings	• Transplant the seedlings at a 25 × 25 cm spacing in 2.5-m rows. Plant a susceptible check (any local variety, TN1, or IR8) and a resistant check (IR40) after every 20 rows of test entries.
5. Irrigating	• Provide continuous irrigation at a 5-cm water depth beginning 2 DT because the flies are hydrophilic and lay eggs on leaves close to the water.
6. Rating damage	• Record damage ratings at 25-30 DT (Fig. 3). Ratings are based on severity of damage. A beginner should grade individual plants but experienced workers can grade by rows. Base ratings on the average damage of hills in a row of an entry. • The test is considered valid when the susceptible check has a rating of at least 7 based on the following scale:

Scale	Damage
0	None
1	One leaf/hill with feeding lesions
3	Two or more leaves/hill but not more than 33% of the leaves with feeding lesions
5	33 to 50% of the leaves with feeding lesions
7	More than 50% of leaves with feeding lesions but no broken leaves
9	More than 50% of leaves with feeding lesions and some broken leaves

3. Screening test varieties for whorl maggot resistance in the field.

MECHANISMS OF RESISTANCE

Methods to determine levels of antixenosis, antibiosis, and tolerance are described.

Antixenosis for oviposition in the greenhouse

Steps	Key points
1. Preparing seedboxes	• Use the compartment seedbox (114 × 69 × 7 cm) (Fig. 1).

Steps	Key points
	• Fill the seedbox with fertile soil to about 3 cm deep. • Each seedbox can accommodate 66 entries; hence, the number of seedboxes depends on the number of test entries and the replications. One seedbox represents one replication. • Have at least six replications.
2. Sowing seed of test entries	• Sow 2-3 seeds of each entry in the compartments, skipping one compartment between entries. Include TN1 as the susceptible check and IR40 as the resistant check. • Cover the seed with fine soil and water to saturation point. • Place seedbox in a water tray. Water the seedlings regularly with a hose sprinkler.
3. Thinning seedlings	• Seven DAS, thin seedlings leaving one healthy seedling per entry in each compartment.
4. Flooding the seedbox	• Run water into the water tray to a level high enough to cover the soil surface in the seedbox.
5. Infesting the seedlings	• At 15-20 DAS, clip all seedlings to the same height. • Cover each seedbox with a nylon mesh cage. • Make sure the leaves of the plants do not touch the sides and top of the cage. • Collect whorl maggot adults in rice paddies using the mylar cage collector (Fig. 2). • Infest each seedbox (66 entries) with 200 freshly field-collected whorl maggot adults. Keep the flies on the seedlings for 48 h.
6. Evaluating	• Count the eggs laid per entry per replication. To count the eggs, remove the seedlings taking care not to damage or dislodge the eggs. Then count the eggs on the leaves. • Compare the number of eggs laid among the entries.

Antibiosis on larval growth and survival

Steps	Key points
1. Preparing the pots	• Fill 8-cm-diam clay pots with fertile soil. The number of pots depends on the number of test entries and the replications. • Replicate each entry 8-10 times.

Steps	Key points
2. Sowing seed of the test entries	• Sow 5 seeds of each test entry in each pot. Include TN1 as the susceptible check and IR40 as the resistant check. • Keep the pots in a water tray in the greenhouse.
3. Collecting whorl maggot adults in the field	• Four days before infestation of the test entries (16 DAS), collect whorl maggot adults in the field. Place them on 20-day-old TN1 plants in a cage and allow them to oviposit for 24 h.
4. Collecting eggs	• Collect eggs from the TN1 plants in the oviposition cage by cutting leaf portions with 1 egg to a size of 1 cm or less. • Place the cut leaf portions with eggs in a petri dish lined with moist filter paper.
5. Incubating eggs	• Incubate the eggs for 2-3 days or up to the dark-head stage at room temperature (22-30° C).
6. Infesting test entries	• Arrange the potted seedlings in a randomized complete block design in a water tray. Make sure the leaves of the seedlings in one pot do not touch the leaves in another pot. This will prevent transfer of larvae from one entry to the other. • Infest the seedlings at the rate of one egg per seedling. Hold the cut leaf portion with forceps and insert it between the stem and leaf sheath of one of the two youngest leaves.
7. Evaluating	• Dissect infested plants at 15 days after infestation (DI). • Collect larvae and pupae from each entry and place them in vials with KAAD mixture. 95% ethyl alcohol 100 ml Kerosene 5 ml Glacial acetic acid 20 ml Dioxane 10 ml • Determine percentage of larval survival. $\%\ \text{survival} = \frac{\text{no. of surviving larvae}}{\text{no. of eggs put on plants}} \times 100$ Compare results among entries. • Classify lavae into small (<2 mm), medium (2-4 mm), and large (>4 mm) and determine the percentage of each classification for each replication.

Steps	Key points
	$\%\ \text{small larvae} = \frac{\text{no. of small larvae}}{\text{total larvae counted}} \times 100$ $\%\ \text{medium larvae} = \frac{\text{no. of medium larvae}}{\text{total larvae counted}} \times 100$ $\%\ \text{large larvae} = \frac{\text{no. of large larvae}}{\text{total larvae counted}} \times 100$ • If pupae are present at time of sampling compute % pupae as $\frac{\text{no. of pupae}}{\text{total larvae and pupae counted}} \times 100$ Those similar to the resistant check are considered resistant; those similar to the susceptible check, susceptible; and those between, moderately resistant.

Tolerance in the field

Steps	Key points
1. Same as steps 1-6 in *Field screening*	• The methods used in field screening are used for the field tolerance test. Replicate each entry four times.
2. Evaluating	• After grading damage which is usually at 25-30 DT, count the tillers on each hill and measure the height of the plants. • Observe 10 hills/plot. • Repeat damage grading 5 days after the first grading and again count the tillers and measure plant height of the same plants sampled in the first observation. • Determine the extent of recovery among the test entries as based on damage grades, tiller number, and plant height and compare with that of the susceptible and resistant checks.

Tolerance in the greenhouse

Steps	Key points
1. Preparing the pots	• Fill 8-cm-diam clay pots with fertile soil.

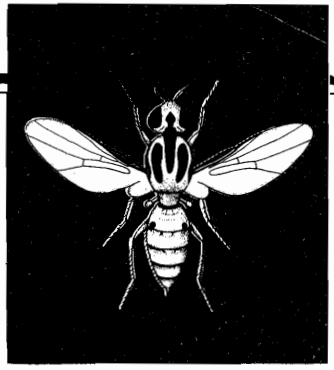
Steps	Key points
2. Sowing seed	• Sow 3-5 seeds in each pot. Use TN1 as a susceptible check and IR40 as a resistant check. Replicate six times.
3. Thinning the seedlings	• Thin to 3 seedlings/pot before caging.
4. Caging and infesting the seedlings	• At 15-20 DAS, cage the potted seedlings (20 pots/cage). • Collect flies in the field and release them at the rate of 5 flies/pot (100 flies/cage). • Allow the flies to oviposit for 48 h, then remove the cage to release them. • Arrange the pots in a randomized complete block design. To prevent transfer of newly hatched larvae, make sure the leaves of each entry do not touch the leaves of another entry.
5. Evaluating	• Grade plant damage at 15-20 DT using the scale as in *Greenhouse screening.* • Count the tillers and measure plant height. • Repeat observations 5 days later, again recording the damage, number of tillers, and plant height. • Determine extent of recovery of the entries and compare results with the susceptible and resistant check.

SOURCES OF RESISTANCE

One variety and several wild rices have resistance to the whorl maggot.

Accessions resistant to the whorl maggot *Hydrellia philippina* at IRRI, 1983.

IRRI accession no.	Name	Origin
	Variety	
36958	IR40	Philippines
	Wild rices	
100115	*O. brachyantha*	Guinea - Conakry
101231	*O. ridleyi*	Sierra Leone
101232	*O. ridleyi*	Sierra Leone
101233	*O. ridleyi*	Sierra Leone
101234	*O. ridleyi*	Sierra Leone
101235	*O. ridleyi*	Sierra Leone
101236	*O. ridleyi*	Sierra Leone



Chapter 4
RICE STEM MAGGOT

The rice stem maggot *Chlorops oryzae* is a pest of rice only in Japan. It also attacks other graminaceous crops. Maggots feed on leaves in the central whorl of plants in the vegetative stage. Maggots of the second and third broods feed on developing grains within the boots and cause yield losses. Methods for screening and determining the mechanisms of resistance have been developed.

FIELD SCREENING

Field screening methods for both direct seeded and transplanted rice have been developed in Japan.

Direct seeded rice

The screening method described for direct seeded rice is a combination of those by Fukuda and Inoue (1962), Koyama (1970), and Koyama and Hirao (1971).

Steps	Key points
1. Preparing seed of test entries	• Include seed of a susceptible (Norin 8) and a resistant (Norin 25) check.
2. Sowing seed	• In Japan sowing is usually during the first week of April to coincide with the abundance of adults from the middle of May to the first week of June. • Sow seed in rows 3 cm apart at 1 seed/hill. For each variety, plant 3 replications each with 90 hills in a 3.3-m^2 area.
3. Infesting plants	• Natural infestation provides a source of insects.
4. Evaluating	• At 120 days after sowing (DAS), select at random 100 plants from each entry per replication. • Count damaged and undamaged leaves, stems, and panicles. • Compute the following: $\%\ \text{damaged leaves} = \frac{\text{no. of damaged leaves}}{\text{total no. of leaves}} \times 100$ $\%\ \text{damaged stems} = \frac{\text{no. of damaged stems}}{\text{total no. of stems}} \times 100$

Steps	Key points
	$\text{\% damaged panicles} = \frac{\text{no. of damaged panicles}}{\text{total no. of panicles}} \times 100$ $\text{Resistance index} = \frac{\text{no. of injured panicles}}{\text{total no. of infested stems}} \times 100$ Classify varieties into three categories based on the resistance index: *Category* — *Resistance index* Resistant — 0-33% Moderately resistant — 34-66% Susceptible — 67-100%

Transplanted rice

The method of Koyama (1970) is used in field screening transplanted rice.

Steps	Key points
1. Preparing seed	• Prepare seed of test entries, including susceptible (Norin 8) and resistant (Norin 25) check varieties.
2. Sowing seed	• Sow seed in seedbeds covered with vinyl or oil paper to provide a good stand of seedlings to expose to ovipositing adults in the field.
3. Transplanting the seedlings	• Transplant seedlings 23 DAS, at 1 seedling/hill, in plots measuring 15 m^2/entry.
4. Infesting the plants	• Natural infestation provides a source of insects.
5. Evaluating	• Follow the procedure used in *Direct seeded rice.*

MECHANISMS OF RESISTANCE

Antibiosis on larval survival in the greenhouse

A method to determine larval survival on rice varieties was developed by Yushima and Tomisawa (1957).

Steps	Key points
1. Preparing seed	• Prepare seed of test entries.

Steps	Key points
2. Sowing seed	• Sow seed in 15-cm-diam pots at 3 seeds/pot. Replicate each entry 10 times. One pot represents one replication. In the greenhouse arrange the pots in a water tray, using a randomized complete block design.
3. Infesting the plants	• Infest plants with first-instar maggots when they have 4-5 leaves. Using a fine brush put one maggot on the uppermost leaf of each plant. To obtain maggots collect adults from nursery beds and allow them to oviposit on leaves in glass jars. Use the maggots hatching from the eggs to infest the test entries.
4. Evaluating	• At 35-40 days after infestation dissect the stems to expose the larvae or pupae and count the surviving insects on each entry. • Compute the following: $\%\ \text{survival} = \frac{\text{no. of larvae + pupae counted}}{\text{no. of larvae infested}} \times 100$ $\%\ \text{pupal formation} = \frac{\text{no. of pupae counted}}{\text{no. of larvae infested}} \times 100$ • Compare results among entries.

Antibiosis on larval survival in the field

The methods described are those developed by Koyama (1970) and Koyama and Hirao (1971).

Steps	Key points
1. Same as steps 1-4 under *Transplanted rice*	
2. Evaluating	• Count 3 times (42, 51, and 68 days after transplanting) the eggs laid on 30 stems/entry per replication. Use colored strings to mark stems with eggs. • Compute % larval survival: $\frac{\text{no. of stems damaged}}{\text{no. of stems with eggs}} \times 100$

Steps	Key points

- Classify varieties:

Classification	*% survival of larvae*
Resistant	< 10
Moderately resistant	10-50
Susceptible	> 50

- Determine the resistance index, which is represented by the survival rate of larvae during the feeding period.

$$\text{Resistance index} = \frac{\text{no. of injured panicles}}{\text{total no. of infested stems}} \times 100$$

Convert the resistance index as follows:

Varietal classification	*Resistance index*
Resistant	0-33%
Moderately resistant	34-66%
Susceptible	67-100%

SOURCES OF RESISTANCE

Sources of resistance to the rice stem maggot are in the table.

Variety	Reference	Variety	Reference
Aichi-asahi	Fukuda and Inoue (1962)	Koshihikari	Fukuda and Inoue (1962)
Akebono	Fukuda and Inoue (1962)	Kuzuyu	Koyama (1970)
Ashai	Koyama and Hirao (1971)	Mihonishiki	Fukuda and Inoue (1962)
Asakaze	Fukuda and Inoue (1962)	Nakasengoku	Fukuda and Inoue (1962)
Ayanishiki	Fukuda and Inoue (1962)	Nakura	Fukuda and Inoue (1962)
Azusa	Fukuda and Inoue (1962)	Norin 10	Koyama (1970)
Chiyohikari	Fukuda and Inoue (1962)	Norin 12	Fukuda and Inoue (1962)
Dokai-shinriki	Fukuda and Inoue (1962)	Norin 18	Fukuda and Inoue (1962)
Etunan 18	Koyama (1970)	Norin 22	Fukuda and Inoue (1962)
Fukuminori	Fukuda and Inoue (1962)	Norin 25	Fukuda and Inoue (1962)
Hamayu	Fukuda and Inoue (1962)		Koyama and Hirao (1971)
Haya-uruti	Koyama (1970)	Norin 35	Fukuda and Inoue (1962)
Hikotaromoti	Koyama (1970)	Norin 38	Fukuda and Inoue (1962)
Hozakae	Fukuda and Inoue (1962)	Norin 39	Fukuda and Inoue (1962)
Ikurin 5	Koyama (1970)	Norin 43	Fukuda and Inoue (1962)
Joshu	Koyama and Hirao (1971)	Norin 44	Fukuda and Inoue (1962)
Kairyo-akita	Fukuda and Inoue (1962)	Norin 49	Fukuda and Inoue (1962)
Kanan-nansen	Koyama 1970	Obanazawa	Koyama (1970)
Kiyosumi	Fukuda and Inoue (1962)	Oha	Koyama and Hirao (1971)
Koganemani	Fukuda and Inoue (1962)	Ou 20	Koyama (1971)

Steps	Key points		
Ou 29	Koyama (1971)	ashahi	
Ou 187	Koyama (1970)	Shin 7	Fukuda and Inoue (1962)
Ou 188	Koyama and Hirao (1971)	Shubo	Fukuda and Inoue (1962)
	Koyama (1970)	Sin 4	Koyama (1970)
Ou 200	Koyama (1970)	Sin 5	Koyama (1970)
Ou 214	Koyama (1970)	Takara	Fukuda and Inoue (1962)
Ou 229	Koyama (1970)	Tohoku 64	Koyama (1970)
Ou 230	Koyama and Hirao (1971)	Tugaru-asahi	Koyama (1970)
	Koyama (1970)	Tyosen	Koyama (1970)
Ou 231	Koyama (1970)	Tyusin 203	Koyama (1970)
Sakaikaneko	Koyama and Hirao (1971)	Yaeho	Fukuda and Inoue (1962)
Sakaikinsu	Koyama (1970)	Yamahonami	Fukuda and Inoue (1962)
Sekiminori	Fukuda and Inoue (1962)	Yamakogane	Fukuda and Inoue (1962)
Senbon-	Fukuda and Inoue (1962)		

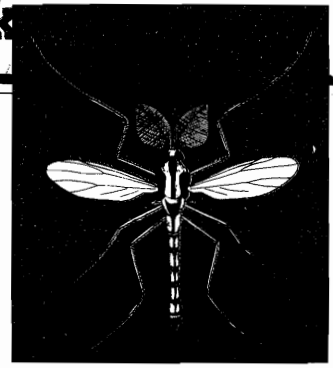
REFERENCES CITED

Fukuda, J., and H. Inoue. 1962. Varietal resistance of rice to the rice stem maggot. Intl. Rice Comm. Newsl. 11(1):8-9.

Koyama, T. 1970. Resistance in rice plant against the rice stem maggot. Bull. Tohoku Agric. Exp. Stn. 39:171-206.

Koyama, T., and J. Hirao. 1971. Varietal resistance of rice to the rice stem maggot. Pages 251-256 *in* Tropical Agricultural Research Center. Proceedings of the symposium on rice insects. Tokyo, Japan.

Yushima, T., and J. Tomisawa. 1957. Problems on insect resistance of rice plant to rice stem maggot. I. A new method for measuring varietal resistance of crops against rice stem maggot. Jpn. J. Appl. Entomol. 1(3):180-183.



Chapter 5
BLOODWORM

Bloodworm *Chironomus tepperi* feeds on newly sown seeds and on the roots of young rice seedlings. Bloodworms are a threat to crop establishment. Seedlings may be uprooted and accumulate near levees. In Australia newly sown rice is most susceptible to bloodworm attack. Drill-sown crops are not affected.

GREENHOUSE SCREENING

Greenhouse screening for resistance to bloodworms has been conducted in Australia (Treverrow 1979).

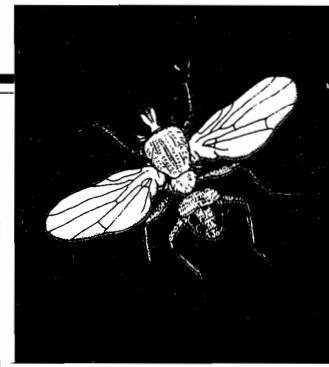
Steps	Key points
1. Preparing seed	• Prepare seed of test entries.
2. Sowing seed	• Sow 5 pregerminated seeds of each entry in a pot. Arrange pots in a randomized complete block design with four replications, each pot representing a replication. Varieties Inga, Dawn, or IR22 can be used as susceptible checks.
3. Infesting plants	• Collect bloodworms in the field and introduce heavy infestations at sowing time. • Include an uninfested control for each entry.
4. Evaluating	• Uproot plants 35 days after infestation. • Dry and weigh each plant. • Compute % damage: $\frac{\text{av dry wt of control plant} - \text{av dry wt of infested plant}}{\text{av dry wt of control plant}} \times 100$ • Varieties with less than 10% damage are considered resistant.

SOURCES OF RESISTANCE

Sources of resistance to bloodworms are Omirt 39 and YR 303D-5 (Treverrow 1979).

REFERENCE CITED

Treverrow, N. 1979. Resistance of rice varieties to attack by bloodworms. Australian Rice Workshop, New South Wales Department of Agriculture, Agricultural Research Centre, Yanco, New South Wales, Australia 2703.



Chapter 6
RICE SEEDLING FLY

The rice seedling fly *Atherigona exigua* Stein is a pest of dryland rice seedlings in South and Southeast Asia. Maggots feeding on the central shoot of rice produce deadhearts. Studies on resistance of rice varieties to the seedling fly have been limited to field screening in Indonesia (Soejitno 1977).

FIELD SCREENING

Field screening is conducted in dryland fields.

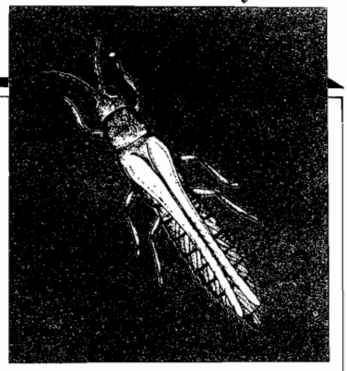
Steps	Key points
1. Preparing seed of test entries	• Secure and prepare seed of the test entries and include susceptible and resistant checks if available.
2. Preparing plant beds	• Construct 1- × 2-m raised beds for every 10 varieties to be tested. Beds should be 40 cm apart. • Have four replications for each variety.
3. Sowing the seed	• Sow about 100 seeds of each variety in1-m rows, with 20-cm spacing between rows.
4. Infesting the test entries	• Infestation depends on the natural field population of the seedling fly.
5. Evaluating	• Count the infested plants at 2, 4, and 6 weeks after sowing. • Compute the percentage of infestation using the formula: $\%\ \text{infestation} = \frac{\text{no. of plants infested}}{\text{total no. of plants observed (infested and healthy plants)}} \times 100$

SOURCES OF RESISTANCE

The sources of resistance identified were Arias and C 22 (Soejitno 1977).

REFERENCE CITED

Soejitno, J. 1977. Reaksi biberapa varietas gogo terhadap lalat bibit padi, *Atherigona exigua* Stein [Indonesian, with English summary]. Laporan Kemajuan Penelitan Seri Hama/Penyakit No. 6:30-35.



Chapter 7
THRIPS

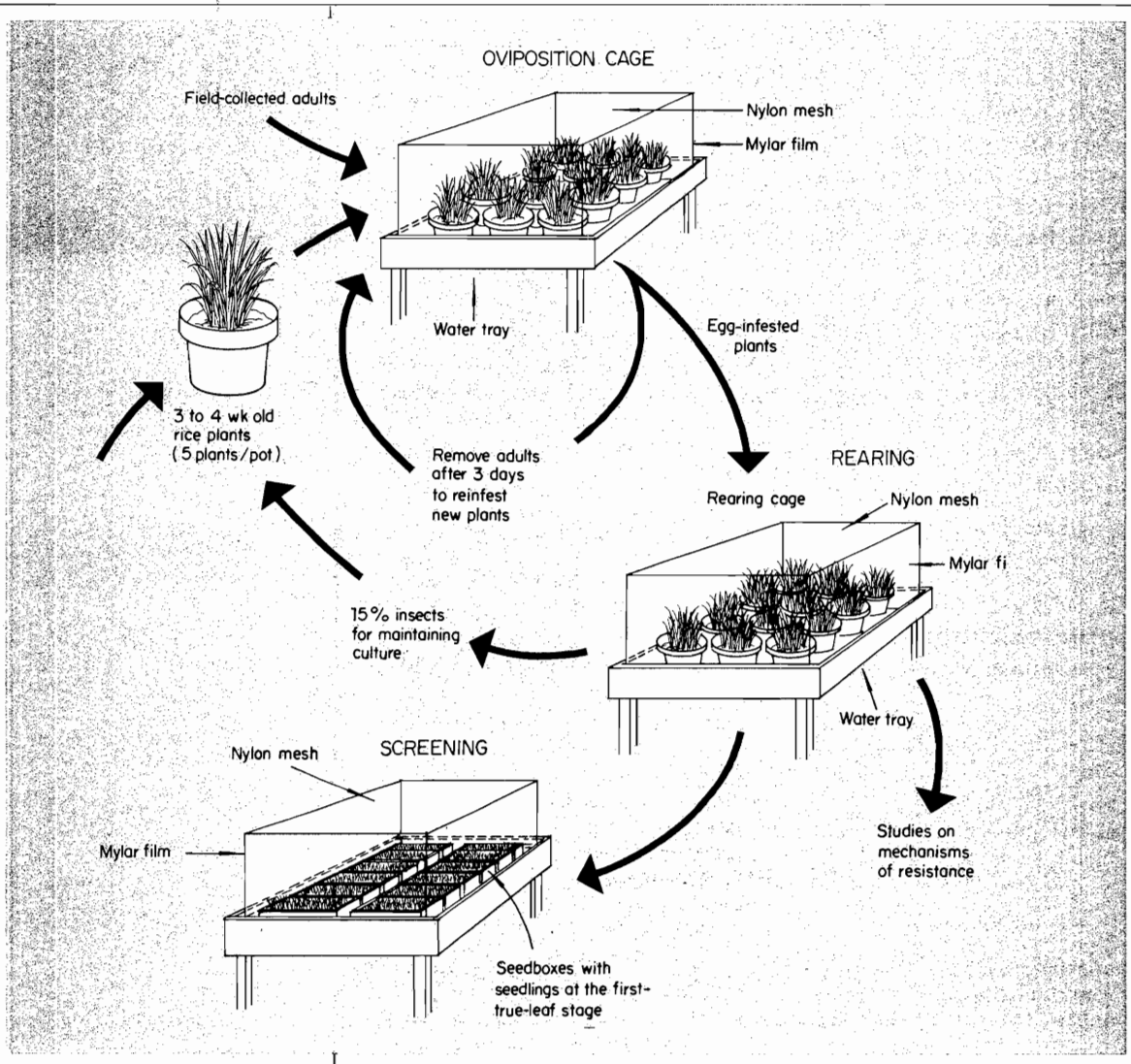
The rice thrips *Stenchaetothrips biformis* (= *Thrips oryzae*) is a small insect which feeds primarily on young rice seedlings in the nursery or in the field. It has been recorded throughout South and Southeast Asia and is considered a pest in Bangladesh, China, India, and Sri Lanka. Damage is most obvious in late crops, especially during drought. Infested leaves curl inward from the margins. Feeding by thrips causes scorching, wilting, and leaf-tip drying, which result in stunted plants or death and poor crop stand.

A thrips rearing technique and a greenhouse screening method have been developed at IRRI. Field screening has been conducted in Bangladesh, India, and Sri Lanka, and several sources of resistance have been selected. Some of the resistant varieties from Sri Lanka have also been found resistant in China.

REARING

Rearing is done in an insectary under controlled temperature (25 ± 2° C) and relative humidity (70 ± 10%). The rearing and greenhouse screening procedures of Nugaliyadde and Heinrichs (1984a,b) are illustrated in Figure 1.

Steps	Key points
1. Growing host plants	• Plant TN1 (or any susceptible variety) at 5 seedlings/12-cm-diam pot. • Grow the potted plants inside a cage to protect them from other pests. Apply fertilizer.
2. Infesting plants with ovipositing adults	• When the plants are about 3-4 weeks old, infest them with field-collected adult thrips at the rate of 4-5 pairs/plant. Use a camel hair brush (size 00) to handle thrips; place them on the youngest leaf. • Place the pots in the insectary in mylar oviposition cages ventilated by nylon mesh tops. • After 3 days, unroll the infested leaves carefully and remove the adults to reinfest another set of plants. Females live about 12 days, so they can be used to oviposit at least 4 times. • Leave the egg-infested plants in mylar rearing cages for 2 weeks. The eggs will hatch and larvae will develop into adults. Each plant will contain about 20 thrips. • Use 15% of the progeny to maintain the culture, and the rest for screening and studies on mechanisms of resistance.



1. Procedure for rearing and greenhouse screening of rice for thrips resistance (Nugaliyadde and Heinrichs 1984b).

GREENHOUSE SCREENING

Steps	Key points
1. Growing test entries	• Pregerminate seeds of 16 entries, including resistant (Dahanala 2220) and susceptible (TN1) checks. • Sow the entries in rows 10 cm long and 4 cm apart in a plastic tray (32 × 24 × 10 cm) filled with soil 5 cm deep. Each seedbox should contain 16 rows in 2 strips. Assign resistant and susceptible checks

Steps	Key points
	randomly with other entries. Replicate each set of entries 3 times (3 seedboxes). Place the plastic tray in a water-filled iron tray in the greenhouse. To maintain soil moisture, add water regularly to the plastic trays. • When the seedlings are at the first-true-leaf stage, thin to 10/row.
2. Infesting test entries	• Infest entries at the first-true-leaf stage. Collect thrips-infested leaves in the rearing cage and evenly distribute them between rows of the entries. To prevent escape, cover the tray with a mylar cage with a nylon mesh top. Adult thrips will move from infested leaves to the unfolded part of the first true leaves of the test entries. There should be 2-3 adults/seedling.
3. Evaluating	• Make damage ratings when the susceptible check has a rating of 9 [about 7 days after infestation (DI)], using the scale given below. The score is based on damage to the 1st and 2d leaves. *Scale* — *Damage* 0 None 1 Rolling of terminal 1/3 of 1st leaf only 3 Rolling of terminal 1/3 to 1/2 of 1st and 2d leaves 5 Rolling of terminal 1/2 of 1st and 2d leaves. Less than 30% of the leaf surface scorched. 7 Rolling of entire length of all leaves. Scorching on 30-60% of the leaf surface. Wilting not pronounced. 9 Rolling of entire length of all leaves. Scorching on more than 60% of leaf surface. Wilting of leaf tips.

FIELD SCREENING

Only a limited number of field screening trials for thrips have been done and evaluation criteria for various areas differ greatly. Methods are given for the reader to determine the most useful under local conditions.

Steps	Key points
1. Planting test entries	• Thrips prefer young seedlings. Use seed sown in the field rather than transplanted seedlings. • Pregerminate seed.

Steps	Key points
	• Sow seed in 0.7-m-long rows in moist soil at 1 seed/cm. Distance between rows should be 20 cm. • Replicate each entry three times with one row per replication. • Use Dahanala 2220 as a resistant check and IR8 or some other susceptible variety as the susceptible check. • At 7 days after sowing (DAS) slightly increase the water and maintain it at the proper level.
2. Infesting plants	• Natural infestation provides a source of insects.
3. Evaluating	• Make damage ratings twice — the first at 14 DAS and the second at 21 DAS (Fig. 2). The following rating systems have been used. Further studies to determine the most useful method are necessary.

Velusamy et al (1975)

Count total leaves and affected leaves on 20 random plants per replication per entry and compute:

$$\% \text{ infestation} = \frac{\text{no. of affected leaves in 20 plants}}{\text{total no. of leaves in 20 plants}} \times 100$$

Then use the following rating system:

Reaction	*% infestation*
Resistant	0-10
Moderately resistant	11-20
Moderately susceptible	21-50
Susceptible	51-100

Kudagamage (1977)

Scale	*Damage*
0	None
1	Leaves healthy except for slight rolling of the 1st and 2d leaves
3	Slight rolling and withering of the 1st and 2d leaves
5	First and 2d leaves withered and dead; upper leaves slightly rolled
7	Upper leaves rolled, withered, and wilted with a few white lesions; lower leaves dead
9	Upper leaves drying with pronounced white lesions; seedlings stunted; lower leaves dead

2. Field screening for thrips resistance at Batalagoda, Sri Lanka. The susceptible variety is in the center.

Steps	Key points
	Kabir and Alam (1981)
	Scale — *Damage*
	0 — None
	1 — Light infestation, tip of leaves rolling
	3 — Medium light infestation, 20-30% leaves rolling
	5 — Medium infestation, 30-50% leaves rolling
	7 — Medium heavy infestation, 50-75% leaves rolling
	9 — Heavy infestation, 75-100% leaves rolling
	Velusamy and Chelliah (1980)
	Scale — *Damage*
	0 — None
	1 — Rolling of terminal third of 1st leaf
	3 — Rolling of terminal third to half of 1st and 2d leaves
	5 — Rolling of terminal half of 1st, 2d, and 3d leaves; yellowing of leaf tips
	7 — Rolling of entire length of all leaves; pronounced yellowing
	9 — Complete plant wilting, yellowing, and scorching

MECHANISMS OF RESISTANCE

Entries with a rating of 0-5 in either the greenhouse or field screening tests are retested in the insectary under controlled temperature and humidity to confirm the level and nature of resistance.

Antixenosis for feeding

Steps	Key points
1. Selecting entries	• Select and test 10 entries at a time, including Dahanala 2220 as a resistant check and TN1 as a susceptible check.
2. Growing test plants	• Pregerminate seed of the test entries. To form a circle of 10 seedlings, sow 1 seed/entry equidistant from the others around a 12-cm-diam clay pot. Replicate each set of entries 10 times.
3. Infesting plants	• When the seedlings are at the first-true-leaf stage, place 1-cm-long pieces of rice leaves infested with a total of 50 adult thrips at the center of each pot. Cover the seedlings with a mylar cylinder (10-cm-diam, 30-cm-high) ventilated by a nylon mesh top.
4. Evaluating	• Count the thrips on each seedling at 2 and 3 DI. Compare results among test entries and resistant and susceptible checks.

Antixenosis on oviposition rate

Steps	Key points
1. Growing test plants	• Plant pregerminated seed in 5-cm-diam clay pots at 1 seed/pot. Replicate each test entry five times. Grow Dahanala 2220 as a resistant check and TN1 as a susceptible check. Apply fertilizer.
2. Infesting plants	• When the seedlings are at the 5th-leaf stage, place 5 pairs of newly emerged adults on the youngest leaf blade. Use a camel hair brush (size 00) to handle the insects. • Cage the seedlings in mylar cylinders (4-cm-diam, 30-cm-high) with a nylon mesh top for ventilation. Arrange the pots in a randomized complete block design in a water-filled tray.
3. Evaluating	• Three days after infestation, examine the youngest leaf blade of each test plant for eggs under a

Steps	Key points
	binocular microscope. Eggs appear as translucent swellings when viewed against light. Determine the total number of eggs on each test entry and compare with that on the resistant and susceptible checks.

Antibiosis on population growth and Degree of plant injury

Steps	Key points
1. Growing test plants	• Grow test plants and checks as described in *Antibiosis on oviposition rate.*
2. Infesting plants	• Infest plants at 4 levels: 0, 2, 4, and 6 pairs per plant. Maintain a control without thrips for each entry. • When the seedlings are at the 5th-leaf stage, arrange the pots in a water-filled tray in a split-plot design, with variety as the main plot and infestation levels as the subplots. • Replicate the experiment five times, with one pot serving as one replication per treatment. • Infest the plants with newly emerged adult pairs with the aid of a camel hair brush and cage in mylar cylinders (4-cm-diam, 30-cm-high) with nylon mesh windows for ventilation.
3. Evaluating	• Fourteen days after infestation, count the total number of eggs, larvae, prepupae, pupae, and adults on each plant. • Remove the plants from the pots, wash thoroughly, and measure plant height. • Determine the dry weight of each plant. • Compare results for the test entries with the resistant and susceptible checks.

SOURCES OF RESISTANCE

The sources of resistance to thrips in *Oryza sativa* as based on the literature (field screening) and greenhouse screening at IRRI (Nugaliyadde and Heinrichs 1984a) are listed in the table.

Variety	Origin	Reference
ADT7	India	Velusamy et al (1975)
ADT22	India	Velusamy et al (1975)
ASD4	India	Velusamy et al (1975)
ASD5	India	Velusamy et al (1975)
ASD7	India	Velusamy and Chelliah (1980)
ASD7 (06303)[a]	India	Nugaliyadde and Heinrichs (1984a)

Variety	Origin	Reference
Babawee	Sri Lanka	Velusamy and Chelliah (1980)
Balamawee	Sri Lanka	Velusamy and Chelliah (1980)
BJ1	Bangladesh	BRRI (1977)
BJ1 (00256)	Bangladesh	Nugaliyadde and Heinrichs (1984a)
BJ1 (03711)	Bangladesh	Nugaliyadde and Heinrichs (1984a)
BW78	Sri Lanka	Kabir and Alam (1981)
Chandina composite	Bangladesh	BRRI (1977)
CO6	India	Velusamy et al (1975)
CO7	India	Velusamy et al (1975)
CO16	India	Velusamy et al (1975)
CO23	India	Velusamy et al (1975)
CO27	India	Velusamy et al (1975)
CO29	India	Velusamy and Chelliah (1980)
Dahanala	Sri Lanka	Ekanayake and Fernando (1977)
Dahanala 682	Sri Lanka	Kudagamage (1977)
Dahanala 682 (50729)	Sri Lanka	Nugaliyadde and Heinrichs (1984a)
Dahanala 2220	Sri Lanka	Kudagamage (1977)
Dahanala 2220 (50730)	Sri Lanka	Nugaliyadde and Heinrichs (1984a)
Demata Kotan	Sri Lanka	Ekanayake and Fernando (1977)
Gangala	India	Velusamy et al (1975)
Gonabaru (07809)	Sri Lanka	Nugaliyadde and Heinrichs (1984a)
H105	Sri Lanka	Velusamy and Chelliah (1980)
Heenati (08964)	Sri Lanka	Nugaliyadde and Heinrichs (1984a)
Heenati (08921)	Sri Lanka	Nugaliyadde and Heinrichs (1984a)
Herath banda	Sri Lanka	Ekanayake and Fernando (1977)
IET3817	India	Kabir and Alam (1981)
IET4506	India	Kabir and Alam (1981)
IET4786	India	Velusamy and Chelliah (1980)
Jeeragasamba	Sri Lanka	Velusamy et al (1975)
Kalubalawee	Sri Lanka	Velusamy et al (1975)
Kalubalawee (07702)	Sri Lanka	Nugaliyadde and Heinrichs (1984a)
Kaluheenati	Sri Lanka	Kudagamage (1977) and Ekanayake, Fernando (1977)
Kaluheenati (15568)	Sri Lanka	Nugaliyadde and Heinrichs (1984a)
Kaoshiung sen yu 185	Taiwan, China	Kabir and Alam (1981)
Madael	Sri Lanka	Ekanayake and Fernando (1977)
MRC505	Philippines	Kabir and Alam (1981)
Nira	USA	Velusamy and Chelliah (1980)
Perunel	Sri Lanka	Velusamy and Chelliah (1980)
Ptb 19	India	Velusamy and Chelliah (1980)
Ptb 33	India	Velusamy et al (1975)
Sinna Sivappu	Sri Lanka	Velusamy and Chelliah (1980)
Sudu Hondarawala	Sri Lanka	Velusamy and Chelliah (1980)
Suduru Samba	Sri Lanka	Velusamy and Chelliah (1980)
Sudurvi 305	Sri Lanka	Velusamy et al (1975)
Sugadas	?	Velusamy et al (1975)
Thirissa	Sri Lanka	Velusamy and Chelliah (1980), Kabir and Alam (1981)
Thunmas hamara	Sri Lanka	Ekanayake and Fernando (1977)
TNR1	India	Velusamy et al (1975)
TNR2	India	Velusamy et al (1975)
TKM2	India	Velusamy et al (1975)
TKM6	India	Kabir and Alam (1981)
Utri Rajapan	Indonesia	Velusamy et al (1981)
Warangal	India	Ekanayake and Fernando (1977)
Wanni Dahanala (11726)	Sri Lanka	Nugaliyadde and Heinrichs (1984a)

[a]IRRI accession numbers are given in parentheses.

Several wild rice species with resistance to rice thrips have been identified.

Species IRRI accession no.	Origin	Reference
O. eichingeri		
101171	Tanzania	Velusamy et al (1981)
101418	Uganda	Nugaliyadde and Heinrichs (1984a)
101422	Uganda	Nugaliyadde and Heinrichs (1984a)
101424	Uganda	Nugaliyadde and Heinrichs (1984a)
O. glaberrima		
103437	Senegal	Velusamy et al (1981)
103438	Senegal	Velusamy et al (1981)
103443	Senegal	Velusamy et al (1981)
O. minuta		
101079	Philippines	Nugaliyadde and Heinrichs (1984a)
101083	Philippines	Nugaliyadde and Heinrichs (1984a)
101097	Philippines	Nugaliyadde and Heinrichs (1984a)
O. nivara		
100897	Burma	Nugaliyadde and Heinrichs (1984a)
101510	India	Velusamy et al (1981)
103836	Bangladesh	Velusamy et al (1981)
O. nivara/O. sativa		
103791	Venezuela	Velusamy et al (1981)
O. officinalis		
100181	Burma	Nugaliyadde and Heinrichs (1984a)
100973	Philippines	Nugaliyadde and Heinrichs (1984a)
101073	Philippines	Nugaliyadde and Heinrichs (1984a)
101117	Philippines	Nugaliyadde and Heinrichs (1984a)
101155	Malaysia	Nugaliyadde and Heinrichs (1984a)
O. perennis		
103849	India	Velusamy et al (1981)
O. rufipogon/O. nivara		
101993	Sri Lanka	Velusamy et al (1981)
O. sativa f. *spontanea*		
103826	Bangladesh	Velusamy et al (1981)
103831	Bangladesh	Velusamy et al (1981)

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Ekanayake, E. O., and H. E. Fernando. 1977. Rice thrips study. Central Agricultural Research Institute, Peradeniya, Sri Lanka. 12 p. (mimeo.)

Kabir, M. A., and M. S. Alam. 1981. Screening rice varieties and breeding lines for thrips resistance in late boro. Int. Rice Res. Newsl. 6(1):7-8.

Kudagamage, C. 1977. Varietal resistance to the rice thrips, *Baliothrips biformis*. Int. Rice Res. Newsl. 2(5):11.

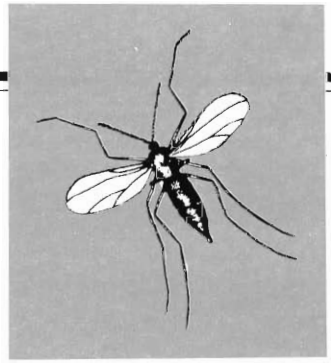
Nugaliyadde, L., and E. A. Heinrichs. 1984a. Resistance of *Oryza* spp. to thrips *Stenchaetothrips biformis* (Bagnall) (Thysanoptera:Thripidae). Crop Protection 3:305-313.

Nugaliyadde, L., and E. A. Heinrichs. 1984b. Biology of rice thrips, *Stenchaetothrips biformis* (Bagnall) (Thysanoptera:Thripidae) and a greenhouse rearing technique. J. Econ. Entomol. 77:1171-1175.

Velusamy, R., I. P. Janaki, A. Subramanian, and J. Chandramohan. 1975. Varietal resistance to rice insect pests. Rice Entomol. Newsl. 3:13-16.

Velusamy, R., and S. Chelliah. 1980. Rice resistance to thrips. Int. Rice Res. Newsl. 5(1):7.

Velusamy, R., S. Chelliah, and J. Chandramohan. 1981. Resistance of rice cultivars and wild rices to thrips. Int. Rice Res. Newsl. 6(6):9.



Chapter 8
GALL MIDGE

The gall midge *Orseolia oryzae* is a serious rice pest in South and Southeast Asia and parts of West Africa. Rice and several grasses serve as its host plants. Upon hatching the larva moves downward under the leaf sheath to the growing point. Its presence causes the leaf sheath to develop into an onion leaf-like gall called an *onion shoot or silvershoot*. Damage results in plant stunting and the affected tillers produce no panicles.

Extensive studies on rice resistance to the gall midge have been conducted in several countries and methods of screening and determining the mechanisms of resistance are well developed. Many sources of resistance have been identified. Because it is difficult to achieve satisfactory control of the gall midge with insecticides, the growing of gall midge-resistant varieties has become a major control tactic in several countries.

REARING

Efficient greenhouse rearing methods have been developed and are used in India, Indonesia, Sri Lanka, and Thailand. The method of Arifin and Vreden (1977) in Indonesia is explained.

Construction and fabrication of equipment

The greenhouse is equipped with water pan trays, rearing cages, and seedboxes.

Steps	Key points
1. Constructing a greenhouse	• A greenhouse typical for gall midge resistance studies has 3 units: a 9- × 5- × 5-m unit for insect rearing, a 13- × 5- × 5-m unit for varietal resistance screening, and a 4- × 5- × 5-m unit for studies on the mechanisms of resistance. • Construct the greenhouse so that there will be optimum lighting.
2. Installing water pan trays in the greenhouse	• Trays for insect rearing and varietal screening can be made of wood and lined with plastic sheets. Use corrugated plastic sheets to protect the plastic lining when the rearing cages are placed on the trays. You can also use galvanized iron trays. The trays must be deep enough to cover the basal parts of the potted plants (about 20 cm). • Place water in the trays to provide optimum humidity and prevent parasitization and predation.

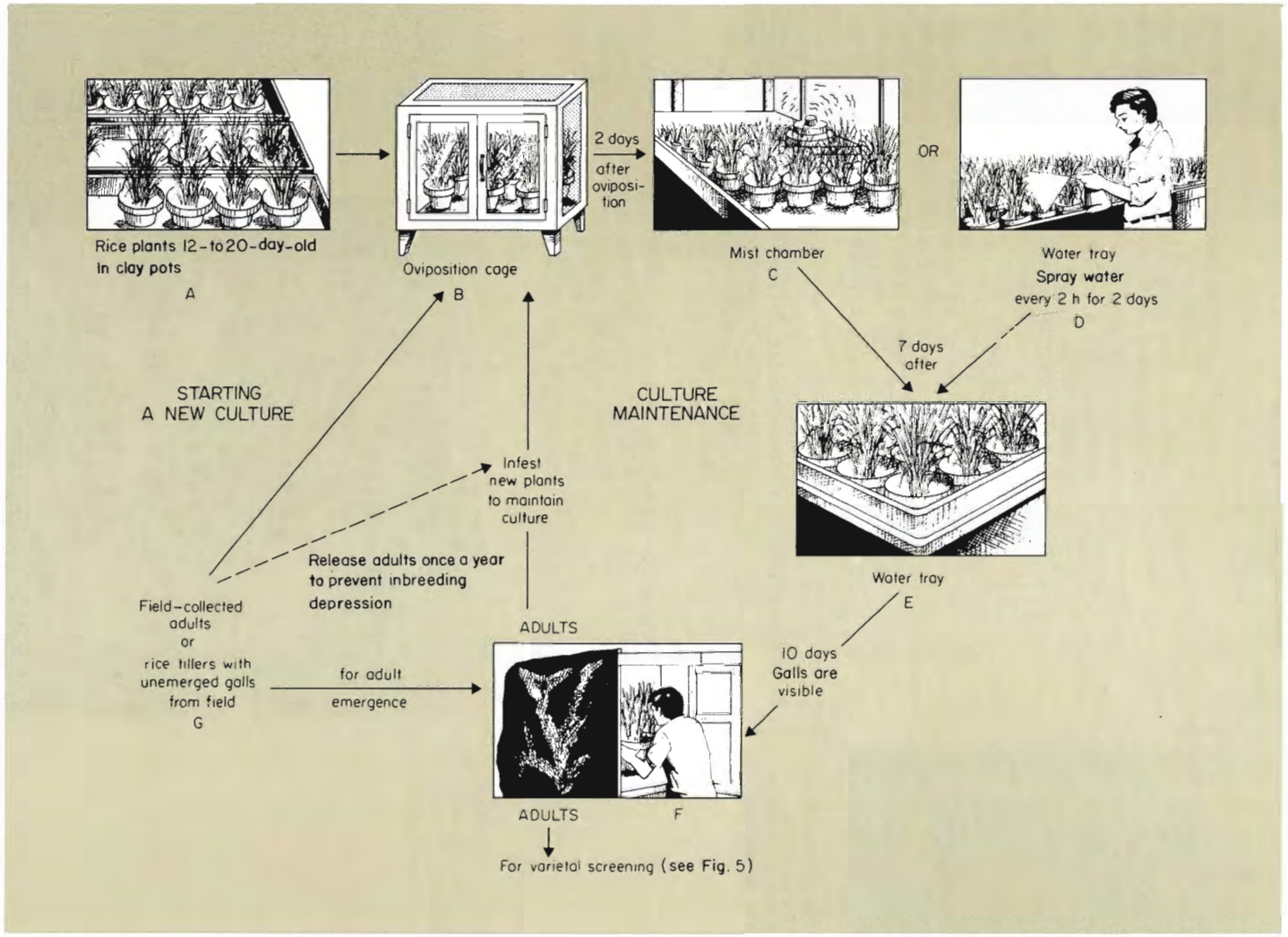
1. Oviposition or fly emergence cage.

	• The size of the trays depends on the size of the greenhouse and on the volume of materials to be screened.
3. Constructing rearing cages	• For rearing cages use glass for roofing, fine mesh screen for sides, and glass for the doors. Leave the bottom open. • Cages should measure 80 × 80 × 100 cm (Fig. 1).
4. Constructing seedboxes	• Make wooden seedboxes 50 × 60 × 7 cm. A seedbox this size will hold 15 rows of test entries including standard checks.

Rearing the gall midge

A scheme to mass rear gall midge is shown in Figure 2. Maintain a continuous supply of 15- to 20-day-old plants of a susceptible variety for the oviposition cage. The potted plants serve as a substrate on which the adult gall midge oviposits and as a food source for the larvae.

Steps	Key points
1. Growing food plants	• Mix fertilizer with soil. • Fill 15- × 15-cm pots with the soil. Twice a week soak seeds of a susceptible variety such as IR8 and TN1 for germination, and sow them in the pots 2 days later at the rate of 75-100 seeds/pot. • Keep the potted plants in the greenhouse with adequate light for 10-15 days after sowing (DAS).
2. Collecting adults in the field to start the culture	• You can start a gall midge culture by two methods: — Collect adults in the field, or — Collect rice tillers with small galls from which adults have not yet emerged (Fig. 2G). • If field-collected adults are used, place them directly in the oviposition cage with 15- to 20-day-old seedlings. • If infested tillers are collected, place them in the adult emergence cage (Fig. 2F). When adults emerge, collect them with an aspirator and put them in the oviposition cage.
3. Egg production	• Place 30 females and 30 males in an oviposition cage containing 16 pots of 15- to 20-day-old seedlings. Infest two cages daily for routine rearing. • Provide the adults with fresh 15- to 20-day-old potted plants daily for oviposition.



2. Procedure for rearing the gall midge.

3. A mist chamber provides a damp environment which facilitates hatching and movement of the gall midge larvae down the leaf to the growing point.

Steps	Key points
	• After 2 days, transfer the potted plants with eggs to a mist chamber (Fig. 2C, 3) and replace the plants with new ones. Keep the plants in the mist chamber for 4 days. Moisture on the plant is necessary for egg hatching and for the newly emerged larva to move down the leaf to the growing point. • If a mist chamber is not available, transfer the plants to a tray filled with water and spray the plants with water every 2 h during the day for 2 days (Fig. 2D, 4). • After 4 days in the mist chamber, transfer the plants to a water tray (Fig. 2E) and raise the water level in the tray to cover the 2-3 cm of the basal part of the plants. This provides optimum humidity and prevents parasitization and predation of larvae.
4. Rearing of adults	• When galls are observed on the plants (about 16-20 days after oviposition), transfer the potted plants from the water tray to the adult emergence cage (Fig. 2F).
5. Collecting adults	• Collect adults every morning or between 1800 and 2100 h. The adults will concentrate near a light if the light is placed at one end of the cage and the other end is covered with black cloth. Carefully collect the adults with an aspirator.

4. Spraying of plants infested with gall midge eggs to facilitate egg hatching and movement of larvae down the leaf to the growing point.

Steps	Key points
	• Use the adults for varietal screening and for culture maintenance. • Transfer adults for culture maintenance to the oviposition cage and repeat steps 3-5.
6. Preventing inbreeding depression of culture	• Add field-collected adults to the culture at least once a year. This will help prevent inbreeding depression and a shift to an unfavorable sex ratio.

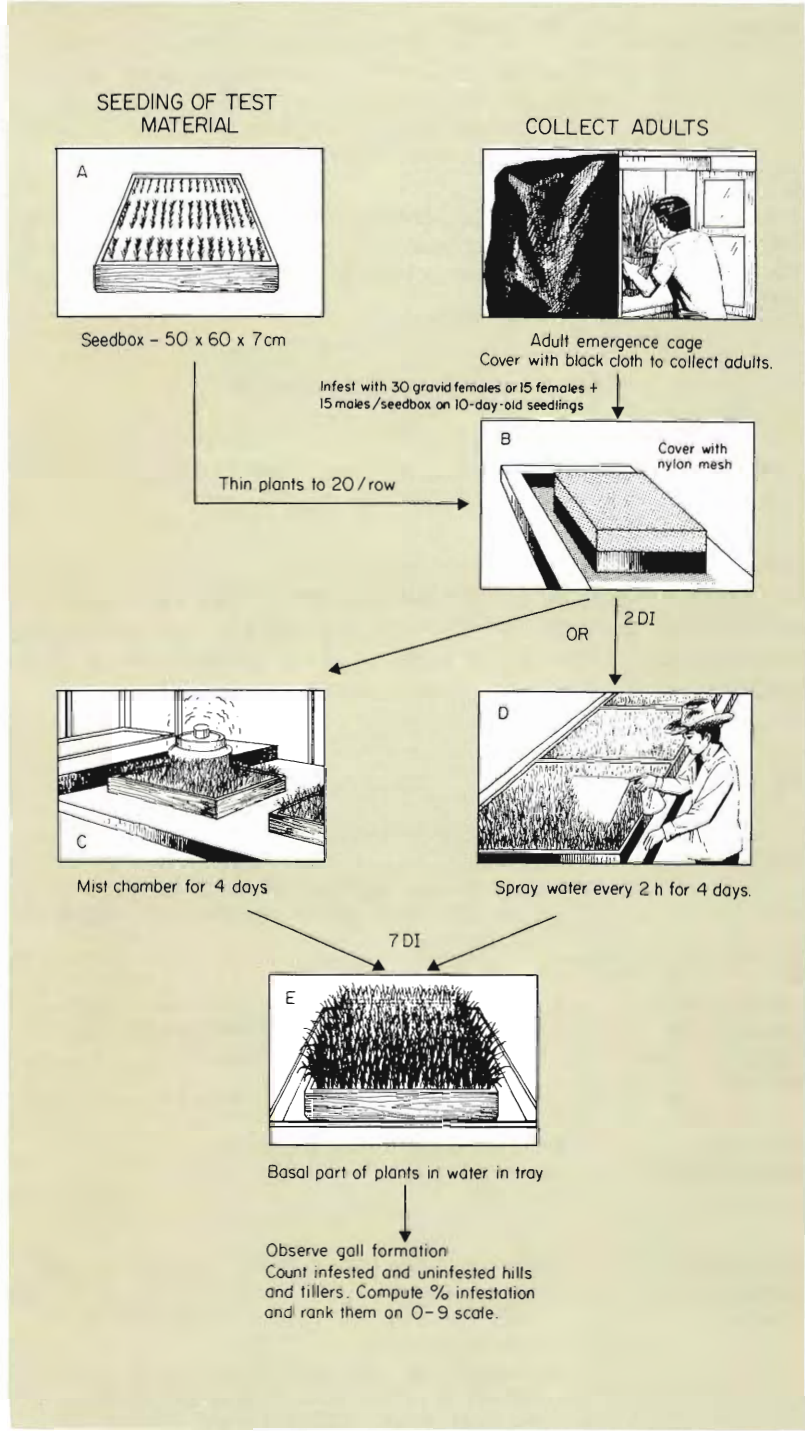
GREENHOUSE SCREENING

Efficient greenhouse screening methods have been developed and are used in China, India, Indonesia, Thailand, and Sri Lanka. The method used in Indonesia (Vreden and Arifin 1977) is described.

Seedling screening

The screening procedure is illustrated in Figure 5. Different methods are used to infest seedlings. Infesting with fertile eggs is cumbersome and time-consuming. Infesting with adults — either gravid females or both females and males (Fig. 5) — is most common.

Steps	Key points
1. Scheduling sowing and infesting	• Sowing and infesting depend on the availability of adults. • Time the sowing so that the seedlings will be 10-15 days old when adults intended for infestation emerge.
2. Listing and preparing all materials to be tested	• List all entries to be tested on the data sheets. • Arrange the envelopes containing test entries by number and experimental layout.
3. Preparing seedboxes for sowing	• Fill seedboxes with fertile soil.
4. Sowing seed of test entries and checks	• Make 3 15-row groups 18 cm long and 3 cm apart in a seedbox 50 × 60 × 7 cm (Fig. 5A). Of the 15 rows, 1 should be a susceptible check (IR8) and 1 a resistant check. • Varieties that can serve as gall midge-resistant checks at various sites considering reactions to the local biotype (IRRI 1981) are:



5. Procedure for the greenhouse screening of test entries for gall midge resistance. DI = days after infestation.

Steps	Key points	
	Biotype	*Variety as resistant check*
	China biotype	Eswarakora, Leuang 152, Ob 677, and their derivatives
	Indonesia biotype	Leuang 152, Siam 29, Muey Nawng 62M, Ob 677, and their derivatives
	Thailand biotype	Eswarakora and its derivatives
	India (Raipur biotype)	Leuang 152, Eswarakora, Siam 29, Ob 677, and their derivatives
	India (Andhra Pradesh biotype)	Leuang 152, Ptb 18, Eswarakora, Siam 29, Ob 677, and their derivatives
	India (Orissa biotype)	Leuang 152, Ptb 18, Siam 29, Ob 677, and their derivatives
	India (Bihar biotype)	Eswarakora and its derivatives (MR)
	Sri Lanka biotype	Leuang 152, Ptb 18, Siam 29, Ob 677, and their derivatives
5. Infesting seedlings with adults	• Thin plants to 20/row before infestation. • At 12-15 DAS, cover the seedlings with nylon mesh to prevent escape of adults (Fig. 5B, 6). • Infest each box with 30 females and 15 males.	
6. Maintaining the infested plants	• Transfer the seedboxes to a mist chamber 2 days after infestation (DI) (Fig. 5C). • If a mist chamber is not available, transfer the seedboxes to an open shallow tray and spray water every 2 h during the day for 2 days (Fig. 5D). • Five days later (or 7 DI), transfer the seedboxes to a water pan tray and raise the water level to 3 cm above the base of the plant (Fig. 5E). This will prevent parasitization and predation of the gall midge larvae.	
7. Evaluating	• At 20-30 DI, examine plants and determine total number of plants and number of infested and healthy plants. The test is considered valid when at least 60% of the plants of the susceptible check have galls. • Cut plants with no visible galls longitudinally and look for small galls. • Calculate percentage of infested plants: $\%\ \text{infested plants} = \frac{\text{no. of infested plants}}{\text{total no. of plants}} \times 100$	

6. Nylon mesh cage used in infesting test plants with ovipositing gall midge adults.

Steps	Key points
	• Convert percentage of infested plants to a 0-9 scale using the SES for rice:

Scale	*Plants with galls*
0	None
1	Less than 1%
3	1-5%
5	6-15%
7	16-50%
9	51-100%

Advantages of greenhouse screening

Greenhouse screening

- provides a controlled and uniform infestation,
- prevents parasitization of the larvae,
- can be conducted throughout the year, and
- allows screening of a large number of entries.

Disadvantage of greenhouse screening

In greenhouse screening, the laborious task of mass rearing is necessary.

FIELD SCREENING

Field screening for gall midge resistance should be in fields with high gall midge populations and should coincide with peak insect abundance as

indicated by previous observations. Methods to increase field populations have been developed by Prakasa Rao (1975). The field screening method described is that reported in IRRI (1981).

Steps	Key points
1. Selecting a *hot spot* and monitoring midge populations in the field to determine correct planting time	• Survey neighboring fields for the presence of larvae, pupae, and silvershoots. • Install light traps to determine the adult populations.
2. Scheduling land preparation, sowing, and transplanting	• As soon as the peak population incidence is estimated, schedule land preparation, sowing of test materials, and transplanting. • Transplanting should coincide with the peak of the gall midge population in the field.
3. Preparing a field layout	• Prepare the layout as a randomized complete block design if entries are replicated. In initial screening, one replication per entry is sufficient, but in retesting or biotype studies, entries should be replicated at least three times.
4. Transplanting	• Plant 2 adjacent 5-m rows of a test entry/replication. There should be 1 seedling/hill and 25 hills/row at 25-cm spacing between rows and 20 cm between hills. • Plant a row of susceptible and resistant checks after every 10 rows of test entries.
5. Attracting gall midge for natural infestation	• Install 100-watt electric lamps — 1 lamp/20 rows of test entries (10 test entries) or 25 m^2 — 3 m above the ground when the crop is 40 days old. Use the lights from 1900 h to 0400 h to attract midge adults to settle and oviposit on the test plants.
6. Maintaining midge populations in the field	• Maintain a constant water level in the field to provide high humidity. • Excessive nitrogenous fertilizer will help increase the field infestation of gall midge.
7. Evaluating screening reactions	• Record silvershoots twice — at 30 and 50 days after transplanting (DT). • Gather data on total number of hills, number of infested and healthy hills, total number of tillers, and number of infested and healthy tillers. The test is considered valid when at least 15% of the

Steps	Key points
	tillers of the susceptible check are infested (see *Considerations)* • Calculate percentage of plants and tillers damaged and convert the result to a 0-9 scale as in step 7 *Greenhouse screening.*
	• Considerations: If the percentage of damaged tillers is low at 50 DT, ratoon the crop. Gall midge larvae feed only at the tillering stage and ratooning will provide plants in the tillering stage. If insects are not sufficient to infest the crop, it is possible that after ratooning, the number of larvae may be enough for valid screening. Note that some varieties that do not form a visible gall may still have damage that causes distorted panicle formation. Thus, if a variety showing resistance is considered for use as a donor, dissect it to determine whether the shoot apex has been destroyed either by the formation of a small gall chamber or by necrosis (decaying of the primordial cells by a small amount of feeding by the larva). Varieties showing necrosis are not suitable donors because their potential yield will be affected if they are attacked.

Advantages of field screening

- Agronomic conditions can be more or less similar to those in farmer fields.
- Breeders' observations on plant type characters are possible.
- Natural infestation with a genetically heterogenous insect population is possible.
- Crop resistance to an insect other than gall midge or to a disease may be detected simultaneously.

Disadvantages of field screening

- If infestation is low, the research program is delayed for 1 year.
- Nontarget pests such as rats and insects may make readings on plant susceptibility impossible.
- The infestation pattern may be irregular and some susceptible entries may escape damage.
- A relatively large area is required and land preparation and maintenance costs are high.

MECHANISMS OF RESISTANCE

The nature and level of resistance to gall midge are determined through antixenosis for oviposition and antibiosis on larval survival and insect

development and population growth. Varieties being considered for use as donors in the breeding program and breeding lines being considered for release are likely candidates for studies on mechanisms of resistance. The methods described here are adapted from those used by Kalode et al (1977), Modder and Alagoda (1972), Perera and Fernando (1969), Fernando (1972), and Arifin (BORIF, Bogor, Indonesia, pers. comm., 1981).

Antixenosis for oviposition

Steps	Key points
1. Listing and preparing seed of test entries	• Select entries based on results of screening tests. • Include susceptible and resistant checks in the list.
2. Scheduling infestation	• Determine the period when adults are available for infestation.
3. Sowing seed of test materials	• Sow 10 seeds/variety in 1 row in a seedbox. • Use three or more seedboxes, one box representing one replication, and arrange entries in a randomized complete block design. Replicate each entry at least five times.
4. Infesting test materials	• Cover each box with a screen net. • Release gravid females in the box at 1 insect/5-7 plants.
5. Evaluating	• Count eggs laid on each plant 2 days after release of adults. • Compare results among entries.

Antibiosis on larval survival

Steps	Key points
1. Preparing test materials	• Choose varieties with different levels of resistance to the gall midge. Such varieties may be chosen from the screening results. • Secure seed of test materials including the susceptible check IR8, and a resistant check (see *Greenhouse screening*).
2. Sowing test materials	• Sow seed of the test varieties in 10-cm-diam pots filled with fertile soil at 5-7 seeds/plot. • Plant 5 pots/variety, 1 pot representing 1 replication. Replicate entries at least five times. • Place the pots in a tray with water at a level below the top edge of the pots.

Steps	Key points
3. Infesting test materials	• Collect eggs about to hatch in the culture cage by clipping off leaf portions with eggs, and place them on agar in a petri dish. • Place a thin film of water over the agar to facilitate egg hatching. • Use 10-14 newly hatched larvae to infest each pot of seedlings at the 5-6 leaf stage or 10-13 DAS. • Place the larvae on the leaf sheath at a height of 3-5 cm from the base of the plant using a soft hair pencil or camel hair brush. • Infest 3-4 larvae/seedling.
4. Maintaining infested plants	• After infesting plants transfer the pots to a water tray and raise the water level in the tray to 3 cm above the base of the plant. • Arrange pots in a randomized complete block design.
5. Evaluating	• At 10-12 DI count the surviving larvae on the plants. • Dissect plants and look for small galls. • Compute survival percentage: $\%\ \text{survival} = \frac{\text{no. of live larvae counted}}{\text{no. of larvae infested}} \times 100$ • Tabulate data and analyze results.

Antibiosis on population growth

Steps	Key points
1. Preparing test materials	• Choose varieties to use based on screening results. • Secure seed of test materials.
2. Sowing seed	• Sow seed of test varieties at 225 seeds/pot in 20-cm-diam pots filled with fertile soil. • Replicate each entry at least five times. A pot represents one replication.
3. Infesting test materials	• At 10-13 DAS, enclose each pot with nylon net or a mylar film cage. • Infest each pot with 20 gravid females. • Remove the cage 48 h after release of adults.
4. Maintaining infested plants	• Transfer the pots to a mist chamber for 4 days to allow hatching of eggs and migration of larvae to

Steps	Key points
	the shoot apices. If a mist chamber is not available, transfer pots to the water tray. Spray the plants with water every 2 h during the day for 2 days. • Return potted plants to their respective cages and maintain in a water tray.
5. Evaluating	• When adults begin emerging (about 17 DI), remove and count daily until emergence is completed. • Determine the total number of adults emerging from each entry and compare results among entries.

SOURCES OF RESISTANCE

Rice varieties reported as having resistance to the gall midge in various screening programs as indicated in the literature from 1955 to 1980 are reported by Pathak and Heinrichs (1982).

Variety	Origin[a]	Reference
Al	India	Krishna (1977)
AC1423	India	Shastry and Seshu (1971)
ARC5810	India	Shastry et al (1971)
ARC5833, 5834	India	Kalode (1980)
ARC5838, 5842	India	Shastry et al (1971)
ARC5848, 5911, 5912, 5918, 5939	India	Kalode (1980)
ARC5947A, 5949, 5951	India	Shastry et al (1971)
ARC5959	India	CRRI (1974)
ARC5984	India	Shastry et al (1971)
ARC5987	India	CRRI (1974)
ARC5988, 6001	India	Kalode (1980)
ARC6010, 6014, 6087	India	Shastry et al (1971)
ARC6103	India	Kalode (1980)
ARC6136, 6140	India	Shastry et al (1971)
ARC6157	India	Kalode (1980)
ARC6158, 6163, 6202, 6210, 6221, 6232, 6234, 6238, 6557, 6605A	India	Shastry et al (1971)
ARC6606, 6607, 6618, 6619	India	Kalode (1980)
ARC6631	India	Shastry et al (1971)
ARC6632	India	CRRI (1974)
ARC7004, 7077, 7138, 7213	India	Shastry et al (1971)
ARC7255, 7292, 7293	India	Kalode (1980)
ARC7308, 7316, 7317, 7318, 7329	India	Shastry et al (1971)
ARC10040, 10227	India	Kalode (1980)
ARC10272, 10295	India	Shastry et al (1971)
ARC10331	India	Kalode (1980)
ARC10345	India	Shastry et al (1971)
ARC10360, 10377	India	Kalode (1980)
ARC10395	India	Shastry et al (1971)
ARC10460	India	Kalode (1980)

Variety	Origin[a]	Reference
ARC10494	India	Shastry et al (1971)
ARC10520	India (Indonesia)	Soehardjan et al (1974)
ARC10534	India	Shastry et al (1971)
ARC10557, 10627	India	Kalode (1980)
ARC10654	India	Shastry et al (1971)
ARC10659, 10660	India	Kalode (1980)
ARC10817	India	Shastry et al (1971)
ARC10932-2	India	Soehardjan et al (1974)
ARC10963, 11210, 11307	India	Shastry et al (1971)
ARC11704	India	Kalode et al (1977)
ARC13500, 13516, 13564, 13902, 13929, 14148, 14378, 14421, 14549, 14725, 14748, 14787	India	CRRI (1974)
ARC14950B	India	Krishna (1977)
ARC15067, 15151, 15159, 15905, 17789, 18595 18596, 18601	India	CRRI (1974)
Bainsa	India	Sen (1957)
Chemban, Chennellu, Chenni Nayakan	India	Kalode et al (1977)
Chuvanna Kumbolan	India	Krishna (1977)
Dahanala 37	India	CRRI (1964)
DNJ45, DV12	India	Shastry and Seshu (1971)
Eswarakora	India	Venkataswamy (1966)
Hochin	India	CRRI (1964)
HR14	India	Khan and Murthy (1955)
HR42	India	Krishnamurthy Rao and Krishnamurthy (1964)
HR63	India	Venkataswamy (1968)
HR64	India	Krishnamurthy Rao and Krishnamurthy (1964)
JB446 (Desi Bayahunda)	India	Shastry and Seshu (1971)
JBS673 (Ratnachudi)	India	Bhat et al (1958)
JBS990, 1224	India	Seshu et al (1974)
Kakatiya	India	IRRI (1977)
Kurutha Vellathan	India	Kalode et al (1977)
Kalijira	India (Bangladesh)	BRRI (1979)
Leuang 152	Thailand (India)	Sastry et al (1975)
Malalwariyan	India	Krishna (1977)
MNP7, 14, 14A, 15, 30, 62, 336, 380B, 448, 457	India	Seshu et al (1974)
MNP471	India	Sastry and Prakasa Rao (1976)
MNP753, 800, 837	India	Seshu et al (1974)
Muey Nawng Fang, Muey Nawng 62, Muey Nawng 62M	Thailand	Kovitvadhi (1963)
Muey Nawng 6	Thailand	Ou and Kanjanasoon (1961)
Neto	India	Sen (1957)
Ob 677, 678	Sri Lanka	IRRI (1977)
Pandi, Parakulam	India	Kalode et al (1977)
Peykeo E53, Peykeo P129	India	CRRI (1964)
Ptb 10	India	Thomas and Chacko (1968)
Ptb 12	India	Kalode et al (1977)
Ptb 18	India	CRRI (1965)
Ptb 19	India	Kalode et al (1977)
Ptb 21	India	CRRI (1965)

Variety	Origin[a]	Reference
Ptb 27	India	CRRI (1964)
Ptb 28, 32	India	Khush (1977)
Siam 29	Thailand (India)	Riley (1881)
T10, 16	India	Kalode et al (1977)
T405, 1162	India	Kalode (1980)
T1421, 1425	India	Kalode et al (1977)
T1426	India	Kalode (1980)
T1432, 1471	India	Kalode et al (1977)
T1479, 2587	India	Kalode (1980)
Valsara Champara	India	Krishna (1977)
Vella Chenipan, Vellathil Cheera, Velutha Chera	India	Kalode et al (1977)
W1251, 1253, 1257, 1263	India	AICRIP (1967)
710	India	Kalode et al (1977)
Wild rice species *O. brachyanta, coarctata, eichingeri, granulata, ridleyi*	India	Israel et al (1963)

[a]The name in parentheses indicates the country where the test for resistance was conducted, if different from the origin of the variety.

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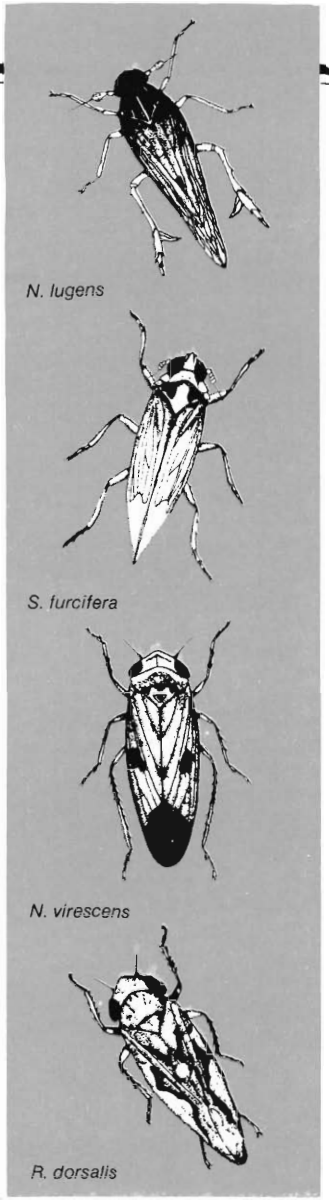
Chapter 9
BROWN PLANTHOPPER, WHITEBACKED PLANTHOPPER, GREEN LEAFHOPPER, AND ZIGZAG LEAFHOPPER



The hoppers are serious pests of rice throughout Asia. The brown planthopper (BPH) *Nilaparvata lugens,* whitebacked planthopper (WBPH) *Sogatella furcifera,* green leafhopper (GLH) *Nephotettix virescens,* and zigzag leafhopper (ZLH) *Recilia dorsalis* feed on rice plants. In addition they also transmit virus diseases that often cause more severe yield reduction than the feeding damage. Because of the economic importance of the hoppers, rearing and varietal resistance screening programs are well established in many countries. Various sources of resistance have been identified and numerous commercial varieties have been released. Because of the selection for biotypes within hopper populations, major emphasis is on the development of additional varieties with new genes for resistance. The methods described here can be used for other hopper species including the rice delphacid *Sogatodes orizicola,* a vector of hoja blanca virus in Central and South America; and the small BPH *Laodelphax striatellus,* a vector of rice striped virus and black-streaked dwarf in China, Japan, and Korea.

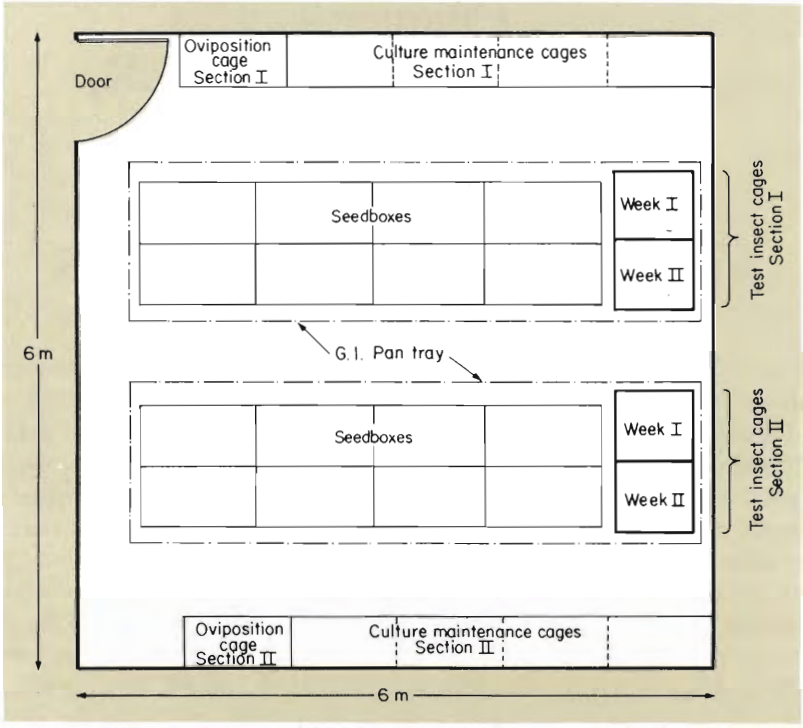
REARING METHODS

The hopper species have been reared extensively throughout Asia. Methods vary in respect to the age of food plants used, type of rearing cages, etc. The methods used to rear hopper species at IRRI are described.

Construction and fabrication of equipment

A greenhouse and rearing cages are the major items required for rearing hoppers. Both insect rearing and screening rice for resistance are done in the greenhouse.

Steps	Key points
1. Constructing a greenhouse	• The greenhouse should be well lighted and should provide a suitable environment for growing plants throughout the year. • Divide the greenhouse into rooms with screen walls (Fig. 1). The various hopper species and biotypes of a species are reared in separate rooms. Screening of test entries against the



1. Diagram of a room for rearing hoppers and for screening rice for resistance to hoppers. The room has two sections (I and II), each consisting of oviposition, culture maintenance, and test-insect rearing cages. Each section provides sufficient insects for the seedboxes in one water tray.

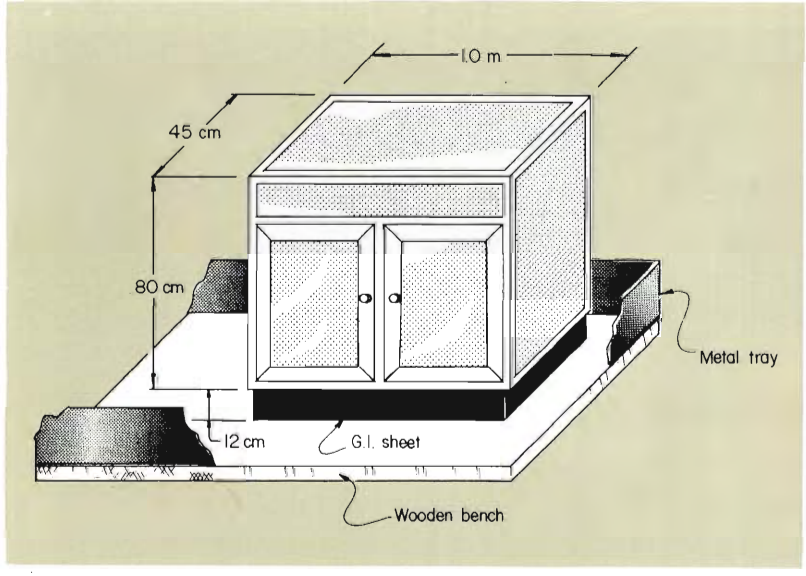
Steps	Key points
	various hopper species and biotypes should be in the same room where the hopper is reared. • A screenhouse (screened roof and sides) can be used instead of a greenhouse (glass roof) during the dry season in the tropics. Some national programs use an open-sided structure with palm leaf roof (Fig. 2). However, plant growth is poor because of low light intensity and plants can remain in the house for only a short period.
2. Constructing a soil bin	• Storing soil near the greenhouse increases the efficiency of the rearing and screening program (Fig. 3). During the wet season when field-collected soil is too wet to use, stored soil becomes a necessity. The soil bin should hold sufficient soil to support the rearing and screening programs throughout the wet season.
3. Obtaining tools	• A pushcart (Fig. 4) and a wheelbarrow are necessary for transporting soil, potted plants and cages for the rearing program, and seedboxes and soil for the screening program.

2. Palm leaf roofed structure used for rearing hoppers and screening plants for hopper resistance.

3. Bin for storing soil. The wire screen sieve is used for pulverizing large soil particles for seedbox screening.

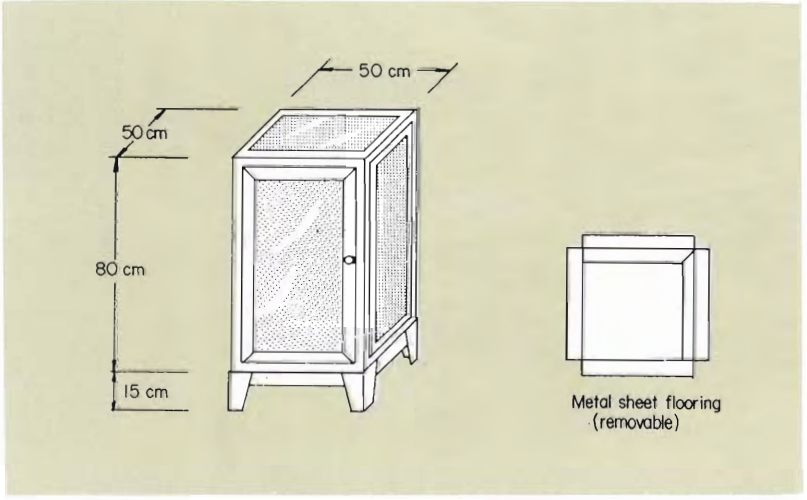
4. Pushcart for moving plant material.

Steps	Key points
4. Constructing rearing cages	• Cages made of water-resistant wood are more durable than steel cages because the latter rust rapidly in reaction to fertilizer and frequent watering. • Hoppers are usually reared on rice plants in one of two types of cages.

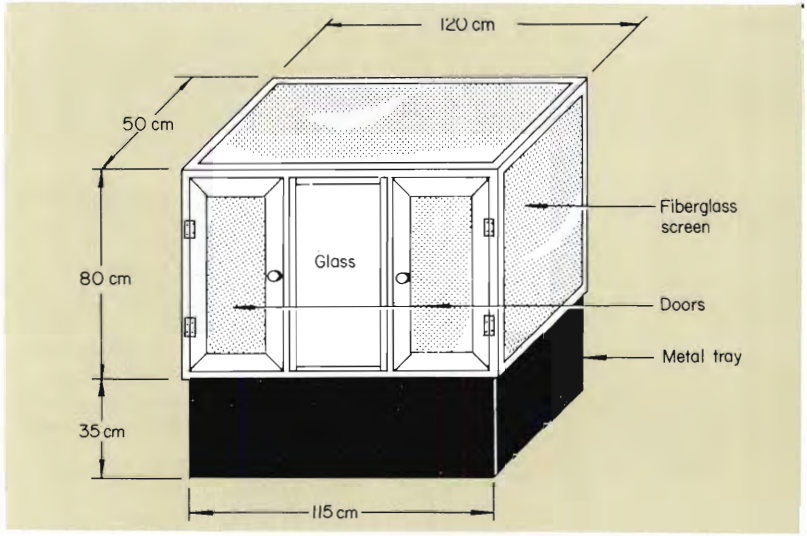


5. Bottomless hopper-rearing cage on a galvanized iron tray. Potted plants sit in about 8 cm of water inside the cage.

Steps	Key points
	— A bottomless cage placed in a galvanized iron water pan tray (Fig. 5) is used primarily at IRRI. The plants sit in water and do not require daily watering. This cage is used primarily for BPH and WBPH. — A rearing cage with four legs and a closed bottom (Fig. 6) can be used. This cage is placed on a table and the potted plants in it must be watered daily. The legs of the table sit in a pan tray with water to prevent ants from entering the cage. Because the insects are not exposed to as high moisture as in the bottomless cage, this cage is used for the GLH and ZLH, which are often attacked by the fungus *Metharrhizium anisopliae*. The cage can also be used for BPH and WBPH.
5. Constructing an insect-transfer cage	• To facilitate the transfer of planthoppers from old to fresh potted plants, a transfer cage is useful (Fig. 7). — The cage holds 30-cm-deep water. — Place old potted plants with hoppers in the cage and slowly add water. — The hoppers move from the base of the plants to the leaf portion and are easily transferred to fresh plants in the rearing cage by tapping the leaves over the fresh plants.
6. Constructing water pan trays	• Keep seedboxes in water pan trays (Fig. 8) to decrease the amount of watering required.



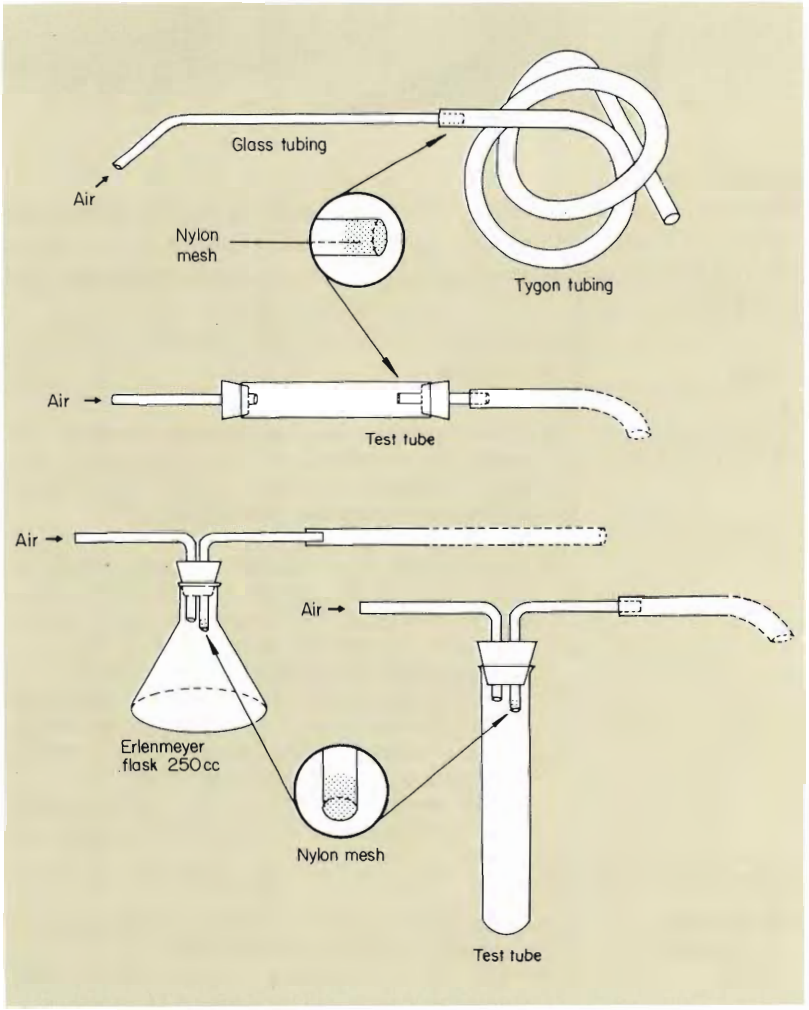
6. Common hopper-rearing cage. Hoppers are reared on potted plants in the cage.



7. Cage used to facilitate the transfer of brown planthoppers. Plants are placed in the metal tray and the water level is raised to cover the base of the plants, forcing the hoppers to move up to the leaves.

8. Seedboxes in water pan trays. Rearing cages in a water pan tray are seen in the background.

Steps	Key points
7. Obtaining mouth aspirators or a suction machine	• Aspirators (Fig. 9) are useful for handling of insects during field collection, removal of predators in the rearing program, and handling of hoppers in studies of mechanisms of resistance. The suction machine (Fig.. 10), although more expensive, is more efficient than the mouth aspirator.
8. Purchasing clay pots	• Clay pots are used in growing test plants for studies on mechanisms of resistance, and food plants for insect culture.



9. Aspirators used in handling leafhoppers and planthoppers.

10. Suction machine for collecting hoppers.

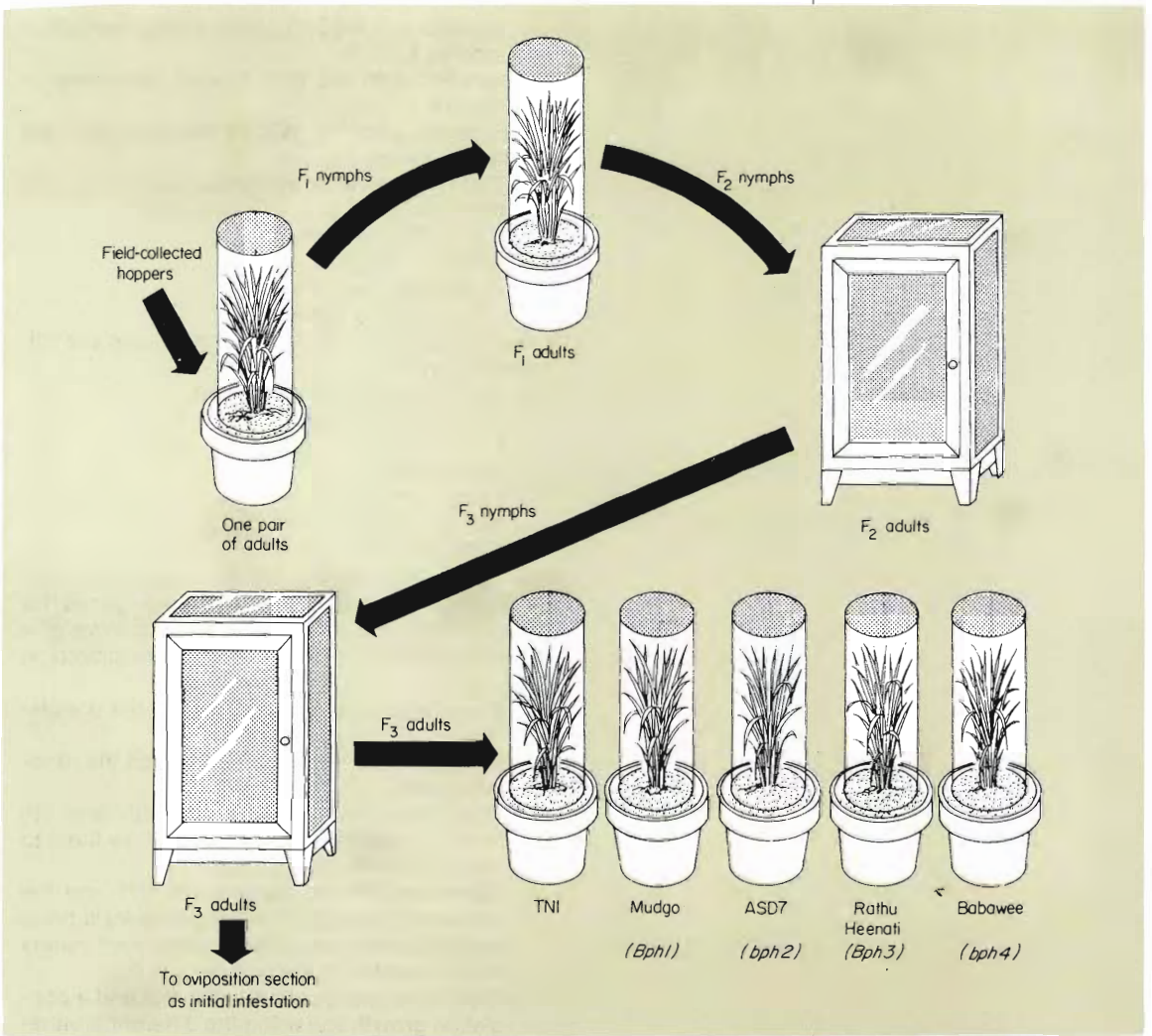
Source of insects

The culture is usually started by collecting hoppers in the field. If a hopper species is not present locally, it can be imported if quarantine regulations allow it. If the hopper is sent long distances, it must be in the egg stage and sent by air.

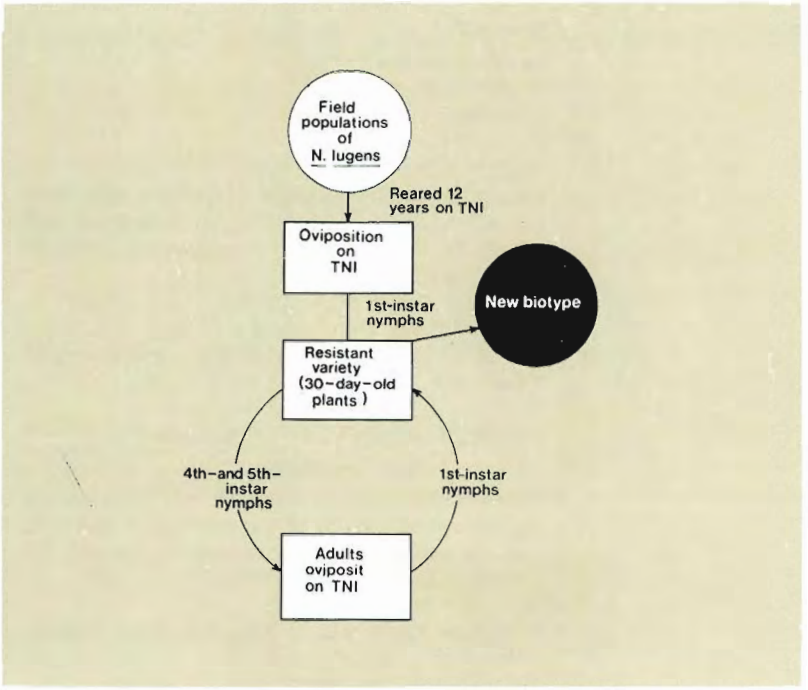
Steps	Key points
1. Collecting hoppers to start the culture	• Collect nymphs and adults from infested rice fields. Use an aspirator if the population is very low. If a large population is present, tap infested plants over a container or an insect net. • Collect adults at lights. • If the hopper is not available locally, import it. Many scientists throughout Asia are rearing BPH, WBPH, and GLH and cultures can be obtained from them. To prepare for hopper shipment — expose plants to gravid females for 24 h; — remove the females, trim the leaves, wash soil from the roots, and wrap roots in moist cotton; — place plants in a polyethylene bag or plastic vials plugged with dry cotton; and — ship immediately so that the material reaches its destination within 7 days after the eggs are laid.
2. Eliminating viruliferous hoppers	• Cage potted plants of the weed *Monochoria vaginalis,* a nonhost of rice viruses. • Place gravid females on the plants. Before they starve the insects will lay eggs on *M. vaginalis.*

Steps	Key points
	• Six days after the females are placed on *M. vaginalis,* introduce young, virus-free potted seedlings of a susceptible rice variety such as Taichung Native 1 (TN1). Nymphs emerging from *M. vaginalis* will feed on TN1. These nymphs are free of rice virus diseases, which are not transovarially transmitted.
3. Identifying biotype reactions	• Isolate 10 pairs of hoppers separately by caging on TN1 (Fig. 11).

11. Method used to determine the biotype of field-collected brown planthoppers. Two pairs (male and female) of newly emerged F_3 adults are placed on potted plants of differential varieties and population growth is determined. F_3 adults are also transferred to the oviposition section (Fig. 13) to start the culture.



Steps	Key points
	• Place F_1 and F_2 progeny of each culture on fresh TN1 plants. • Test the F_3 using the seedbox screening test and population growth study for the BPH on the various differential varieties given in Figure 11. The insect can be classified into biotypes if they develop a population (about 250/female) on and kill any of the varieties below: — TN1 only = biotype 1 — varieties with the *Bph 1* gene (Mudgo, IR26, IR29, IR30, and IR34) = biotype 2 — varieties with the *bph 2* gene (ASD7) = biotype 3 — varieties with the *Bph 3* gene (Rathu Heenati) = biotype 4 — varieties with the *bph 4* gene (Babawee) = biotype 5 • For identification of WBPH biotypes, use the following varieties: — TN1 = no gene for resistance — N22 = *Wbph 1* — ARC10239 = *Wbph 2* — ADR52 = *Wbph 3* — Podiwi-A8 = *wbph 4* — N'Diang Marie = *Wbph 5* • For identification of GLH biotypes, use the following varieties: — TN1 = no gene for resistance — Pankhari 203 = *Glh 1* — ASD7 = *Glh 2* — IR8 = *Glh 3* — Ptb 8 = *glh 4* — ASD8 = *Glh 5* — TAPL No. 796 = *Glh 6* — Moddai Karuppan = *Glh 7* • If survival on varieties with known genes for resistance is too low for starting a culture of a desired biotype, use the scheme illustrated in Figure 12. — Place insects on TN1 to increase the population. — Place them as 1st-instar nymphs on the resistant variety. — When the survivors reach the 4th and 5th instars, place them on TN1 and allow them to become adults and lay eggs. — Continue alternate feeding on TN1 and the resistant variety until there is sufficient survival and population growth on the resistant variety alone to maintain the culture. — Conduct a seedbox screening test and a population growth test using the differential varie-



12. Method used to select for a brown planthopper biotype that is virulent on a previously resistant variety (Pathak and Heinrichs 1982).

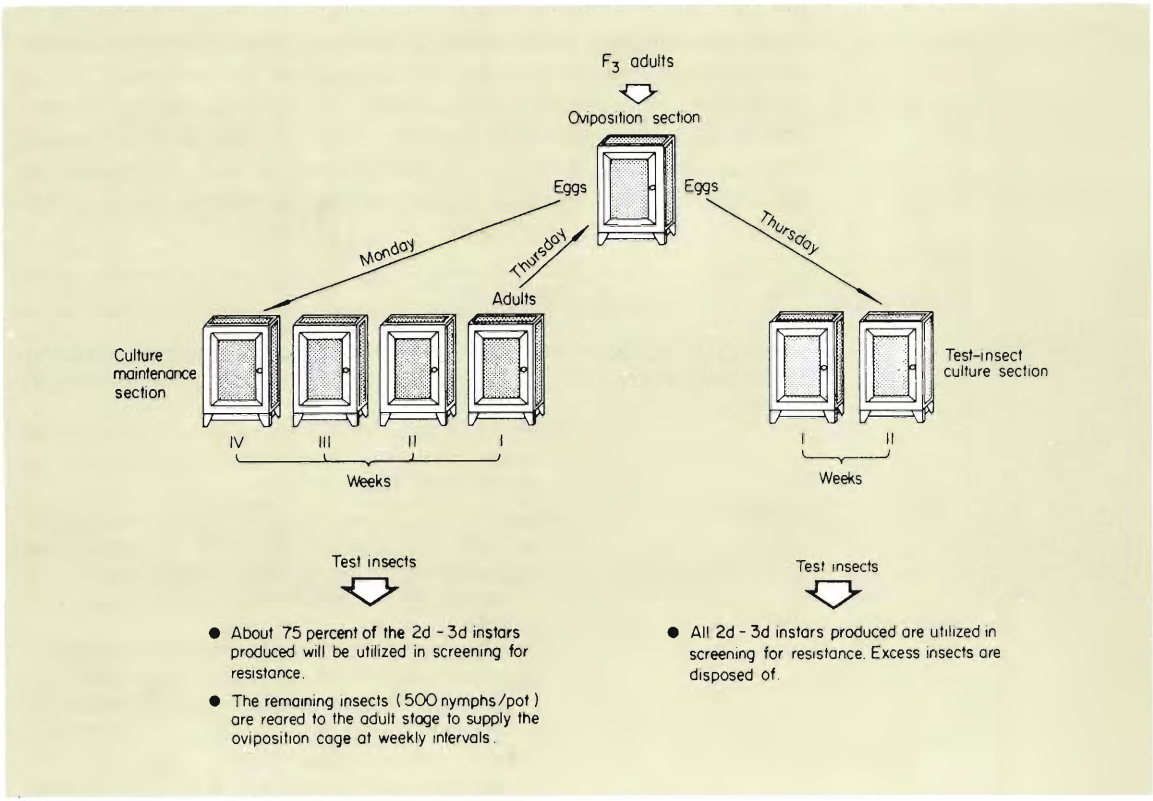
Steps	Key points
	ties. Compare the selected population and that reared on TN1 (check). When growth of the hoppers on the previously resistant variety is equal to that on the susceptible TN1, the population is considered a biotype that is virulent on the variety.

Food source for hoppers

A continuous supply of plants should be available for mass rearing of hoppers.

Steps	Key points
1. Estimating number of pots to be planted per week for rearing test insects	• The number of pots required is based on the number of insects required each week. Calculate the number of required insects by the following formula: Seedboxes to be screened per week × entries (compartments) per seedbox × seedlings/compartment × insects/seedling. • The approximate number of insects per seedling is as follows: — BPH biotype 1 = 10

Steps	Key points
	— BPH biotype 2 = 10 — BPH biotype 3 = 10 — GLH = 3 — WBPH = 8 — ZLH = 3 • Example: For BPH biotype 1, 4 seedboxes, each containing 252 compartments, are evaluated each week. At 20 seedlings/compartment and 10 insects/seedling: $4 \times 252 \times 20 \times 10 = 201{,}600$ 201,600 insects are required. • About 5,600 nymphs hatch from the plants in 1 pot. • $\frac{\text{201,600 2d- and 3d- instar insects required}}{\text{5,600 nymphs/pot}} = 36$ pots • For every two pots with egg-infested plants, add a pot with fresh plants to the test-insect cages to provide sufficient plant material for rearing the insects to the 2d and 3d instar. • 36 + 18 = 54 pots. • If hoppers are reared to the adult stage, change pots twice (once in the 4th and once in the 5th instar) before the insects become adults.
2. Estimating the number of pots to be planted weekly to supply the culture maintenance section	• Transfer egg-infested potted plants from the oviposition cage to a culture maintenance cage weekly (Fig. 13). The hoppers require about 14 days to become adults so replace one-half of the pots with fresh potted plants twice a week. Thus, for the common rearing cage (Fig. 6) which holds 9 pots, 72 pots are required per week: 9 for each of the 4 cages plus 36 pots of egg-infested plants for a total of 72 pots.
3. Growing food plants	
a) for ZLH	• TN1 seedlings 8 to 10 days old are used to rear the ZLH. — Plant twice a week. — Fill 12-cm-diam clay pots with soil and sow about 250 seeds/pot. — Water gently with a sprinkler twice a day.
b) for BPH, WBPH, and GLH	• To grow food plants to 50 days old, sow about 350 seeds — TN1 for BPH biotype 1, WBPH, and GLH; Mudgo for BPH biotype 2; and ASD7 for BPH biotype 3 — in 12-cm-diam clay pots filled with soil. When seedlings are 10 days old, transplant 4 seedlings into the clay pots. Place the pots in a water tray inside a screened room. If the tray will be placed outside the room, enclose it with a net



13. Test-insect production for screening tests. Arrows indicate movement of hoppers to various sections.

Steps	Key points
	to protect the plants from insect pests and the culture from parasites and predators. If infestation still occurs, remove the pests immediately by splashing the plant with water and by gently tapping each potted plant to dislodge the arthropods. • Fertilize the plants as necessary. • Remove lower leaf sheaths 8 days before placing the potted plants in the rearing cages to eliminate possible hopper eggs. — Just before placing the potted plants in the rearing cages, remove any arthropods present by splashing the plants with water.

Changing food plants and movement of insects

Cages are grouped according to function:

- oviposition,
- culture maintenance, and
- test insect (Fig. 13).

For each cage holding 9 pots in the oviposition section, you need 4 culture maintenance cages to produce about 18,000 test insects/day 4 days a week. The required number of test insect cages depends on the age to which the test insects must be reared for specific studies. The common rearing cage can hold 9 12-cm-diam pots, each pot containing 4 to 5 plants at the booting stage as food for the hoppers. The changing of food plants and the movement of the insects as in rearing the BPH at IRRI is described. The method is the same for the WBPH, ZLH, and GLH.

Steps	Key points
1. Starting the culture in the oviposition cage	• To start the culture place 9 (common rearing cage) or 18 (bottomless cage) pots with plants in the oviposition cage. • Expose plants for 3 days to about 2,000 gravid females selected from the F_3 of the biotype identification study (Fig. 11).
a. BPH	• Remove the potted plants containing ovipositing hoppers and hopper eggs from the oviposition cage and place them in the transfer cage (Fig. 7). — Slowly fill bottom of the transfer cage with water. — While waiting for the bottom to fill with water, place nine pots of fresh plants into the oviposition cage. There should be at least 10 tillers/pot. — Next remove plants from the transfer cage and return them to the oviposition cage. Hold the old plants with hoppers over the fresh plants and tap the leaves gently to dislodge the hoppers from the leaves onto the fresh plants. — Infest the fresh plants with 15-20 female hoppers/tiller. With an aspirator, blow off or remove insects that remain on the potted plants after tapping. • Place in the culture maintenance cage the egg-infested plants from which the hoppers were removed. • If a transfer cage is not available, you can use the following method described for WBPH, GLH, and ZLH.
b. WBPH, GLH, and ZLH	• Adult GLH and ZLH are sensitive, active insects. At a slight disturbance inside the cage they fly and jump in all directions, especially toward the open door or any source of light. Use black cloth to cover the GLH or ZLH cages when transferring egg-infested plants from the oviposition to the culture maintenance cage and when placing fresh plants in the cage for oviposition. • The black cloth is not necessary for WBPH, which is not as active. — Cover the cage door with black cloth before opening it.

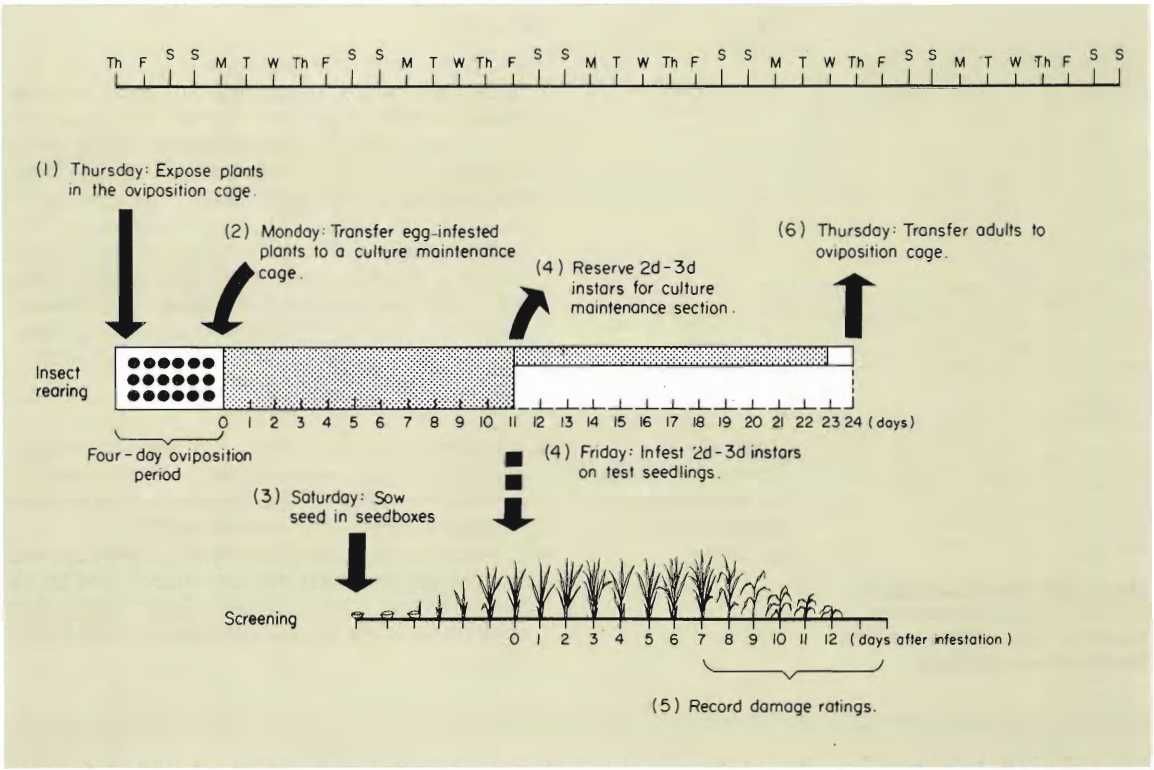
Steps	Key points
	— Tap the plants so that the hoppers will leave the plants and move to the cage walls. — Remove the egg-infested plants from the oviposition cage and transfer them to the culture maintenance cage. — Place fresh 50-day-old plants in the oviposition cage.
2. Rearing insects in the culture maintenance cage	• The culture maintenance cage is used to rear insects to the adult stage to supply adults back to the oviposition cage and maintain the culture. It is also used to rear 2d- to 3d-instar nymphs as test insects for mass screening tests. These insects (2d-3d instars) are ready for screening tests 11 days after egg-infested plants are introduced from the oviposition to the culture maintenance cage.
a. BPH	• Change plants in the culture maintenance cages twice a week. Remove about half of the pots each time and replace them with fresh potted plants. — Slowly remove all dried and wilting potted plants together with hoppers from the cage. — Arrange the remaining healthy potted plants on one side of the cage and put fresh potted plants in the vacated area. — Return the dried and wilted potted plants with the hoppers to the culture maintenance cage and tap the plants gently to dislodge the hoppers and distribute them onto the fresh potted plants. • About 24 days after the eggs are placed in the culture maintenance cage, the insects will have developed to the adult gravid stage and will be ready for transfer to the oviposition cage. To transfer the adults to the oviposition cage use the transfer cage method described in step 1 for BPH.
b. WBPH, GLH, and ZLH	• Change plants in the culture maintenance cages twice a week. — Cover the cage door with black cloth before opening it. — To add fresh plants, slightly lift the black cloth and lift the pots with wilting and dried leaves. Tap the plants gently to dislodge the hoppers before removing the plants from the cage. The hoppers will move to the cage walls or to the other plants in the cage. — When all the wilted plants have been removed, place the remaining plants on one side of the cage and put the fresh plants in the vacated area. The hoppers will redistribute themselves on the fresh plants. • To transfer hopper adults from the culture maintenance cage to the oviposition cage

Steps	Key points
	— Tap the plants to dislodge the insects, leave the insects in the cage, and remove all the potted plants. — Individually collect the hoppers off the walls of the cage with a mouth aspirator or suction machine and transfer them to the oviposition cage. Remove all predators, parasites, and other arthropods present in the culture. • Another way of transferring adults from the culture maintenance cage to the oviposition cage is to: — Move the culture maintenance cage next to the oviposition cage. — Open the doors of both cages and join the two cages at the door opening. — Cover the culture maintenance cage containing the adults with black cloth. — Tap the sides of the culture maintenance cage. The adults will transfer from the dark culture-maintenance cage to the light oviposition cage. — Collect any hoppers that remain with a mouth aspirator or suction machine.
3. Rearing insects in the test-insect cage	• Once the culture is established and sufficient egg-infested plants are produced to supply the culture maintenance cages, the excess egg-infested plants from the oviposition cage can be used to supply the test-insect cages. Insects in these cages are reared to 2d- to 3d-instar nymphs, for screening tests or other specific studies.

Weekly cage rotational schedule

When the F_3 adults from the biotype identification study become available (Fig. 11), the mass rearing program can be started (Fig. 13). The rearing system used at IRRI is based on a 5-day work week and is explained on that basis. Specific days of the week are given for illustrative purposes only and the program can be modified to suit the specific needs of any varietal resistance program.

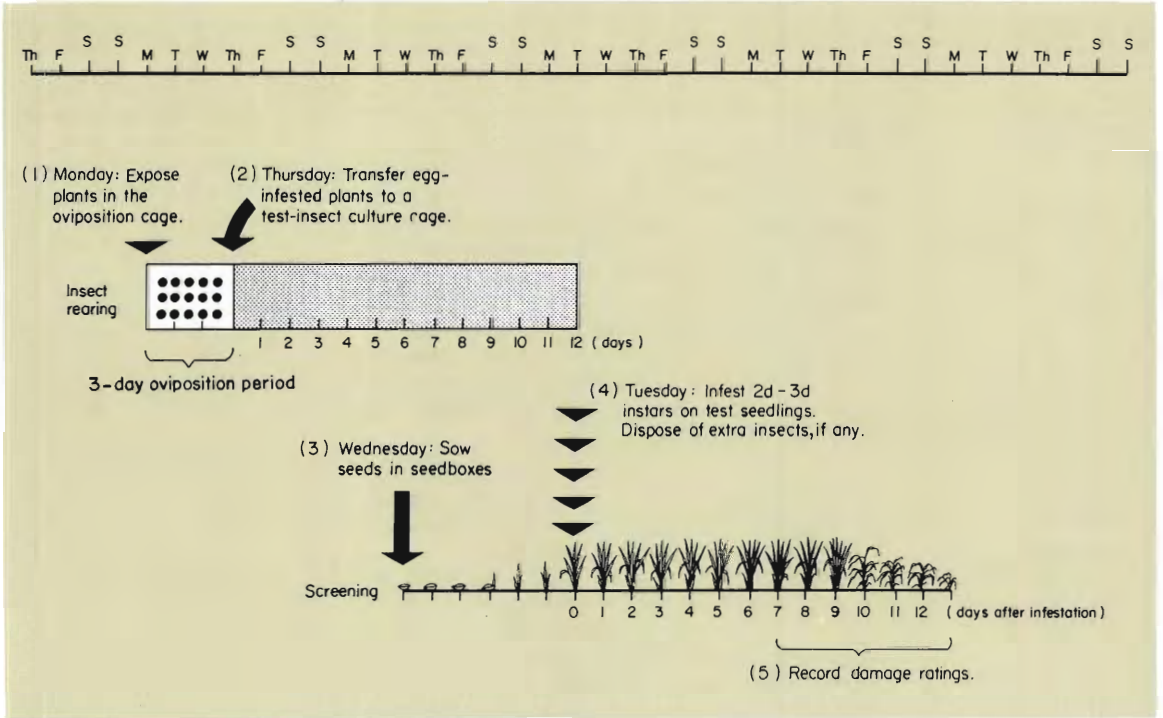
Steps	Key points
1. Maintaining the culture	• A complete cycle of egg to egg takes about 4 weeks. For each oviposition cage four culture maintenance cages are required: one cage is filled with egg-infested plants each week. • Expose plants in the oviposition cage to ovipositing hopper females on Thursday (Fig. 13, 14). On Monday (after a 4-day oviposition period) transfer the egg-infested plants to a culture main-



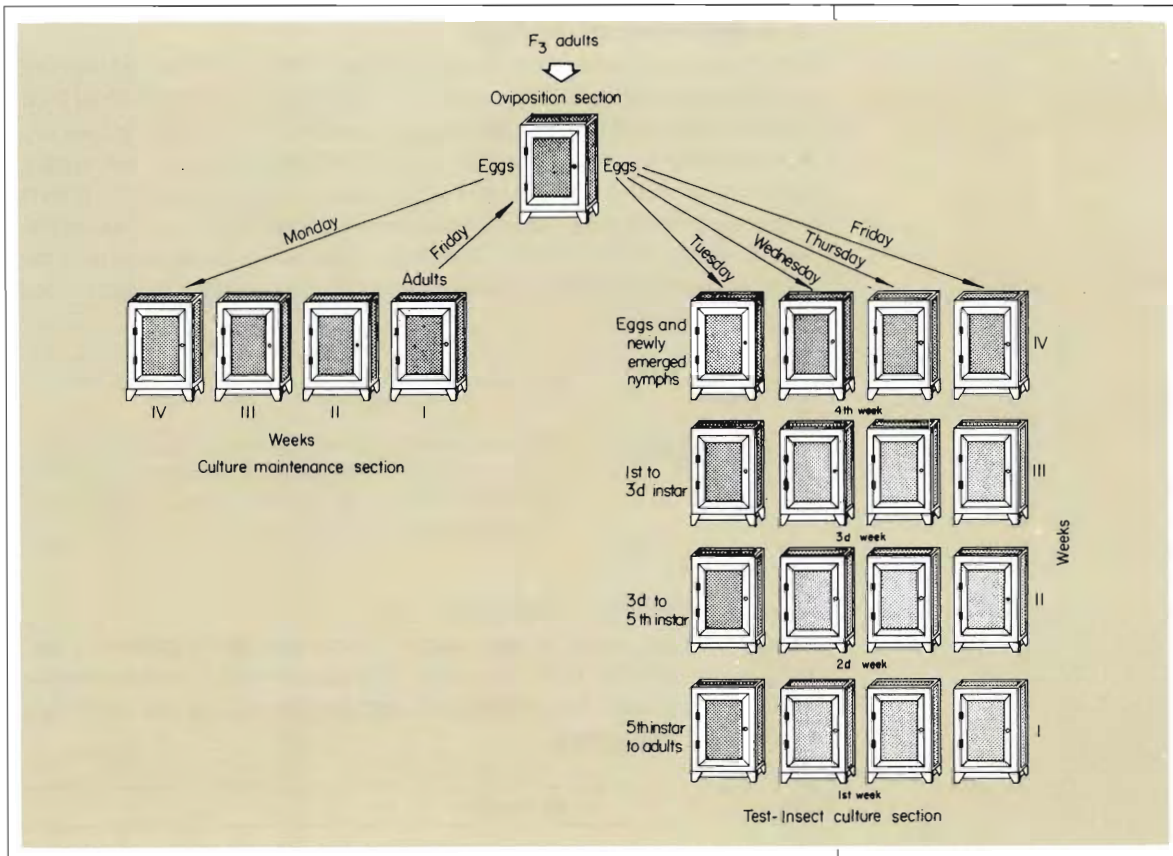
Steps	Key points
	tenance cage. Because it takes about 20 days between the time the eggs are laid and the adult stage, transfer the adults to the oviposition cage the fourth Thursday (24 days) after the egg-infested plants from the oviposition cage are placed in the culture maintenance cage. Thus, the culture maintenance cage that receives egg-infested plants from the oviposition cage on week 1 (on Monday) (Fig. 13) has adults ready for transfer to the oviposition cage on Thursday, 24 days later (Fig. 14).
2. Rearing test insects for screening	• For each oviposition cage two test-insect cages are required (Fig. 13). Fill one cage with egg-infested plants Thursday of each week. • A complete cycle to produce 2d- to 3d-instar nymphs takes 2 weeks. • When egg-infested plants from the oviposition cage are moved to the culture maintenance cage on Monday, place a fresh batch of plants in the oviposition cage on which the adults in the cage

14. Schedule of activities related to culture maintenance and screening. Circled numbers indicate procedural steps in chronological order.

Steps	Key points
	will again feed and lay eggs (Fig. 13, 15). On Thursday after a 3-day oviposition period, transfer the egg-infested plants to a test-insect culture cage. There are 12 to 15 days between the time the eggs are laid and the 2d- to 3d-instar stage. Thus, the nymphs are ready for use as test insects for screening the second Tuesday (12 days) after the egg-infested plants from the oviposition cage are placed in the test-insect culture cage. Thus, the test-insect culture cage that receives egg-infested plants on week 1 (Thursday) has nymphs ready for infestation on plants in seedboxes on Tuesday, 12 days later (Fig. 15, step 4).
3. Rearing test insects for studies on mechanisms of resistance and screening	• If a large screening program is maintained and insects of various ages are regularly required for studies on mechanisms of resistance, the scheme illustrated in Figure 16 can be used. • The required number of test-insect cages depends on the age to which the test insects are to be reared before being utilized. Each oviposition cage requires the following number of test-insect cages:



15. Schedule of activities related to test-insect culture and screening. Circled numbers indicate procedural steps in chronological order.



Steps	Key points
	— Four cages for newly emerged nymphs. At 25° C the eggs in the plants which were placed in a test-insect rearing cage on Tuesday will hatch the following Tuesday. This cage can again be filled with egg-infested plants. — Eight cages for 1st- to 2d-instar nymphs which are generally used in screening for resistance 12 and 15 days from time of oviposition. — Twelve cages for 3d- to 5th-instar nymphs. — Sixteen cages for newly emerged to 5-day-old adults, 20 to 26 days after oviposition. • On Monday each week, egg-infested plants are transferred from the oviposition cage to the culture maintenance cage. • On Tuesday, Wednesday, Thursday, and Friday of the same week, egg-infested plants are transferred from the oviposition cage to the test-insect cages to rear hoppers of known ages for various studies (Fig. 16).

16. Production of test insects of known ages for various studies.

Overpopulation in rearing cages

Overpopulation occurs when there are about 20 insects/10-day-old seedling or 40 insects/tiller on 50-day-old plants. Overpopulation on individual food plants in the rearing cages should be prevented. If it occurs, plants dry quicker and must be changed more often, thus depleting the food supply sooner than planned. The growth and reproductive potential of the insects on overpopulated plants also are adversely affected. Wilting of food plants within 3 days after insect infestation and appearance of planthoppers on the leaves because of crowded conditions at the base of the plants are indications of overpopulation.

Step	Key point
1. Reducing insect density	• When overpopulation occurs, reduce insect density by transferring them to other potted plants in the same cage or to those in another cage containing insects of the same age or discard.

Removal of unwanted arthropods

Other biotypes, other hopper species, and other pests, parasites, and predators may infest the rearing cages. This is a common problem when the potted plants used for oviposition and feeding are grown outdoors, unprotected with netting.

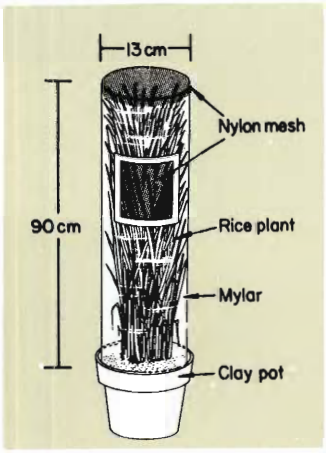
Steps	Key points
1. Vacating the oviposition cage	• If a high population of other arthropods develops in the rearing cages or a mixture of biotypes of a given species occurs, remove all potted plants. To kill the arthropods, leave the cage vacant for 3 days. Introduce fresh potted plants and infest them with individually collected adults. • If the population of the unwanted arthropods is low, remove them individually with a mouth aspirator or a suction machine. Plants need not be removed.
2. Protecting the hoppers from ants	• Place each foot of the cage or the table on which the cages are set in trays containing water (Fig. 8).

Condition of the culture

Fungal diseases, which occur during rainy and low-temperature (below 20° C) periods, limit the production of insects. *Metharrhizium anisopliae* is especially serious in the ZLH culture. Although inbreeding depression has not been a problem in hopper rearing at IRRI, the culture should be checked once a year to determine whether production is decreasing.

Steps	Key points
1. Controlling fungal disease	• Place 100W incandescent lamps over the rearing cages. Leave the lamps on day and night during the rainy season (Fig. 17).
2. Checking for inbreeding depression	• Conduct a population growth study. — Transplant 3 10-day-old TN1 seedlings in a 16-cm-diam clay pot. Each pot serves as a replication and the study should be replicated 10 times. — At 30 days after transplanting (DT) enclose the plants with a 13- × 90-cm mylar cage (Fig. 18). — At 40 DT when the plants are 50 days old, remove all arthropods on the caged plants. — Infest each cage with two pairs (male and female) of newly emerged adults. — When the progeny resulting from the two pairs have hatched, count all the nymphs in each cage and calculate the number of offspring per

17. Lights over zigzag leafhopper rearing cages.



18. Potted rice plants for a population growth study, enclosed in a mylar film cage.

19. Greenhouse room where insects are reared and seedlings are screened for resistance to BPH.

Steps	Key points
	female adult. The following number of progeny per pair is considered normal: BPH = 300 WBPH = 200 GLH = 200 ZLH = 100

SCREENING METHODS

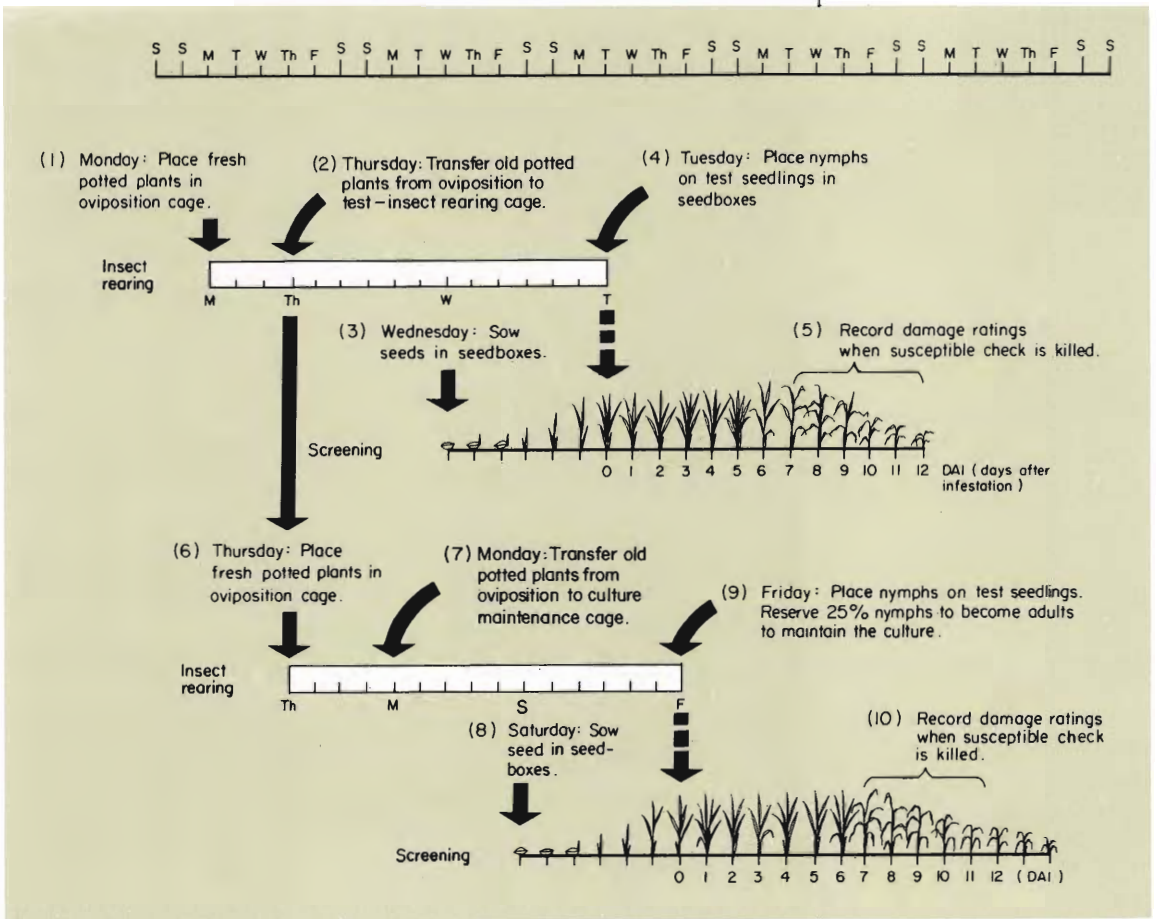
Cultivar screening for hopper resistance can be done in the greenhouse or in the field. Greenhouse screening is the rapid method for evaluating a large number of breeding lines, but the resistant entries should also be screened in the field before releasing them as varieties. Special methods have been developed to increase field populations to provide sufficient insect pressure for valid screening. Two methods of greenhouse screening used at IRRI are described.

Greenhouse screening by the conventional seedbox test

The conventional seedbox screening test is a common method used for greenhouse screening throughout Asia. More than 60,000 entries/year are

evaluated for BPH resistance in one greenhouse room at IRRI (Fig. 19). The conventional seedbox test is a rapid method of screening large volumes of material for qualitative resistance. However, it often cannot detect moderately resistant varieties and rates them as susceptible.

Steps	Key points
1. Scheduling seed sowing and seedling infestation	• Sowing and infesting should be timed according to the hopper rearing schedule. • Sow seed 5 to 6 days after the infested plants containing eggs are removed from the oviposition cage (Fig. 20). • At 6 days after sowing (DAS), the seedlings are ready for infestation. The nymphs will be in the 2d and 3d instars at this time (12-15 days after oviposition).



20. Sequence of activities for rearing hoppers and screening rice plants for resistance.

Steps	Key points
2. Listing test materials	• List all the entries to be tested on data sheets (Fig. 21, 22) for unreplicated and replicated tests. In the initial evaluation of the germplasm collection, test entries are not replicated. Entries selected in the unreplicated tests are retested with three replications. • Arrange the seed envelopes in numerical order based on the entry numbers. • Record entry numbers from each envelope on the data sheet and bundle the envelopes of every 10 consecutive entries with a rubber band.

21. Data sheet used for unreplicated greenhouse tests for brown planthopper. Example of the July 1983 pedigree nursery screening against biotype 2.

BPH SCREENING OF

SOWN: August 10, 1983 — July 1983 Pedigree Nursery — TO BPH Biotype 2

INFESTED: August 17 — TOTAL TESTED: 175 — SELECTED: 25

GRADED: August 26 — CODE NO START: 75,876 — END: 76050

BOX-ROW ENTRY NO.	GRADE	BOX-ROW ENTRY NO.	GRADE	BOX-ROW ENTRY NO.	GRADE	BOX-ROW ENTRY NO.	GRADE	BOX-ROW ENTRY NO.	GRADE	BOX-ROW ENTRY NO.	GRADE
		75,901	9	75,931	9	75,961	9	75,991	9	76,021	9
		02	7	32	1	62	5	92	7	22	7
		03	7	33	7	63	9	23	3	23	3
		04	7	34	7	64	9	94	7	24	7
		05	9	35	9	65	7	95	7	25	NG
75,876	9	06	9	36	NS	66	7	96	9	26	7
77	9	07	3	37	7	67	7	97	9	27	9
78	7	08	7	38	7	68	7	98	7	28	5
79	9	09	7	39	1	69	9	99	7	29	7
80	1	10	9	40	9	70	9	76,000	1	30	9
81	7	11	NS	41	9	71	7	01	7	31	9
82	7	12	7	42	7	72	9	02	9	32	7
83	9	13	7	43	7	73	3	03	7	33	7
84	5	14	7	44	7	74	7	04	7	34	9
85	9	15	9	45	3	75	9	05	7	35	9
86	9	16	9	46	9	76	9	06	5	36	7
87	7	17	9	47	9	77	7	07	9	37	7
88	3	18	7	48	NG	78	7	08	9	38	7
89	9	19	3	49	7	79	1	09	9	39	9
90	NG	20	9	50	1	80	7	10	3	40	1
91	7	21	7	51	9	81	NG	11	7	41	9
92	7	22	9	52	9	82	7	12	9	42	7
93	7	23	7	53	9	83	9	13	7	43	9
94	9	24	NG	54	7	84	9	14	5	44	NG
95	9	25	7	55	3	85	7	15	9	45	7
96	7	26	7	56	7	86	7	16	9	46	5
97	9	27	7	57	9	87	3	17	9	47	7
98	7	28	3	58	7	88	9	18	7	48	7
99	9	29	9	59	5	89	7	19	9	49	9
75,900	7	30	7	60	7	90	9	20	7	50	9

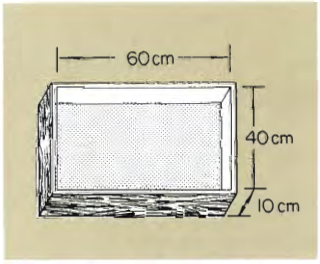
NG = NO GERMINATION NS = NO SEED

SCREENING RESISTANCE TO BROWN PLANTHOPPER, IRRI GREENHOUSE, 19 83.

Biotype 2 Sown July 5 Material Retesting of Selected
Tray 8 Infested July 11 Germplasm Collection Accessions
Batch 1 Graded July 20 No. tested 120 No. selected 73

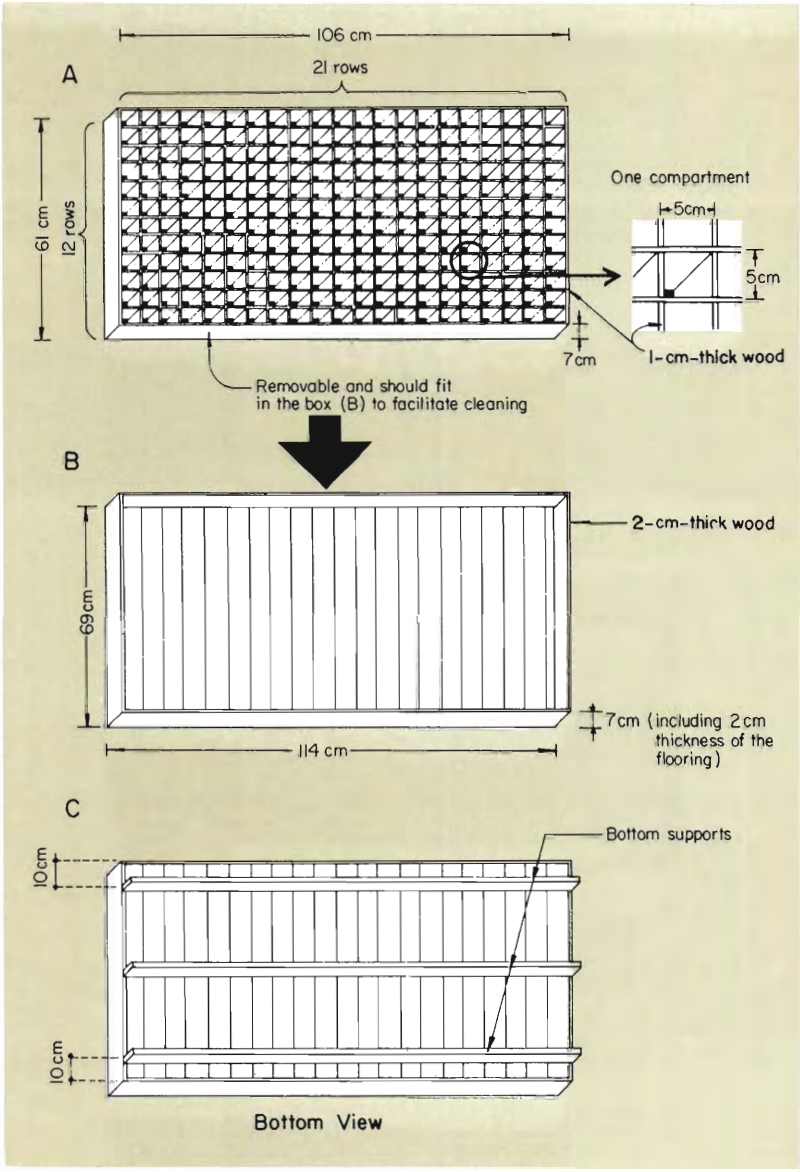
IRRI Acc. No.	Replicate I	II	III	$\bar{x}$		Replicate I	II	III	$\bar{x}$		Replicate I	II	III	$\bar{x}$
15,182	5	5	5	5	15,297	3	3	5	3	15,490	1	1	1	1
190	3	1	3	3	298	3	3	3	3	491	3	5	3	3
192	3	3	3	3	299	5	5	3	5	498	9	9	9	9
195	1	1	1	1	304	3	3	7	5	503	7	7	7	7
200	7	5	3	5	306	9	7	9	9	508	3	3	5	3
201	1	1	1	1	310	9	5	7	7	509	1	3	3	3
203	9	9	7	9	316	9	9	9	9	513	5	3	5	5
206	1	1	3	1	320	9	7	9	9	514	7	3	5	5
207	3	3	5	3	322	1	3	3	3	516	9	9	9	9
209	5	3	5	5	332	1	1	1	1	520	9	9	9	9
212	3	3	3	3	338	9	9	9	9	522	7	9	9	9
213	1	1	1	1	347	9	9	9	9	532	3	3	3	3
217	9	9	9	9	354	9	9	9	9	533	3	5	3	3
219	1	1	1	1	370	9	9	7	9	534	1	1	1	1
220	9	9	9	9	376	9	7	9	9	535	3	3	5	3
221	5	5	5	5	389	7	9	9	1	536	1	1	1	1
222	3	1	1	1	397	5	5	3	5	541	1	1	1	1
224	3	1	3	3	399	1	1	1	1	545	1	1	1	1
226	9	7	9	9	402	9	9	9	9	551	1	1	1	1
228	9	9	7	9	406	3	7	5	5	552	9	9	9	9
230	1	1	1	1	412	5	5	5	5	555	7	9	9	9
232	9	7	9	9	421	3	3	1	3	557	3	1	1	1
233	3	3	3	3	423	9	9	7	9	568	9	7	9	9
237	9	7	9	9	426	5	3	7	5	563	5	5	7	5
239	7	7	7	7	428	9	9	9	9	582	9	9	9	9
244	1	1	1	1	441	3	1	1	1	585	9	9	7	9
215	9	9	1	9	442	3	3	3	3	589	9	7	9	9
257	9	7	9	9	444	1	1	1	1	590	5	7	7	7
259	3	3	3	3	452	3	3	3	3	601	5	5	5	5
260	1	1	3	1	454	7	7	5	7	604	9	9	9	9
276	1	1	1	1	457	1	1	1	1	617	1	1	3	1
279	1	1	1	1	463	9	9	9	9	618	3	1	3	3
280	9	9	9	9	466	5	7	5	5	634	1	1	1	1
281	3	1	3	3	467	3	3	5	3	656	9	9	9	9
285	3	1	1	1	471	3	3	3	3	658	7	9	9	9
286	1	3	3	3	472	9	9	9	9	677	9	9	9	9
292	9	9	9	9	477	9	7	9	9	700	1	1	1	1
293	9	9	9	9	481	9	9	9	9	719	9	7	9	9
295	5	7	3	5	488	3	1	3	3	736	1	1	1	1
296	3	1	3	3	489	1	3	1	1	744	1	3	1	1

22. Data sheet used for replicated greenhouse screening tests for brown planthopper biotype 2.



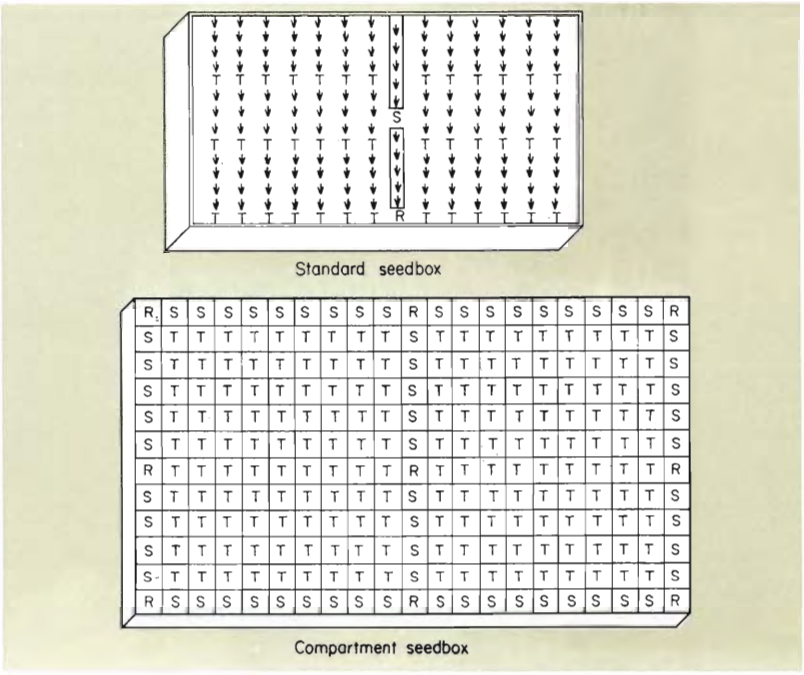
23. Standard wooden seedbox (inside dimensions).

Steps	Key points
3. Selecting seedboxes	• Two types of seedboxes are used: the standard seedbox (Fig. 23) and the compartment seedbox (Fig. 24). — The standard seedbox measures 60 × 40 × 10 cm. Because seeds are sown in rows (Fig. 25) the number of insects per seedling can easily be determined if data on nonpreference are also taken during screening. Thirty-nine test entries can be screened in a box.



24. Compartment seedbox. A) compartment structure consisting of 252 compartments measuring 5 × 5 cm, B) top view of the seedbox, and C) bottom view.

Steps	Key points
	— In the compartment seedbox the compartments serve as a guide in sowing the seeds of test entries and prevent dispersing of seed while watering. This seedbox measures 114 × 69 × 7 cm and 180 test entries can be screened per seedbox. About twice as many entries can be screened in the compartment type as in the

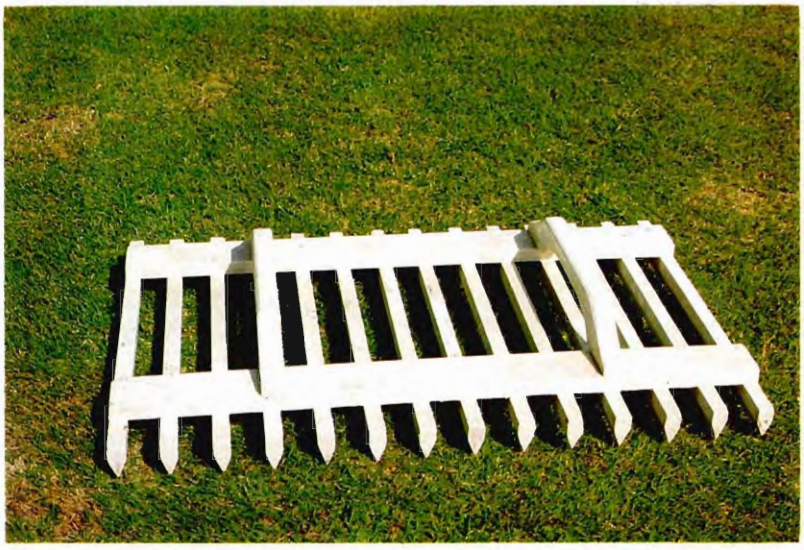


25. Placement of test (T) and check entries (R = resistant, S = susceptible) in the standard (A) and compartment (B) seedboxes.

Steps	Key points
	standard seedbox. Thus, if greenhouse space is limited use the compartment seedbox. A disadvantage is the difficulty of counting the insects per seedling if nonpreference data are to be recorded.
4. Preparing soil for the seedbox	• It is important to use soil that promotes rapid and even growth throughout the seedbox. Use soils that have high organic matter content and are free from pathogenic microorganisms. Avoid using toxic soils or soils that are deficient in nutrients. • Air-dry and grind the soil with a grinder (Fig. 26) or pass it through a screen (Fig. 3) to produce small particles. • If organic fertilizer or mineral elements are needed, mix them with the soil before filling the seedboxes.
5. Putting soil in the seedbox	• For the standard seedbox use a 3-cm soil depth. Level the soil and use a row marker (Fig. 27) to make rows which will serve as a sowing guide. • For the compartment seedbox add soil to a 3-cm depth and level. Place the compartment structure (Fig. 24A) in the seedbox. Label where the susceptible and resistant check varieties are to be planted in the seedboxes.

26. Electric soil grinder.

Steps	Key points
6. Sowing seed and rearing the test seedlings	• Identify, with the pot labels along the side of the seedboxes, the first entry of each bundle of 13 seed envelopes (Fig. 28).



27. Row marker for making marks in the soil in seedboxes.

28. A pot label is placed on each bundle of 13 seed envelopes. The biotype, entry or code no. of the first envelope in the series of 13, and the number of the seedbox are indicated on the label. The rows have been marked with the row marker (Fig. 27) and seeds of the susceptible check and resistant check have been sown.

Steps	Key points

- The following varieties can be used as susceptible and resistant checks for the given insects.

Hopper	*Susceptible check*	*Resistant check*
BPH biotype 1	TN1	ASD7, IR26, or IR29
BPH biotype 2	Mudgo, IR26, or IR29	ASD7
BPH biotype 3	ASD7	IR26 or IR29
BPH South Asian biotype	TN1	ARC10550, Ptb 33, or Sinna Sivappu
WBPH	TN1	IR2035-117-3
GLH	TN1	IR29
ZLH	TN1	Ptb 21 or Ptb 33

- Sow 39 test entries in 12-cm rows running the width of the seedbox with 3 entries in each row.
- Sow 25 seeds of each test entry in the 12-cm row.
- Cover the seeds with a thin layer of fine soil.
- Use a screen cage to protect the seeds from rats and birds (Fig. 29).
- Sprinkle seedboxes with sufficient water to just saturate the soil. Continue sprinkling one to two times daily depending on the sunlight and temperature, until the seeds have germinated. Too much water will cause flooding and cause uneven germination.
- After germination, increase the frequency of watering or place seedboxes in a water pan, if available.

29. Screen cage to protect newly sown seed from rat and bird damage.

Steps	Key points
b. Compartment seedbox	• The compartment structure is used as a sowing guide. The compartment seedbox contains 9 compartments with a resistant check and 63 compartments with a susceptible check located around the edge and across the center (Fig. 25B). The susceptible and resistant checks are the same as those used in the standard seedbox. • Sow 180 test entries in the remaining compartments. Sow about 25 seeds/compartment. • Place a thin layer of soil over the seeds. • Protect the box from rats and birds, and water one to two times daily. After each watering, slightly lift the compartment so that it will not sink into the soil. If it sinks into the soil, it will be difficult to remove without disturbing the seedlings. • About 5 DAS when seedlings have roots, gently remove the sowing guide from the seedbox. The guide can then be used in another seedbox.
7. Infesting seedlings in seedboxes with hoppers	• At 7 DAS, when seedlings are in the 3-leaf stage (growth stage 1), place the seedboxes in a water pan inside a screened room. Remove the weeds and thin the seedlings to about 20/row or compartment. • Fill the pan with water to a 5-cm depth. • Remove from the test-insect cage infested plants containing 2d- and 3d-instar nymphs. • Distribute the nymphs uniformly on the test seedlings by holding the base of the pot and lightly tapping the plants and blowing on them (Fig. 30). • The approximate number of insects per seedling is 10 BPH, 8 WBPH, and 3 GLH or ZLH. If older test seedlings are used, increase the number of insects per seedling. Also, when you observe that the susceptible check is not being damaged as rapidly as it should, add insects to accelerate the rate of hopper burning.
8. Grading plants for damage	• Grade the entries in each seedbox when the susceptible check seedlings in that box are about 90% dead. • Grade for plant damage using the Standard Evaluation System for Rice (SES) scale (IRRI 1980).
a. BPH	*Scale* — *Damage* 0 — None 1 — Very slight damage 3 — First and second leaves of most plants partially yellowing 5 — Pronounced yellowing and stunting or about half of the plants wilting or dead

30. Infesting seedlings with hoppers from the test-insect rearing cage.

Steps	Key points	
	7	More than half the plants wilting or dead and remaining plants severely stunted
	9	All plants dead
b. WBPH	*Scale*	*Damage*
	0	None
	1	Very slight damage
	3	First and second leaves with orange tips; slight stunting
	5	More than half the leaves with yellow-orange tips; pronounced stunting
	7	More than half the plants dead; remaining plants severely stunted and wilted
	9	All plants dead
c. GLH and ZLH	*Scale*	*Damage*
	0	None
	1	Very slight damage
	3	First and second leaves yellowing
	5	All leaves yellow, pronounced stunting or both
	7	More than half the plants dead; remaining plants wilting, severely stunted
	9	All plants dead
	• Record damage ratings for each entry on data sheets (Fig. 21, 22).	
	• When entries are replicated the grade is based on the average of the replications.	

Greenhouse screening by the modified seedbox test

Because the conventional seedbox screening test is mostly qualitative, entries with moderate levels of resistance because of tolerance or because of low levels of antibiosis or nonpreference are usually susceptible. Thus, the conventional test is modified to detect varieties with moderate levels of resistance.

In the modified test, the plants are older at time of infestation and fewer hoppers per seedling are placed on the plants. In addition, the progeny rather than the initial source of infestation are the insects that cause the plant damage. The modified method described here has primarily been used to evaluate varieties for BPH resistance. With modifications the method can be used for other hopper species.

The methods used in the conventional test are followed, except for modifications as described.

Steps	Key points
1. Thinning seedlings	• Replicate each entry three times. • Sow about 20 seeds in the standard seedbox in 18-cm rows. At 5 DAS, thin the seedlings to 10/row.
2. Infesting seedlings with hoppers	• At 10 DAS infest plants with 2d- and 3d-instar hoppers at the rate of 3-5/seedling. • Cover the whole seedbox of infested seedlings with a fiberglass screen cage (65 × 45 × 90 cm) to prevent the insects from escaping.
3. Grading plant damage	• When the susceptible check has a rating of 7 due to damage by the progeny of the initial infestation (which occurs about 28 DAS depending on temperature), begin grading. Repeat the grading process 3 more times at 2-day intervals.

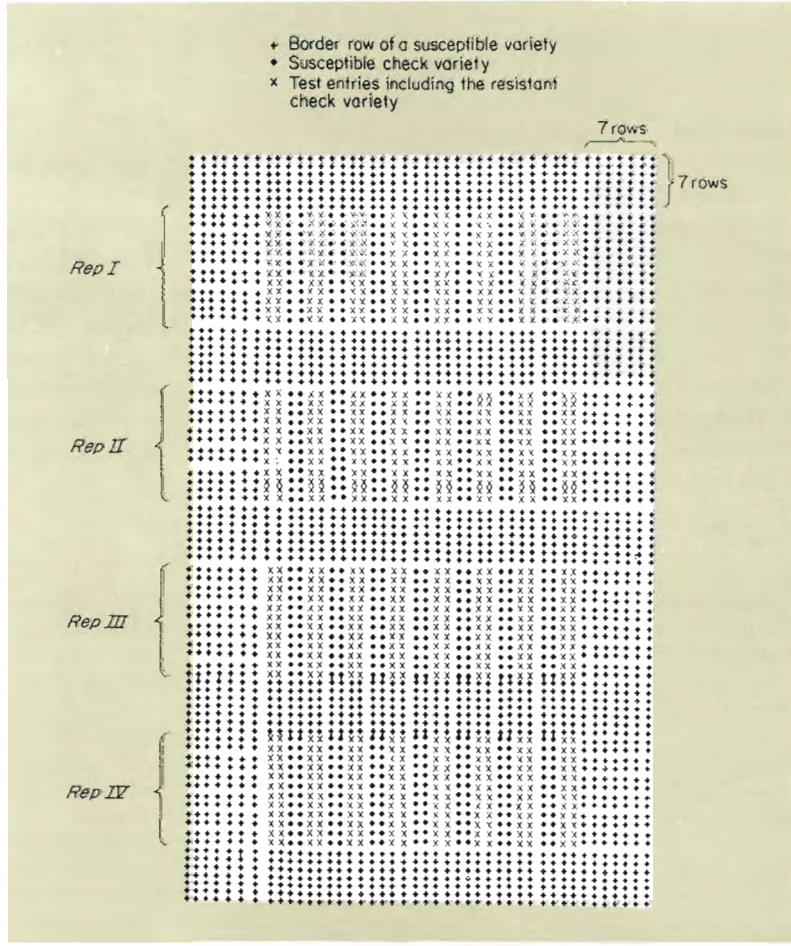
Field screening using the resurgence technique

Breeding lines identified as resistant in the greenhouse should be tested in the field before their release as varieties.

Techniques for field evaluation of rice for BPH have been developed. The methods described for BPH can be used for the WBPH, GLH, and ZLH in sites where field populations of these hoppers are high. Select a screening site and time of the year when BPH are abundant. Sometimes, BPH populations are often too low or too unevenly distributed for reliable field screening. These handicaps to field screening can be overcome by causing BPH population increases through the application of resurgence-inducing insecticides (Chelliah and Heinrichs 1980; Heinrichs et al 1982a, b; Reissig et al 1982a, b).

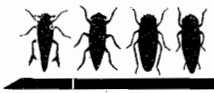
Steps	Key points
1. Rearing hoppers to supplement natural field infestations	• If a BPH culture is maintained for greenhouse screening, the insects can be used to supplement natural field populations. If no culture is maintained, collect hoppers and rear them on plants for later release in the field. — About 2 months before the scheduled day of transplanting, place BPH on caged 80-day-old TN1 plants in the greenhouse. Rear them on the plants, adding fresh plants as needed. — Within 2 generations (about 2 months) the progeny from each female is sufficient to infest a 25-lm strip of susceptible border rows 1.4 m wide.

Steps	Key points
2. Preparing the field layout	• In preliminary testing, replicate each entry once. In retesting, replicate entries four times and arrange them in a randomized complete block design (Fig. 31). • Each replication consists of 2 rows 2.4 m long for each test entry and a resistant and susceptible check with 20-cm spacing between hills. Provide border rows around each replication, by planting a susceptible variety in 1.4-m strips of 7 rows each.
3. Sowing seed	• Sow in a wetbed nursery.
4. Preparing the field	• Use normal tillage practices. Apply fertilizer basally.



31. Field plot layout used for screening rice for brown planthopper resistance.

Steps	Key points
	• Soil-incorporate a systemic insecticide at 1 kg ai/ha for control of tungro virus and seedling pests and for vigorous crop growth. The BPH population is higher in dense, healthy crop stands where tiller counts are high. The insecticide will not be deleterious to the BPH as the residue levels are low by the time the BPH begin feeding. If an insecticide is not soil-incorporated, broadcast a granular formulation 5 DT.
5. Transplanting	• At 21 DAS in the nursery, transplant the susceptible border rows, test entries, and susceptible and resistant checks. — Transplant two adjacent rows of each test entry and two rows of a resistant check in each replication. Sinna Sivappu and Ptb 33 are resistant at most sites and can be used as resistant checks. Transplant two rows of a susceptible check such as TN1 on each side of each test entry and the resistant check. The susceptible check can be the same variety as that used for the border rows. However, a variety that can maintain high BPH populations and has resistance to viruses is generally used as the border variety. — Transplant a susceptible border of 7 rows around each replication.
6. Applying insecticide to border rows to induce BPH resurgence	• To kill natural enemies apply as a foliar spray a resurgence-inducing insecticide such as decamethrin (30 g ai/ha), methyl parathion, triazophos, or diazinon (500 g ai/ha) to the susceptible border rows beginning at 20 DT. Repeat the sprays at 10-day intervals to 70 DT, if necessary. Observe the base of the plants after each spraying to determine whether spiders, mirid bugs, and other predators are still abundant. If so, repeat the spray application the next day.
7. Infesting the plants artificially with BPH	• After the first application of the resurgence-inducing insecticide at 20 DT, evenly distribute late-instar nymphs at the rate of 5 insects/hill in the susceptible border rows throughout the field. If natural populations of BPH are high, this step may be omitted.
8. Counting BPH	• Although the number of BPH is not necessary in determining the damage rating, this information is useful in determining preference and tolerance. — Make a visual observation of the number of BPH on 5 hills/plot starting at about 40 DT.



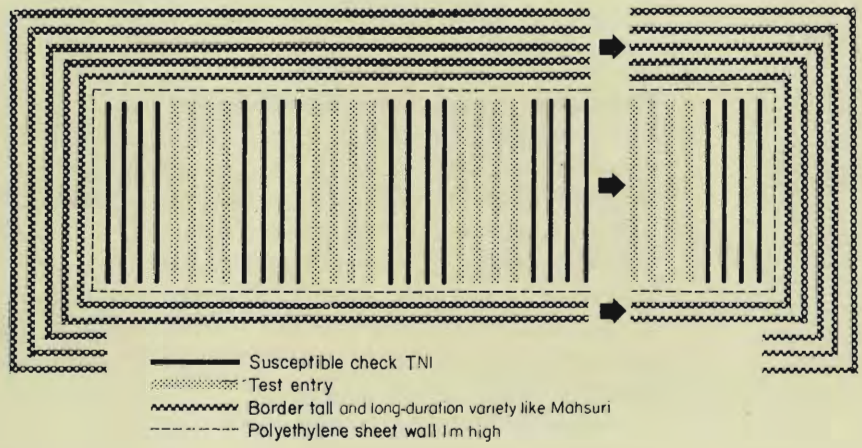
Steps	Key points
	— In selecting hills to be sampled, use the same sites — based on the locations of the hills within the two rows — for all entries of a given replication. In the next replication, sample different hills. This procedure is repeated for the four replications. For example, if the 5th, 9th, and 11th hills of 1 row and the 4th and 8th hills of the second row in replication 1 are sampled, repeat this sequence for all entries in replication 1 including the susceptible and resistant checks. Do not count the BPH on the two hills at each end of the row. — When there are more than 1,000 BPH/hill, count only 5 representative tillers/hill and multiply the number of BPH per tiller by the number of tillers in the hill.
9. Grading plant damage	• When plants in one of the susceptible checks start wilting, begin grading all entries for damage. Generally the following BPH populations are necessary to provide sufficient plant damage for a valid test: — 25 BPH female adults/hill at maximum tillering — 100 BPH female adults/hill at flowering • Grade the entries on a row basis. Use the SES scale for damage rating.

Scale	*Damage*
0	None
1	Slight yellowing of a few plants
3	Leaves partially yellow but with no hopperburn
5	Leaves with pronounced yellowing and some stunting or wilting and 10-25% of plants with hopperburn, remaining plants severely stunted
7	More than half the plants wilting or with hopperburn, remaining plants severely stunted
9	All plants dead

Field screening using the polyethylene barrier technique

In India the BPH population have at times been so low that the resurgence technique described in the preceding section did not increase populations to a level where hopperburn occurred. The resurgence technique was modified by Kalode et al (1982). A polyethylene sheet is placed around small field plots to prevent movement of BPH nymphs outside the plot and to prevent predators such as spiders from entering the plot.

Steps	Key points
1. Rearing hoppers to supplement the natural field infestation	• Rear hoppers to infest the field plots. Begin the rearing program so that hoppers are available by 30 DT.
2. Preparing the field layout	• The field layout is illustrated in Figure 32. — Transplant around the test entries 5 border rows of a tall and long-duration variety (such as Mahsuri) spaced 10 cm between hills. — Within the border rows, plant 4 rows of 10 hills each/entry at a spacing of 25 cm between hills. — Adjacent to each test entry, plant 4 rows of a susceptible check at a 25-cm spacing between hills.
3. Sowing seed	• Sow seed in a wetbed nursery.
4. Preparing the field	• Use recommended tillage practices. • Apply fertilizer basally. • Soil-incorporate a systemic insecticide before transplanting. If insecticide is not soil-incorporated, apply a granular formulation 5 DT to promote good crop growth.
5. Transplanting the seedlings	• At 21 DAS the seedlings are ready for transplanting. Transplant the susceptible border rows, susceptible check, and test entries.



32. Polyethylene barrier technique for field screening for rice brown planthopper resistance (Kalode 1982).

Steps	Key points
	— Plant 4 rows of each test entry. — Alternately plant 4 rows of the susceptible check next to the test entry. — Plant the 5 border rows of Mahsuri around the plots.
6. Applying insecticide to the entire plot to induce BPH resurgence	• Beginning at 10 DT, spray the entire plot (border rows, test entries, and the susceptible check) with 0.02% methyl parathion (200 g ai/1,000 liters water per ha) at 10-day intervals, or with 0.005% deca-methrin (50 g ai/1,000 liters water per ha) at 15-day intervals.
7. Enclosing test entries and susceptible checks with a polyethylene sheet	• At 30 DT enclose the test entries and susceptible check with a 76-cm-high polyethylene sheet with its top open. Use bamboo or wood stakes and metal wire to support the sheet.
8. Infesting the plots with BPH	• As soon as the polyethylene sheet is in place, infest the plots with laboratory-reared or field-collected BPH. • Repeat BPH releases in the plots until a sufficient population is established.
9. Grading plant damage	• When plants in one of the susceptible checks begin wilting, start grading entries for damage. Use the SES scale described under *Field screening using the resurgence technique.*

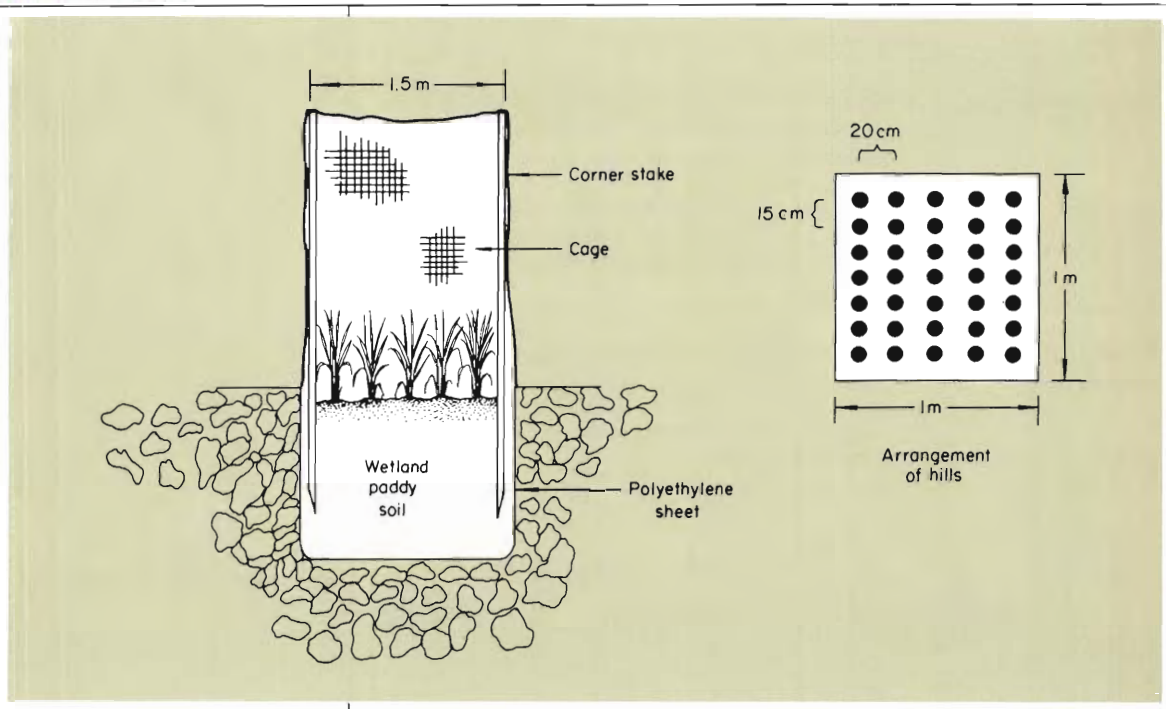
Field screening using the microplot technique

The microplot technique simulates field screening. This technique is especially useful in areas where it is preferable not to induce hopperburn because of damage to neighboring research plots or nearby farmer fields. A cage placed over a small plot consisting of one entry induces the buildup of artificially infested BPH populations by preventing natural enemies from entering the plots and preventing BPH from escaping to the nearby research plots or fields where they are not wanted. Although the microplot has been used only for BPH screening it may be useful for screening rice against other hopper species.

In the microplot technique, 1.5 × 1.5 × 2.0 m cages are placed either over small plots in a wetland field (Fig. 33), or in specially prepared pits around the greenhouse or in a dryland field. Use of the latter is described.

Steps	Key points
1. Preparing the pits	• Dig 1.5-× 1.5-m pits to a depth of 1.0 m (Fig. 34) in a convenient place near a building where water is available and the cages will be protected from wind and animals. • Line each pit with a polyethylene sheet. • Fill the pits with wetland paddy soil up to 15 cm below ground level to create typical wetland soil conditions.
2. Preparing the cages	• Make fiberglass mesh cages measuring 1.5 × 1.5 × 2.0 m. • Cage supports consist of an iron frame and bamboo or wood stakes placed in each of the four corners of the pit.

33. Cage used in the microplot technique for screening for brown planthopper resistance in a wetland rice field at IRRI.



34. Pit used in the microplot technique for screening for brown planthopper resistance.

Steps	Key points
	• The number of cages depends on the number of entries to be tested and the number of replications. One cage accommodates one entry. In retesting, replicate each entry four times and arrange the entries in a randomized complete block design. Include one susceptible and one resistant check varieties.
3. Sowing seed	• Sow seed in a wetbed nursery. Have enough plants to transplant in each cage 35 hills of 3 seedlings each.
4. Preparing the soil in the microplots	• Prepare the soil before transplanting. • Apply fertilizer basally.
5. Transplanting	• At 21 DAS transplant seedlings into the plots at a spacing of 15 cm between hills and 20 cm between rows. • Immediately after transplanting, set the cages over the plots to prevent insects and natural enemies from entering.
6. Replanting	• At 7 DT replant all poorly growing or dead hills.

Steps	Key points
7. Removing insect pests and natural enemies	• At 15 DT spray the plants in the cage with a short residual insecticide such as decamethrin, which has high toxicity to spiders and other natural enemies and low toxicity to BPH.
8. Preparing insects for infestation	• At 19 DT prepare the insects for field infestation. To transport insects to the field, place 10 pairs (male and female) of greenhouse-reared BPH adults (2- to 3-day-old) in a test tube with 1 TN1 seedling. Each plot requires 7 test tubes (7 test tubes × 10 pairs = 70 pairs).
9. Infesting the caged plots with hoppers	• At 20 DT remove natural enemies from the caged plots by hand. Then release BPH from the test tubes, 2 pairs per hill or 70 pairs per cage.
10. Applying fertilizer	• Apply N fertilizer to the plots at 30 and 60 DT.
11. Removing natural enemies	• Check cages for natural enemies weekly and spray plants with a 0.002% concentration of decamethrin for control, when needed.
12. Grading plant damage	• When about 50% of the susceptible check plants in one cage are wilting or are hopperburned, begin grading damage of all entries. • Repeat damage ratings at 4-day intervals until there is no further increase in damage. Follow the same damage rating described in *Field screening using the resurgence technique.*
13. Recording yield	• Take yield data by harvesting the plots at maturity. Correct to 14% grain moisture and calculate yield by $\frac{100 - \%\text{ moisture of sample}}{86} \times \text{grain wt in g}$ Yield is expressed in grams per square meter.

MECHANISMS OF RESISTANCE

The greenhouse and field screening tests described in the section *Screening methods* — where entries are rated based on the damage caused by the hoppers — are used to eliminate the most susceptible varieties. Through further testing in replicated tests, more entries are eliminated. Selected entries, such as varieties being considered for use as resistance donors in the

breeding program or elite breeding lines being considered for release as recommended varieties, should be further evaluated to determine their levels of resistance as affected by plant-insect interactions. Although an understanding of the mechanisms of resistance is not necessary in the development of resistant varieties, the information will be useful to entomologists and breeders as they work together to develop varieties with the most effective type of resistance against pest populations.

Various tests have been developed to determine such components of resistance as antixenosis (nonpreference), antibiosis, and tolerance. Antixenosis tests for nymphs on seedlings and adult hoppers on older plants are described. Methodology is described for four tests that study the effect of level of antibiosis on 1) hopper population growth, 2) survival of nymphs, 3) feeding rates, and 4) location of feeding sites. Greenhouse and field methods have been developed to measure levels of tolerance for the brown planthopper.

Antixenosis of nymphs on seedlings

Studies on the level of antixenosis of nymphs on rice seedlings can be conducted simultaneously with the conventional seedbox screening test.

Steps	Key points
1. Steps 1-7 in the conventional seedbox screening test	• Use the 60- × 40- × 10-cm seedbox. • Replicate treatments at least four times. • Thin seedlings to 20/row and infest with hoppers.
2. Evaluating	• At 48 h after infestation, count the nymphs on 10 seedlings/row and express insect number on a seedling basis.

Antixenosis of adults for plants in the vegetative stage and for oviposition

Two tests to evaluate the level of antixenosis elicited by varieties in the vegetative stage of growth are described. The tests differ only in the arrangement of the varieties within the seedbox. In one, plants are arranged in a 10- × 10-cm spacing; in the other, they are arranged in a circle.

Steps	Key points
a. 10- × 10-cm spacing	
1. Setting the date for the test	• Set the date for infesting the plants. Seed sowing and insect rearing schedules will depend on that date.

Steps	Key points
2. Sowing seed	• Thirty-three days before the scheduled test, soak the seed of the varieties selected in water for 24 h. Include a resistant and a susceptible check. Incubate the seed for 48 h. • Sow 2 germinated seeds/hill at a 10- × 10-cm spacing in a 60- × 40- × 10-cm seedbox (Fig. 23). Replicate the entries at least four times in a randomized complete block design using one seedbox for each replication. One hill represents a replication.
3. Rearing insects	• Twenty-five days before the test, place gravid females in an oviposition cage for 24 h to lay eggs on 50-day-old potted TN1 plants. The nymphs that hatch from the eggs will become adults and be gravid on the day of infestation if reared at 25-30° C.
4. Infesting the plants	• At 30 DAS remove the dried or yellowing outer leaf sheaths and prune the plants to 2 tillers/hill. • Infest the plants in the seedbox at the rate of 10 adult female hoppers/hill. • Place a fiberglass mesh cage over the seedbox to prevent hoppers from escaping.
5. Counting the hoppers	• At 24 h after infestation, count the insects on each hill. Repeat at 48 and 72 h. Compute the average of replications for the 3 countings and compare to determine the levels of antixenosis among varieties.
6. Counting the eggs	• After the 72 h counting, cut the plants close to the soil. • Take the plants to the laboratory, dissect them under a microscope, and count the eggs to determine the levels of antixenosis for oviposition among varieties.
b. Circular arrangement	
1. Sowing seed of test entries	• Sow 10 seeds of each entry in 1 hill, 1 hill representing a replication. Set the hills 4 cm apart in a 60-cm-diam circle in a seedbox. Replicate the entries four times, one circle representing a replication. • At 10 DAS thin seedlings to 5/entry. Place a mylar cage over the plants (Fig. 35).

35. Mylar cage over seedlings in a seedbox to study antixenosis. Plants were arranged in a 60-cm-diam circle.

Steps	Key points
2. Releasing insects	• Release 2-day-old adult males and females of equal proportions at the rate of 3/seedling at 30 DAS. Place the adults in a petri dish in the middle of the circle.
3. Evaluating	• Count the insects on each entry at 24, 48, and 72 h after infestation. Compute the average of replications for the three countings and compare among varieties.

Antixenosis for feeding

You can determine the probing activity of the hoppers by counting the feeding probes or stylet sheaths left by the hoppers in plant tissues. Varieties with high levels of antixenosis for feeding generally have more feeding probes when exposed to hoppers because the insect is not satisfied with the food source and moves around, making numerous probes in search of preferred food. In varieties lacking antixenosis for feeding, few probes are made as the insect continues feeding in one site over a long period. To determine the number of probes follow these steps.

Steps	Key points
1. Enclosing insects on leaves or leaf sheaths	• Enclose one or two adults in a parafilm sachet attached to the leaves (leafhoppers) or leaf sheaths (planthoppers) of the rice plant. Replicate each entry 10 times, each replication consisting of 1 plant. • Allow the insect to feed for 24 h.
2. Cutting leaf or leaf sheath portions	• Cut the portion of the leaves or leaf sheath where the insects have fed (2-2.5 cm long).
3. Staining the stylet sheaths	• Dip the cut leaves in a staining solution of 0.1% rhodamine or erythrosin dye for 10-15 min. • Stylet sheaths are stained pink by the dye.
4. Counting the stylet sheaths	• Examine the leaf portions under a microscope and count the stylet sheaths. Determine the average number of stylet sheaths per variety. • Compare the number of stylet sheaths among varieties.

Antibiosis on population growth

The hopper population growth study conducted in the greenhouse takes several weeks; however, it will provide the most accurate indication of the

levels of antibiosis in a rice variety and will give a good indication of what the effect of the variety will be on the hopper population in the field. Both the antixenosis and antibiosis factors involving the various insect stages will affect the rate of population growth. The population growth study allows identification of varieties with moderate levels of resistance due to moderate levels of antixenosis and antibiosis.

The methods described are those used for the BPH at IRRI. With slight modifications in timing of various steps, the same procedure can be used for other hopper species.

Steps	Key points
1. Sowing seed	• At 35 days before the scheduled insect infestation, sow 3 pregerminated seeds each of the test entries including a resistant and susceptible check in a 16-cm-diam clay pot. Plant 5 pots/variety, each pot representing a replication. • Water the pots daily and protect them from insects. • Apply fertilizer to the soil in the pots if needed. Uniformly apply the same amount in pots of all test entries.
2. Rearing test insects	• Use either nymphs or adults for population growth studies. — If adults are used, place gravid females in an oviposition cage (Fig. 5 or 6) for 24 h, 23 days before infestation. Nymphs that hatch from the eggs will become adults in about 3 days before the test begins. — If first-instar nymphs are to be used as the initial population, place gravid females in an oviposition cage for 24 h, 9 days before infestation. Nymphs that hatch from the eggs will be about 1 day old at the start of the test.
3. Infesting test varieties with hoppers	• Thirty-five days after sowing, clean the base of the plants and enclose the plants of each pot with a 13-cm-diam, 90-cm-high mylar film cage (Fig. 18). Remove any insects or spiders from the plants. • Infest plants in each cage with 10 1-day-old nymphs or 2 pairs (2 males + 2 females) of 3-day-old adults. — Place the insects near the base of the plants with the aid of a mouth aspirator (Fig. 9) inserted through a slit or a 1-cm-diam hole in the mylar cages. If a hole is used, plug it with a cotton wad after infestation. — It is extremely important that the infestation procedure be done carefully so that the insects

Steps	Key points
	are not injured in handling. If any of the hoppers are killed during the infestation process, results will not be reliable. Handling of the hoppers, especially the newly emerged nymphs, requires practice. — Early on the day after infestation, check the cages for any dead hoppers. Replace the dead hoppers.
4. Counting the hoppers	• The number of days after infestation when the population is to be counted depends on the temperature, which affects the rate of insect development, and the stage of the insect used for infesting. — If nymphs are used as the initial source of infestation, the F_1 nymphs will have hatched by about 23 days after the initial infestation at 25-30°C. Count the population 30 days after infestation (DI). — If adults are the initial source, F_2 nymphs will hatch about 32 DI. Count the population at 25 DI for the F_1 or 40 DI for the F_2. • If the susceptible check wilts before the scheduled day for counting, count the insects immediately and terminate the experiment. Otherwise, insect mortality will make the counts unreliable. • Record on the data sheets the number of DI at which counting is conducted. • Two methods are used for counting, depending on whether insect weights are also needed. If insect weights are not needed proceed as follows: — About 3 days before counting, discontinue watering the potted plants to allow the soil surface to be almost dry but not yet cracked. — Apply a household aerosol spray such as DDVP (= dichlorvos) (2,2-Dichlorovinyl dimethyl phosphate) in the mylar cage. — Tap the cage to dislodge any remaining insects on the plants so that they will fall on the soil at the base of the plants. — Remove the mylar cage and first count the insects sticking to the inner wall of the cage and those clinging to plants. — Next make marks on the soil with a scalpel, dividing the area into equal divisions. Count the insects in each division. • If insect weights are also needed, follow these procedures: — Place the pot inside a 40-cm-diam, 50-cm-high plastic container. — Tap the cage slightly and remove the mylar cage.

Steps	Key points
	— Tilt the pot, tap the plant slightly, and blow insects off and into the bucket. — Tilt the bucket and tap so that the hoppers will slide to one area. — Collect the hoppers with a suction machine (Fig. 10). — Count the insects. — To determine total biomass per cage or average weights of the hoppers, place the hoppers in an oven at 60 °C for 12 h and weigh on a 0.001-mg sensitivity balance.

Antibiosis on survival of nymphs

The study of nymphal survival requires less time to conduct, but it does not give differences in levels of antibiosis that are as distinct as those indicated by the population growth study. It determines the effect of antibiosis factors on only one stage, the nymphs. First-instar nymphs are used as test insects.

Steps	Key points
1. Sowing seed of test varieties	• Thirty days before the scheduled date of infesting test varieties with hoppers, sow 2 pregerminated seeds of the test entries, including a resistant and a susceptible check variety, in a 16-cm-diam clay pot. • Have 10 pots for each variety, each pot representing a replication.
2. Rearing test hoppers	• Place gravid female adults in an oviposition cage (Fig. 5 or 6) for 24 h, 8 days before putting hoppers on the test entries. • At 25-30° C the nymphs will hatch on the infestation date. — Thin the seedlings 5 DAS. Leave one healthy seedling per pot. — Water the seedlings daily and protect them from insects. — Apply fertilizer, if necessary, equally to all pots. — Just before infestation remove the yellowing and dried outer leaf sheaths.
4. Infesting plants with hoppers	• Enclose each potted plant with an 8-cm-diam × 60-cm-high mylar film cage. • Through a slit or hole on the side of cage, place 10 1st-instar nymphs in each cage.
5. Counting the hoppers	• At 20 DI count the live hoppers in each cage and calculate survival percentage.

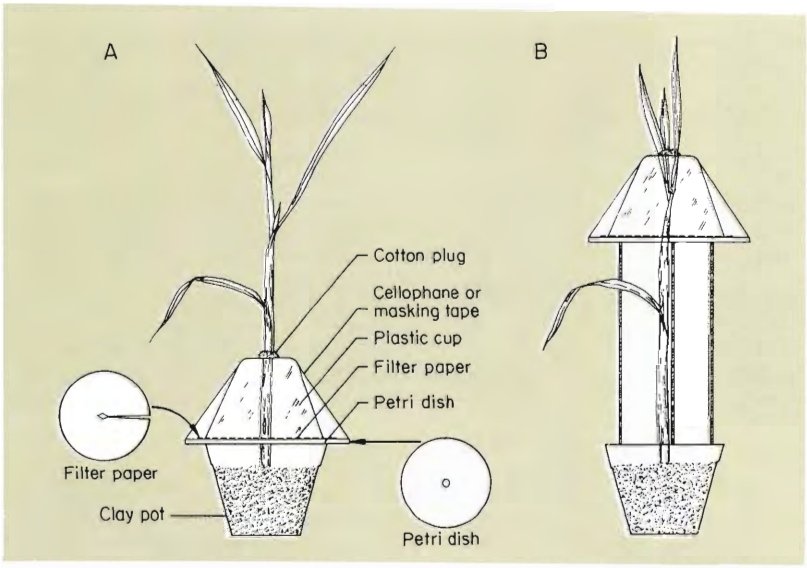
Antibiosis on feeding rates

Several methods to determine the feeding activity of hoppers have been developed (Paguia et al 1980). They are all indirect methods and measure the amount of honeydew excreted by the hoppers. Methods involve the collection of hopper-excreted honeydew on filter paper and on parafilm or in parafilm sachets. The amount of honeydew excreted is measured by determining the area, weight, or volume of honeydew excreted, and using colorimetric methods and measurement of radioactivity in the honeydew. Colorimetric and radioactivity techniques have been developed by Velusamy and Chelliah at Tamil Nadu Agricultural University, Coimbatore, India (pers. comm., 1982).

The ninhydrin-treated filter paper method is first described. This method is useful only in measuring the amount of amino acids in the honeydew and thus indicates feeding activity only in the phloem as the xylem contains little or no amino acid content. Other techniques also described should be used for the GLH, which feeds in both the phloem and the xylem. The ninhydrin techniques are most useful for the BPH, which is a phloem feeder.

Steps	Key points
Ninhydrin-treated filter paper methods	
1. Scheduling date of the feeding study	• Feeding studies can be conducted on plants of any age. If plants of 2 or more ages are to be compared, stagger the planting so that the plants can be tested simultaneously. Thus, if 30- and 60-day-old plants are to be compared, sow the seed of the 60-day-old plants 30 days before sowing those of the 30-day-old plants.
2. Sowing seeds of test entries	• Germinate about 50 seeds of each entry in petri dishes containing moist filter paper. • Maintain the moisture in the petri dishes. • Sow the germinated seed in a 60- × 40- × 10-cm seedbox. After 7 days, the seedlings are ready for transplanting.
3. Putting soil in pots	• One day before transplanting or 6 DAS, place puddled soil in clay pots. Each entry should have 10 pots, each pot serving as a replication. Pot size depends on the age of the plants to be used.
4. Transplanting the seedlings	• At 7 DAS transplant healthy seedlings into fertilized puddled soil in clay pots at 3 seedlings/pot. — Plant 10 pots of each entry. — Handle the seedlings with forceps, taking care not to cause any damage.

Steps	Key points
5. Protecting plants from arthropod infestation	• Place the potted seedlings where there is ample sunlight and where they are protected from arthropod pests and predators.
6. Rearing test hoppers	• Twenty-five days before the scheduled day of hopper infestation, place gravid females on plants in an oviposition cage for 24 h. • Transfer the egg-infested plants to a nymph-rearing cage and rear the nymphs to the 5th instar. Limit the population in the rearing cage to 5 nymphs/tiller. • Collect nymphs that reach the 5th instar at the same time and transfer them to another cage containing fresh potted plants. • The adults that emerge at the same time are used as test insects.
7. Collecting hoppers and starving them	• Collect 4-day-old female adults of uniform size between 0900 and 1000 h. • Place them in a container with moist filter or tissue paper and starve them for 4 h.
8. Preparing the feeding chambers	• The feeding chamber is placed at the base of the plant for BPH and WBPH (Fig. 36A) and over the middle portion of the leaves for GLH and ZLH (Fig. 36B).



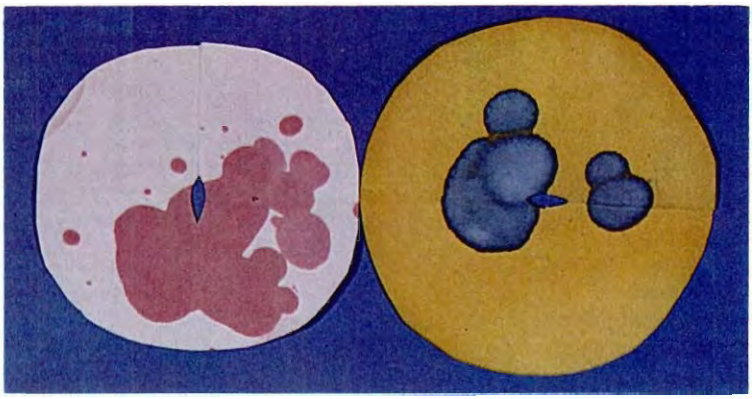
36. Diagram of the feeding chamber used for collecting honeydew in a feeding study. A: for BPH and WBPH, B: for GLH and ZLH.

Steps	Key points
	— Thin the plants in the pot to one per pot. — The outer leaf sheath, which is loose, usually produces moisture when it comes in contact with the filter paper. Remove it from the stem by cutting it at its base. — Invert a plastic petri dish on each pot and guide the leaf tips through the hole in the center. Then invert a plastic cup on the petri dish and guide the leaves through the hole at its bottom. — After fixing the petri dishes and plastic cups on all test plants, wipe the upper surface of the petri dishes, the inner sides of the plastic cups, and the stem of the plants with tissue paper to remove any possible moisture. — Wash hands thoroughly with soap and water, then wipe with clean dry cloth before handling the filter papers. Slightly lift the inverted plastic cup and place the circular filter paper on the petri dish. The stem of the plant will pass through the slit cut toward the center of the filter paper in which a hole is made to accommodate the stem. — Take care not to touch the filter paper with the fingers. Use forceps or carefully hold the edges of the filter paper with your fingertips. — Fasten the cup by fixing cellophane or masking tape on two opposite sides of the petri dish. — Run a pencil around the edge of the plastic cup lightly marking the outline of the cup on the filter paper. The area inside the circle is the area on which honeydew falls and the spots outside the circle are due to contaminants. — Treatments, replications, or code numbers and other markings should be written in lead pencil on the margin of the filter paper outside the plastic cup.
9. Infesting test entries with hoppers	• Introduce five female hoppers in each plastic cup through the top where the stem of the plant passes through. Plug this opening with cotton to prevent escape of the insects.
10. Collecting filter paper	• Allow the insects to feed for 24 h; then collect the filter papers with forceps and arrange them in a single layer on white cartolina or cardboard. Air-dry.
11. Spraying filter paper with ninhydrin	• Spray the filter papers with 0.01% ninhydrin in acetone solution. Then oven-dry them for 6 min at 100°C. The honeydew spots will appear as violet or purple due to their amino acid content.

Steps	Key points
	• The amount of honeydew excreted can be determined by any one of three methods. — Measure the area of the honeydew spots on the filter paper. — Cut out and weigh the spots. — Elute the spots and determine their density in a colorimeter.
a. Area of spots	• Shortly after the ninhydrin treatment, place tracing paper over the treated filter paper and trace the spots. Do not delay the procedure for more than 3 days because the spots fade and will be hard to delineate. • Place the tracing paper with the spots drawn on it over graph paper with mm^2 grids. • Count the number of mm^2 occupied by the spots and express feeding activity as mm^2 of spot area.
b. Weight of spots	• Cut out the portions of the filter paper containing spots. • Weigh the cutout portions and express feeding activity in mg of filter paper stained with honeydew.
c. Color intensity	• The color intensity method measures the amount of ninhydrin reacting amino acids in the honeydew and feeding activity is expressed as μg leucine equivalents/filter paper based on colorimeter readings. First, prepare the following chemicals and develop a standard curve. — *Citrate buffer* (0.2M, pH 5.0). Dissolve 21 g citric acid monohydrate in 200 ml 1.0N NaOH and dilute to 500 ml with water. — *Ninhydrin solution.* Dissolve 20 g ninhydrin in 500 ml methyl cellusolve. Next dissolve 0.8 g $SnCl{\cdot}2H_2O$ in 500 ml of the citrate buffer. Add this solution to the ninhydrin-methyl cellusolve solution. — *Diluent solvent.* Mix equal volumes of H_2O and *n*-propanol. — *Leucine solution (2.0mM stock solution).* 26.24 g leucine/100 ml of 80% ethyl alcohol. • From the 2.0mM leucine stock solution prepare leucine solutions of 6 concentrations ranging from 0.5mM to 2.0mM (0.5, 0.6, 0.7, 0.8, 1.0, and 2.0mM) by mixing with 80% ethyl alcohol. • From each of the leucine concentrations, remove 0.1 ml, place it in a test tube, and add 1 ml of the ninhydrin solution. Mix and heat in a boiling water bath for 20 min. After cooling add 5 ml of the 50% n-propanol diluent solvent. Repeat this process for each of the leucine concentrations 0.5mM, 0.6mM, 2.0mM.

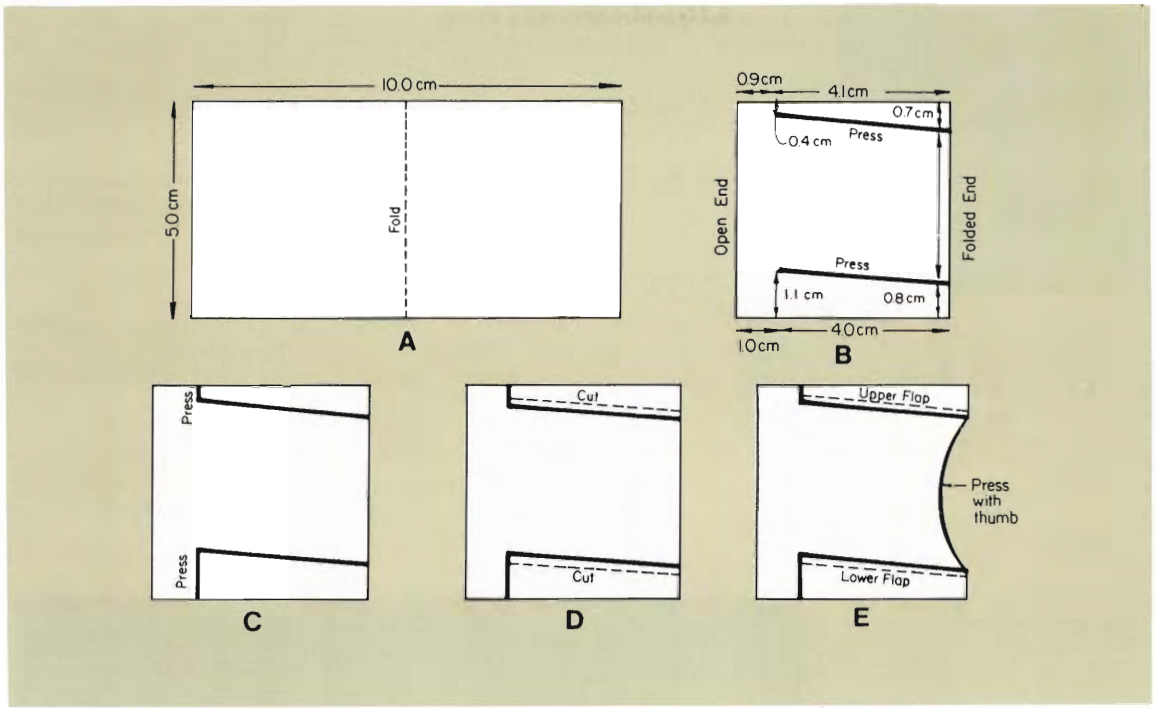
Steps	Key points
	• Read the optical density of each leucine concentration in a colorimeter set at 570 nm using a blue filter. • In developing the standard curve, indicate the optical density on the Y axis and the concentration of leucine on the X axis. Plot the optical density value against the corresponding concentration of leucine. • Elute each filter paper previously sprayed with ninhydrin solution in 5.0 ml of a solution prepared by mixing 0.8 ml of 1.2% copper sulfate and 4.2 ml of 85% alcohol. After the color is removed, place the eluate in a test tube and measure the color intensity in a colorimeter set at 570 nm using a blue filter. • Compare the color intensity of each sample eluted with the standard curve. Determine the concentration as based on the optical density and express it as μg of leucine equivalents/filter paper.
^{32}P activity in honeydew and hoppers	
1. Rearing plants and hoppers	• Follow steps 1-6 under *Ninhydrin-treated filter paper methods.*
2. Labeling plants	• The use of radioactivity to determine the feeding rate of hoppers has been utilized by Velusamy and Chelliah at Tamil Nadu Agricultural University, Coimbatore, India (pers. comm., 1982), as based on Chelliah and Heinrichs (1980). Label the plants as follows: — Immerse the roots of 25-day-old plants of each entry in a flask containing 100 ml of distilled water with 0.5 μc/ml ^{32}P for 24 h. — Transfer each plant to a 100-ml flask containing distilled water.
3. Setting up the cage	• Insert the stem of each plant through a central hole in an inverted plastic petri dish (10-cm-diam). • Place a Whatman No. 40 filter paper (9-cm-diam) around the stem and over the petri dish. • Place an inverted plastic cup cage over the filter paper.
4. Releasing the hoppers	• Release 5 4-day-old female hoppers that have been previously starved for 4 h into each cage through the hole at the top through which the plant is inserted. • Allow the hoppers to feed on the labeled plants for 24 h.

Steps	Key points
5. Measuring radioactivity	• Remove the insects and the filter paper. • Anaesthetize the hoppers. • Record the counts of samples consisting of 5 hoppers and 1 filter paper in a GM counter for 50 s. • The total sap ingested by the hoppers is indirectly determined by summing the radioactivity in the hoppers and in the filter paper.
Bromocresol green-treated filter papers	
1. Steps 1-6 in *Ninhydrin-treated filter paper methods*	• The bromocresol green technique overcomes the disadvantages of the ninhydrin technique and also detects xylem feeding. The disadvantages of the ninhydrin technique follow: 1. Ninhydrin is applied after the feeding test and it is not possible to estimate feeding activity when the test is in progress; and 2. The spots fade rapidly necessitating the measurement of spot areas within a few days after completion of the test. • Prepare the bromocresol green solution by mixing 2 mg bromocresol green powder in 1 ml ethanol (Pathak and Heinrichs 1982b). • Dip filter paper in the bromocresol green solution for 2 min and air-dry for 1 h. • Repeat the dipping process and allow to dry for at least 1 h. The filter paper turns yellow-orange when treated with the solution. Bromocresol green is a pH indicator that is yellow at pH 3.8 and blue-green at pH 5.4.
2. Enclosing plants in cages	• Follow step 8, *Ninhydrin-treated filter paper methods.* It is important that no moisture other than the honeydew touch the filter paper in handling or after it is put in the cage. To prevent moisture from entering through the hole in the center of the inverted petri dish, place cellophane tape over the hole and around the stem of the plant.
3. Arranging the cages	• Arrange cages in a randomized complete block design.
4. Infesting test entries with hoppers	• Introduce 5 female hoppers into each cage through the top of the inverted plastic cup where the stem of the plant passes through. Plug the opening with cotton to prevent escape of the



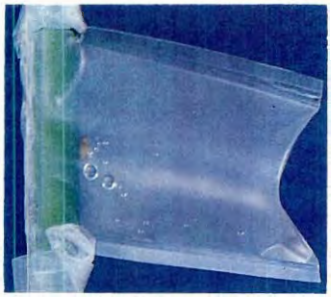
37. Left: apparatus used for the collection of honeydew with bromocresol green-treated filter paper. Right: blue spots produced by the honeydew of the brown planthopper on bromocresol green-treated filter paper and purple spots on a ninhydrin-treated filter paper.

Steps	Key points
	insects. Spots on the filter paper will be evident as soon as honeydew is excreted (Fig. 37).
5. Collecting the filter paper	• After the hoppers have fed for 24 h, remove the filter paper from the cage. Handle the filter paper with forceps so that no moisture from the hands will come in contact with it.
6. Evaluating the area of the honeydew spots	• Because the spots are permanent and will not fade, the area can be measured at your convenience. Keep the filter papers in plastic bags until measurements are to be made. • Proceed to measure the area or determine the weight of the spots in the same manner as described in step 11 in *Ninhydrin-treated filter paper methods.*
Parafilm sachet methods	
1. Rearing plant hoppers	• The parafilm sachet can be used for any hopper species. Follow steps 1-6, *Ninhydrin-treated filter paper methods.*
2. Preparing parafilm sachets	• Parafilm sachets are used for the collection and quantitative determination of honeydew. The amount of honeydew excreted by the hoppers feeding inside the sachets can be determined by the volume and weight of the sachet or through colorimetric analysis. Prepare the parafilm sachets as follows: — Cut parafilm into 10- × 5-cm pieces and fold in the middle (Fig. 38A). — Place the pieces on a smooth surface and seal by pressing with the edge of a microslide to



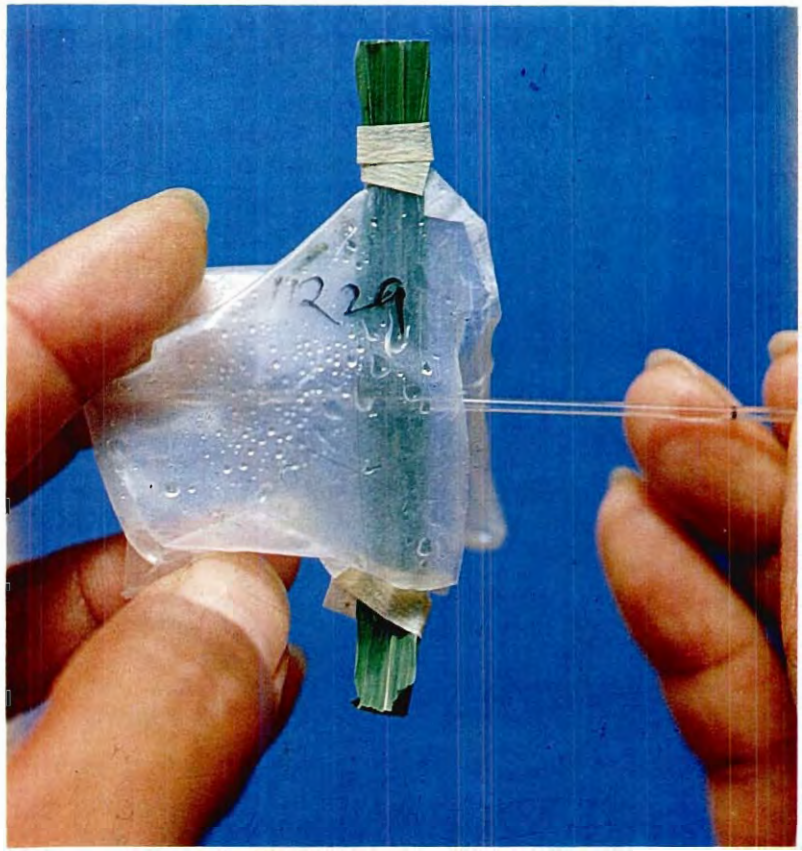
Steps	Key points
	form grooves at specified distances (Fig. 38 B, C). — Cut the upper and lower flap along and parallel to the oblique grooves with scissors (Fig. 38D). — Dilate the sachet by inserting the blunt end of a pencil through the open end and pressing the folded end with the thumb (Fig. 38E). This process increases the capacity of the sachet and permits the test hopper to move freely.
3. Placing sachet on the plant	• Place the edges of the open end around the stem for BPH and at the middle portion of the leaves for WBPH, GLH, and ZLH. Press together the lower portions of the sachet. Stretch the lower flap and wrap it around the stem. Place one sachet/plant, five plants/variety.
4. Releasing hoppers	• With an aspirator, introduce 1 previously starved 2-day-old adult female through the open upper portion of the sachet.
5. Sealing the sachet	• Seal the sachet by pressing its upper open edges together and by winding the upper flap around the

38. Steps in preparing a parafilm sachet for collecting honeydew excreted by planthoppers and leafhoppers feeding on rice plants (Pathak et al 1982).

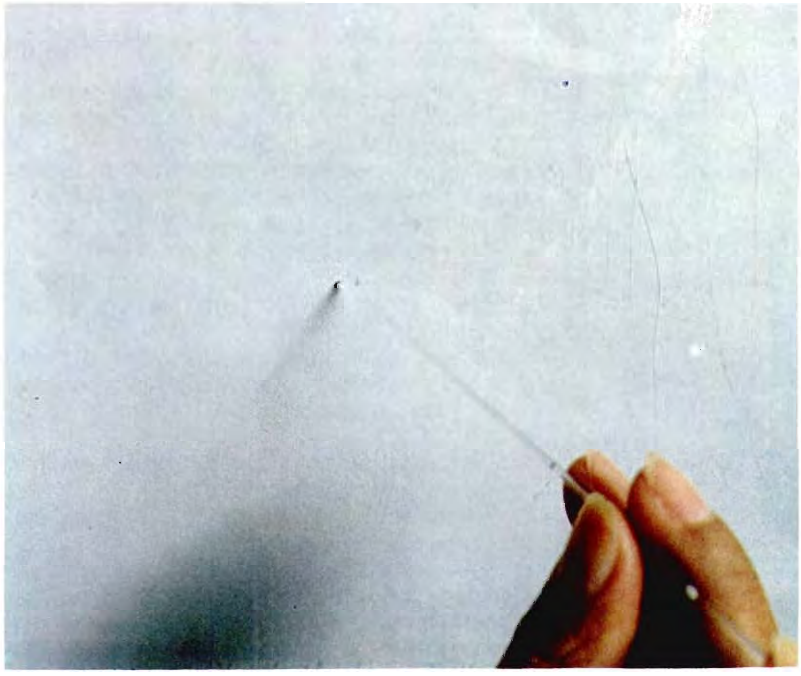


39. Parafilm sachet with the upper and lower flap wrapped around the stem and the open end sealed around the stem. Honeydew excreted by one brown planthopper feeding on a susceptible variety is seen as droplets in the sachet.

Steps	Key points
	stem (Fig. 39). Allow the hopper to feed for 24 h. The excreted honeydew builds up in the outer lower corner of the sachet (Fig. 39).
6. Removing the hopper	• When the feeding period is completed, remove the insect by introducing an aspirator tube through a vertical incision made in the sachet parallel to the stem.
7. Determining the quantity of honeydew excreted a. Weight	• Remove the sachet containing the honeydew from the plant. Handle the sachet with forceps to prevent moisture from the hand from coming in contact with the sachet. • Weigh the sachet with the honeydew. • Blot out the honeydew with filter paper. • Reweigh the sachet after blotting. The difference between the two weights is the weight of the honeydew.



40. Removing green leafhopper honeydew from a parafilm sachet with a micropipette.



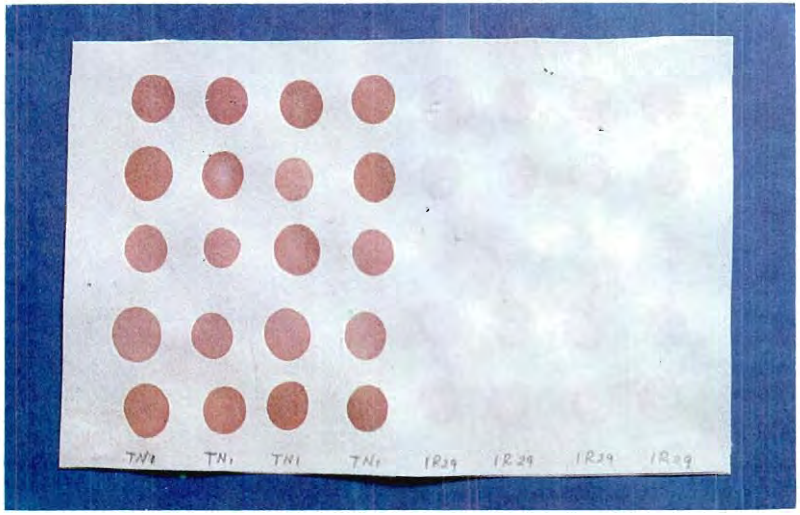
41. Placing 5-μl samples of honeydew on filter paper before treatment with ninhydrin.

Steps	Key points
b. Volume and weight	• Leave the sachet on the plant and through the incision collect the honeydew with calibrated micropipettes of various sizes (1-100 μl). — Weigh the micropipettes before and after collection of honeydew on a 0.001-mg sensitivity balance. — Record the volume of honeydew as based on the measurements with the micropipette. — Determine the specific gravity of the honeydew by comparing its weight/volume with that of distilled water. The weight will be about 1.009 g (Pathak et al 1982). — Multiply the known volume of honeydew as measured with the micropipette by its specific gravity to estimate its weight.
c. Colorimetric method - quantitative estimation of amino acid and sugar content	• The morning after the sachet is placed on the plant, collect 5 samples of 5 μl honeydew from each of the 5 sachets (Fig. 40). The total of 25 samples will provide honeydew for 25 spots of 5 μl each/variety. Consider each spot a replication. • Place a filter paper sheet (20 × 23 cm) over a piece of graph paper. • Deposit 5-μl samples as spots in rows at the intersection of 2 lines on the graph paper until 25 spots are made (Fig. 41)

Steps	Key points
	• Air-dry the honeydew deposits. • For amino acid determination dip the sheet in 0.25% ninhydrin in acetone. Let it develop overnight at room temperature (Fig. 42). • For sugar determination dip in 2% aniline hydrochloride in ethyl alcohol or a benzidine mixture: 0.5 g benzidine in 5 ml acetic acid 4.0 g trichloroacetic acid in 5 ml water 90.0 ml acetone Air-dry in a fume hood for 5 min, and place in an oven at 100° C for 5 min. • Cut sheets in strips and get optical density readings of the spots by total scanning with a densitometer such as Fujiox, model FD-A IV, with a 530-nm filter. • Adjust optical density to the darkest color with a maximum light transmission of 80%. • Determine mean optical density readings per variety by analyzing ninhydrin and aniline hydrochloride or benzidine-treated papers separately. • Compare optical densities among varieties. The higher the optical density, the higher the amino acid and/or sugar content of the honeydew and the higher the feeding activity in the phloem.

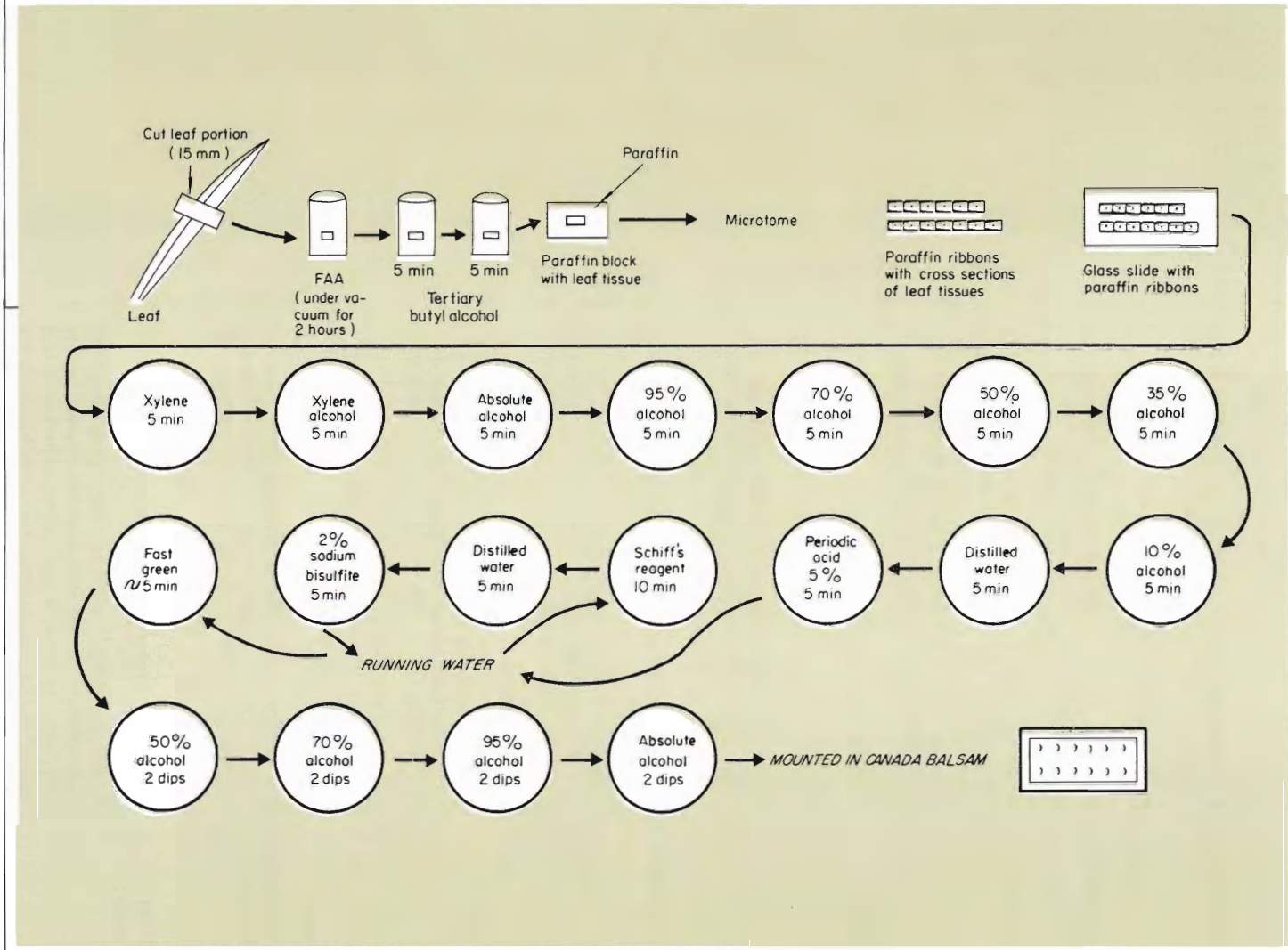
Antibiosis on location of feeding sites

The method used by Pilar Paguia, Dr. R. Maag Ltd., CH-8157 Dielsdorf, Switzerland (pers. comm., 1982), at IRRI is described.



42. Five-μl spots of green leafhopper honeydew from susceptible TN1 (left) and from resistant IR29 (right) after treatment with ninhydrin.

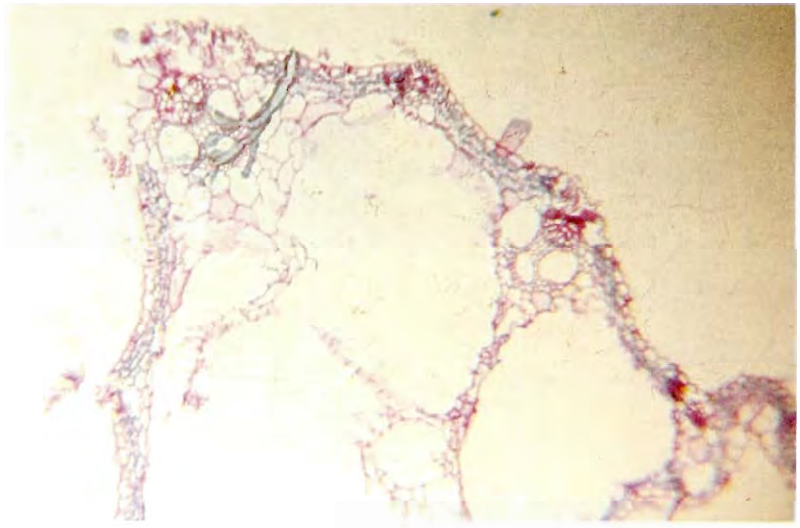
Steps	Key points
1. Allowing insects to feed	• Cage 2 adults in a parafilm sachet attached to the leaves (WBPH, GLH, and ZLH) or leaf sheaths (BPH). • Allow the insects to feed for 24 h.
2. Cutting leaf pieces	• Cut the portion of the leaf or leaf sheath where the insects fed into 15-mm pieces.
3. Fixing the leaf tissues	• Fix the leaf tissues in FAA (40% formalin, 10% acetic acid, and 50% ethyl alcohol) under vacuum for 2 h (Fig. 43). • Dehydrate the tissues in 2 series of tertiary butyl alcohol of 5 min each.
4. Embedding leaf tissues in paraffin wax	• Embed leaf or leaf sheath portions in paraffin wax.
5. Sectioning tissue blocks	• Section tissue blocks at 15 microns using a microtome. Mount paraffin ribbons on a microscope glass slide.
6. Staining the tissue sections	• Use the periodic acid-Schiff (PAS) reaction (Fig. 43, 44). Periodic acid periodic acid, HIO_4 — 1.0 g 90% ethyl alcohol — 100.0 ml Schiff's reagent basic fuchsin, C. I. 42500 — 0.5-1.0 g distilled water — 85.0 ml sodium metabisulfite ($NA_2S_2O_5$) — 1.9 g N HCL — 15.0 ml Sodium bisulfite sodium metabisulfite ($NA_2S_2O_5$) — 0.5 g distilled water — 100.0 ml
7. Mounting tissue on the microscope slide	• If specimens are to be examined later and permanent slides are needed, mount the stained tissue in Canada balsam mounting medium. Place cover slip over the medium.
8. Determining feeding sites	• Examine specimens under a compound microscope. • Look for stylet sheaths and determine the site of termination. In the PAS reaction, stylet sheaths appear red-violet. When differentiated with fast green during the process, the sheaths pick up the



43. Procedure for making paraffin sections and staining rice leaf or leaf sheath sections using the periodic acid-Schiff reactions for feeding site determinations (Humason 1962). The stylet sheaths left by the hoppers take up the stain. The time in each jar is indicated.

Steps	Key points
	green stain (Fig. 45). Figures 46-48 illustrate stylet sheaths of the GLH in different parts of a leaf and leaf sheath and a close-up of a vascular bundle with stylet sheaths.

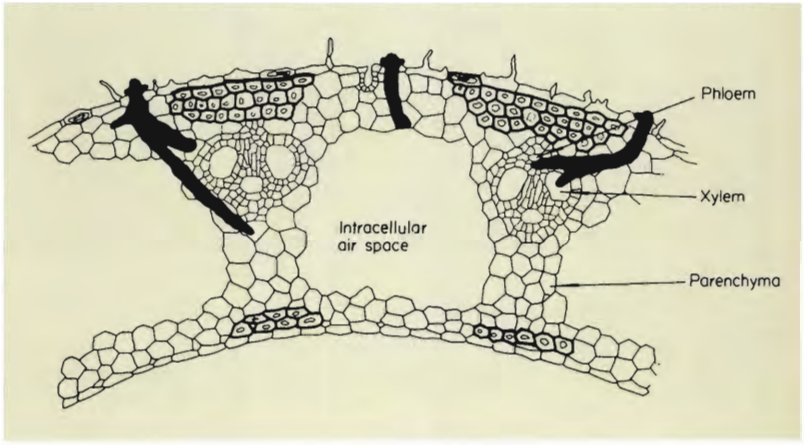
44. Arrangement of jars used in the staining of rice leaf or leaf sheaths when using the periodic acid-Schiff reaction.



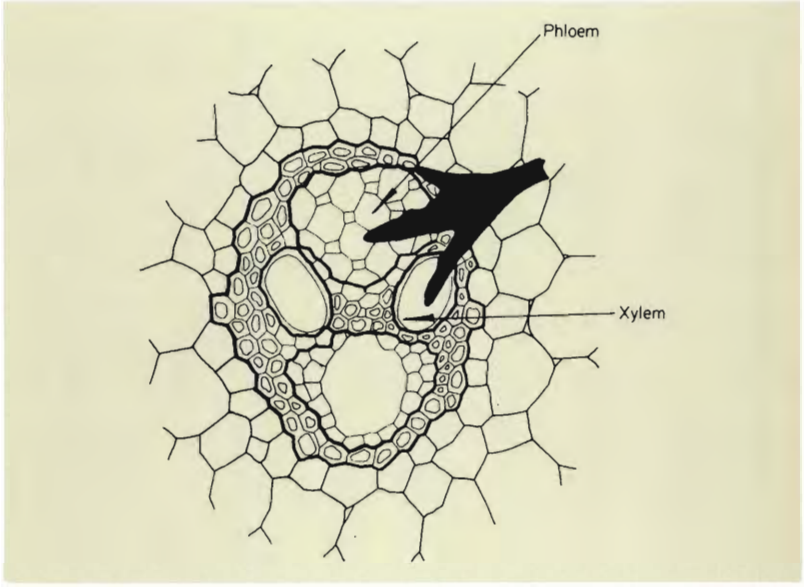
45. Green leafhopper stylet sheath in the leaf of a resistant variety Palasithari 601 stained with periodic acid-Schiff reaction. Note the forking of the terminal portion of the stylet sheath, indicating a search for a desirable feeding location.

46. Cross section of a rice leaf with stylet sheaths of the green leafhopper *Nephotettix virescens* in the mesophyll (left) xylem and phloem (center), and vascular bundle (right).

47. Cross section of a rice leafsheath with stylet sheaths of green leafhopper, *Nephotettix virescens* ending in the parenchyma (left and center) and phloem and xylem (right).



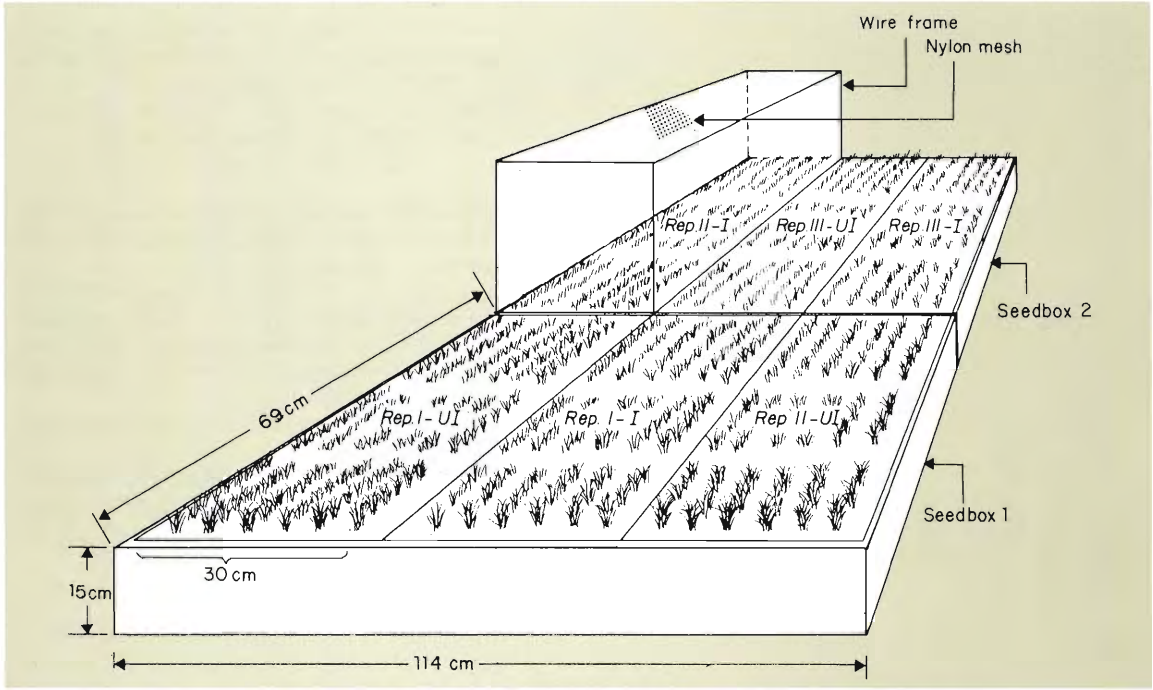
48. Cross section of rice plant vascular bundle with stylet sheaths of green leafhopper *Nephotettix virescens*.



Tolerance in seedbox screening in the greenhouse

Seedbox screening of seedlings in rows is the most rapid method of mass screening for tolerance (Panda and Heinrichs 1983).

Steps	Key points
1. Identifying potentially tolerant entries	• Use the modified seedbox screening test to select entries with damage ratings of 1-5.
2. Preparing seed of the test entries	• For the tolerance test, use remnant seed from the entries selected in the modified seedbox screening test. — Select healthy seed of uniform size. — Include a susceptible check and a resistant check having antibiosis. — Pregerminate 200 seeds per entry.
3. Preparing seedboxes	• Screen 18 entries including a resistant and a susceptible check in 2 adjacent seedboxes, each measuring 114 × 69 × 15 cm (Fig. 49). • Line the inner bottom portion of the seedbox with a polyethylene sheet to regulate moisture for proper plant growth and root system development.



49. Arrangement of varieties 1-18 and replications I-III in two seedboxes in the seedbox screening for tolerance. I = infested with BPH, UI = uninfested, rep = replication.

Steps	Key points
	• Mix complete fertilizer with a soil mixture that consists of 2/3 fertile soil and 1/3 sand and place it in the seedbox. The sand enables you to remove the plants with ease at the end of the test. • Place a mylar film wall (15-cm-high) around the inner wall of the seedbox to prevent water leakage.
4. Sowing seed	• Prepare six blocks in two adjacent seedboxes (Fig. 49). Infest three blocks with hoppers. • Divide each block into 3 sections to accommodate 18 20-cm rows (6 rows/section) with a 5-cm spacing between rows. The 18 test entries are allocated at random to the 18 rows in each block. Randomization is done 6 times — once for each block. Each entry is replicated 3 times under each infestation level. • Sow 15 pregerminated seeds/row. • Place seedboxes in a water pan tray containing water 7 cm deep.
5. Thinning seedlings	• Thin the seedlings to 10/row 7 DAS.
6. Infesting seedlings with BPH	• At 12 DAS, infest the seedlings with nymphs at the rate of 6 nymphs (1- to 2-day-old)/seedling by uniformly distributing 360 nymphs over each group of 6 rows (6 rows × 10 plants × 6 nymphs/plant = 360 nymphs) or 1,080 nymphs (18 rows × 10 plants × 6 nymphs/plant = 1,080) in each block. Place a nylon mesh cage over each block.
7. Evaluating	• When the BPH become adults on the susceptible check entry in any of the blocks (about 14-16 DI), collect them separately on each row of an entry using an aspirator. To prevent movement of BPH from 1 row to another, place a 20-cm-long and 18-cm-high mylar film sheet along each side of the row and a 30-cm-long and 18-cm-high sheet at the end of the rows between the 2 sections. • Grade the plants for damage on a row basis using the SES scale. • Dry the BPH in an oven at 60° C for 48 h. Weigh the BPH to determine the dry weight on a row basis. • Soak the soil with water. Remove the entire plants per row. Take care not to damage the roots. — Wash the soil from the roots. — Air-dry the plants for 2 h. — Dry the plants in an oven at 70° C for 48 h. — Weigh the plants to determine their dry weight.

Steps	Key points
8. Computing the functional plant loss index (FPLI)	• In the computation of each FPLI, the *dry weight of infested plants* and *dry weight of uninfested plants* must come from the same replication. • If the damage rating of a row is 3 or below, compute the FPLI as $\left[1-\left(\frac{\text{dry weight of infested plants}_i}{\text{dry weight of uninfested plants}}\right)\right]\times 100$ If the damage rating is more than 3, compute FPLI as $\left[1-\left(\frac{\text{dry weight of infested plants}}{\text{dry weight of uninfested plants}}\right)\left(1-\frac{\text{damage rating}}{9}\right)\right]\times 100$
9. Analyzing data and identifying tolerant lines	• Run an analysis of variance and Duncan's multiple range test (DMRT) on the FPLI and BPH dry weight (see Gomez and Gomez 1984). These analyses are used to determine the tolerant lines (lines with a significantly lower FPLI than, and a nonsignificantly different BPH dry weight from, the susceptible **check**).

Tolerance of selected entries in pots in the greenhouse

Entries selected from the seedbox screening are further evaluated to confirm which variety possesses antibiosis, tolerance, or both. In this method older potted plants are used and several BPH population levels, rather than just one level, are tested.

Steps	Key points
1. Selecting test entries	• Select test entries with a tolerant reaction as indicated by the seedbox screening method (See *Tolerance in seedbox screening in the greenhouse.*)
2. Preparing seed of test entries	• Use remnant seed from the seedbox screening test. — Select healthy seed of uniform size. — Include a susceptible and a resistant check entry having antibiosis. — Pregerminate 100 seeds/entry.

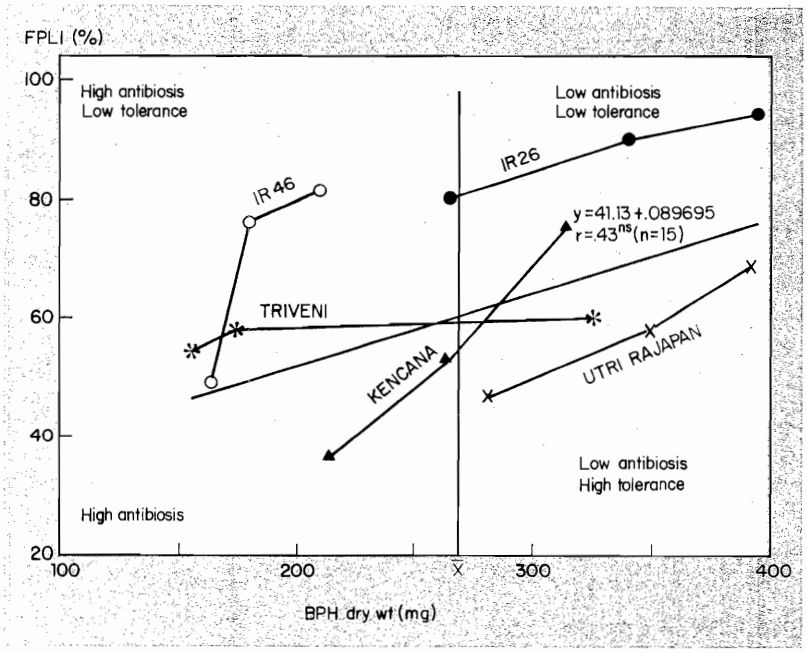
Steps	Key points
3. Preparing pots	• Prepare 5 clay pots (16-cm-diam, 20-cm-high) for each entry and BPH population level (5 pots × 4 BPH population levels = 20 pots for each entry). Each pot is an experimental unit. • Fill the pots with fertile soil. • Sow 4 pregerminated seeds in each pot at a 0.5 to 0.75 cm depth. Sow the 4 seeds 6 cm apart.
4. Watering plants	• Water the soil in the pots as required by using a sprinkler hose or by placing the pots in a water pan tray.
5. Thinning seedlings	• At 7 DAS thin the seedlings to 2/pot. Select test seedlings that are growing normally.
6. Caging the plants	• At 21 DAS wash the plants thoroughly to remove all arthropods and place a 13- × 90-cm mylar film cage over the 2 plants in each pot.
7. Removing arthropods and infesting the plants with BPH	• At 30 DAS again remove any arthropods in the cage. Using an aspirator, place 1st-instar nymphs of uniform age on the test plants through a slit in the cage. • Infest 5 pots of each entry at each of the following 4 levels of BPH populations: 0, 25, 50, and 100 per plant. • Arrange the entries in a completely randomized design in 5 blocks, each block representing a replication. • Split each entry into 4 subplots, each representing the 4 levels of BPH populations.
8. Grading the plants for damage and removing the BPH	• At 30 DI (60 DAS) grade the plants for damage based on the SES scale. Remove the BPH by placing the potted plants in a 40-cm-diam and 50-cm-high plastic container. Remove the mylar cage and tap the plants to dislodge the BPH. Remove any remaining BPH with a camel hair brush and place them in a vial. Remove the BPH at the bottom of the pail with a suction machine and put them in the vial. Do not water the pots 2 days before insect collection because the BPH will stick to the wet pots and make collection difficult.
9. Determining the dry weight of the plants	• Thoroughly wet the soil in the pots. Remove the entire portion of both plants taking care not to damage the roots by removing the soil along with the roots. Remove the soil by washing the roots in running water. Dry the 2 plants in an oven at 70° C, for 60 h. Weigh after drying.

Steps	Key points
10. Determining the dry weight of the BPH	• Dry the insects in an oven at 60° C for 48 h. Weigh after drying. • Determine the FPLI as indicated in step 8 of *Tolerance in seedbox screening in the greenhouse.*
11. Drawing the graph	• Plot a scatter diagram with FPLI on the Y-axis and BPH dry weight on the X-axis. There are 3 points for each entry: each point refers to the mean value averaged over the 5 replications and corresponding to each of the BPH levels (ie, 25, 50, and 100 BPH per plant) — as illustrated in Figure 50.
12. Computing the regression line	• First compute a simple linear regression line $\overline{Y} = a + bx$, where Y = FPLI and X = BPH dry weight, based on all the points in the scatter diagram (see Gomez and Gomez 1984.).

Step 1. Prepare the data table:

Variety	Observation no.	X	Y	X^2	Y^2	XY
V_1	1	X_{11}	Y_{11}			
	2	X_{12}	Y_{12}			
	.	.	.			
	.	.	.			
	.	.	.			
	n_1	X_1n_1	Y_1n_1			
V_2	1	X_{21}	Y_{21}			
	2	X_{22}	Y_{22}			
	.	.	.			
	.	.	.			
	.	.	.			
	.		.			
	n_2	X_2n_2	Y_2n_2			
.						
.						
.						
.						
V_v	1	Xv_1	Yv_1			
	2	Xv_2	Yv_2			
	.	.	.			
	.	.	.			
	.	.	.			
	n_v	Xvn_v	Yvn_v			
Total	n	ΣX	ΣY	ΣX^2	ΣY^2	ΣXY

where X^2 and Y^2 are the squared values of each observation of X and Y, respectively. XY are the product of X and Y values.



50. Identification of components of resistance to *N. lugens* (BPH) using dry weight (antibiosis factor) and functional plant loss index (FPLI) (tolerance indicator) in rice varieties (Panda and Heinrichs 1983).

Step 2. Compute the corrected sum of squares and cross products.

$$\text{The sum of squares: } \Sigma x^2 = \Sigma X^2 - \frac{(\Sigma X)^2}{n}$$

$$\Sigma y^2 = \Sigma Y^2 - \frac{(\Sigma Y)^2}{n}$$

$$\text{The cross product: } \Sigma xy = \Sigma XY - \frac{\Sigma X \, \Sigma Y}{n}$$

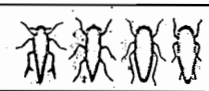
Step 3. Compute the coefficients of the regression lines and the correlation coefficients.

$$\text{The slope: } \quad b = \frac{\Sigma xy}{\Sigma x^2}$$

$$\text{The intercept: } a = \bar{Y} - b\bar{X} \text{ where } \bar{Y} = \frac{\Sigma Y}{n} \text{ and } \bar{X} = \frac{\Sigma X}{n}$$

$$\text{The correlation coefficient: } r = \frac{\Sigma xy}{\sqrt{\Sigma x^2 \, \Sigma y^2}}$$

$$\text{Hence, the estimated line is: } \hat{Y} = a + bX$$



Step 4. Compute the variances necessary for the test of the coefficients.

$$MSE = \frac{\Sigma y^2 - b\Sigma xy}{n-2}$$

$$MSR = b\Sigma xy$$

Step 5. Test the coefficients for significance.

For b: $t_b = \frac{b}{\sqrt{\frac{MSE}{\Sigma x^2}}}$ with $df = n-2$

For r: $t_r = r\sqrt{\frac{n-2}{1-r^2}}$ with $df = n-2$

Step 6. Plot the observed points on a graph distinguishing the varieties by giving symbols for each (Fig. 50).

Step 7. Draw the estimated line on the scatter diagram done in step 6.

Step 8. Draw a vertical line to indicate the mean of the BPH dry weight ($\bar{X}$).

Steps	Key points
13. Interpreting the graph	• Results from an experiment using the above procedures are illustrated in Figure 50. • Utri Rajapan, whose points are located in the lower right section of the graph, has low plant loss (FPLI), despite its ability to support a high BPH population equal to IR36, the susceptible check. This indicates that this variety has tolerance but little antibiosis. The highest levels of tolerance and lowest levels of antibiosis are those varieties whose points would fall in the extreme right and lower section of the graph. • The points of IR26 occur in the upper right section of the graph, which indicates that the variety has a high BPH population and a high FPLI. This means that IR26 has little or no tolerance or antibiosis. Varieties having points in the extreme upper right hand corner would have the least tolerance and antibiosis. • IR46, Kencana, and Triveni, whose points fall mostly to the left of the vertical line (near BPH dry weight), have various levels of antibiosis. The further to the left of the vertical line at which the points occur, the higher the level of antibiosis. Varieties whose points fall in the extreme upper left hand portion of the graph have the highest levels of antibiosis and low or no tolerance.

Tolerance in field evaluation in microplots

Entries that possess tolerance in pot experiments in the greenhouse are further tested in the field. Parameters measured in field testing are plant damage, BPH populations, and grain yield reduction. Field testing allows measurement of a variety's ability to produce grain despite the BPH population it supports. To obtain sufficient BPH populations for valid field testing, infest plants artificially with BPH and place cages over the plots to keep out natural enemies.

Steps	Key points
1. Selecting test entries	• Select test entries based on greenhouse tolerance tests and from breeding lines that are being considered for release to farmers as commercial varieties.
2. Preparing the field layout	• Arrange the microplots in a split plot design with entries as main plots and a BPH-infested and an uninfested treatment as subplots. Replicate the treatments 3 times. • Leave at least a 1-m space between cages (Fig. 33). A 24-× 31-m area (0.07 ha) will accommodate 108 cages (18 entries × 2 infestation rates × 3 replications).
3. Preparing seed	• Use remnant seed from entries selected for tolerance in greenhouse tests or previous field tests, if they are available. Include — a susceptible check, — a resistant check with antibiosis, and — a resistant check with tolerance.
4. Preparing the cages	• Make fiberglass mesh cages measuring 1.5-× 1.5-× 2.0-m (Fig. 33). • Place bamboo stakes at each of the four corners to serve as cage supports.
5. Rearing hoppers	• To artificially infest the plants, use 70 pairs (male and female) per cage or 2 pairs/hill.
6. Sowing seed	• Sow seed in a wetbed nursery. Sufficient seed is necessary to plant 105 seedlings (35 hills × 3 seedlings/hill) in each cage. • Cover the nursery with fiberglass mesh to protect the seedlings from virus vectors.
7. Preparing the field for transplanting	• Prepare the soil before transplanting. • Apply one-third of the recommended rate of

Steps	Key points
	nitrogen fertilizer per crop basally at the last harrowing.
8. Transplanting	• At 14 DAS transplant seedlings into 1- × 1.05-m plots consisting of 5 rows with 7 hills/row at 15-cm spacing between hills and 20-cm between rows.
9. Placing cages over plots	• Immediately after transplanting, cover each plot with fiberglass mesh cage (Fig. 33).
10. Replanting	• At 7 DT replant all poorly growing or dead hills.
11. Removing insect pests and natural enemies	• At 15 DT spray the plants with a short residual insecticide such as decamethrin, which is highly toxic to spiders and other natural enemies but of low toxicity to BPH.
12. Preparing insects for infestation	• At 19 DT place the required number of pairs (male and female) of greenhouse-reared BPH adults (2- to 3-day-old) for each cage, in containers with 30-day-old TN1 seedlings.
13. Infesting the caged plants with hoppers	• At 20 DT remove natural enemies by hand from all the cages. Then, release 70 pairs of hoppers (2 pairs/hill) into each cage that is to receive the BPH-infested treatment.
14. Applying fertilizer	• Broadcast one-third of the recommended rate of fertilizer per crop to the plots 30 DT and another one-third 60 DT.
15. Spraying to control natural enemies	• Check cages for natural enemies weekly. If natural enemies are present, spray plants with a 0.002% concentration of decamethrin.
16. Evaluating	• When the susceptible check begins to wilt, do the following on a weekly basis: — Record the number of BPH/hill on 15 hills at 3 center rows and 5 center hills/row. Express the number as an average of the 15 hills. — Grade the 15 hills for damage using the 0-9 SES scale. — Harvest the entire microplot at maturity. When computing the grain yield adjust grain weight to 14% moisture by $\frac{100 - \text{percent moisture content of grain}}{86} \times \text{weight of harvested grain}$

Steps	Key points
17. Computing yield loss	• Determine percent yield loss for each entry of each replication as $\left(1 - \frac{\text{yield in infested plot}}{\text{yield in uninfested plot}}\right) 100$ • Run an analysis of variance and Duncan's multiple range test to compare — percent yield loss among entries; — BPH populations among entries (select the peak population for each entry); and — plant damage ratings among entries for each observation date.
18. Interpreting the analyses	• Entries that have a BPH population not significantly different from that of the susceptible check have little or no antibiosis. Of the entries with little or no antibiosis, those with significantly lower damage ratings and lower yield reduction than the susceptible check have tolerance. Degree of tolerance is based on amount of yield loss and can be compared among entries based on the separation of means with the DMRT.

SOURCES OF RESISTANCE

Many varieties have been identified as resistant to BPH, WBPH, GLH, and ZLH in IRRI greenhouse screening studies. The variety name, accession number, and origin are given in the following tables.

Varieties resistant or moderately resistant to brown planthopper *Nilaparvata lugens* at IRRI, 1983.

IRRI accession no.	Name	Origin	Reaction[a] to BPH biotypes		
			1	2	3
156	H5	Sri Lanka	R	MR	S
158	H105	Sri Lanka	R	R	S
233	MTU15	India	R	S	R
674	HR12	Sri Lanka	MR	S	R
3472	Murunga 307	Sri Lanka	R	S	R
3473	Murunga 308	Sri Lanka	R	S	R
3475	Sudurvi 305	Sri Lanka	R	S	R
3697	CO 25	India	R	S	R
4897	CO 13	India	MR	S	S
4901	HR19	India	R	S	R

IRRI accession no.	Name	Origin	Reaction[a] to BPH biotypes		
			1	2	3
5920	Ptb 20	India	R	R	R
6069	Anbaw C7	Burma	R	R	S
6113	Ptb 21 (Tekkan)	India	R	R	R
6218	MGL 2	India	R	S	R
6261	CO 10	India	R	S	R
6303	ASD7	India	R	R	S
6365	MTU15	India	R	MR	R
6380	ASD9	India	R	MR	S
6400	CO 22	India	R	S	R
6663	Mudgo	India	R	S	R
7299	Su-yai 20	China	R	S	R
7728	Podimawee	Sri Lanka	R	R	S
7730	Hathiel	Sri Lanka	R	R	R
7731	Kuruhondarawala	Sri Lanka	R	R	R
7733	Gangala	Sri Lanka	R	R	R
7734	Thirissa	Sri Lanka	R	R	R
7752	Balamawee	Sri Lanka	R	R	R
7814	Dikwee	Nigeria	R	R	S
7815	V1	Sri Lanka	MR	R	S
7833	M302/Mas 24 (1900)	Sri Lanka	MR	R	S
7919	MTU9	India	R	S	R
8809	DV9	Bangladesh	MR	S	MR
8900	Sudhubalawee	Sri Lanka	R	S	R
8919	Balamawee	Sri Lanka	R	R	R
8937	Pannetti	Sri Lanka	R	MR	S
8943	Murungabalawee	Sri Lanka	R	S	R
8955	Murungakayan	Sri Lanka	R	R	S
8956	Vellailangayan	Sri Lanka	R	R	S
8957	Muthumanikam	Sri Lanka	R	R	S
8960	Muthumanikam	Sri Lanka	R	MR	R
8967	Berawee	Sri Lanka	R	MR	S
8978	Babawee	Sri Lanka	R	R	R
8990	Seruvellai	Sri Lanka	R	R	S
9073	JC154	Philippines	MR	S	R
9607	XB5	Australia	R	S	MR
9939	Batia Sira	India	MR	MR	S
10633	CO 13	India	R	S	R
10639	AC1730	India	R	MR	R
11054	WC1251	India	R	S	R
11055	WC1253	India	MR	S	R
11056	WC1257	India	R	S	R
11057	WC1263	India	R	S	R
11096	Murungakayan 307	Sri Lanka	R	S	R
11097	Murungakayan 302	Sri Lanka	R	MR	S
11098	Suduwi 306	Sri Lanka	R	S	R
11107	Murungakayan 308	Sri Lanka	R	S	R
11670	Murungan	Sri Lanka	R	MR	S
11673	Bala Murunga	Sri Lanka	R	MR	S
11677	Kosatawee	Sri Lanka	R	R	S
11687	Sinnakayam	Sri Lanka	R	S	R
11697	Sinnasuappu	Sri Lanka	R	R	S
11703	PK-1	Sri Lanka	R	R	S
11716	Malkora	Sri Lanka	R	MR	S
11729	Sirappu Paleusithari	Sri Lanka	R	MR	S
11730	Rathu Heenati	Sri Lanka	R	R	R

IRRI accession no.	Name	Origin	Reaction[a] to BPH biotypes		
			1	2	3
11731	Sinna Karuppan	Sri Lanka	R	R	S
11935	Periamorungan	Sri Lanka	R	R	S
11938	Podiwee	Sri Lanka	R	MR	S
11983	Pawakkulama	Sri Lanka	R	S	R
11956	Mahadikwee	Sri Lanka	R	MR	S
11963	Ovarkaruppan	Sri Lanka	R	R	S
11969	Tibiriwewa	Sri Lanka	R	S	R
11974	Andaragahawewa	Sri Lanka	R	S	R
11978	Heenukkulama	Sri Lanka	R	S	R
12001	Madayal	Sri Lanka	R	MR	S
12066	Kuruluthuduwee B-13	Sri Lanka	R	R	MR
12069	Palasithari 601	Sri Lanka	R	R	S
12071	Murungakayan 3	Sri Lanka	R	MR	S
12072	Murungakayan 101	Sri Lanka	R	R	S
12073	Murungakayan 304	Sri Lanka	R	R	S
12074	Murungakayan 303	Sri Lanka	R	MR	S
12075	Hondarawala 502	Sri Lanka	R	R	R
12076	Hondarawala 378	Sri Lanka	R	R	R
12078	Murungakayan 104	Sri Lanka	R	R	S
12087	Dikwee 328	Sri Lanka	R	R	S
12089	M. I. 329	Sri Lanka	R	MR	S
12094	Maha Murungan, Badulla	Sri Lanka	R	R	S
12308	ARC 6650	India	MR	MR	S
12648	ARC 10834	India	MR	S	MR
13743	WC1252	India	R	S	R
13744	WC1256	India	MR	S	R
13748	Phourel	India	R	S	R
15190	Hondarawala	Sri Lanka	MR	R	R
15192	PK-1	Sri Lanka	R	R	S
15195	Hathili	Sri Lanka	R	R	S
15200	Hathili	Sri Lanka	R	MR	S
15201	Podiwi A8	Sri Lanka	R	R	S
15203	Malalwariyan	Sri Lanka	R	S	MR
15206	Hotel Samba	Sri Lanka	R	R	R
15207	Gangala	Sri Lanka	MR	R	S
15209	Muththessa	Sri Lanka	MR	MR	S
15212	Bathkiriel	Sri Lanka	R	MR	S
15213	Mahamawee	Sri Lanka	R	R	S
15219	Mawee	Sri Lanka	R	R	S
15221	Kalu Mudukiriyal	Sri Lanka	R	MR	S
15222	Molagu Samba	Sri Lanka	R	R	S
15224	Balaratawee	Sri Lanka	R	R	S
15230	Rathna Samba	Sri Lanka	R	R	S
15232	Rathu Balawee	Sri Lanka	R	S	R
15233	Vellai Illankali	Sri Lanka	MR	R	S
15239	Sulai	Sri Lanka	R	R	S
15244	Koththamalli Samba	Sri Lanka	R	R	S
15259	Gangala	Sri Lanka	MR	R	S
15260	Hathiyal	Sri Lanka	R	R	S
15279	Kalukuruwee	Sri Lanka	MR	R	R
15281	Senawee	Sri Lanka	R	MR	R
15285	Kirimurungawee	Sri Lanka	R	R	S
15286	Heenhoranamawee	Sri Lanka	MR	R	MR
15295	Batapala	Sri Lanka	MR	MR	S
15297	Kahata Samba	Sri Lanka	R	R	R

IRRI accession no.	Name	Origin	Reaction[a] to BPH biotypes		
			1	2	3
15298	Kalu Samba	Sri Lanka	R	R	R
15299	Malwaran	Sri Lanka	R	MR	S
15304	Herath Banda	Sri Lanka	MR	MR	MR
15322	Mahamawee	Sri Lanka	MR	R	S
15332	Horanamawee	Sri Lanka	R	R	R
15389	Lekam Samba	Sri Lanka	R	R	R
15397	Muthumanikan	Sri Lanka	R	MR	R
15399	Dampata Podiwee	Sri Lanka	R	R	S
15406	Gambada Samba	Sri Lanka	MR	MR	R
15412	Lekam Samba	Sri Lanka	R	R	R
15421	Sulai	Sri Lanka	R	R	R
15428	Murunga	Sri Lanka	R	S	R
15442	Dikwee	Sri Lanka	R	R	S
15444	Sinna Sivappu	Sri Lanka	R	R	R
15452	Chembavalai Samba	Sri Lanka	R	R	S
15457	Paravaikalayan	Sri Lanka	R	R	S
15467	Murunkan	Sri Lanka	R	R	S
15471	Moddai Karuppan	Sri Lanka	R	R	S
15477	Moddai Karuppan	Sri Lanka	R	R	S
15488	Sudu Hathiyal	Sri Lanka	R	R	S
15489	Mudu Kiriyal	Sri Lanka	R	R	R
15490	Kalu Hathiyal	Sri Lanka	MR	R	S
15491	Gires	Sri Lanka	MR	R	S
15508	Karuppan	Sri Lanka	R	R	S
15513	Perum Karuppan	Sri Lanka	MR	MR	S
15514	Sedukkan Samba	Sri Lanka	MR	MR	S
15516	Vellai Seenetti	Sri Lanka	R	S	R
15520	Vellai Seenadi	Sri Lanka	R	S	R
15532	Hapudevaraja	Sri Lanka	R	R	S
15533	Malwaththawee	Sri Lanka	R	R	S
15534	Godael	Sri Lanka	R	R	S
15535	Puwak eta Hathiyal	Sri Lanka	R	R	S
15536	Sudu Madael	Sri Lanka	MR	R	S
15541	Sudu Hondarawala	Sri Lanka	R	R	R
15545	Muthumanikam	Sri Lanka	R	R	S
15551	Loku Samba	Sri Lanka	R	R	S
15557	Hathiyal	Sri Lanka	R	R	S
15558	Kirikunda	Sri Lanka	R	S	R
15563	Hathiyal	Sri Lanka	R	MR	S
15581	Sudu Madael	Sri Lanka	R	S	R
15582	Kiri Murunga	Sri Lanka	R	S	R
15590	Muppangan	Sri Lanka	R	S	S
15601	Sudu Murunga	Sri Lanka	R	MR	S
15602	Pokkali	Sri Lanka	R	S	R
15604	Japan Heenati	Sri Lanka	R	S	R
15605	Japan Wee	Sri Lanka	R	S	R
15617	Paragahakale	Sri Lanka	MR	R	S
15634	Hondarawala	Sri Lanka	R	R	R
15677	Vellai Seenati	Sri Lanka	MR	S	R
15700	Paravakalayan	Sri Lanka	R	R	S
15719	Mudukiriel	Sri Lanka	R	S	R
15725	Murunga Balawee	Sri Lanka	R	S	R
15726	Hatadawee	Sri Lanka	R	S	R
15744	Hondarawala	Sri Lanka	R	R	R
15749	Sudu Heenati	Sri Lanka	R	S	MR

IRRI accession no.	Name	Origin	Reaction[a] to BPH biotypes		
			1	2	3
15775	CR57-29	Sri Lanka	MR	R	MR
15791	CR94-13	India	R	R	S
19299	HR12	India	R	S	R
19300	M. C. M. 1	Sri Lanka	R	S	R
19301	M. C. M. 2	Sri Lanka	R	S	R
19325	Ptb 33	India	R	R	R
19676	ARC5929	India	R	S	R
19678	ARC5981A	India	R	S	R
19684	Anethoda	Nigeria	R	MR	MR
19930	Karekagga 78	India	MR	R	R
20221	ARC5757	India	MR	S	MR
20237	ARC5785	India	MR	S	R
23806	Bakai	Nepal	R	R	S
24154	IR26	Philippines	R	S	R
26838	ASD14	India	MR	S	R
26955	Chianung Sen 11	Taiwan	R	R	S
30411	IR28	Philippines	R	S	R
30412	IR29	Philippines	R	S	R
30413	IR30	Philippines	R	S	R
30414	IR32	Philippines	R	R	MR
30415	IR34	Philippines	R	S	R
30416	IR36	Philippines	R	R	R
31368	Bata Suduwee	Sri Lanka	R	S	R
31409	Heenmurunga	Sri Lanka	R	S	R
31415	Honderawala	Sri Lanka	MR	R	R
31418	Ismail	Sri Lanka	MR	S	MR
31419	Kachepota	Sri Lanka	R	S	R
31474	Malkora	Sri Lanka	R	R	R
31477	Masmudu Kiriyal	Sri Lanka	R	MR	R
31482	Mawee	Sri Lanka	R	R	R
31501	Murungawee	Sri Lanka	MR	S	R
31517	Polayal	Sri Lanka	R	R	R
31544	Sudu Honderawala	Sri Lanka	R	R	R
31697	Bachoi Kolom	Bangladesh	R	MR	S
31706	Balam Sail	Bangladesh	R	S	R
31816	Jhual Kata	Bangladesh	MR	S	MR
32004	Dong Pueng 147-9-32	Thailand	R	S	R
32536	IR38	Philippines	R	R	R
32630	IR1614-389-1-1	Philippines	MR	S	R
32638	IR1702-74-3	Philippines	R	S	R
32639	IR1702-158-3	Philippines	R	S	R
32642	IR1707-69-3-1	Philippines	MR	S	MR
32670	IR2003-P7-7-4	Philippines	MR	S	MR
32671	IR2003-P7-7-4-2	Philippines	R	S	MR
32674	IR2003-P21-17-3	Philippines	R	S	MR
32675	IR2006-P12-12-2	Philippines	R	S	MR
32676	IR2016-P4-5-3	Philippines	R	S	MR
32680	IR2031-238-5-2-6-2	Philippines	MR	MR	MR
32684	IR2034-289-1-1-1	Philippines	MR	MR	R
32695	IR46	Philippines	R	MR	R
32700	IR2070-401-3-2-4	Philippines	MR	S	MR
32701	IR2070-464-1-3-6	Philippines	R	R	MR
32702	IR2070-834-1-2-2	Philippines	R	MR	R
32703	IR2071-77-9-3-5	Philippines	MR	S	MR
32709	IR2071-559-2-1-3	Philippines	R	MR	S

IRRI accession no.	Name	Origin	Reaction[a] to BPH biotypes		
			1	2	3
32710	IR2071-588-2-5-1	Philippines	MR	S	MR
32720	IR2172-61-2-3	Philippines	MR	MR	MR
32721	IR2172-64	Philippines	R	S	R
32736	IR3265-193-3	Philippines	MR	S	MR
35172	SXC108	India	MR	S	MR
36218	Alewee	Sri Lanka	R	S	R
36223	Ballawala	Sri Lanka	MR	S	R
36228	Demala Balawee	Sri Lanka	R	S	R
36230	Dhemaswee	Sri Lanka	R	S	R
36234	Galhonderawalu	Sri Lanka	R	S	R
36245	Hapurajah	Sri Lanka	R	S	R
36246	Hatapanduru	Sri Lanka	R	S	R
36247	Hathe pas dawase wee	Sri Lanka	R	S	R
36248	Hathial	Sri Lanka	R	R	R
36257	Iriwee	Sri Lanka	R	S	R
36264	Kalu Balawee	Sri Lanka	MR	S	R
36267	Kalu Heenati	Sri Lanka	R	S	R
36288	Katahata Hamba	Sri Lanka	R	MR	R
36293	Kohu Mawee	Sri Lanka	R	R	R
36295	Kolayal	Sri Lanka	R	R	R
36303	Kuru Hondarawalu	Sri Lanka	R	R	R
36319	Maha Mawee	Sri Lanka	R	S	R
36320	Maha Samba	Sri Lanka	MR	R	R
36322	Manchel Paragahakaleyan	Sri Lanka	R	S	R
36323	Mas Handiran	Sri Lanka	R	R	R
36324	Molligu Samba	Sri Lanka	R	R	R
36342	Noorti Onru	Sri Lanka	MR	S	MR
36343	Perun Karuppan	Sri Lanka	MR	S	MR
36352	Poliyal	Sri Lanka	R	MR	R
36374	Sayan	Sri Lanka	MR	S	R
36396	Ugu Samba	Sri Lanka	R	MR	MR
36412	Anekoda	Sri Lanka	MR	R	S
36421	Chella Nayagam	Sri Lanka	MR	MR	MR
36424	Dewaradha	Sri Lanka	R	R	MR
36445	Heta Punduru Wee	Sri Lanka	R	R	MR
36446	H. M. C.	Sri Lanka	R	MR	S
36463	Kokkali	Sri Lanka	R	S	MR
36478	Maha Kathiyal	Sri Lanka	R	MR	S
36479	Maharajah Wee	Sri Lanka	MR	MR	S
36480	Maharathkunda Wee	Sri Lanka	R	S	R
36482	Malawariya	Sri Lanka	R	S	R
36958	IR40	Philippines	R	MR	S
36959	IR42	Philippines	R	MR	S
39247	CR157-392-4	India	R	R	R
39244	CR126-42-5	India	R	R	R
39292	IR2071-625-1-252	Philippines	R	R	MR
39341	IR44	Philippines	R	R	R
39621	CR190-162-13	India	R	R	MR
39645	IR2071-747-6-3-2	Philippines	R	R	R
40947	ARC11321	India	R	MR	MR
41016	ARC12176	India	MR	MR	MR
41541	ARC14395	India	R	MR	MR
41566	ARC14453	India	R	MR	R
41569	ARC14465	India	R	MR	MR
41580	ARC14515	India	R	R	MR

IRRI accession no.	Name	Origin	Reaction[a] to BPH biotypes		
			1	2	3
41598	ARC14548	India	R	MR	MR
41603	ARC14559	India	R	MR	MR
41672	ARC14664	India	R	MR	MR
41676	ARC14668	India	R	MR	MR
41677	ARC14669	India	R	MR	MR
41678	ARC14670	India	MR	MR	MR
41679	ARC14671	India	R	MR	MR
41732	ARC14740	India	R	MR	MR
41739	ARC14764	India	MR	MR	MR
41747	ARC14774	India	R	R	MR
41759	ARC14807	India	MR	MR	MR
41978	ARC15242	India	MR	MR	MR
41997	ARC15285	India	R	MR	R
41998	ARC15286	India	R	MR	R
42018	ARC15339	India	MR	MR	MR
42033	ARC15381	India	R	R	MR
42041	ARC15457	India	MR	MR	MR
47425	Sivuru Wee	Sri Lanka	R	R	R
47675	IR45	Philippines	R	R	R
49174	Chotasoia	Bangladesh	R	R	MR
49192	Guiaroi	Bangladesh	R	R	R
	C62-1-230	Taiwan	R	R	S
	C62-1-373	Taiwan	MR	R	S
	RP9-6	India	MR	S	R
	IR1561-288	Philippines	R	S	R
54260	Padi Bureng	Indonesia	R	[b]	–
54639	Arivakkari	India	R	–	–
54641	Aryan Manjeri	India	R	–	–
54647	Chara	India	R	–	–
54675	IC142	India	R	–	–
54676	IC148	India	R	–	–
54696	J1-35	India	R	–	–
54724	JH5	India	R	–	–
54727	Kalladachamban	India	R	–	–
54729	Karappan	India	R	–	–
54739	Kayamma	India	R	–	–
54744	Kurippala	India	R	–	–
54754	Navara	India	R	–	–
54777	T213	India	R	–	–
54778	T231	India	R	–	–
54780	T243	India	R	–	–
54822	T10022	India	R	–	–
54826	Thulunadan	India	R	–	–
54834	Veluthakaruka	India	R	–	–
54867	K47	India	R	–	–
54872	K54	India	R	–	–
54882	K68	India	R	–	–
54892	K288	India	R	–	–
54953	K619	India	R	–	–
54987	K815-5	India	R	–	–
55010	K1008	India	R	–	–
55055	ADR43	India	R	–	–
55056	ADR54	India	R	–	–
55057	ADR68	India	R	–	–
55070	Chemban	India	R	–	–
55182	Veluthancheera	India	R	–	–

IRRI accession no.	Name	Origin	Reaction[a] to BPH biotypes		
			1	2	3
55189	231	India	R	–	–
55196	1371	India	R	–	–
55198	1437	India	R	–	–
55199	1464	India	R	–	–
55204	3069	India	R	–	–
55205	5352	India	R	–	–
55206	8378	India	R	–	–
55209	8880	India	R	–	–
55210	8895	India	R	–	–

[a]R = resistant, MR = moderately resistant, S = susceptible. [b]Not tested.

Wild rice species resistant to brown planthopper biotypes 1, 2, and 3 at IRRI, 1983.

IRRI accession no.	Species	Origin	Reaction[a] to biotypes		
			1	2	3
100115	*O. brachyantha*	Africa	R	R	R
100165	*O. latifolia*	Guatemala	R	R	S
100167	*O. latifolia*	Costa Rica	R	MR	R
100168	*O. latifolia*	Costa Rica	R	S	MR
100169	*O. latifolia*	Costa Rica	R	R	R
100170	*O. latifolia*	Costa Rica	R	R	R
100171	*O. latifolia*	Guatemala	R	S	R
100172	*O. latifolia*	Guatemala	S	S	MR
100179	*O. officinalis*	Japan	R	R	R
100180	*O. officinalis*	Malaya	R	R	R
100181	*O. officinalis*	Burma	R	R	R
100195	*O. nivara*	Burma	S	R	R
100211	*O. sativa/O. nivara*	India	R	R	R
100224	*O. barthii*	Gambia	R	R	R
100820	*O. ridleyi*	Japan	R	MR	MR
100821	*O. ridleyi*	Japan	R	MR	MR
100878	*O. officinalis*	Thailand	R	R	R
100882	*O. australiensis*	India	R	R	R
100883	*O. officinalis*	India	R	R	R
100886	*O. punctata*	India	R	R	MR
100893	*O. brachyantha*	India	R	R	R
100895	*O. latifolia*	USA	R	MR	R
100896	*O. officinalis*	Thailand	R	R	R
100914	*O. latifolia*	Mexico	R	S	R
100920	*O. nivara/O. rufipogon*	Malaysia	R	R	S
100937	*O. punctata*	Ghana	R	R	R
100947	*O. officinalis*	India	R	R	R
100948	*O. officinalis*	India	R	R	R
100953	*O. officinalis*	India	R	R	R
100954	*O. punctata*	India	R	R	R
100956	*O. latifolia*	India	R	MR	S
100959	*O. latifolia*	Mexico	R	S	S
100962	*O. latifolia*	Guatemala	R	MR	MR
100963	*O. latifolia*	Guatemala	R	MR	S
100964	*O. latifolia*	Guatemala	R	S	MR

IRRI accession no.	Species	Origin	Reaction[a] to biotypes		
			1	2	3
100965	*O. latifolia*	Costa Rica	R	S	S
100966	*O. latifolia*	Panama	R	S	S
100973	*O. officinalis*	Philippines	R	R	R
101073	*O. officinalis*	Philippines	R	R	R
101074	*O. officinalis*	Philippines	R	R	R
101077	*O. officinalis*	Philippines	R	R	R
101078	*O. officinalis*	Philippines	R	R	R
101079	*O. minuta*	Philippines	R	R	R
101081	*O. minuta*	Philippines	R	R	R
101082	*O. minuta*	Philippines	R	R	R
101083	*O. minuta*	Philippines	R	R	R
101084	*O. minuta*	Philippines	R	R	R
101086	*O. minuta*	Philippines	R	R	R
101089	*O. minuta*	Philippines	R	R	R
101092	*O. minuta*	Philippines	R	R	R
101094	*O. minuta*	Philippines	R	R	R
101096	*O. minuta*	Philippines	R	R	R
101097	*O. minuta*	Philippines	R	R	R
101099	*O. minuta*	Philippines	R	R	R
101100	*O. minuta*	Philippines	R	R	R
101101	*O. minuta*	Philippines	R	R	R
101112	*O. officinalis*	Philippines	R	R	R
101113	*O. officinalis*	Philippines	R	R	R
101114	*O. officinalis*	Philippines	R	R	R
101115	*O. officinalis*	Philippines	R	R	R
101116	*O. officinalis*	Philippines	R	R	R
101117	*O. officinalis*	Philippines	R	R	R
101118	*O. officinalis*	Philippines	R	R	R
101121	*O. officinalis*	Philippines	R	R	R
101122	*O. minuta*	Philippines	R	R	R
101123	*O. minuta*	Philippines	R	R	R
101124	*O. minuta*	Philippines	R	R	R
101125	*O. minuta*	Philippines	R	R	R
101126	*O. minuta*	Philippines	R	R	R
101128	*O. minuta*	Philippines	R	R	R
101129	*O. minuta*	Philippines	R	R	R
101132	*O. minuta*	Philippines	R	R	R
101133	*O. minuta*	Philippines	R	R	R
101134	*O. minuta*	Philippines	R	R	R
101135	*O. paraguaensis*	Philippines	R	R	R
101137	*O. officinalis*	Philippines	R	R	R
101139	*O. officinalis*	Philippines	R	R	R
101141	*O. minuta*	Philippines	R	R	R
101142	*O. officinalis*	Philippines	R	R	R
101143	*O. minuta*	Philippines	R	R	R
101144	*O. australiensis*	Australia	R	R	R
101149	*O. officinalis*	Malaysia	R	R	R
101150	*O. officinalis*	Malaysia	R	R	R
101151	*O. officinalis*	Malaysia	R	R	R
101152	*O. officinalis*	Malaysia	R	R	R
101154	*O. officinalis*	Malaysia	R	R	R
101155	*O. officinalis*	Malaysia	R	S	R
101166	*O. officinalis*	Philippines	R	R	R
101171	*O. punctata*	Tanzania	MR	R	S
101200	*O. barthii*	Nigeria	R	S	R
101243	*O. barthii*	Mali	MR	R	R

IRRI accession no.	Species	Origin	Reaction[a] to biotypes		
			1	2	3
101329	*O. punctata*	Nigeria	R	R	S
101386	*O. minuta*	Japan	R	R	R
101387	*O. minuta*	Japan	R	R	R
101392	*O. latifolia*	Guatemala	MR	R	S
101395	*O. alta*	USA (USDA)	R	R	R
101397	*O. australiensis*	USA (USDA)	R	R	R
101399	*O. officinalis*	Vietnam	R	R	R
101408	*O. punctata*	Ghana	R	R	R
101409	*O. punctata*	Ghana	R	R	S
101410	*O. australiensis*	Australia	R	R	R
101412	*O. officinalis*	India	R	R	R
101414	*O. officinalis*	India	R	S	R
101417	*O. punctata*	Kenya	R	MR	S
101418	*O. eichingeri*	Uganda	R	R	R
101421	*O. eichingeri*	Uganda	R	R	R
101422	*O. eichingeri*	Uganda	MR	R	S
101424	*O. eichingeri*	Uganda	R	R	R
101426	*O. eichingeri*	Uganda	R	R	MR
101429	*O. eichingeri*	Uganda	R	R	R
101430	*O. punctata*	Uganda	R	R	S
101434	*O. punctata*	Tanzania	R	R	R
101439	*O. punctata*	Ghana	MR	R	R
101441	*O. punctata*	–	R	R	R
101443	*O. latifolia*	Morocco	R	S	S
101524	*O. nivara*	India	MR	S	MR
101943	*O. rufipogon/O. nivara*	Taiwan	MR	MR	MR
101973	*O. nivara*	India	MR	S	S
102164	*O. nivara*	India	R	R	R
102165	*O. nivara*	India	R	R	R
102166	*O. nivara*	India	R	R	R
102167	*O. nivara*	India	R	R	R
102168	*O. nivara/O. rufipogon*	India	R	R	R
102169	*O. nivara*	India	R	R	R
102175	*O. nivara*	India	MR	MR	MR
102176	*O. nivara*	India	R	R	R
102178	*O. nivara*	India	R	R	R
102179	*O. nivara*	India	R	R	R
102382	*O. officinalis*	Indonesia	R	R	R
102385	*O. officinalis*	Indonesia	R	R	R
102481	*O. latifolia*	Nicaragua	R	S	S

[a]R = resistant, MR = moderately resistant, S = susceptible.

Varieties resistant to whitebacked planthopper *Sogatella furcifera* at IRRI, 1983.[a]

IRRI accession no.	Name	Origin	IRRI accession no.	Name	Origin
2459	Kinko	Japan	6829	Ratua 81	Pakistan
3391	T1	Guyana	6834	Toga 378	Pakistan
3520	CI5662-2	Japan	6836	JKW S39	Pakistan
3650	CI6008-1	India	6837	Toga 356	Pakistan
3651	CI6008-2	India	6838	CM7-6	Pakistan
3652	Mushkan	India	6839	Palman 46	Pakistan
3653	CI6010-1	India	6840	JKW141	Pakistan
3657	Safed	India	7466	C5560	Thailand
3658	CI6015-1	India	7733	Gangala	Sri Lanka
3660	CI6018-1	India	7792	Kogbati 3	Sierra Leone
3661	CI6018-2	India	8607	DD 4	Bangladesh
3662	CI6018-3	India	8978	Babawee	Sri Lanka
3663	Bansi	India	9014	Jhona 160	Pakistan
3664	CI6037-1	India	9016	Jhona 5704	Pakistan
3665	CI6037-2	India	9017	Jhona 5715	Pakistan
3666	CI6037-3	India	9018	Jhona 5716	Pakistan
3702	NP130	India	9019	Jhona 5723	Pakistan
3718	T1	India	9020	Jhona 5867	Pakistan
3720	T43	India	9021	Jhona 6029	Pakistan
3721	T136	India	9022	Jhona 6036	Pakistan
3722	T137	India	9023	Jhona 6053	Pakistan
3724	Ambeniohor 12	India	9024	Jhona RF 57	Pakistan
3737	S67	India	9032	Basmati 5854	Pakistan
3738	GS 398	India	9037	Basmati 6129	Pakistan
3742	Jhona	India	10573	1 (PI 184675-6)	Iran
3743	346 Mahlar	India	10623	Mahlar 346	India
3980	Desi 2	India	10625	C5-17	India
4819	N22	India	10626	S 12 D. Z. K.	India
4820	T1	India	10630	Muchkan 41	India
5821	AR1	India	10637	AC 1443	India
6040	ADT16	India	10642	C-5281	India
6144	FR13A	India	10643	C-5298	India
6255	Early Sutarsar 39	India	10771	Siam Garden (Chiclayo)	Peru
6264	N22	India	13742	WC 1240	India
6273	T43	India	14418	IARI 5804	India
6294	T1	India	14547	N-136 (PI 337328)	India
6307	Jhona 349	India	14548	Suda (PI 337329)	India
6337	B76	India	14758	Hevino	Ghana
6394	HR22	India	15173	Miro Miro	Senegal
6418	Mushkan 41	Pakistan	15190	Hondarawala	Sri Lanka
6419	Toga 28	Pakistan	15420	Sivuruwee	Sri Lanka
6520	Son 14	Pakistan	15421	Sulai	Sri Lanka
6425	Ratua 394	Pakistan	15433	Weli Handiran	Sri Lanka
6427	Mahlar 346	Pakistan	15735	Heen Rath	Sri Lanka
6429	Palman 46	Pakistan	15859	N'diang Marie	Senegal
6436	T137	India	16140	Oha	Nepal
6662	Colombo	India	16195	Gokhue Saier	Nepal
6663	Mudgo	India	16197	Chaia Anaser	Nepal
6818	Kaki Ratii 421	Pakistan	16249	Katuyhar Dhan	Nepal
6821	Jhona 349	Pakistan	16251	Thalie	Nepal
6822	JKW S20	Pakistan	16254	Nakhi	Nepal
6823	RDS19	Pakistan	16262	Ghaiya	Nepal
6824	JKWS18	Pakistan	16270	Lalki Saro	Nepal

IRRI accession no.	Name	Origin
16277	Kataunsa	Nepal
16278	Nakhi	Nepal
16637	Tjempo Tolo	Indonesia
17039	Dasal	India
17048	Bohai	Taiwan
17099	Hawm Jampah	Thailand
17179	Badigul	Indonesia
17331	Brondol	Indonesia
17411	Cere Merah	Indonesia
19283	Indel	Indonesia
19577	Kannazhakan 0.69-78	India
19673	RP 79-13	India
19682	Mettasannalu	India
19915	Forastero "Cacao"	Colombia
19916	Forastero (Mono bola blanco)	Colombia
19928	Cheriviruppu	India
19977	Kretek Emas	India
19978	Kumpulan	Indonesia
20803	ARC 10239	India
23813	Bansphul	Nepal
23860	Dharia	Nepal
23861	Dhera Dun Basmati	Nepal
23871	Faram Bajari	Nepal
23939	Karmuli	Nepal
23941	Khasi (Prasad)	Nepal
23990	Nemai	Nepal
23994	Padhanee	Nepal
24008	Raj Bhog	Nepal
24013	Ram Kajara	Nepal
24017	Rango	Nepal
24049	Sonkhas Cha	Nepal
24750	Mundam Kuriak	Indonesia
27925	Dhan 263	Pakistan
27929	Dhan 300	Pakistan
27930	Dhan 403	Pakistan
27933	Dud Malai 198A	Pakistan
27939	Hansraj S-15	Pakistan
27942	Hansraj 54	Pakistan
27943	Hansraj 54	Pakistan
27960	Jawari Surkh 328	Pakistan
27961	Jhona Babri K. W. 418	Pakistan
27963	Jhona Mamun 117	Pakistan
27964	Jhona Sherbati 371	Pakistan
27965	Jhona Sufaid 67	Pakistan
27966	Jhona White Nehri 42	Pakistan
27967	Jhona 26	Pakistan
27968	Jhona 44	Pakistan
27970	Jhona 76	Pakistan
27971	Jhona 79S	Pakistan
27972	Jhona 82C	Pakistan
27973	Jhona 83	Pakistan
27974	Jhona 86	Pakistan
27975	Jhona 91A	Pakistan
27976	Jhona 101	Pakistan
27977	Jhona 104S	Pakistan
27979	Jhona 116A	Pakistan
27980	Jhona 129	Pakistan
27981	Jhona 129	Pakistan
27983	Jhona 153	Pakistan
27984	Jhona 155	Pakistan
27985	Jhona 156S	Pakistan
27986	Jhona 178	Pakistan
27987	Jhona 183	Pakistan
27988	Jhona 191	Pakistan
27989	Jhona 192	Pakistan
27990	Jhona 193	Pakistan
27991	Jhona 213	Pakistan
27993	Jhona 298	Pakistan
27994	Jhona 393	Pakistan
27995	Jhona 426/37	Pakistan
28007	Kangni 27	Pakistan
28013	Kasarawala & Mundara 116	Pakistan
28022	Lal Dhan 304	Pakistan
28023	Lal Dhan 304	Pakistan
28035	Malhar 314/5	Pakistan
28042	Munji Sufaid 23A	Pakistan
28052	Munji 389	Pakistan
28083	NC 3/6	Pakistan
28107	P 603	Pakistan
28192	Ratua 32-2	Pakistan
28265	Siah Nakidar 195	Pakistan
28299	Sufaida 172	Pakistan
28326	Ziri Palman 434	Pakistan
28333	2/38 1	Pakistan
28359	24A/10	Pakistan
28361	26A	Pakistan
28423	173	Pakistan
28431	224	Pakistan
28439	274	Pakistan
28442	293	Pakistan
28448	357	Pakistan
28449	360	Pakistan
28451	368	Pakistan
33832	Unnamed	Burma
35072	NP 97	India
36282	Kalu Mahatmaya	Sri Lanka
36342	Noorti Onru	Sri Lanka
36354	Pukuru Pansi	Sri Lanka
38607	R 149	Nepal
38655	Coarse White 40	Pakistan
39739	RP 107-15	India
40880	Sudu Weda Heenatti	Sri Lanka
40946	ARC 11316 (brown furrows)	India
44181	UPRB 5	India
44182	UPRB 6	India
44183	UPRB 7	India
44184	UPRB 8	India
44205	UPRB 29	India

IRRI accession no.	Name	Origin
44246	UPRB 74	India
44247	UPRB 75	India
44255	UPRB 84	India
44256	UPRB 85	India
44819	Black Gora S. N. 32	India
44862	Brown Gora S. N. 112	India
44868	SN 118	India
44869	White Gora S. N. 2	India
44870	White Gora S. N. 5	India
44871	White Gora S. N. 6	India
44873	White Gora S. N. 13	India
44891	White Gora S. N. 116	India
44893	White Gora S. N. 119	India
44936	Anupama	India
44941	Arjunsail	India
44991	AC 511	India
45093	Basantobahar	India
45234	Brown Gora	India
45408	CR 113-74	India
45428	CR 141-4004-1-191	India
45429	CR 141-4004-2-192	India
45500	Deola	India
45555	Dhusuri	India
45628	DA 27-C	India
45631	DW 2	India
45924	Jhulma	India
45987	Kalanuniabhog	India
45992	Kali Aus	India
46318	Maru	India
46473	OC 134	India
46477	OC 1393 Horimoi	India
46478	OC 1393 Mut. 29	India
46491	Pafuri	India
46510	Dhala Bokra	India
46536	Phenidhula	India
46538	Postasilet	India
46544	PAU 225B-79	India
46550	Radjakata	India
46561	Ramajowan	India
46585	Rupsail	India
46586	Rupsail	India
46598	RP 79-3	India
46603	RP 79-27	India
46604	RP 79-28	India
46612	Sachi	India
46625	Sadalaghu	India
46628	Safa	India
46631	Sahebsail	India
46632	Saket 1	India
46633	Saket 1	India
46643	Sankarjata	India
46647	Sankarkalma	India
46654	Sarunagra	India
46660	Sathi 34-36	India
46661	Sathi 34-36	India
46662	Sathi 34-46	India
46678	Sindurmukhi	India
46736	Teenpakhia	India
46787	W 398	India
46854	Unnamed	India
46855	Unnamed	India
46863	Unnamed	India
46917	Unnamed	India
46919	Unnamed	India
46949	Unnamed	India
47011	IS 38	Ivory Coast
47407	Pokkalian	Sri Lanka
48151	Khao Niaw Leuang	Thailand
49172	Chengri	Bangladesh
49256	Ratoi	Bangladesh
49539	C. P. 12	India
49544	Cross 1	Australia
49564	Cuttack 13	India
49681	Hansraj	India
50240	Vellathan	India
50241	Vellayatham	India
50740	Desi Munji	India
50742	Desi Munji	India
50987	Horana Wee	Sri Lanka
51762	NCS 2	India
51829	NCS 75	India
51895	NCS 148	India
51918	NCS 178	India
51923	NCS 183	India
51926	NCS 188	India
51938	NCS 201	India
52052	Babigocha	India
52151	Jorai Vadhu	India
52164	Kesari Chipti	India
52180	Kumorohadanti	India
52256	Tella Kalchlu	India
52271	Badansal	India
52276	Bhavadya	India
52283	Davadya	India
52313	Jaigaon 5	India
52338	Kusali Sal	India
52353	Nagpur 22	India
52403	Asha	India
52411	Basmati Sal	India
52416	Cippi	India
52420	Colam (Dhanuri)	India
52422	Dangar	India
52425	Danger Sal	India
52433	Dholi Sal	India
52436	Dilli Sutar	India
52437	Dilli Sutar	India
52459	Lakkar	India
52467	Mamra Jadi Sal	India

IRRI accession no.	Name	Origin	IRRI accession no.	Name	Origin
52471	Parmal	India	52478	Safed Badi Sal	India
52474	Patli Sal	India	52560	Lakkar	India

[a]Varieties with ratings of 1 and 3 based on damage scale 1-9:1 = resistant, 9 = susceptible.

Wild rice species resistant to whitebacked planthopper at IRRI, 1983.

IRRI accession no.	Species	Origin	IRRI accession no.	Species	Origin
100168	*O. latifolia*	Costa Rica	101122	*O. minuta*	Philippines
100172	*O. latifolia*	Guatemala	101123	*O. minuta*	Philippines
100179	*O. officinalis*	Japan	101124	*O. minuta*	Philippines
100180	*O. officinalis*	Malaya	101125	*O. minuta*	Philippines
100181	*O. officinalis*	Burma	101126	*O. minuta*	Philippines
100820	*O. ridleyi*	Japan	101128	*O. minuta*	Philippines
100821	*O. ridleyi*	Japan	101129	*O. minuta*	Philippines
100878	*O. officinalis*	Thailand	101132	*O. minuta*	Philippines
100883	*O. officinalis*	India	101133	*O. minuta*	Philippines
100916	*O. rufipogon*	China	101134	*O. minuta*	Philippines
100959	*O. latifolia*	Mexico	101135	*O. paraguaensis*	Philippines
100962	*O. latifolia*	Guatemala	101137	*O. officinalis*	Philippines
100963	*O. latifolia*	Guatemala	101139	*O. officinalis*	Philippines
100964	*O. latifolia*	Guatemala	101141	*O. minuta*	Philippines
100965	*O. latifolia*	Costa Rica	101142	*O. officinalis*	Philippines
100973	*O. officinalis*	Philippines	101143	*O. minuta*	Philippines
101073	*O. officinalis*	Philippines	101149	*O. officinalis*	Malaysia
101074	*O. officinalis*	Philippines	101150	*O. officinalis*	Malaysia
101077	*O. officinalis*	Philippines	101151	*O. officinalis*	Malaysia
101078	*O. officinalis*	Philippines	101152	*O. officinalis*	Malaysia
101079	*O. minuta*	Philippines	101154	*O. officinalis*	Malaysia
101081	*O. minuta*	Philippines	101155	*O. officinalis*	Malaysia
101082	*O. minuta*	Philippines	101166	*O. officinalis*	Philippines
101083	*O. minuta*	Philippines	101171	*O. punctata*	Tanzania
101084	*O. minuta*	Philippines	101329	*O. punctata*	Nigeria
101086	*O. minuta*	Philippines	101386	*O. minuta*	Japan
101089	*O. minuta*	Philippines	101387	*O. minuta*	Japan
101092	*O. minuta*	Philippines	101399	*O. officinalis*	Vietnam
101094	*O. minuta*	Philippines	101412	*O. officinalis*	India
101096	*O. minuta*	Philippines	101414	*O. officinalis*	India
101097	*O. minuta*	Philippines	101417	*O. punctata*	Kenya
101099	*O. minuta*	Philippines	101418	*O. eichingeri*	Uganda
101100	*O. minuta*	Philippines	101421	*O. eichingeri*	Uganda
101101	*O. minuta*	Philippines	101422	*O. eichingeri*	Uganda
101112	*O. officinalis*	Philippines	101424	*O. eichingeri*	Uganda
101113	*O. officinalis*	Philippines	101426	*O. eichingeri*	Uganda
101114	*O. officinalis*	Philippines	101429	*O. eichingeri*	Uganda
101115	*O. officinalis*	Philippines	101430	*O. punctata*	Uganda
101116	*O. officinalis*	Philippines	101434	*O. punctata*	Uganda
101117	*O. officinalis*	Philippines	101979	*O. nivara*	India
101118	*O. officinalis*	Philippines	102382	*O. officinalis*	Indonesia
101121	*O. officinalis*	Philippines	102385	*O. officinalis*	Indonesia

Varieties resistant and moderately resistant to the green leafhopper *Nephotettix virescens* at IRRI, 1983.

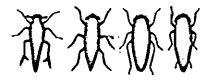
IRRI accession no.	Name	Origin[a]	IRRI accession no.	Name	Origin
33	Bengawan	Indonesia	4435	Tung-can-ro-mao	China
34	Tjere Mas	Indonesia	4537	Lenkan-mi-thou-goo	China
41	B436	Philippines	4539	Lien-tsan 50	China
45	AC435	Taiwan	4553	Gu-wen-gu-gi-goo	China
46	C34	–	4660	Wan-li-shun	China
48	FB86	Philippines	4661	Shu-ya-tsan	China
66	T1242	India	4910	India pa lil 46	Sierra Leone
160	59-325 (B 11/Mas)	Sri Lanka	4919	Bambako	Malawi
210	Nang Keo	Vietnam	5089	4105	Iran
315	RPP49	Philippines	5147	Kanan 13 Original	Taiwan
326	TP MIL 53	Philippines	5263	FB 123	Philippines
611	Sigadis	Indonesia	5324	Sigadis	Indonesia
679	Remadja	Indonesia	5325	Remadja	Indonesia
681	Tilakkachray	India	5326	Djelita	Indonesia
831	Gam Pai 30-12-15	Thailand	5333	Peta	Indonesia
834	Pin Gew Bow 17-4-27	Thailand	5351	221 BC4-45-3	Indonesia
846	Gam Pai 30-12-41	Thailand	5408	RT 80	Indonesia
869	Gow Ruang 21-1-59	Thailand	5410	Simbando (Local)	Indonesia
876	Gam Pai 30-12-78	Thailand	5411	Lead	Senegal
1563	Ping Shan Ta Tswen Ku	China	5452	Campena	Philippines
2113	Calrose Sel.	USA	5458	AC2382 (Siam 29)	–
3392	T1	Guyana	5462	FB 26	Philippines
3397	Hashikalmi	Surinam	5464	FW 182	Philippines
3453	PI 223456	Afghanistan	5475	Gantang	Malaysia
3454	1150-S	Afghanistan	5779	TP/R (76889-160)	USA
3511	PI 184676	Iran	5806	D99	Malawi
3512	Dom Siah	Iran	5811	Baishbish	Pakistan
3514	Sadri Rice 1	Iran	5815	Chinsurah 19	India
3609	Bengawan	Indonesia	6101	PTB27	India
3622	Intan 2400	Indonesia	6105	PTB18	India
3627	Mas 2401	Indonesia	6113	PTB21	India
3629	Peta 2802	Indonesia	6129	Baiang 6	Indonesia
3630	Pulut Nangka 016	Indonesia	6267	ASD1	India
3633	Remadja	Indonesia	6288	Chinsurah 2	India
3634	Peta (PI 233289)	Indonesia	6303	ASD7	India
3673	Bhura Rata 2	India	6331	CO 18	India
3690	CO 9	India	6380	ASD9	India
4021	Binicol	Philippines	6393	ASD8	India
4095	Sigadia	Indonesia	6412	ADT14	India
4120	Cateto 22	Haiti	6433	T23	India
4142	Hnanwa Phingauk	Burma	6445	D204-1	India
4143	Khayangya	Burma	6497	Laki 495	Bangladesh
4216	Ladang	Indonesia	6498	Katia Bagder 13-20	Bangladesh
4285	Ta-poo-cho z	China	6504	Khama 49-8	Bangladesh
4326	Leu-wei-thou	China	6518	Fulkari 653	Bangladesh
4334	Bir-mee-tsan	China	6520	Laki 115	Bangladesh
4335	Bir-tsan 3	China	6528	Laki 544	Bangladesh
4348	Bir-co-ru-mao	China	6539	Fulkari 715	Bangladesh
4349	Bir-co-se-mao 7	China	6541	Bhadoia 233	Bangladesh
4353	Bir-mur-z	China	6552	Laramon 720	Bangladesh
4356	Bir-chau-gee	China	6554	Laki 720	Bangladesh
4395	Si-shan-z-thou	China	6562	Badal 292	Bangladesh
4414	Sa-tsan 35	China	6566	Baguamon 436	Bangladesh

IRRI accession no.	Name	Origin	IRRI accession no.	Name	Origin
6576	Laki 383	Bangladesh	8834	DV71	Bangladesh
6578	Khama 49-2	Bangladesh	8839	DV85	Bangladesh
6590	Baguamon 14	Bangladesh	8840	DV86	Bangladesh
6607	Laki 462	Bangladesh	8870	DV139	Bangladesh
6611	Bagiamon 202	Bangladesh	8888	T412	India
6613	Kalimekri 77-5	Bangladesh	8899	Maharatawee	Sri Lanka
6621	Laki 396	Bangladesh	9113	JC68	India
6622	Laki 659	Bangladesh	9135	Ham Moon	Hongkong
6624	Laki 146	Bangladesh	9152	JC41	India
6642	HD19	Australia	9957	Koimurali	India
6643	HD20	Australia	10024	H. P. 334	India
6865	221 BC4-1-45-10	Indonesia	10030	Rangadoria	India
6867	221 BC4-178-13	Indonesia	10059	Addey	India
7279	Nawn	Thailand	10076	H. P. 21	India
7299	Su-yai 20	China	10079	Malsara	India
7367	PI 184675-3	Iran	10085	H. P. 34	India
7368	PI 184675-4	Iran	10086	H. P. 35	India
7555	Kanfor 13	Philippines	10106	H. P. 68	India
7556	Kanfor M3 S 1	Philippines	10123	Boka	India
7564	221 BC4-1-45-6-12	Indonesia	10144	H. P. 112	India
7566	221 BC4-1-45-8-1	Indonesia	10295	PB 76-66D-113	Philippines
7567	221 BC4-1-45-8-2	Indonesia	10298	C-18	Philippines
7571	221 BC4-1-45-10-3	Indonesia	10300	C-13	Philippines
7579	221 BC4-1-178-6-3	Indonesia	10301	Dom-Sia	–
7730	Hathiel	Sri Lanka	10569	2	Afghanistan
8184	Betong	Indonesia	10570	1 (PI 184675-1)	Iran
8321	Tilockkachari	Bangladesh	10593	Shala	Indonesia
8336	Jhingasail	Bangladesh	10594	Sigardis	Indonesia
8342	Godalki	Bangladesh	10596	Dara	Indonesia
8366	DNJ95	Bangladesh	10597	Sibudijang	Indonesia
8368	DNJ85	Bangladesh	10612	Ch. 47	India
8392	DNJ27	Bangladesh	10617	G. S. 784	India
8393	DNJ25	Bangladesh	10779	40c/1.5.07	Surinam
8401	DNJ11	Bangladesh	10817	295/8/1/5/2	Indonesia
8403	DNJ9	Bangladesh	10819	295/8/1/9/3	Indonesia
8421	DNJ153	Bangladesh	10823	Line 8	Malaysia
8454	DNJ97	Bangladesh	10825	Line 5	Malaysia
8459	DJ115	Bangladesh	10827	Line 19	Malaysia
8479	DJ90	Bangladesh	10828	294/4/1/10/1/3	Indonesia
8511	DJ9	Bangladesh	10872	Daw Leaung 133-3-88	Thailand
8548	DZ104	Bangladesh	11052	PTB18	India
8553	DZ84	Bangladesh	11134	Hybrid 1	India
8559	DZ54	Bangladesh	11142	Ngakywe Taung pyan	Burma
8567	DZ21	Bangladesh	11184	Lasaw	Philippines
8573	DS1	Bangladesh	11357	IR127-80-1-10	Philippines
8574	DK1	Bangladesh	11361	IR253-4-1-2-1	Philippines
8589	D. IN 7	Bangladesh	11533	Do Khao	Laos
8594	DL 7	Bangladesh	11616	Chao Pho Kha	Laos
8679	CTG428	Bangladesh	11697	Sinnasuappu	Sri Lanka
8682	CTG534	Bangladesh	11700	Rathu Honderawala	Sri Lanka
8732	UCP32	Bangladesh	12069	Palasithari 601	Sri Lanka
8760	DM19	Bangladesh	12070	Vanam	Sri Lanka
8794	UCP122	Bangladesh	12105	Kile-Ame	Vietnam
8796	DM77	Bangladesh	12114	Muoy Roy Chey	Cambodia
8802	UCP195	Bangladesh	12177	ARC6006	India
8816	DV29	Bangladesh	12186	ARC6038	India

IRRI accession no.	Name	Origin
12192	ARC6046	India
12194	ARC6050	India
12205	ARC6076	India
12216	ARC6102	India
12239	ARC6160	India
12249	ARC6180	India
12293	ARC6608	India
12314	ARC7012	India
12316	ARC7059	India
12361	ARC7306	India
12396	ARC10229	India
12404	ARC10243	India
12420	ARC10281	India
12559	ARC10656	India
12599	ARC10746	India
12635	ARC10804	India
12646	ARC10826	India
12669	ARC10905	India
13017	T442-90	Philippines
13018	T442-1-58	Philippines
13387	Fatehpur-3	Pakistan
13502	C8442	Indonesia
13535	Tjere Omas	Indonesia
14438	IARI-10376	India
14526	Lawangin	Afghanistan
14640	Selak	Indonesia
14702	CPA6809-74	Thailand
14730	Hybrid line 1	Iran
14733	Hybrid line 4	Iran
14748	Mani Coyo X Gbali	Ghana
14749	Mango	Ghana
14751	Umelia	Ghana
14755	Kobi Kamo	Ghana
14756	Bawku Red	Ghana
14758	Hevino	Ghana
14759	Mmo Kokoo	Ghana
14761	Mmo Tumtum	Ghana
14762	Mani Coyo	Ghana
14775	Harlan Coll. -25B	Nigeria
14777	Kaosen Yu 12	Philippines
14880	96	Liberia
14976	IR272-4-1-2 (Mala)	Philippines
14978	Bignou 11	Senegal
15164	Avieble	Ivory Coast
15166	Kpecekre N	Ivory Coast
15167	Kpecekre P	Ivory Coast
15169	Sakpale	Ivory Coast
15183	Hathili	Sri Lanka
15195	Hathili	Sri Lanka
15197	Sambawee	Sri Lanka
15200	Hathili	Sri Lanka
15260	Hathiyal	Sri Lanka
15281	Senawee	Sri Lanka
15286	Heenhoranamawee	Sri Lanka
15452	Chembavalai Samba	Sri Lanka
15489	Mudu Kiriyal	Sri Lanka
15541	Sudu Hondarawala	Sri Lanka
15549	Podi Samba	Sri Lanka
15551	Loku Samba	Sri Lanka
15554	Muthu Samba	Sri Lanka
15555	Sigardis	Sri Lanka
15567	Honderawala	Sri Lanka
15679	Heenati	Sri Lanka
15683	Kulawalai	Sri Lanka
15687	Sivappu Palaisithari	Sri Lanka
15717	Sulai	Sri Lanka
15720	Rathkal	Sri Lanka
15735	Heen Rath	Sri Lanka
15769	Lawangeen	Afghanistan
15771	ARC7119	India
15774	ARC7316	India
15802	Limbanfing	Senegal
15804	Sicoumba-Sigueuleme	Senegal
15806	Thiorna Balde	Senegal
15807	Bignou 1	Senegal
15808	Bignou 2	Senegal
15814	Kalor	Senegal
15823	Diorna Essadoya	Senegal
15832	Kamossor 2	Senegal
15836	Tendeng	Senegal
15881	Lasane	Senegal
15882	Rhissa	Senegal
15883	Sefa	Senegal
15884	Ernest	Senegal
15917	Oulaigue Yanari	Senegal
15948	Amoule Borome	Senegal
15964	Kabero	Senegal
15987	Bolon Bala	Senegal
16001	Minding Messing	Senegal
16012	Wansiline	Senegal
16014	Sidou	Senegal
16193	Bageri	Nepal
16246	Gadur	Nepal
16250	Sokan Dhan	Nepal
16259	Ghaiya	Nepal
16404	Dua Satu	Indonesia
16439	Kentjlam	Indonesia
16456	Ketan Kretek Mas	Indonesia
16598	Tjempo Kenongo	Indonesia
16630	Tjempo Sintho	Indonesia
17007	Nep Dau	Vietnam
17091	Fawng Nam	Thailand
17115	Khao Prajuab	Thailand
17134	Soi Daruni	Thailand
17194	Balap Merah	Indonesia
17195	Balap Putih	Indonesia
17273	Bengawan Omas	Indonesia
17474	Djaliah Merah	Indonesia
17575	Gadis Ciamis	Indonesia
17586	Gaja Baru	Indonesia
17602	Gapita	Indonesia
17614	Gedi	Indonesia

IRRI accession no.	Name	Origin
17733	Hegar Manah	Indonesia
17748	Indel	Indonesia
17751	Inten	Indonesia
18003	Klenyam	Indonesia
18258	Menur Tjempa	Indonesia
18498	PN4. Rambangan	Indonesia
18615	Rayat	Indonesia
18632	Retant	Indonesia
18645	Rondo Jeblot	Indonesia
18712	Sedan Mundur	Indonesia
18736	Segon Perak	Indonesia
18824	Sidara	Indonesia
18832	Siem	Indonesia
18863	Sintawati	Indonesia
18870	Sipendek	Indonesia
18930	Srogel Abang	Indonesia
18948	Swadaja	Indonesia
18977	Terong	Indonesia
19012	Tjempo Djelita	Indonesia
19032	Tjempo Makmur	Indonesia
19128	Tjere Sentral	Indonesia
19142	Tjo Unus	Indonesia
19196	Intan	Indonesia
19216	Bakka Batjere	Indonesia
19223	Bakka Lompo	Indonesia
19265	Cere Emas	Indonesia
19267	Dewi Arimbi	Indonesia
19317	PTB-24	India
19331	Bilugyun Ngasein C 32	–
19333	CO 13 × CO 4 6522 B4	–
19357	Shannyein C 24-71	–
19358	Siam 11	–
19525	Kalias	Malaysia
19577	Kannazhakan 0.69-28 (PI 365271)	India
19930	Karekagga 78	India
20105	Rambu	Indonesia
20260	ARC 5842	India
20353	ARC 6132	India
20498	ARC 7098	India
20599	ARC 7320	India
20845	ARC 10313	India
21037	ARC 10697	India
21055	ARC 10733	India
21165	ARC 10981	India
21174	ARC 10995	India
21175	ARC 10998	India
21182	ARC 11060	India
21211	ARC 11130	India
21235	ARC 11218	India
21266	ARC 11264	India
21311	ARC 11317	India
21322	ARC 11330	India
21473	ARC 11554	India
21476	ARC 11558	India
21960	ARC 12172	India
22723	Ang Sar K 118	Cambodia
22774	Chhmar Khmao	Cambodia
22775	Chhmar Kraham	Cambodia
22877	Kang Soy	Cambodia
22898	Kaun Trei K. T.	Cambodia
22930	Kon Khmao	Cambodia
23108	Neang Saath	Cambodia
23788	Andi	Nepal
23811	Bango Masino	Nepal
23816	Bayarni	Nepal
23823	Bhoolani	Nepal
23835	Chiruakhu	Nepal
23874	Gajali	Nepal
23876	Gardi	Nepal
23886	Gurra	Nepal
24048	Sokan	Nepal
24076	White Atte	Nepal
24141	RD 5	Thailand
24142	Sokowa	India
24174	BKN 6625-109-1	Thailand
24175	BKN 6802 (1)-3-2	Thailand
24197	Long Tong	Vietnam
24242	ARC-614	India
24263	ARC 11208	India
24329	Gam Pai 30-12-30	Thailand
24331	Gow Ruang 21-1-3	Thailand
24332	Gow Ruang 88	Thailand
24346	Jek Gradod 85-4-34	Thailand
24348	Khao Cha Ngae 54-6-2	Thailand
24356	Khao Leaung 50-4-163	Thailand
24400	Leaung Yai 49-2-119	Thailand
24412	Ma Kheua Leu 133-2-36	Thailand
24413	Ma Kheua Leu 133-2-63	Thailand
24431	Nawn 51-9-79	Thailand
24448	Pah Leaung 29-11-112	Thailand
24453	Pah Puang 73-4-19	Thailand
24468	Sai Bua 57-106-2	Thailand
24474	Tewadah	Thailand
24481	IR5491 (GLH_1)	Philippines
24482	IR5492 (GLH_2)	Philippines
24537	Ase Hindi	Indonesia
24687	Sintha	Indonesia
24688	Teja Sari 2	Indonesia
24806	Sipuluik Uleik	Indonesia
24902	Cendana	Indonesia
24908	Dempet Terong	Indonesia
24924	Gadis	Indonesia
25075	Pare Mas	Indonesia
25149	Sri Gaya	Indonesia
25168	Apolo	Indonesia
25309	Tokaluku	Indonesia
25487	Padi Cinta	Indonesia
25494	Padi Grundul 2	Indonesia
25548	Padi Sudara	Indonesia
25645	Rawa	Indonesia
25703	Sikaloko	Indonesia

IRRI accession no.	Name	Origin	IRRI accession no.	Name	Origin
25753	Sipilian	Indonesia	26401	Moni Mukul	Bangladesh
25827	Aghanisail	Bangladesh	26407	Neel Huri	Bangladesh
25830	Arai	Bangladesh	26412	Porichok	Bangladesh
25831	Ashni	Bangladesh	26419	Shor Shori	Bangladesh
25833	Aus Jhari	Bangladesh	26420	Suna Digha	Bangladesh
25834	Aus Murali	Bangladesh	26426	Ajoldiga	Bangladesh
25836	Bakoi	Bangladesh	26427	Arichadiga	Bangladesh
25838	Bathuri	Bangladesh	26435	Bhug	Bangladesh
25842	Bondyl	Bangladesh	26437	Bile Boira	Bangladesh
25844	Chamka	Bangladesh	26438	Boira Luta	Bangladesh
25847	Chengri	Bangladesh	26439	Bora Diga	Bangladesh
25848	Chiknal	Bangladesh	26445	Chanda Amon	Bangladesh
25851	Dhalashaita	Bangladesh	26454	Digha	Bangladesh
25852	Dumai	Bangladesh	26456	Dudh Bazal	Bangladesh
25854	Garia	Bangladesh	26475	Jatra Motuk	Bangladesh
25855	Chungur Bali	Bangladesh	26478	Jhul Diga	Bangladesh
25858	Gungur Murali	Bangladesh	26479	Joli Amon	Bangladesh
25861	Hasha	Bangladesh	26492	Khoiamotor	Bangladesh
25862	Hijolee	Bangladesh	26494	Kola Mona	Bangladesh
25863	Holai	Bangladesh	26498	Lal Amon	Bangladesh
25907	Muijuri	Bangladesh	26499	Lal Chamara	Bangladesh
25911	Pankhiraj	Bangladesh	26507	Moish Mira	Bangladesh
25920	Sampatti	Bangladesh	26515	Nedpasha	Bangladesh
25926	Terabali	Bangladesh	26517	Raj Bhawalia	Bangladesh
25957	Bombilla	Brazil	26519	Rangi Khama	Bangladesh
26288	Asmaita	Bangladesh	26521	Sada Bazal (2)	Bangladesh
26289	Aswina	Bangladesh	26524	Shuli Dhan	Bangladesh
26291	Badal 2	Bangladesh	26529	Sungail	Bangladesh
26292	Bagdar	Bangladesh	26530	Sungwala	Bangladesh
26295	Bale Betor	Bangladesh	26533	Akand Sail	Bangladesh
26300	Bawoi Jhak	Bangladesh	26548	Baliya Aman	Bangladesh
26301	Bazal	Bangladesh	26560	Bharat	Bangladesh
26304	Betoir	Bangladesh	26578	Borojyot	Bangladesh
26306	Bhaira Nota	Bangladesh	26585	Buta Sail	Bangladesh
26307	Bhawalia	Bangladesh	26601	Dinga Manik	Bangladesh
26308	Bhawalia Diga	Bangladesh	26604	Dhol Kochuri	Bangladesh
26310	Bhoro Diga	Bangladesh	26605	Dulaiti	Bangladesh
26311	Bhoro Nepa	Bangladesh	26610	Dudraj	Bangladesh
26318	Boron 2	Bangladesh	26611	Dudsar	Bangladesh
26320	Chamar	Bangladesh	26612	Dushor	Bangladesh
26322	Chingair	Bangladesh	26622	Gia Dhan	Bangladesh
26323	Choron Bawla	Bangladesh	26623	Goar Sail	Bangladesh
26325	Chota Digha	Bangladesh	26630	Gopal Bhog	Bangladesh
26329	Dev Moni	Bangladesh	26633	Gurdoi	Bangladesh
26332	Digha (2)	Bangladesh	26645	Indra Sail	Bangladesh
26333	Dol Kochu	Bangladesh	26661	Kabra Balam	Bangladesh
26334	Dol Kochu (2)	Bangladesh	26663	Kaisha Binni	Bangladesh
26341	Fulkari	Bangladesh	26665	Kalabinni	Bangladesh
26345	Goria	Bangladesh	26677	Karti Gotak	Bangladesh
26347	Gour Kajol	Bangladesh	26682	Kashia Binni	Bangladesh
26349	Gutok	Bangladesh	26686	Khakkol	Bangladesh
26350	Hijli Digha	Bangladesh	26689	Khirloni	Bangladesh
26382	Khoya Motor	Bangladesh	26691	Khorma	Bangladesh
26383	Koia Maguri	Bangladesh	26693	Kochudhola	Bangladesh
26389	Laki	Bangladesh	26694	Koha Binni	Bangladesh
26400	Mohish Kani	Bangladesh	26707	Lal Binni	Bangladesh



IRRI accession no.	Name	Origin
26712	Lalpat Jat	Bangladesh
26717	Loiatag	Bangladesh
26719	Lota Sail	Bangladesh
26736	Moishabida	Bangladesh
26737	Moisha Mira	Bangladesh
26745	Muta Gorol	Bangladesh
26748	Nak Pasi	Bangladesh
26755	Nidan Sail	Bangladesh
26757	Nona Sail	Bangladesh
26771	Pat Jat	Bangladesh
26772	Pura Binni	Bangladesh
26796	Shul Kumor	Bangladesh
26803	Surjamukhi	Bangladesh
26861	SLO 4	India
26910	MK 47-22	India
26914	Chitadium	Bangladesh
26916	LD125	Sri Lanka
26947	ARC614-25 (b)	India
26952	Biplab	Bangladesh
26955	Chianung Sen 11	Taiwan
26957	CR1113	Costa Rica
26959	Goa Ruang 88	Thailand
26967	Milagrosa Mutant	Philippines
27011	Ase Garis	Indonesia
27019	Ase Lagading	Indonesia
27077	Bakka Sunggu	Indonesia
27493	Tosidongi	Indonesia
27520	Agauh	Bangladesh
27625	Nahng Nuan	Thailand
27759	Khao Ti-nueng	Thailand
28029	Magoi Sem 75	Pakistan
28030	Mahla 422	Pakistan
28040	Mundara Dhan 90S	Pakistan
28164	P718	Pakistan
28166	P729	Pakistan
28173	P740	Pakistan
28180	Ratria	Pakistan
28181	Ratrio	Pakistan
29153	Aus 367	Bangladesh
29433	Tupa 353	Bangladesh
29444	Tupa 729	Bangladesh
29766	Khao Chao Phokka	Laos
29829	Khao Pouka	Laos
30557	Menyarhunei	Liberia
31304	Liberian Coll. B 26	Liberia
31583	Jholi Aus	Bangladesh
31612	Nuncha	Bangladesh
31614	Porangi	Bangladesh
31633	Sona Biron	Bangladesh
31657	Iron	Bangladesh
31666	Koara	Bangladesh
31667	Keora	Bangladesh
31670	Lal Aswina	Bangladesh
31675	Molla Diga	Bangladesh
31740	Biron	Bangladesh
31744	Biruin	Bangladesh
31746	Bish Katari	Bangladesh
31751	Chakkol	Bangladesh
31770	Digha	Bangladesh
31774	Dudh Kolom	Bangladesh
31780	Dumai Sail	Bangladesh
31790	Gorfa	Bangladesh
31831	Kala Kura	Bangladesh
31839	Kapa Diga	Bangladesh
31840	Karim Sail	Bangladesh
31843	Kartik Srimuin	Bangladesh
31872	Mainamoti	Bangladesh
31874	Maloti	Bangladesh
31884	Moina Sail	Bangladesh
31886	Montonga	Bangladesh
31888	Moyna Moti	Bangladesh
31904	Pahari Balam	Bangladesh
32007	Jao Daeng 63-6-19	Thailand
32022	PW 201	Thailand
32290	Damsiah	Iran
32291	Domsiah	Iran
32311	Hassan Tareme	Iran
32313	Massan Mulat	Iran
32315	Mulai	Iran
32316	Mulai	Iran
32317	Mulai	Iran
32318	Mulai	Iran
32320	Mussatarem	Iran
32323	Mussa Tareme	Iran
32325	Mussa Tareme	Iran
32326	Rishakder	Iran
32328	Rishdar	Iran
32329	Sadri	Iran
32330	Sadri	Iran
32333	Sadri	Iran
32335	Sadri	Iran
32337	Sadri	Iran
32339	Sadri	Iran
32340	Sadri	Iran
32359	Tareme	Iran
32361	Tareme	Iran
32369	Tchampa	Iran
32374	Tchampa	Iran
32375	Valiseh	Iran
32379	Zardrome	Iran
32696	IR2061-464-2	Philippines
32740	AC9-57-2	Bangladesh
32752	AC18-1-3-1	Bangladesh
32753	AC19-1-1	Bangladesh
32757	Aswina 307	Bangladesh
32758	Aswina 308	Bangladesh
32759	Aswina 309	Bangladesh
32761	Aswina 327	Bangladesh
32762	Badal 25-15	Bangladesh
32766	Badal 103	Bangladesh
32778	Badal 702	Bangladesh
32783	Badal 816	Bangladesh

IRRI accession no.	Name	Origin
32786	Badal 962	Bangladesh
32790	Badal 991	Bangladesh
32791	Badal 993	Bangladesh
32792	Badal 1003	Bangladesh
32799	Bagu Aman 199	Bangladesh
32800	Bagu Aman 202	Bangladesh
32801	Bagu Aman 235	Bangladesh
32804	Bagu Aman 436	Bangladesh
23805	Bazail 65	Bangladesh
32806	Bazail 197	Bangladesh
32807	Bazail 372	Bangladesh
32808	Bazail 407	Bangladesh
32810	Bazail 414	Bangladesh
32811	Bazail 424	Bangladesh
32812	Bazail 468	Bangladesh
32814	Bazail 592	Bangladesh
32815	Bazail 924	Bangladesh
32816	Bazail 935	Bangladesh
32818	Bazail 1047	Bangladesh
32819	Bazail 1187	Bangladesh
32820	Bhadoia 687	Bangladesh
32821	Biruin 315	Bangladesh
32822	Biruin 384	Bangladesh
32824	Biruin 405	Bangladesh
32826	Biruin 519	Bangladesh
32827	Biruin 539	Bangladesh
32830	Chala Aman 51-3	Bangladesh
32850	Chala Aman 719	Bangladesh
32874	Gowai 279	Bangladesh
32908	Kala Aman 961	Bangladesh
32972	Bawyurin	Burma
33045	D59-6	Burma
33069	Gauk	Burma
33072	Gauk	Burma
33108	Hnankar Htun	Burma
33120	Hnanwa Meegalk	Burma
33250	Khesaba	Burma
33424	Namankyauk	Burma
33438	Natpyihmwe	Burma
33976	AC 7030	India
35065	NC 1184	India
35075	Nungi	India
35099	Peswari	India
35123	Sada Parangi	India
35150	Shatika	India
35167	SXC20	India
35168	SXC67	India
35170	SXC92	India
35172	SXC108	India
35200	Thalanayar-1	India
36139	Boro Black	India
36563	Jao Nah	Thailand
37154	Karsail (Nato)	Bangladesh
37256	Pakkaraj	Bangladesh
37257	Pakkiraj	Bangladesh
37295	Shabani	Bangladesh
37303	Shatia	Bangladesh
37377	Baturi	Bangladesh
37388	Binnatoha	Bangladesh
37403	Chini Atap	Bangladesh
37405	Choroasaita	Bangladesh
37414	Dhalgoria	Bangladesh
37418	Digha	Bangladesh
37419	Dudh Bazal	Bangladesh
37422	Duni	Bangladesh
37424	Ellai	Bangladesh
37448	Holde Shaiti	Bangladesh
37480	Kalogaria	Bangladesh
37484	Karachi	Bangladesh
37488	Kashiabinni	Bangladesh
37491	Katijan	Bangladesh
37522	Lalmoti	Bangladesh
37523	Lalpaika	Bangladesh
37536	Maloti	Bangladesh
37544	Moshiamira	Bangladesh
37615	Murali	Bangladesh
37649	Kalimekri 77-5	Bangladesh
37651	Kalimekri 391	Bangladesh
37653	Kalimekri 402	Bangladesh
37664	Khama 2	Bangladesh
37669	Khama 55-22	Bangladesh
37671	Khama 149	Bangladesh
37679	Khama 1066	Bangladesh
37682	Khama 1177	Bangladesh
37683	Khama 1180	Bangladesh
37684	Khama 1181	Bangladesh
37688	Khama 12-21	Bangladesh
37690	Laki 1-14	Bangladesh
37697	Laki 58-2	Bangladesh
37708	Laki 142	Bangladesh
37709	Laki 146	Bangladesh
37717	Laki 209	Bangladesh
37725	Laki 492	Bangladesh
37727	Laki 567	Bangladesh
37731	Laki 581	Bangladesh
37733	Laki 651	Bangladesh
37736	Laki 724	Bangladesh
37738	Laki 839	Bangladesh
37742	Laki 937	Bangladesh
37743	Laki 968	Bangladesh
37746	Laki 1011	Bangladesh
37802	Basmati 9-96	–
37807	Black 28-556	Bangladesh
37831	Black 28-580	Bangladesh
37843	Cylindrical 30-603	Bangladesh
37844	Cylindrical 30-604	Bangladesh
37863	Cylindrical 30-624	Bangladesh
37864	Cylindrical 30-625	Bangladesh
37866	Cylindrical 30-627	Bangladesh
37870	Cylindrical 30-631	Bangladesh
37881	Cylindrical 30-642	Bangladesh
37883	Cylindrical 30-644	Bangladesh

IRRI accession no.	Name	Origin
37889	Cylindrical 30-650	Bangladesh
37890	Cylindrical 30-651	Bangladesh
37915	Deep Straw 24-508	Bangladesh
37918	Deep Straw 24-511	Bangladesh
37920	Deep Straw 24-513	Bangladesh
37932	Deep Straw 24-525	Bangladesh
37933	Deep Straw 24-526	Bangladesh
37944	Deep Straw 24-537	Bangladesh
37956	Dudshwar 15-146	Bangladesh
37957	Dudshwar 15-147	Bangladesh
37974	Dudshwar 15-164	Bangladesh
38009	Jessobalam 3-23	Bangladesh
38010	Jessobalam 3-24	Bangladesh
38029	Jessobalam 3-45	Bangladesh
38031	Jessobalam 3-47	Bangladesh
38053	Kataribhog 7-87	Bangladesh
38082	Latisail 11-122	Bangladesh
38083	Latisail 11-123	Bangladesh
38107	Nizersail 32-734	Bangladesh
38186	Rajasail 2-8	Bangladesh
38187	Rajasail 2-9	Bangladesh
38189	Rajasail 2-12	Bangladesh
38190	Rajasail 2-13	Bangladesh
38191	Rajasail 2-14	Bangladesh
38192	Rajasail 2-15	Bangladesh
38194	Rajasail 2-17	Bangladesh
38197	Rajasail 2-21	Bangladesh
38199	Red-pie-bold 17-206	Bangladesh
38200	Red-pie-bold 17-207	Bangladesh
38201	Red-pie-bold 17-208	Bangladesh
38206	Red-pie-bold 17-213	Bangladesh
38207	Red-pie-bold 17-214	Bangladesh
38208	Red-pie-bold 17-215	Bangladesh
38245	Sadajira (Deep Straw) 16-188	Bangladesh
38362	Sadajira 19-340	Bangladesh
38371	Straw Fine 22-378	Bangladesh
38386	Straw Fine 22-296	Bangladesh
38388	Straw 23-398	Bangladesh
38392	Straw 23-403	Bangladesh
38394	Straw 23-405	Bangladesh
38403	Straw 23-415	Bangladesh
38441	Straw 23-454	Bangladesh
38448	Straw 23-463	Bangladesh
38449	Straw 23-464	Bangladesh
38454	Straw 23-469	Bangladesh
38458	Straw 23-473	Bangladesh
38474	Straw 23-489	Bangladesh
38479	Tilockachari 33-742	Bangladesh
38480	Tilockachari 33-743	Bangladesh
38481	Tilockachari 33-744	Bangladesh
38483	Tilockachari 33-746	Bangladesh
38488	Tilockachari 33-751	Bangladesh
38489	Tilockachari 33-752	Bangladesh
38495	Tilockachari 33-758	Bangladesh
38496	Tilockachari 33-759	Bangladesh
38531	TK-Red 35-795	Bangladesh
38571	IR4432-63-5	Philippines
38572	IR5785-188-2-1	Philippines
38573	IR5853-165-1	Philippines
38606	R 146	Nepal
38627	BR 6	Bangladesh
38636	BR 167-26-2-1	Bangladesh
38658	Daudzai (PI 412789)	Pakistan
38660	Daudzai (PI 412792)	Pakistan
38661	Daudzai (PI 412793)	Pakistan
38734	California 2	
38756	Dinalaga	Philippines
38773	Iniba-bain	Philippines
38778	Kamil 24	Philippines
38831	Ratossa	Philippines
38854	C 4	Philippines
38895	Taichung Sen-Shih 218	Taiwan
38898	Taichung Sen-Shih 223	Taiwan
38922	B-1205C-SM-5-2	Indonesia
38925	B-2039C-KN-7-2-3-2-2	Indonesia
38966	B-2714C-PN-1-107-2	Indonesia
38973	B-3752-8-PN-2-2	Indonesia
39163	BKN-FR 76023	Thailand
39168	BKN 6986-66-2	Thailand
39236	CNL 31	India
39240	CNL 241	India
39258	Dumsiah 81	Iran
39287	IR2061-214-3-3-17-2	Philippines
39289	IR2061-522-6-9	Philippines
39290	IR2061-628-1-6-4-3	Philippines
39360	IR3941-9-2	Philippines
39361	IR3941-14-2-2-3	Philippines
39363	IR3941-25-1	Philippines
39364	IR3941-27-1	Philippines
39368	IR3941-40-3	Philippines
39371	IR3941-58-3	Philippines
39374	IR3941-68-1-3-2	Philippines
39382	IR3941-97-1	Philippines
39383	IR4219-35-3-3	Philippines
39384	IR4422-98-3-6-1	Philippines
39385	IR4432-52-6-4	Philippines
39386	IR4625-132-1-2	Philippines
39422	IR5825-41-2-P1	Philippines
39423	IR5825-41-2-P2	Philippines
39424	IR5825-41-2-P4	Philippines
39425	IR5853-135-3-P3	Philippines
39426	IR5853-135-4-P3	Philippines
39434	IR5865-32-3	Philippines
39441	IR5896-10-2	Philippines
39486	KN1B-361-8-6-9-2-7	Indonesia
39520	MRC603-303	Philippines
39554	Beguamon 202	Bangladesh
39555	Beguamon 349	Bangladesh
39634	FH566	India
39635	FH568	India
39697	PAU41-306-2-1	India
39811	RP974-56-3-3-1	India

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39812	RP974-97-2-6-1-1	India
39947	BKN6809-74-6	–
40041	RP825-70	–
40668	Leaung Rai	Thailand
40772	Khe Tom Khao	Thailand
40881	Sudu Wee	Sri Lanka
41082	ARC12654	India
41266	ARC13856	India
41296	ARC13899	India
41538	ARC14387	India
41558	ARC14430	India
41564	ARC14447	India
41673	ARC14665	India
41674	ARC14666	India
41689	ARC14689	India
41722	ARC14730	India
41723	ARC14731	India
41724	ARC14732	India
41766	ARC14815	India
41786	ARC14850	India
41829	ARC14943	India
41830	ARC14944	India
41831	ARC14948	India
41839	ARC14961	India
41840	ARC14962	India
41841	ARC14963	India
41842	ARC14965	India
41843	ARC14966	India
41844	ARC14969	India
41845	ARC14971	India
41854	ARC14981	India
41877	ARC15013	India
41885	ARC15036	India
41897	ARC15048	India
41898	ARC15049	India
41938	ARC15129	India
41945	ARC15165	India
42099	ARC15603	India
42275	ARC18113	India
42439	ARC18517	India
42460	ARC18580	India
42502	ARC6114	India
42593	ARC10692	India
42608	ARC10969	India
42609	ARC10970	India
42612	ARC10980	India
42648	ARC11241	India
42650	ARC11243	India
42738	ARC13525	India
42775	ARC13766	India
42819	ARC13980	India
42980	ARC14744	India
42994	ARC14787	India
42999	ARC14801	India
43017	ARC14903	India
43079	ARC15130	India
43109	ARC15169	India
43146	ARC15294	India
43149	ARC15301	India
43159	ARC15361	India
43160	ARC15362	India
43177	ARC15391	India
43181	ARC15399	India
43193	ARC15426	India
43198	ARC15440	India
43293	ARC18582	India
43295	ARC18586	India
43572	Remaja	Indonesia
43796	Bow Pagal	Bangladesh
43798	Chakulia	Bangladesh
43813	Dhalgora	Bangladesh
43838	Gori	Bangladesh
43840	Gouri Hatti	Bangladesh
43841	Gouri Saita	Bangladesh
43851	Hanpa (Black)	Bangladesh
43897	Kocho Olola	Bangladesh
43921	Maguria	Bangladesh
43925	Molica	Bangladesh
43945	Pukhi	Bangladesh
44015	Hashikalmi	Bangladesh
44029	SPT 6830-681-20	Thailand
44136	Gow Ruang 17-2-206	Thailand
44137	Gow Ruang 17-2-118	Thailand
44157	Leaung Noi 31-1-39	Thailand
44313	Bengawan	–
44444	Hinon	Philippines
44449	Ilon-ilon	Philippines
44474	Intan	Philippines
44708	Purtok	Philippines
44713	Ramadia	Philippines
44819	Black Gora S. N. 32	India
44857	Brown Gora S. N. 95	India
44890	White Gora S. N. 113	India
44895	Achra 108-1	India
44904	Ajan	India
44910	Ajandhula	India
44911	Ajanmete	India
44929	Altapati	India
44932	Amla	India
44963	Ausdhan	India
44975	Aus Paddy (Black)	India
44976	Aus Paddy (Black)	India
44977	Aus Paddy (Red)	India
44983	Aviram	India
44998	A 29-23	Burma
45010	Badkalma	India
45023	Baisbish	Bangladesh
45030	Bakmal	India
45031	Bakoi	India
45032	Bakoi	India
45051	BAM 3	India
45143	Bhajana	India

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45146	Bhapsail	India
45153	Bhasamanik	India
45169	Bhojna	India
45187	Biatisail	India
45204	Boldar	India
45223	Borakarikatke	India
45230	Bowrash	Bangladesh
45231	Boyan	India
45267	Champakushi	India
45277	Chandramukhi	India
45280	Chapasail	India
45281	Chapasail	India
45288	Chengursail	India
45297	Chile Boro	India
45310	Chotasoni	India
45340	C 22	India
45342	C 26	India
45388	CR3	India
45501	Derraeddari	Sri Lanka
45517	Dhala Moti	India
45548	Dhuli	India
45550	Dhulia	India
45607	Dudsail	India
45609	Dudsail	India
45634	DW6255	India
45677	Gasti Kalma	India
45681	Getui	India
45688	Ghumarsail	India
45689	Ghungarsail	India
45695	Gobrasail	India
45699	Gohana	India
45701	Gokulganja	India
45733	G25	India
45734	G26	India
45813	HS 19/Taichu 65 (VKM-12)	India
45848	I. B. Rose/Latisail	India
45856	Jahari (5240)	India
45928	Jhulur	India
45964	Kaladubraj	India
45966	Kalajiji D-15	Burma
46030	Kanakchur	India
46045	Kanthiladan	India
46062	Kartiksail	India
46094	Kaya	India
46096	Kelash	India
46098	Kelash	India
46099	Kele	India
46109	Kelpi	India
46169	Kumed	India
46255	Leulikelash	India
46263	LK22	India
46278	Maghidhan	India
46280	Magursail	India
46294	Malabati	India
46300	Malia Bhangar 1	India
46301	Maliabhangar 2	India
46303	Malsira	India
46304	Malsira	India
46311	Mansara	India
46332	Meghal Patnai	India
46360	Mohonbhog	India
46375	Motihar	India
46376	Moul	India
46426	Narikalma	India
46430	Nayankalma	India
46556	Rajamanik	India
46613	Sachinona	India
46620	Sadaboldar	India
46621	Sadadhula	India
46743	Thupi Jhingasail	India
46800	Unnamed	India
46801	Unnamed	India
46829	Unnamed	India
46841	Unnamed	India
46856	Unnamed	India
46890	Unnamed	India
46912	Unnamed	India
46935	Unnamed	India
46976	Koimurali	Korea
47007	IS34	Ivory Coast
47011	IS38	Ivory Coast
47017	IS49	Ivory Coast
47025	IS57	Ivory Coast
47052	1-IS 45-C 77	Ivory Coast
47364	Allaipiddi	Srī Lanka
47373	Kalu Heenetti	Sri Lanka
48019	Jao Kong	Thailand
48130	Khao Ma Led Yao	Thailand
48311	Leaung Yai	Thailand
48332	Mah Eng	Thailand
48477	Ta Wan Khuen	Thailand
49221	Khisail	Bangladesh
49233	Longbadal	Bangladesh
49243	Motorsail	Bangladesh
49390	Arroz Cristol	Mozambique
49424	Bandi	India
49442	Bawrashmurali	India
49452	Bhmbalo	West Africa
49483	Chakkana Thandan	India
49490	Cheera	India
49529	Choorapaudy	India
49620	EAT164 F	USSR
49636	Eravapandy	India
49685	Hatwho	Japan
49756	Kanmba Karabi R. 295	India
49766	Karsample Reo	India
49767	Kar sample White	India
49810	Koombalai A. 24	India
49837	Kuthi Kondappan	India
49877	Manavari	India
49878	Manavari	India
50004	Pamani Samba	India

IRRI accession no.	Name	Origin	IRRI accession no.	Name	Origin
50009	Para Nellu	India	52725	NDAU 147	India
50035	Pisini	India	52791	Athikkira Mudan	India
50083	Ropotor	Gambia	52793	Campavu	India
50116	Sarosamavali 2N	India	52815	Germal	India
50135	Senthinayagam	India	52839	Vadlo Kendal	India
50142	"Shima"	–	53500	Ejali	Bangladesh
50161	Sornavari or Arupathamkodai	India	53558	Origachi	Bangladesh
50170	Surli (Dye Brown)	India	53580	Tenguri	Bangladesh
50212	USSR Black	USSR	53581	Uradigha	Bangladesh
50277	Yon-gon	Burma	53584	Vawalia	Bangladesh
50380	Suweon 290	Korea	54773	S1904	India
50677	Pattambi 1727	India	54780	T243	India
50678	Pattambi 1751	India	54985	K812-4	India
50686	RNR24263	India	54987	K815-5	India
50695	BG380-1	Sri Lanka	55055	ADR43	India
50698	BG402-2	Sri Lanka	55062	Ariyari	India
50701	KMP12	India	55086	Chuvannakumbolan	India
50703	KMP38	India	55098	GS758	India
50705	KMP41	India	55105	Karikattalam	India
50933	H497	India	55142	PTB	India
50980	Gonabaru	Sri Lanka	55146	Swarna Chempan	India
50987	Horana Wee	Sri Lanka	55148	T16	India
51008	Kokku Vellai	Sri Lanka	55149	T19	India
51032	Maddai Vathiar Nellu	Sri Lanka	55150	T20	India
51048	Pandi Valan	Sri Lanka	55170	Thirinjavella	India
51078	Theva Rathiri	Sri Lanka	55179	Vellakal I	India
51153	P8968-7-3-2-1	Cuba	55182	Veluthancheera	India
51540	Mallai	USSR	55192	710	India
51648	WIR2139	USSR	55196	1371	India
51993	Mahulong	India	55200	1477	India
52020	Phougak	India	55203	2914	India
52095	Chainger	India	55206	8378	India
52104	Chinga	India	55209	8880	India
52183	Kusum Kathi	India	55325	Koor Karuppan	Sri Lanka
52195	Matikiba	India	55364	Valan	Sri Lanka
52242	Sathika	India	55407	MRC12-24E 100	Philippines
52510	Alur Sanna	India	55926	BKN-BR-1031-41-2-6	Thailand
52554	Kankansali	India	55964	IR5677-17-3-1-1	Philippines
52578	Maskati	India	57375	Nahng Kam Pai	Thailand
52587	Ponnani	India	58563	Pa Lamina	Sierra Leone
52594	Sanna Billeri	India	58725	Dhala Gora	Bangladesh
52664	Karahani	India			

[a]A dash (–) indicates that origin is unknown.

Wild rice species resistant to the green leafhopper *Nephotettix virescens* at IRRI, 1983.

IRRI accession no.	Species	Origin	IRRI accession no.	Species	Origin[a]
100168	*O. officinalis*	Costa Rica	100180	*O. officinalis*	Malaya
100172	*O. latifolia*	Guatemala	100181	*O. officinalis*	Burma
100179	*O. officinalis*	Japan	100183	*O. sativa*/*O. rufipogon*	India

IRRI accession no.	Species	Origin	IRRI accession no.	Species	Origin
100195	*O. nivara*	Burma	101114	*O. minuta*	Philippines
100211	*O. perennis*	India	101115	*O. minuta*	Philippines
100224	*O. barthii*	Gambia	101116	*O. minuta*	Philippines
100820	*O. ridleyi*	Japan	101117	*O. minuta*	Philippines
100821	*O. ridleyi*	Japan	101118	*O. minuta*	Philippines
100878	*O. officinalis*	Thailand	101121	*O. minuta*	Philippines
100883	*O. officinalis*	India	101122	*O. minuta*	Philippines
100885	*O. latifolia*	India	101123	*O. minuta*	Philippines
100887	*O. minuta*	India	101124	*O. minuta*	Philippines
100890	*O. latifolia*	India	101125	*O. minuta*	Philippines
100891	*O. latifolia*	India	101126	*O. minuta*	Philippines
100892	*O. punctata*	India	101128	*O. minuta*	Philippines
100895	*O. latifolia*	USA	101129	*O. minuta*	Philippines
100896	*O. officinalis*	Thailand	101132	*O. minuta*	Philippines
100897	*O. nivara*	India	101133	*O. minuta*	Philippines
100899	*O. nivara*	India	101134	*O. minuta*	Philippines
100903	*O. nivara*	India	101135	*O. paraguaensis*	Philippines
100904	*O. rufipogon*	Thailand	101137	*O. officinalis*	Philippines
100914	*O. latifolia*	Mexico	101139	*O. officinalis*	Philippines
100916	*O. rufipogon*	China	101141	*O. minuta*	Philippines
100917	*O. sativa/O. nivara*	Cambodia	101142	*O. officinalis*	Philippines
100937	*O. punctata*	Ghana	101143	*O. minuta*	Philippines
100947	*O. officinalis*	India	101149	*O. officinalis*	Malaysia
100948	*O. officinalis*	India	101150	*O. officinalis*	Malaysia
100953	*O. officinalis*	India	101151	*O. officinalis*	Malaysia
100956	*O. latifolia*	India	101152	*O. officinalis*	Malaysia
100959	*O. latifolia*	Mexico	101154	*O. officinalis*	Malaysia
100962	*O. latifolia*	Guatemala	101155	*O. officinalis*	Malaysia
100963	*O. latifolia*	Guatemala	101166	*O. officinalis*	Philippines
100964	*O. latifolia*	Guatemala	101171	*O. punctata*	Tanzania
100965	*O. latifolia*	Costa Rica	101243	*O. barthii*	Mali
100966	*O. latifolia*	Panama	101329	*O. punctata*	Nigeria
100973	*O. officinalis*	Philippines	101386	*O. minuta*	Japan
101073	*O. officinalis*	Philippines	101387	*O. minuta*	Japan
101074	*O. officinalis*	Philippines	101392	*O. latifolia*	Guatemala
101077	*O. officinalis*	Philippines	101395	*O. alta*	USA (USDA)
101078	*O. officinalis*	Philippines	101399	*O. officinalis*	Vietnam
101079	*O. minuta*	Philippines	101408	*O. punctata*	Ghana
101081	*O. minuta*	Philippines	101409	*O. punctata*	Ghana
101082	*O. minuta*	Philippines	101412	*O. officinalis*	India
101083	*O. minuta*	Philippines	101414	*O. officinalis*	India
101084	*O. minuta*	Philippines	101417	*O. punctata*	Kenya
101086	*O. minuta*	Philippines	101421	*O. eichingeri*	Uganda
101089	*O. minuta*	Philippines	101422	*O. eichingeri*	Uganda
101092	*O. minuta*	Philippines	101424	*O. eichingeri*	Uganda
101094	*O. minuta*	Philippines	101426	*O. eichingeri*	Uganda
101096	*O. minuta*	Philippines	101434	*O. punctata*	Tanzania
101097	*O. minuta*	Philippines	101439	*O. punctata*	Ghana
101099	*O. minuta*	Philippines	101441	*O. punctata*	–
101100	*O. minuta*	Philippines	101443	*O. latifolia*	Morocco
101101	*O. minuta*	Philippines	102178	*O. nivara*	India
101112	*O. minuta*	Philippines	102382	*O. officinalis*	Indonesia
101113	*O. minuta*	Philippines	102481	*O. latifolia*	Nicaragua

[a]A dash (–) indicates that origin is unknown.

Varieties resistant to the zigzag leafhopper *Recilia dorsalis* at IRRI, 1982.

IRRI accession no.	Variety	Origin	IRRI accession no.	Variety	Origin
05334	Mas	Indonesia	30064	Khao Pa Siu	Laos
06101	Ptb 27	India	30065	Khao Phanh	Laos
06259	RDR2	India	30079	Khao Phei Deng	Laos
07752	Balamawee	Sri Lanka	30089	Khao Vieng	Laos
08731	UCP31	Bangladesh	30091	Khao Ykhao	Laos
08801	UCP188	Bangladesh	30095	Khi Tom	Laos
11053	Ptb 21	India	30118	Makhing	Laos
11530	Mak Thoua	Laos	30123	Mak Nam	Laos
11531	Sam Houang	Laos	30145	Unnamed	Laos
30003	Khao Khai	Laos	30146	Unnamed	Laos
30006	Khao Khao	Laos	30169	Phei Hom	Laos
30010	Khaokhao	Laos	30201	Y-Doun	Laos
30042	Khao Meun Lan	Laos	30320	Gangasale Bhatta	Mexico
30049	Khaonondeng	Laos	30351	ROK2	Sierra Leone
30054	Khao None	Laos	30361	Kao Ipoua	Laos
30055	Khao None	Laos	30364	Khao Bohina	Laos
30058	Khao Noy	Laos	30366	Khao Iphoua	Laos
30063	Khao Pa Siu	Laos	30370	Khao Maknam	Laos

Wild rice species resistant to the zigzag leafhopper *Recilla dorsalis* at IRRI, 1982.

IRRI accession no.	Species	Origin	IRRI accession no.	Species	Origin
100168	*O. latifolia*	Costa Rica	100915	*O. sativa/O. nivara*	India
100172	*O. latifolia*	Guatemala	100920	*O. nivara/O. rufipogon*	Malaysia
100179	*O. latifolia*	Japan	100937	*O. punctata*	Ghana
100180	*O. officinalis*	Malaysia	100942	*O. sativa/O. rufipogon*	India
100181	*O. officinalis*	Burma	100947	*O. officinalis*	India
100192	*O. sativa/O. rufipogon*	Malaysia	100948	*O. officinalis*	India
100211	*O. perennis*	India	100953	*O. officinalis*	India
100595	*O. nivara/O. rufipogon*	Taiwan	100954	*O. punctata*	India
100598	*O. sativa/rufipogon*	Taiwan	100956	*O. latifolia*	India
100685	*O. perennis*	Taiwan	100959	*O. latifolia*	Mexico
100820	*O. ridleyi*	Japan	100962	*O. latifolia*	Guatemala
100821	*O. ridleyi*	Japan	100963	*O. latifolia*	Guatemala
100878	*O. officinalis*	Thailand	100964	*O. latifolia*	Guatemala
100883	*O. officinalis*	India	100965	*O. latifolia*	Costa Rica
100885	*O. latifolia*	India	100966	*O. latifolia*	Panama
100887	*O. minuta*	India	100973	*O. officinalis*	Philippines
100890	*O. latifolia*	India	101073	*O. officinalis*	Philippines
100891	*O. latifolia*	India	101074	*O. officinalis*	Philippines
100892	*O. punctata*	India	101077	*O. officinalis*	Philippines
100895	*O. latifolia*	USA	101078	*O. officinalis*	Philippines
100896	*O. officinalis*	Thailand	101079	*O. minuta*	Philippines
100901	*O. sativa/O. nivara*	India	101081	*O. minuta*	Philippines
100911	*O. nivara/O. rufipogon*	Thailand	101082	*O. minuta*	Philippines
100912	*O. rufipogon*	Thailand	101083	*O. minuta*	Philippines
100914	*O. latifolia*	Mexico	101084	*O. minuta*	Philippines

IRRI accession no.	Species	Origin	IRRI accession no.	Species	Origin
101086	*O. minuta*	Philippines	101149	*O. officinalis*	Malaysia
101089	*O. minuta*	Philippines	101150	*O. officinalis*	Malaysia
101092	*O. minuta*	Philippines	101151	*O. officinalis*	Malaysia
101094	*O. minuta*	Philippines	101152	*O. officinalis*	Malaysia
101096	*O. minuta*	Philippines	101154	*O. officinalis*	Malaysia
101097	*O. minuta*	Philippines	101155	*O. officinalis*	Malaysia
101099	*O. minuta*	Philippines	101166	*O. officinalis*	Philippines
101100	*O. minuta*	Philippines	101171	*O. punctata*	Tanzania
101101	*O. minuta*	Philippines	101243	*O. barthii*	Mali
101112	*O. officinalis*	Philippines	101329	*O. punctata*	Nigeria
101113	*O. officinalis*	Philippines	101386	*O. minuta*	Japan
101114	*O. officinalis*	Philippines	101387	*O. minuta*	Japan
101115	*O. officinalis*	Philippines	101392	*O. latifolia*	Guatemala
101116	*O. officinalis*	Philippines	101395	*O. alta*	USA (USDA)
101117	*O. officinalis*	Philippines	101399	*O. officinalis*	Vietnam
101118	*O. officinalis*	Philippines	101408	*O. punctata*	Ghana
101121	*O. officinalis*	Philippines	101409	*O. punctata*	Ghana
101122	*O. minuta*	Philippines	101412	*O. officinalis*	India
101123	*O. minuta*	Philippines	101414	*O. officinalis*	India
101124	*O. minuta*	Philippines	101417	*O. punctata*	Kenya
101125	*O. minuta*	Philippines	101418	*O. eichingeri*	Uganda
101126	*O. minuta*	Philippines	101422	*O. eichingeri*	Uganda
101128	*O. minuta*	Philippines	101424	*O. eichingeri*	Uganda
101129	*O. minuta*	Philippines	101429	*O. eichingeri*	Uganda
101132	*O. minuta*	Philippines	101430	*O. punctata*	Uganda
101133	*O. minuta*	Philippines	101434	*O. punctata*	Tanzania
101134	*O. minuta*	Philippines	101439	*O. punctata*	Ghana
101135	*O. paraguaensis*	Philippines	101443	*O. latifolia*	Morocco
101137	*O. officinalis*	Philippines	102178	*O. nivara*	India
101139	*O. officinalis*	Philippines	102382	*O. officinalis*	Indonesia
101141	*O. minuta*	Philippines	102385	*O. officinalis*	Indonesia
101142	*O. officinalis*	Philippines	102481	*O. latifolia*	Nicaragua
101143	*O. minuta*	Philippines			

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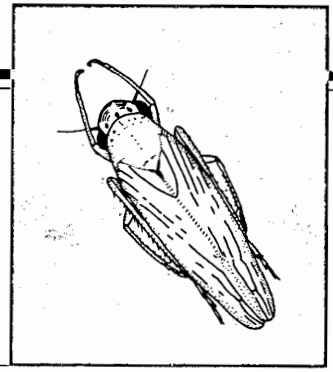

Chapter 10
WHITE LEAFHOPPER

The white leafhopper *Cofana spectra* is widely distributed throughout South and Southeast Asia. Nymphs and adults remove plant sap and cause leaf yellowing and typical hopperburn of a rice crop. Rearing and greenhouse screening methods have been developed by Sekar and Chelliah (1983). Field screening methods were developed by Velusamy et al (1975).

REARING

Hoppers required for greenhouse screening are reared on potted TN1 plants.

Steps	Key points
1. Growing rice plants as a food source	• Transplant 10-day-old TN1 rice seedlings in 10-cm-diam clay pots at 3 seedlings/pot. • When the plants are 45-50 days old, place them in polyester pans filled with water. Confine the potted plants in oviposition cages 45 cm long, 45 cm wide, and 60 cm high with a wooden frame and wire mesh sides. Each cage accommodates 4 potted plants.
2. Collecting adults in the field	• When the food plants are ready, use a sweep net to collect white leafhopper adults in rice fields. • Release the collected hoppers on the caged plants at 50-70 hoppers/cage.
3. Allowing the adults to oviposit	• Keep the adults in the cage for 2-3 days of oviposition.
4. Rearing the insect	• Transfer egg-infested plants to a test-insect rearing cage. — In 10 days, nymphs appear. Maintain the nymphs on the same egg-infested plants until the plants wilt. — Tap the wilting plants gently inside the cage to dislodge the nymphs. Remove the plants from the cage and replace with fresh TN1 plants. — Continue the same process until adult emergence (about 28 days). Some adults are used in the screening tests and the remainder are placed in the oviposition cage to maintain the culture. Each female has an average fecundity of 44 eggs and longevity of 11 days.

SCREENING

Screening for white leafhopper has been limited. The methodology used by Sam et al (1982) at the Tamil Nadu Agricultural University, Coimbatore, India, is described.

Greenhouse screening

In the greenhouse screening, seedlings are screened in seedboxes.

Steps	Key points
1. Preparing seed	• Prepare seed of test materials.
2. Soaking seed for germination	• Soak the seed for 48 h in water. • Remove seed from the water and put in a dark-room for germination.
3. Sowing seed	• Three days after soaking, sow the germinating seeds in 40-cm rows 5 cm apart in seedboxes 50 × 40 × 10 cm and filled with soil to a 5-7 cm depth. Each seedbox accommodates 8 test entries, a resistant check (ASD8), and a susceptible check (TN1). • Thin the seedlings to 20/row and place the seed-boxes in galvanized iron trays (62 × 47 × 15 cm) filled with water 10 cm deep.
4. Infesting the seedlings	• Fifteen days after sowing, cover each seedbox with a nylon mesh cage (55 × 45 × 50 cm). Release white leafhopper adults at the rate of 2 hoppers/seedling.
5. Evaluating	• Grade the seedlings for plant damage on a row basis when 90% of TN1 seedlings are dead. Use the following scale: *Scale* — *Damage* 0 None 1 First leaf tip yellowing 3 First leaf drying and second leaf tip yellowing 5 Yellowing of old leaves or pronounced stunting or both 7 Wilting/drying of more than 50% of the plants 9 All plants dead

Field screening

In field screening for white leafhopper resistance, varieties are graded according to the hopper population as indicated by sweep net counts.

Steps	Key points
1. Preparing field plots	• Prepare field plots of 5 × 0.6 m (3 rows, 5 m long) arranged in a randomized complete block design with 3 replications.
2. Transplanting test entries	• Transplant 25-day-old seedlings of the test entries at a spacing of 20 × 10 cm at 1 seedling/hill.
3. Infesting plants	• Hopper infestation will depend on natural field populations.
4. Evaluating	• Assess the population of leafhoppers by making five sweeps of an insect net in each plot early in the morning. Begin observations at 21 days after transplanting (DT) and continue at weekly intervals to 49 DT. • Classify the varieties as resistant, moderately resistant, moderately susceptible, and susceptible based on the mean population counts/5 sweeps in each variety.

Rating	*Av no./5 sweeps*
Resistant	0 to 5.9
Moderately resistant	6.0-9.9
Moderately susceptible	10.0-12.0
Susceptible	more than 12.0

SOURCES OF RESISTANCE

Six sources of white leafhopper resistance have been reported (Velusamy et al 1975): ADT 14, ASD3, ASD8, CH2, CO 2, and CO 29.

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Chapter 11
BLUE LEAFHOPPER

The blue leafhopper *Empoascanara maculifrons* is a minor rice pest in South and Southeast Asia. By feeding on rice leaves it reduces the vigor of the crop in the nursery and in the early transplanted stages. The blue leafhopper has recently become a serious threat to rice in Tamil Nadu, India.

SCREENING

Screening for resistance has been done in the greenhouse and in the field by Jayaraj (1976) and Velayutham and Jayaraj (1981).

Greenhouse screening

Steps	Key points
1. Preparing seed of test varieties	• Prepare test varieties using CO 33 as the susceptible check and ASD5 as the resistant check.
2. Sowing seed	• Sow seed of test varieties in 50- × 40- × 10-cm seedboxes. Each seedbox accommodates 10 test varieties sown in rows (5 cm apart) with at least 10 seedlings/row.
3. Infesting the seedlings	• Cage each seedbox and release 5-10 hoppers/seedling.
4. Evaluating	• Assess leaf area damaged in 10 seedlings/variety using a damage rating scale of 0-8 as follows:

Scale	*Leaf area damaged (%)*
0	None
1	1-10
2	11-25
3	26-33
4	34-50
5	51-66
6	67-75
7	76-100
8	Plants are dead

Field screening of direct seeded rice

For field screening test entries are sown directly in the field to coincide with peak population of the blue leafhopper.

Steps	Key points
1. Preparing seed of test materials	• Obtain test varieties with CO 33 as the susceptible check and ASD5 as the resistant check.
2. Sowing the seed	• Sow seed of each variety at monthly intervals during the peak population of the blue leafhopper. Use 1- × 1-m plots laid out in a randomized complete block design with 3 replications.
3. Infesting the plants	• Infestation depends on natural populations.
4. Evaluating	• Assess leafhopper populations in each plot by taking 5 sweeps early in the morning and counting the hoppers. Sweep 3 times at weekly intervals from 1 week after sowing. • Group the varieties into resistant, moderately resistant, moderately susceptible, and susceptible based on the population counts: *Group* — *Hoppers/5 sweeps* Resistant — Less than 5 Moderately resistant — 5-13 Moderately susceptible — 14-17 Susceptible — More than 17 • Also assess the extent of damage by scoring leaf area damage in 10 seedlings selected at random in each plot. Base extent of damage on the 0-8 scale used in the greenhouse screening.

Field screening of transplanted rice

Steps	Key points
1. Preparing seed of test materials	• Obtain seed of test varieties. Include CO 33 as the susceptible check and ASD5 as the resistant check.
2. Scheduling sowing	• Screen during periods of peak populations of hoppers. • Schedule sowing of test materials so that the seedlings will be 24 days old at transplanting.
3. Raising seedlings	• Use the wetbed method to grow seedlings.
4. Preparing the field plots	• Prepare 3- × 2-m field plots for each test entry. Replicate each entry three times in a randomized complete block design.

Steps	Key points
5. Transplanting the seedlings	• Transplant 24-day-old seedlings at a spacing of 20 × 10 cm at 2 seedlings/hill.
6. Infesting the plants	• Infestation depends on natural populations.
7. Evaluating	• Assess leafhopper populations by taking 5 random sweeps with a standard net (20-cm-diam)/plot. Make assessments early in the morning at 2-week intervals starting 15 DT until 5 or more observations are made. Group the varieties as in step 3, *Field screening of direct seeded rice.* • Assess extent of damage using the 0-8 scale as in the greenhouse screening.

SOURCES OF RESISTANCE

Six varieties are known as sources of resistance to blue leafhopper:

Variety	Reference
ADT22	Jayaraj (1976)
ASD5	Velayutham and Jayaraj (1981)
CO 25	Velayutham and Jayaraj (1981)
CO 29	Velayutham and Jayaraj (1981)
TKM2	Velayutham and Jayaraj (1981)
TKM6	Velayutham and Jayaraj (1981)

REFERENCES CITED

Jayaraj, S. 1976. Research on resistance in plants to insects and mites in Tamil Nadu. Tamil Nadu Agricultural University, Coimbatore, India.

Velayutham, B., and S. Jayaraj. 1981. Studies on the resistance in rice to blue leafhopper, *Zygina maculifrons* (Motsch.)(Cicadellidae: Homoptera). Seminar on rice pest management, Tamil Nadu Agric. University, Coimbatore, India, 1980.

Chapter 12
STRIPED STEM BORER

The striped stem borer *Chilo suppressalis* is distributed throughout South and Southeast Asia, the temperate areas of China, Japan, and Korea, and in northern Australia and southern Russia. Various grasses serve as alternative host plants. The boring of the larvae within the stem causes the development of deadhearts and whiteheads. Several larvae may be present in one stem.

In contrast to the planthoppers, mass-rearing techniques for the stem borers have not been well developed. Stem borers can be reared on rice plants, but this method, as so far developed, has not been efficient and requires a large space to produce sufficient insects for the screening of large numbers (600) of entries at one time.

The striped stem borer *Chilo suppressalis* can be reared on cut rice stalks or on an artificial diet. Neither method is practical for screening that requires a large number of larvae. They are used, however, to provide insects for studies on mechanisms of resistance, which require fewer insects.

For screenhouse screening, stem borers for infestation of test entries come from eggs laid by moths. The moths are propagated in beds planted to a susceptible variety.

Field screening is dependent on a high natural infestation of preferably the striped stem borer. Although more laborious than relying solely on natural infestation, artificial inoculation of the plants with first-instar larvae supplements the natural population.

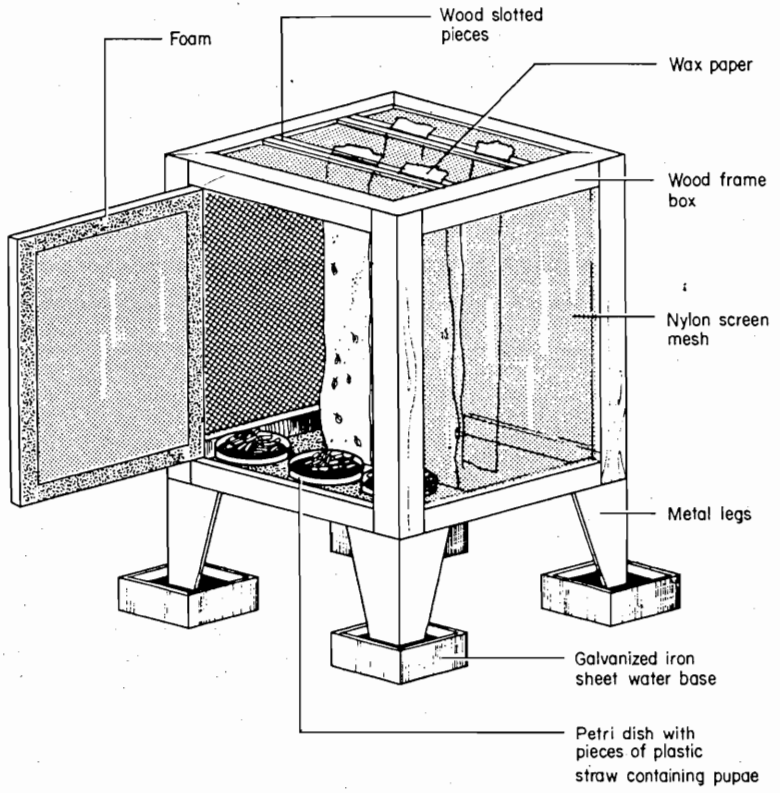
Methods described for rearing and screening the striped stem borer can also be used for the darkheaded stem borer *Chilo polychrysus.*

REARING METHODS

The striped stem borer can be reared on cut rice stalks or on artificial diet.

Cut stalk method

Steps	Key points
1. Starting an initial stem borer population	• Plant regularly (monthly) in the field a susceptible variety such as Rexoro or IR8 to serve as a continuous source of stem borers. • Cut infested rice stubble and bring it to the laboratory. • Dissect the stubble and collect larvae and pupae. • Place pupae (about 100/petri dish) directly in the oviposition cage (Fig. 1). The cage may contain potted plants of Rexoro, IR8, or any other susceptible variety about 30 days old, or strips of wax paper, which serve as a substrate on which the



1. Oviposition cage for stem borers. Wood portions are covered with foam rubber to prevent oviposition on them. Eggs are laid on the wax paper.

Steps	Key points
	eggs are laid. Moths can also be caught in light traps at night and placed directly in the oviposition cage the same night they are collected because most of the eggs are laid on the first night of capture.
	• Place small or early-instar larvae and late-instar larvae collected from the stubble in separate glass jars containing cut Rexoro or IR8 stems (15-20 cm long). Cover the jars with screened screw tops and place them in a water tray to prevent predation by ants.

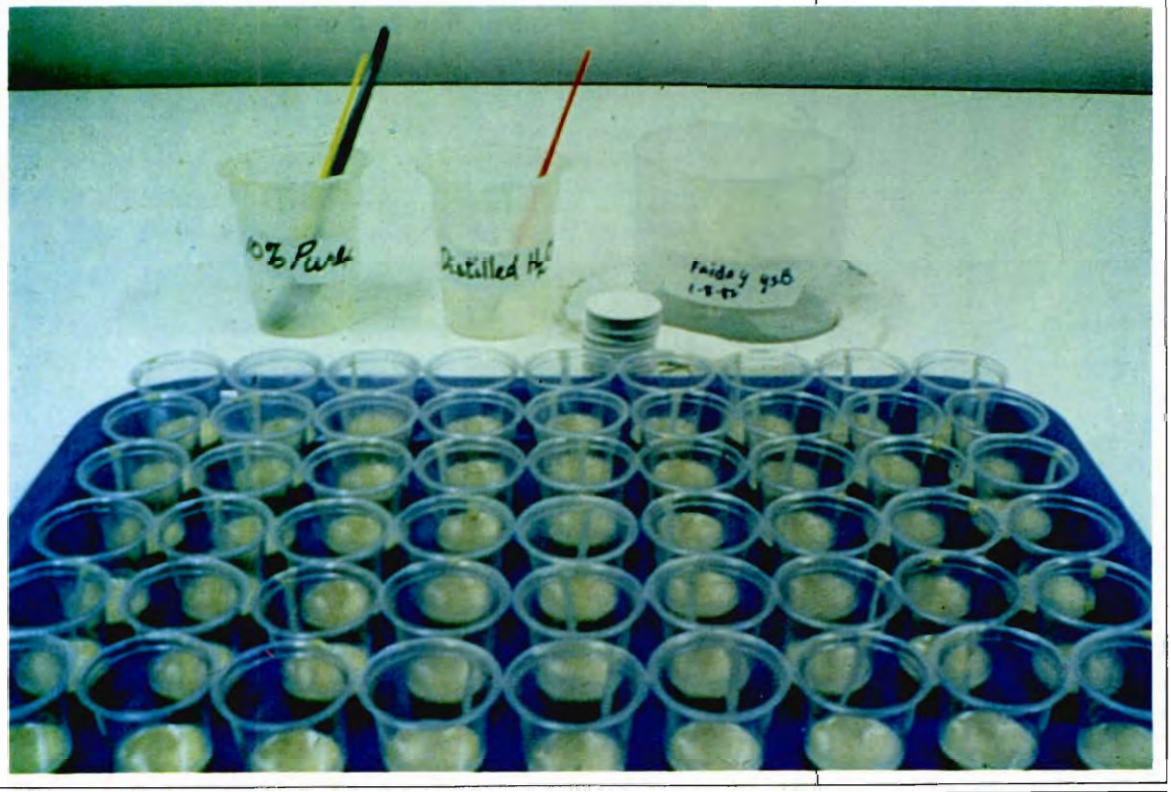
Steps	Key points
	• Transfer larvae to fresh rice stems weekly until pupation is observed. • Open the stems to collect pupae. • Place the pupae in petri dishes and place the petri dish in the oviposition cage (Fig. 1). Moths begin to emerge after 5-7 days in the cage. • Begin collecting egg masses on the third day after moth emergence begins. Cut the leaf or wax paper portions containing eggs and place the eggs in petri dishes lined with moist filter paper. Incubate at 23-30° C until the blackhead stage.
2. Preparing rice stalks	• Cut rice stalks of a susceptible variety in 12- to 15-cm sections. Be sure that each cut stalk has a node about 2-3 cm from the bottom. • Place the stalks tightly in glass jars or clay pots (preferably clay pots) and place them in metal trays with water 3 cm deep. The water will permeate the bottom of the clay pots and provide high relative humidity while the upper halves of the pots remain relatively dry. This gives the borers a gradient of humidity.
3. Allowing eggs to hatch	• Place six egg masses at the blackhead stage in each pot. At egg hatch the larvae bore into the stems at their cut ends. Later the stalks become brown and dry and the borers leave the stalks and crawl to the rims of the pots and search for food.
4. Changing stalks	• When the larvae are at the rim of the pots, pick them up with a fine camel hair brush or soft forceps and place them in new pots with fresh stalks. • Change stalks about two times a week. • When the larvae pupate, avoid disturbing them even if the stalks are drying. Wait for the moths to emerge directly from the dried stalks.
5. Collecting eggs	• Place the moths in oviposition cages. • Collect egg masses every morning and place them in petri dishes lined with moist filter paper and hold at 23-30° C. If the eggs are not immediately needed they can be kept at room temperature until the blackhead stage, then kept in an incubator at 5-8° C up to 10 days, but not longer, without affecting hatching. • Repeat the process to have a continuous supply of insects.

Artificial diet

A modification of an artificial diet developed by Kamano (1971) and the modified southwestern corn borer diet can be used to rear striped stem borer.

Steps	Key points
1. Preparing the Kamano diet	• Prepare the following ingredients. The quantity listed is sufficient for one 250-ml Erlenmeyer flask. Hence, the amount of each ingredient depends on the number of flasks to be used. I. Cellulose powder 1.0 g Wheat bran 2.0 g II. Agar powder 1.0 g Glucose 0.5 g Sucrose 0.5 g Casein 1.5 g Dry yeast 1.0 g Becks salt mixture 0.2 g Cholesterol 0.02 g III. a. Choline chloride 0.05 g b. Sodium dehydroacetate 0.01 g c. Water 50.00 ml IV. a. Sodium ascorbate 0.2 g b. 1% formalin solution 2.00 ml • Weigh cellulose and wheat bran (I) and place in a flask. • Grind and mix ingredients of II thoroughly with mortar and pestle and place in the flask. • Weigh IIIa and IIIb and place in a beaker. Add water and stir to dissolve. Add to flask. • After adding solution III, shake the flask well and autoclave immediately at 112°C and 0.6 kg/cm² pressure for 30 min. After autoclaving, transfer flask into a hood to cool. • Dissolve IVa in IVb, then add solution to the flask. • Remove flask from hood and shake slightly while immersed in cold water to solidify diet and attain uniform distribution of ingredients. Then keep flask in a refrigerator at 15°C until ready for inoculation with larvae.
2. Preparing the modified southwestern corn borer diet (Davis 1976)	• Prepare the following ingredients to make up about 1 liter of the diet: I. Distilled water 911.0 ml II. Agar 17.2 g III. Toasted wheat germ 21.1 g Vitamin-free casein 25.0 g Sucrose 25.0 g

Steps	Key points	
	Salt mix Wesson	7.0 g
	Linseed oil	0.2 g
	Cholesterol	0.1 g
	Maize cob grits (autoclaved)	37.0 g
	Methyl parabenzoate	1.5 g
	Sorbic acid	0.5 g
	Cornstarch	35.2 g
	IV. Vanderzant vitamin mix	5.3 g
	Ascorbic acid	2.1 g
	Formaldehyde (10%)	4.0 g
	• Bring 545 ml distilled water to boil. Add 17.2 g agar and stir until most of the agar has dissolved.	
	• Pour water-agar mixture into a blender.	
	• Add the mixture of III, and blend at high speed for 3 min.	
	• Add 366 ml of cold distilled water and mix for 1 min.	
	• Add the mixture of IV and mix for 1 min.	
	• Pour 6 g of the diet into each of 28-g plastic rearing containers (Fig. 2).	



2. Rearing containers with artificial diet. The clear styrene cups have a 4.5-cm-diam top and 2.5-cm-diam bottom, and are 7.5-cm tall. The camel hair brush is washed in 10% Purex (sodium hydrochlorite) and rinsed in distilled water before its use for picking up larvae from the container at the right.

Steps	Key points
3. Collecting eggs or moths in the field	• Collect egg masses in the field (they are most abundant 50-70 days after transplanting [DT]) or adults at lights at night. Eggs collected in the field are often heavily parasitized. Place moths in oviposition cages the same night they are collected. Place the egg masses collected in the field or from the oviposition cages in petri dishes lined with moist filter paper and keep at a temperature of 25-28° C.
4. Surface-sterilizing the eggs	• Place 480 ml distilled water in a plastic container and add 20 ml Purex (5.25% sodium hypochloride). • Place egg masses into the Purex-water mixture. • Shake gently for 5 min keeping egg masses submerged. • Wash egg masses 5 times with distilled water for 5 min. • Place the eggs in a petri dish lined with moist filter paper.
5. Putting eggs on the Kamano diet	• Lay the diet flasks to be infested on their sides. • Place the eggs (30 eggs/flask) at the blackhead stage on the lower side of the flask. Plug the flasks with sterile cotton and place them in the rearing room at 25-28° C.
6. Putting larvae on the modified southwestern corn borer diet	• When larvae hatch in the petri dish place 2 larvae on the diet in the plastic container.
7. Collecting larvae from diet for studies	• Larvae at any age can be collected from the diet for studies on mechanisms of resistance.
8. Producing adults	• Allow some larvae to pupate in 2.5-cm pieces of plastic or paper straw. Place the plastic or paper straw over the diet when the larvae are in the last instar. When most of the larvae have pupated, remove the old diet and return pupae in straws to the container. Return larvae that have not pupated, with some empty straw pieces, to the container. • Emerging adults can be used for release on the susceptible variety in the screenhouse for screening. Some can be placed in the oviposition cage to maintain the culture.

SCREENING METHODS

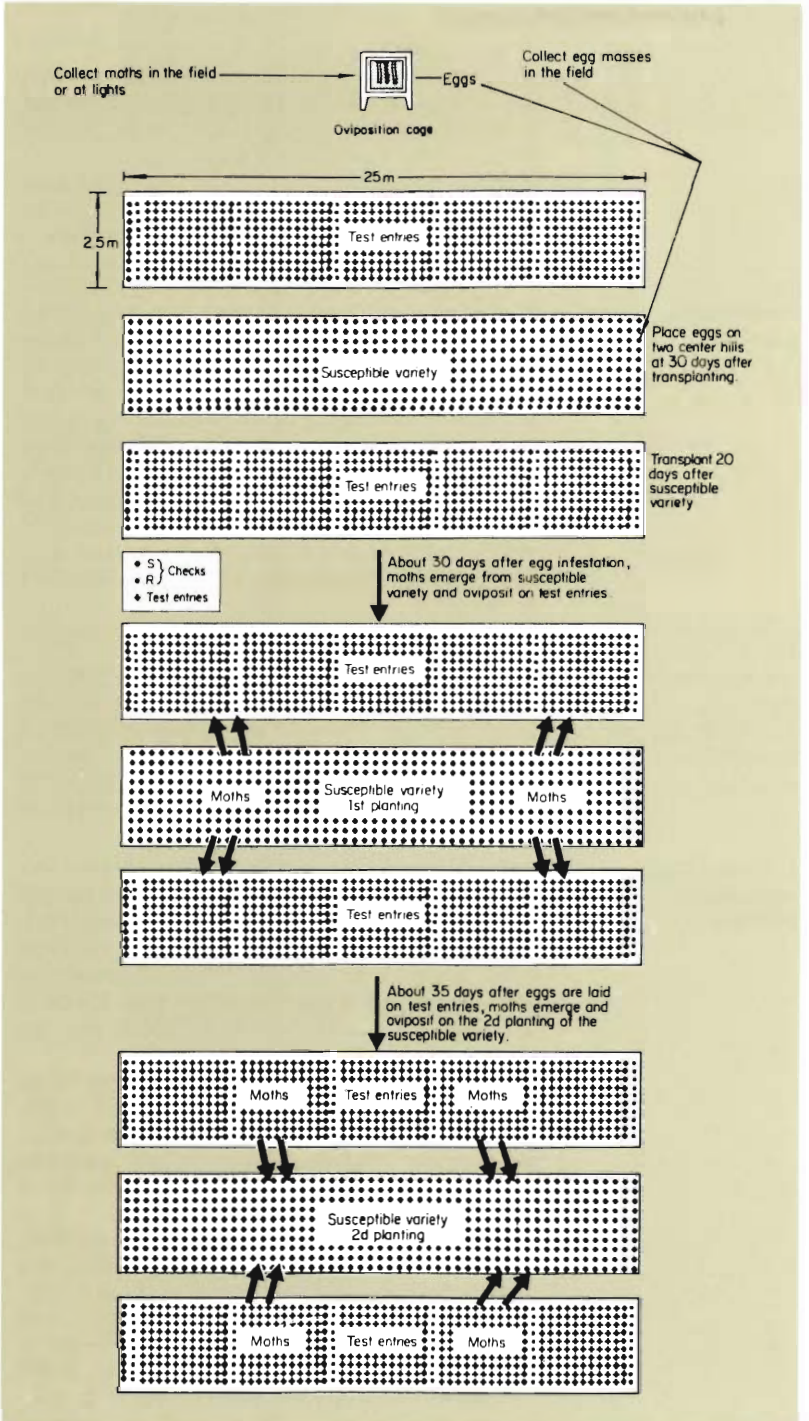
Screenhouse screening

The technique used for screenhouse screening of rices for resistance against the striped stem borer involves the development of a stem borer population on a plant bed of a susceptible variety. Moths emerging from the plants will infest adjacent beds of test entries.

Steps	Key points
1. Constructing a screenhouse	• Construct a screenhouse 28 × 22 × 2.5 m. This size can accommodate 6 25- × 2.5-m beds, separated by concrete pathways. It provides the capacity to evaluate 600 entries at one time in nonreplicated studies. The size of the screenhouse and plant beds can be adjusted to suit the needs of a screening program.
2. Preparing test materials	• Obtain seed of the test entries. Include Rexoro or IR8 as the susceptible check and TKM6 as the resistant check.
3. Scheduling planting	• Plant when eggs (or other insect stages) for infestation are available.
4. Preparing the plant beds	• Prepare 25- × 2.5-m beds separated by pathways (Fig. 3). One of every three plant beds is for the susceptible variety (as a source of infestation for the test entries) and two (one on each side) are for the test entries (Fig. 4).
5. Sowing seed of the susceptible variety	• Sow about 100 g of IR8 or Rexoro seed intended for each susceptible plant bed 20 days ahead of the test entries.
6. Transplanting the susceptible variety to serve as infestation source	• Transplant the susceptible variety 20 days after sowing (DAS) at a distance of 20 cm between rows and 10 cm between plants at 2 seedlings/hill. One hundred twenty-five 2.5-m rows are planted in the bed.
7. Sowing seed of the test entries	• On the same day that the beds of the susceptible variety are planted, sow seed of each test entry (2 g for a 2.5-m row). Sow the same amount of seed of the susceptible and resistant checks.
8. Transplanting test entries	• Transplant 20-day-old seedlings of the test entries in their respective plant beds at a 20 × 20 cm spacing at 1 seedling/hill (transplant Rexoro at 2 seedlings/hill because it has low tillering ability).

3. Screenhouse interior.

Steps	Key points
	After every 10 rows of test entries, plant a row of the susceptible check and a row of the resistant check (Fig. 4). Each plant bed will hold 105 rows (2.5-m-long) of test entries and 11 rows each of the susceptible and resistant checks.
9. Labeling the entries	• Label each entry properly with a wooden tag tied to a bamboo stick.
10. Infesting the plant bed of the susceptible variety	• The method commonly used at IRRI is to infest the plants of the susceptible variety with egg masses. At 30 DT (the test entries are 10 DT at this time), cut to about 1/2 the leaves of the 2 plants in each row to be infested to prevent dislodging of larvae as they move up after hatching. Larvae tend to move upward to the leaf tip for a short period immediately upon hatching before they go down to the leaf sheath.



4. Procedure for screening varieties for resistance to the striped stem borer in the greenhouse.

Steps	Key points
	• Infest each of the two plants in the middle of each row with one egg mass. To do this, attach the leaf or wax paper with the egg mass to the middle portion of the leaves with a paper clip. • Another method of infesting the bed of the susceptible variety is by releasing the moths or by placing pupae on petri dishes near the bed.
11. Preventing egg predation	• Predators such as ants, spiders, and dragonflies feed on stem borer eggs. Ants also feed on larvae and pupae. To prevent ant damage make sure no leaves touch the edge of the bed. Maintain water in the bed at all times and remove floating plants because ants can walk on them. Dragonflies should be caught with an insect net and spiders removed by hand. To control other insect pests of the rice plants, a short-term residual insecticide can be used 10 days before egg infestation. Later, other lepidopterous defoliators may be removed by hand.
12. Infesting the test entries a. With moths from susceptible varieties	• At 30 days after infestation (DI) of the susceptible Rexoro or IR8 plants (test entries are 40 DT) moths emerge. The moths lay eggs on the test entries. When all moths have emerged, the Rexoro or IR8 plants are cut and a new batch of the same variety is planted in the same area in preparation for screening another set of test entries.
b. By the Davis inoculation method	• Instead of planting a susceptible variety and infesting it to serve as source of moths that will lay eggs on the test entries, the Davis inoculator (Fig. 5) can be used to infest the test entries with first-instar larvae. The inoculation method allows the screening of more test entries because the beds otherwise devoted to the susceptible variety can be planted with the test entries. • Collect moths in light traps at night and place them in the oviposition cage the same night. Collect the wax paper with egg masses each morning and incubate the egg masses in a jar with moist filter paper. When eggs hatch, prepare to inoculate the plants. — Mix finely ground maize cob grits with the first-instar larvae by pouring about 100 ml of the grits onto a wax paper containing the larvae. Pour the mixture into a 1-liter supply bottle and repeat the mixing process until the bottle is half full. Calibrate to deliver 10 larvae/hill. If the inoculator delivers too many larvae, add maize cob grits to the mixture until the desired

5. Using the Davis inoculator to dispense about 10 first-instar stem borer larvae/hill.

Steps	Key points
	number of larvae per delivery is obtained. If too few larvae are delivered, add larvae to the bottle and shake well. — Drain the water from the bed and leave a muddy surface. Inoculate the larvae by dropping them on the mud at the base of the hill. Larvae will crawl onto the plants to feed. — Before inoculating the next hill shake the contents of the inoculator.
13. Evaluating a. Oviposition preference	• Oviposition preference can be determined when moths reared from the bed of susceptible varieties are used to infest the test entries. • Count the egg masses on the test entries 35 DI of the susceptible plants or about 5 days after adult emergence is observed. • Do a second counting 15 days after the first. During the first counting, encircle egg masses with a marker pen to avoid double counting during the second counting. • Compare the number of egg masses per variety.
b. Deadhearts	• Count deadhearts on test entries 20 days after oviposition is observed or 14 days after inoculation when the Davis inoculator is used. • Do a second counting 2 weeks after the first. • The screening test is considered valid when the deadhearts on the susceptible check are at least 25%.

Steps	Key points
	• Calculate percentage of deadhearts for each entry: $\%\ \text{deadhearts} = \frac{\text{no. of deadhearts counted}}{\text{total no. of tillers observed}} \times 100$ • Convert percentage of deadhearts to a new figure (D) which corrects for the level of infestation: $D = \frac{\%\ \text{deadhearts in test entry}}{\%\ \text{deadhearts in susceptible check (av of checks on each side of the test entry)}} \times 100$ • Transform the corrected figure (D) to a 0-9 scale.

Scale	*Percent deadhearts (D)*
0	None
1	1-20
3	21-40
5	41-60
7	61-80
9	81-100

c. Whiteheads	• Whitehead damage is usually not considered in screenhouse screening. To determine resistance to whitehead damage, the plants should be infested at the booting stage. Percentage of whiteheads can be estimated at plant maturity using the procedure described under *Field screening.*

Field screening

Field screening is more common than screenhouse screening because of the lack of screenhouse facilities. Screenhouses usually accommodate a few hundred test entries at one time whereas about 3,000 entries can be screened in a 1-ha field. Field screening uses natural infestation and the laborious artificial infestation procedure is not necessary. When natural infestations are low and labor is available, however, first-instar larvae can be added through the Davis inoculator. Field screening is often difficult because of lack of sufficient natural infestation, mixtures of two or more stem borer species, and destruction of the plots by pests other than stem borers.

Steps	Key points
1. Selecting a hot spot	• Select a site where the population level is extremely high (hot spot) and where the insect population consists primarily of the striped stem borer. Select a time of the year when populations are high.
2. Scheduling planting	• Plant when rice stem borer moths are in peak abundance. The plants should be in the vegetative stage when maximum moth flight occurs. Available light trap data or previous experience can be used to determine the proper planting date to obtain maximum infestation. At IRRI the moth population is usually highest in March and October and field plantings are made in February and September.
3. Sowing seed of test entries	• Prepare 1- × 10-m raised seedbeds (wet-bed method) in the field. Sow about 5 g of each entry in a 50-cm row with 10 cm between rows.
4. Transplanting the seedlings	• Before transplanting, apply fertilizer to the field at a rate of 60 kg N, plus 30 kg P_2O_5 and 30 kg K_2O/ha if necessary. • Transplant 20-day-old seedlings in 5-m rows and at a 25- × 25-cm distance (20 hills/row). For initial screening of the germplasm collection plant one row per variety, but for retesting plant four rows with each row as a replication. • After every 10 rows of test entries, plant 1 row each of a susceptible check (Rexoro) and a resistant check (TKM6). • Plant all entries at one seedling per hill except for Rexoro, which should be at two seedlings per hill.
5. Labeling the entries	• Label entries with stakes as in the screenhouse screening.
6. Evaluating	
a. Deadhearts	• Deadhearts are observed 30 and 50 DT on 16 plants in each row. Do not include the two plants at both ends of each row. • Convert percentage of deadhearts to a new figure (D) using the formula given in *Screenhouse screening.* • Transform the corrected figure (D) to a 0-9 scale.
b. Whiteheads	• Count the whiteheads when plants reach maturity. Whitehead counts are usually taken only when breeding lines are screened because of extreme maturity date differences in varieties from the germplasm collection.

Steps	Key points
	• Compute percentage of whiteheads: $\%\ \text{whiteheads} = \frac{\text{no. of whiteheads}}{\text{total productive tillers (those with whiteheads and those with norma panicles)}} \times 100$
	The test is considered valid when whiteheads in the susceptible check average at least 10%.
	• Convert percentage of whiteheads to D as in *Screenhouse screening,* and transform D to a 0-9 scale.

Scale	*Percent whiteheads (D)*
0	None
1	1-10
3	11-25
5	26-40
7	41-60
9	61-100

MECHANISMS OF RESISTANCE

Selected varieties with low deadheart or whitehead infestation can be further evaluated to determine the mechanisms of resistance. The general methods described can be used for both the striped stem borer and darkheaded stem borer *Chilo polychrysus.*

Antixenosis for oviposition in the greenhouse

Steps	Key points
1. Preparing the test materials	• Obtain seed of test varieties. Include IR8 or Rexoro as the susceptible check and TKM6 as the resistant check.
2. Sowing seed	• Sow seed of the test varieties in wooden seedboxes.
3. Transplanting the seedlings	• At 15 DAS, transplant 4 seedlings in each of 4 pots for each variety. One pot represents one replication. Label each pot properly with a garden stake. • Arrange pots in a randomized complete block design in metal trays filled with water.
4. Infesting the plants	• Cage (with nylon or fiberglass mesh) the potted plants using one cage per replication.

Steps	Key points
	• Release 20 newly emerged adults per pot at 1 female:1 male, into each cage. If moths are not sufficient to infest all replications at one time, stagger infestation by releasing 20 moths (1 female:1 male) in each replication every night for 10 consecutive nights.
5. Evaluating	• Count egg masses 3 DI or 3 days after last moth infestation when infestation is done on a staggered basis. • Compare number of egg masses among varieties.

Antixenosis for oviposition in the screenhouse

Steps	Key points
1. Preparing test materials	• Follow the procedure of the greenhouse test.
2. Sowing seeds	• Follow the procedure of the greenhouse test.
3. Transplanting seedlings	• Transplant 15-day-old seedlings of the test varieties in concrete beds in a screenhouse. Plant 4 2.5-m rows (20 × 20 cm) of each variety at 1 seedling/hill. A row represents one replication. Arrange the varieties in a randomized complete block design.
4. Caging plants	• Place one cage over each replication.
5. Infesting the plants	• Release 60 newly emerged adults per row of 12 plants at 1 female:1 male.
6. Evaluating	• Make observations as in the greenhouse test.

Antibiosis on larval survival

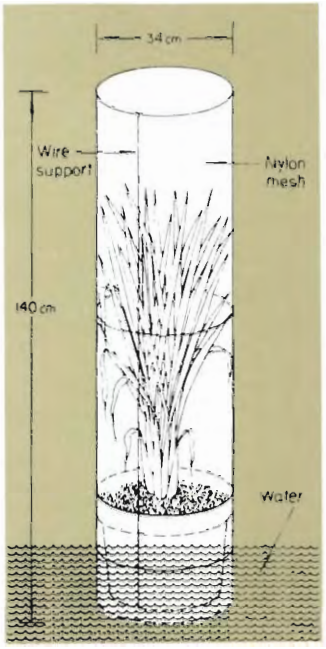
Steps	Key points
1. Planting	• Transplant 15-day-old seedlings in 38-cm-diam clay pots at 5 seedlings/pot. Plant 5-10 pots for each test entry, 1 pot representing 1 replication. Include a resistant and a susceptible check. • Arrange the pots in a randomized complete block design in a water pan in the greenhouse. • If other insects such as leafhoppers and planthoppers are abundant in the test area and are potential pests, cage the plants immediately after transplanting.

Steps	Key points
2. Infesting	• Prune the tillers at 45 DT so that the tillers in each pot number the same and the larvae used to infest each pot also number the same. Infest with newly hatched larvae at one larva per tiller. • Place the larva near the auricles of the youngest leaf using a fine camel hair brush (Fig. 6). • Put each pot in a nylon mesh cage (Fig. 7) to prevent the larva from escaping or transferring to another plant. If cages are not available, trim the leaves so that the leaves of one variety do not touch those of another. That will discourage larval transfer from one plant to another.
3. Evaluating	• Dissect the plants at 30 DI and count the larvae and pupae. • Weigh the larvae and pupae separately per hill or pot and express weight in milligrams per larva or pupa. • Calculate percentage of survival: $\% \text{ survival} = \frac{\text{no. of live larvae and pupae counted}}{\text{no. of initial larvae infested}} \times 100$ • Compare results of the various entries with those of the susceptible and resistant checks.

6. Putting a stem borer larva on the auricle of the rice plant.

Antibiosis on population growth

Steps	Key points
1. Preparing test materials	• Follow procedure in *Antixenosis for oviposition in the greenhouse.*
2. Sowing seeds of test entries	• Follow procedure in *Antixenosis for oviposition in the greenhouse.*
3. Transplanting the seedlings	• Transplant 15-day-old seedlings of the test varieties in 38-cm-diam pots with 3 hills, at 2 seedlings/hill. Plant 10 pots for each variety, 1 pot representing 1 replication. • Urea may be applied to increase tillering capacity of the plants. • Cage each pot with mylar or nylon mesh cages (Fig. 7).
4. Infesting the plants	• Before infestation at 40 DT, count the tillers per hill or pot. Prune to maintain the same number of tillers for all varieties. • Trim the leaves of the plants. Use first-instar larvae, at one larva/tiller.
5. Evaluating	• Record number of deadhearts at 7, 14, and 28 DI; record whiteheads at harvest. • Collect emerging adults daily and record the number. Place them on caged plants (30-40 DT) of the same variety. • Collect egg masses daily from each pot and place them in petri dishes lined with moist filter paper. One petri dish corresponds to one pot. • Count larvae hatching from each dish daily until hatching is finished. • Base population growth on the number of larvae that hatch from the eggs. • Compare results of the entries with those of the susceptible and resistant checks.



7. A cylindrical frame (140 cm × 34 cm diam) of heavy wire supports a nylon mesh cage of similar dimension. The bottom portion of the nylon mesh extends into water to prevent insect escape (Waldbauer and Marciano 1979).

SOURCES OF RESISTANCE

About 15,000 entries from the IRRI germplasm collection have been screened for striped stem borer resistance. Of these, 23 have been selected as resistant (see tables).

Rice varieties or lines resistant to the striped stem borer (*Chilo suppressalis* Walker) at IRRI, 1982.

IRRI accession no.	Variety or line	Origin
237	TKM6	India
99	Taitung 16	Taiwan
3082	Yabami montakhab	Egypt
89	Chianan 2	Taiwan
7299	Su-yai 20	China
7300	Szu-maio	China
1863	Bmt 53 R3536	USA
958	CI 5339	China
1099	36/14 Kaskham	China
39270	IET2845	India
	IET5540	India
32614	IR1514A-E666	Philippines
	IR2328-491-1-1-1	Philippines
	IR2798-143-3-2	Philippines
	IR4791-89	Philippines
	IR5201-122-2	Philippines
39247	CR157-392-4	India
39382	IR3941-97-1	Philippines
11355	IR20	Philippines
30416	IR36	Philippines
36958	IR40	Philippines
39341	IR44	Philippines
47688	IR50	Philippines

Sources of resistance to the striped stem borer *Chilo suppressalis* as listed by Patanakamjorn and Pathak (1967) are:

IRRI accession no.	Variety or line	Origin
00485	Kinki 47	Japan
00489	Miho III	Japan
01397	Pu Tauch Pu Chih Chi 1, Hao	China
00520	Tedori-wase	Japan
00522	Tone-wase	Japan

Wild rice species resistant to the striped stem borer *Chilo suppressalis* at IRRI, 1983.

IRRI accession no.	Species	Origin
100878	*O. officinalis*	Thailand
101079	*O. minuta*	Philippines
101129	*O. minuta*	Philippines
101141	*O. minuta*	Philippines
101154	*O. officinalis*	Malaysia
101417	*O. punctata*	Kenya
102168	*O. nivara*/*O. rufipogon*	India

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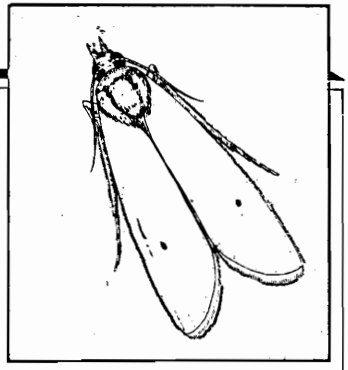


Chapter 13
YELLOW STEM BORER

The yellow stem borer *Scirpophaga incertulas* is the most important stem borer attacking rice and is widely distributed throughout South and Southeast Asia. It develops only on rice and feeds within the stem, causing deadhearts and whiteheads. It has become most abundant where multiple rice cropping is practiced.

No methods for rearing the insect have been developed, but screenhouse and field methods in screening for resistance are available.

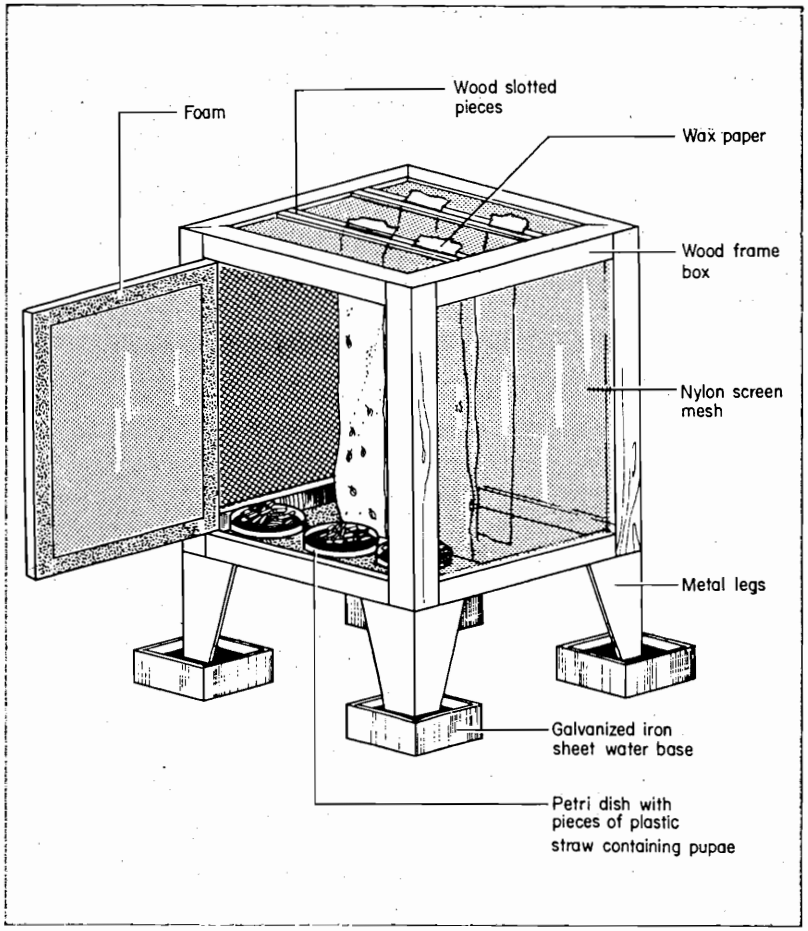
Methods described for the yellow stem borer can also be used for the white stem borer *S. innotata.*

SCREENING METHODS

Screenhouse screening

Yellow stem borer oviposition is not restricted to the rice plant. Because eggs may be laid on the screen wire walls and roof of the screenhouse, and are poorly distributed on the test entries, the plants must be artificially infested with first-instar larvae. To obtain larvae for artificial infestation, egg masses in the field or adult females in light traps at night are collected. First-instar larvae come from the field-collected egg masses and from adults in oviposition cages (Fig. 1). The screenhouse screening technique is illustrated in Figure 2.

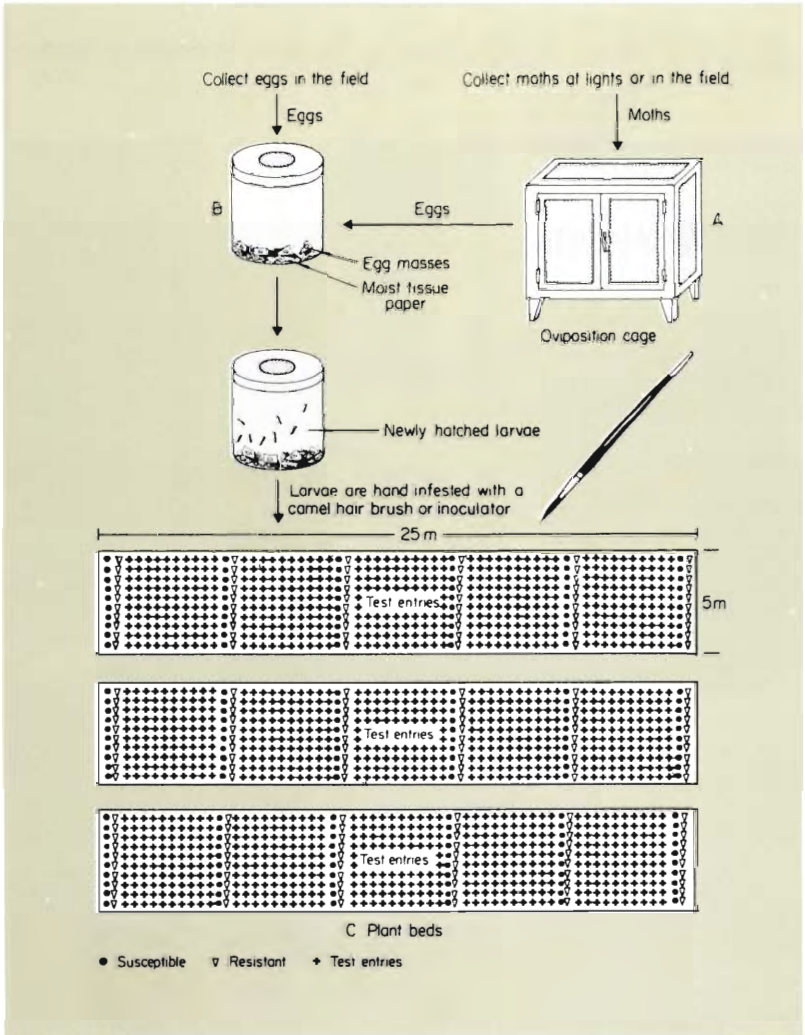
Steps	Key points
1. Constructing a screenhouse	• Construct a screenhouse 28 × 22 × 2.5 m. A screenhouse of this size accommodates 6 25- × 2.5-m beds separated by pathways and provides the capacity to evaluate 600 entries at one time in nonreplicated studies. The size of the screenhouse and plant beds can be adjusted to suit the screening program.
2. Scheduling planting	• Check light trap records to determine seasonal abundance of moths. At IRRI, peaks occur in March and October. • Schedule planting to coincide with the peak abundance of moths and egg masses in the field. Egg masses are abundant on seedlings 2-3 weeks after transplanting.
3. Collecting moths or eggs in the field	• Collect eggs in the field or collect moths near lights in the evening or in the field and place them in oviposition cages containing wax paper or rice



1. Oviposition cage for stem borers. Wood portions are covered with foam rubber to prevent oviposition on them. Eggs are laid on the wax paper.

Steps	Key points
	plants that serve as an oviposition substrate (Fig. 1, Fig. 2A). Remove eggs from the cages every morning. Cut off portions of wax paper or leaves with newly laid eggs and place them in jars with a screen cover and moist cotton at the bottom (Fig. 2B). If eggs are not needed for a few weeks, incubate them at 15-20° C. Newly laid eggs can be kept at this temperature for about 2 weeks without a decrease in hatchability. However, eggs about to hatch at time of collection will hatch at 15-20° C. To hatch stored eggs, remove them from the 15-20° C temperature and put them in a temperature of 25-30° C. Storing eggs allows a large number of larvae hatching the same day for infesting the plants.

Steps	Key points
4. Sowing test materials	• At 14 days before the planned transplanting time, sow around 2 g of seed (for 1 replication) of each test entry in a 20-cm row with 10 cm between rows in 60- × 40- × 10-cm wooden seedboxes with 5-cm-deep soil. Include a susceptible check (Rexoro) and a resistant check (IR1820-52-2-4-1).
5. Planting test materials	• Fourteen days after sowing, transplant the seedlings in the plant beds (Fig. 2C) at 20- × 20-cm spacing.



2. Screenhouse screening for yellow stem borer resistance.

Steps	Key points
	• Plant one row for each variety. For every 10 rows of test entries, plant a row each of the susceptible and the resistant check. Plant one seedling per hill except for Rexoro, which needs two seedlings per hill because of low tillering capacity. • Plant one replication (one row) for initial screening tests. For retesting, replicate entries four times (four rows) in a randomized complete block design. • Label each row or entry with a wooden tag tied to a bamboo stick. • Apply fertilizer at the rate of 60 kg N/ha 1 week after transplanting.
6. Infesting the test entries	• Allow previously collected egg masses to hatch in rearing jars.

3. Placing a yellow stem borer larva on the auricle of a rice plant.

4. Infesting rice seedlings with first-instar yellow stem borer larvae with the Davis inoculator.

Steps	Key points
a. Camel hair brush method	• When the test entries are ready for infestation bring the jars containing hatching larvae to the screenhouse. • Infest the test plants with newly hatched larvae (10 larvae/hill) 14 days after transplanting (DT). Using a fine camel hair brush, place the larvae on the youngest leaf or auricles (Fig. 3) of one tiller.
b. Davis inoculation method	• Use of the inoculator (Fig. 4) is less laborious than that of the camel hair brush and is about 10 times faster. However, the infestation level is less accurate.

Steps	Key points
	• Collect wax paper with egg masses each morning and incubate the egg masses in a jar with moist filter paper. When eggs hatch, prepare to inoculate the plants. — Mix finely ground maize cob grits with the first-instar larvae by pouring about 100 ml of the grits onto wax paper containing the larvae. Pour the mixture into a 1-liter supply bottle and repeat the mixing process until the bottle is half full. Calibrate to deliver 10 larvae/hill. If the inoculator delivers too many larvae, add grits to the mixture until the desired number of larvae per delivery is obtained. If too few larvae are delivered, add larvae to the bottle and shake well. — Drain the water from the bed, leaving a muddy surface. — Inoculate the larvae by dropping them on the mud at the base of the hill. They will crawl onto the plants to feed. — Before inoculating the next hill, revolve the inoculator three times to maintain a uniform distribution of the larvae in the grits.
7. Evaluating a. Deadhearts	• Count the number of deadhearts twice: 2 and 4 weeks after infestation with larvae. • The test is considered valid when percentage of deadhearts on the susceptible check is at least 25. • Compute the percentage of deadhearts for each entry: $\%\ \text{deadhearts} = \frac{\text{no. of deadhearts counted}}{\text{total no. of tillers observed}} \times 100$ • Convert percentage of deadhearts to a new figure (D), which corrects for the level of infestation. $D = \frac{\%\ \text{deadhearts in test entry}}{\%\ \text{deadhearts in susceptible check (av of checks on each side of the test entry)}} \times 100$ • Transform the converted figure to a 0-9 scale.

Scale	*Percent/deadhearts (D)*
0	None
1	1-20
3	21-40
5	41-60
7	61-80
9	81-100

Steps	Key points
b. Whiteheads	• Whitehead damage is usually not considered in screenhouse screening. For determining resistance to whitehead damage, infest the plants at the booting stage. Percentage of whiteheads can be determined at plant maturity using the procedure described in *Field screening.*

Field screening

Field screening is more common than screenhouse screening because of the shortage of screenhouse space. Screenhouses usually can accommodate a few hundred test entries at one time whereas about 3,000 entries can be screened in a 1-ha field. For field screening, larval infestation is natural and the laborious artificial infestation is unnecessary. However, when natural infestations are low and labor is available, infestation with first-instar larvae can be done with the Davis inoculator.

Field screening is often difficult because of insufficient natural infestation, mixtures of two or more stem borer species, and destruction of the plots by other pests.

Steps	Key points
1. Selecting a hot spot	• Select an area where the population level is extremely high (*hot spot*) and where the insect population consists primarily of the yellow stem borer. Select a time of the year when populations are high.
2. Scheduling planting	• Plant during peak abundance of rice stem borer moths. The plants should be in the vegetative stage when maximum moth flight occurs. Light trap data, if available, or previous experience can be used to determine the proper planting date to obtain maximum infestation. At IRRI the moth population is usually highest in March and October and field plantings are made in February and September.
3. Sowing seed of test entries	• Prepare 1- × 10-m raised seedbeds (wet-bed method) in the field. Sow about 5 g of each entry in a 50-cm row with 10 cm between rows.
4. Transplanting the seedlings	• Before transplanting, apply fertilizer to the field at a rate of 60 kg N, plus 30 kg P_2O_5 and 30 kg K_2O/ha if necessary. • Transplant 20-day-old seedlings in 5-m rows and at 25- × 25-cm distance (20 hills/row). For initial germplasm screening, plant one row per variety,

Steps	Key points
	but for retesting, plant four rows with each row as a replication. • After every 10 rows of test entries, plant a row each of a susceptible check (Rexoro) and a resistant check (IR1820-52-2-4-1). • Plant all entries at one seedling per hill except for Rexoro, which should be planted at two seedlings per hill.
5. Labeling the entries	• Label entries with stakes as in the screenhouse screening.
6. Evaluating for deadheart and whitehead damage	• Observe for deadhearts 30 and 50 DT on 16 plants in each row. Do not include the two at both ends of each row. • Compute percentage of deadhearts and convert to a new figure (D) as in screenhouse screening. • Convert the corrected figure (D) to a 0-9 scale as in screenhouse screening. • Count whiteheads before harvest (around 90 DT). This is done only when testing breeding lines because of maturity date differences when evaluating a germplasm collection. • Compute percentage of whiteheads: $\%\ \text{whiteheads} = \frac{\text{no. of whiteheads}}{\text{total productive tillers (those with whiteheads and those with normal panicles)}} \times 100$ • The test is considered valid when whiteheads in the susceptible check average at least 10%. • Convert percentage of whiteheads using the formula for deadhearts and use the following to place whitehead damage on a 0-9 scale.

Scale	*Percent whiteheads (D)*
0	No damage
1	1-10
3	11-25
5	26-40
7	41-60
9	61-100

MECHANISMS OF RESISTANCE

Selected varieties with low deadheart or whitehead infestation can be further evaluated to determine the mechanisms of resistance. The general methods described can be used for both the yellow and the white stem borer.

Antixenosis for oviposition in the greenhouse

Steps	Key points
1. Preparing test materials	• Obtain seed of test varieties. Include IR8 or Rexoro (susceptible check) and IR1820-52-2-4-1 (resistant check).
2. Sowing seed	• Sow seed of the test varieties in wooden seedboxes.
3. Transplanting the seedlings	• At 15 days after sowing (DAS), transplant 4 seedlings in each of 4 pots for every variety. One pot represents one replication. Label each pot properly with a garden stake. • Arrange pots in a randomized complete block design in metal trays filled with water.
4. Infesting the plants	• Cage with nylon or fiberglass mesh the potted plants. One cage is one replication. • Release 20 newly emerged adults per pot, at 1 female:1 male, into each cage. If there are not enough moths to infest all replications at one time, stagger infestation by releasing 20 moths (2 females:1 male) in each replication every night for 10 consecutive nights.
5. Evaluating	• Count egg masses 3 days after infestation (DI) or 3 days after last moth infestation when infesting on a staggered basis. • Compare the number of egg masses among varieties.

Antixenosis for oviposition in the screenhouse

Steps	Key points
1. Preparing test materials	• Follow the procedure of the greenhouse test.
2. Sowing seed	• Follow the procedure of the greenhouse test.
3. Transplanting the seedlings	• Transplant 15-day-old seedlings of the test varieties in concrete beds in a screenhouse. Plant 4 2.5-m rows (20 × 20 cm) of each variety, at 1 seedling/hill. A row represents one replication. Arrange the varieties in a randomized complete block design.
4. Caging plants	• Place one cage over each replication.

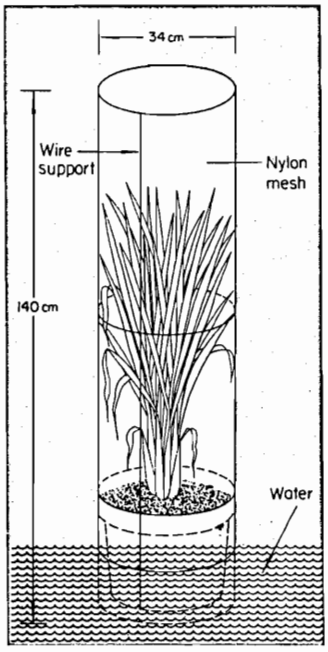
Steps	Key points
5. Infesting the plants	• Release 60 newly emerged adults per row of 12 plants at 1 female:1 male.
6. Evaluating	• Make observations as in the greenhouse test.

Antibiosis on larval survival

Steps	Key points
1. Planting	• Transplant 15-day-old seedlings in 38-cm-diam clay pots at 5 seedlings/pot. Plant 10 pots for each test entry, with 1 pot representing 1 replication. Include a resistant and a susceptible check. • Arrange the pots in a randomized complete block design in a water pan in the greenhouse. • If other insects such as leafhoppers and planthoppers are abundant in the test area and are potential pests, cage the plants immediately after transplanting.
2. Infesting the plants	• Prune the tillers 45 DT so that the number of tillers in each pot is the same and the number of larvae used for infesting each pot is similar. • Infest the plants with newly hatched larvae at one larva per tiller. • Place the larva near the auricles of the youngest leaf using a fine camel hair brush (Fig. 3). • Put each pot in a nylon mesh cage (Fig. 5) to prevent the larvae from escaping or transferring from one plant to another. If cages are not available, trim the leaves so that the leaves of one variety do not touch the leaves of another. This will discourage larval transfer.
3. Evaluating	• Dissect the plants 30 DI and count the larvae and pupae. • Weigh the larvae and pupae separately per hill or pot and express weight in milligrams. • Calculate percentage of survival: $\% \text{ survival} = \frac{\text{no. of live larvae and pupae counted}}{\text{no. of initial larvae infested}} \times 100$ • Compare results of the various entries with those of the susceptible and the resistant checks.

Antibiosis on population growth

Steps	Key points
1. Preparing test materials	• Follow the procedure in *Antixenosis for oviposition in the greenhouse.*
2. Sowing seed of test entries	• Follow the procedure in *Antixenosis for oviposition in the greenhouse.*
3. Transplanting the seedlings	• Transplant 15-day-old seedlings of the test varieties in 38-cm-diam pots with 3 hills at 2 seedlings/hill. Plant 10 pots for each variety, 1 pot representing one replication. • Urea may be applied to improve the tillering capacity of the plants. • Cage each pot with mylar film or nylon mesh cages (Fig. 5).
4. Infesting the plants	• Before infestation at 40 DT, count the tillers per hill or pot. If possible, maintain the same number of tillers in each pot for all varieties. • Trim the leaves of the plants. Infest plants with first-instar larvae, one larva/tiller.
5. Evaluating	• Record the number of deadhearts at 7, 14, and 28 DI; record whiteheads at harvest. • When adults start to emerge, collect emerging adults daily and record the number. Place them on caged plants (30-40 DT) of the same variety. • Collect egg masses daily from each pot and place them in petri dishes lined with moist filter paper. One petri dish corresponds to one pot. • Count larvae hatching from each dish daily until hatching is finished. • Base population growth on the number of larvae that hatch from the eggs. • Compare results of the entries with those of the susceptible and resistant checks.



5. A cylindrical frame (140 cm × 34 cm diam) of heavy wire supports a nylon mesh cage of similar dimension. The nylon mesh extends to the water to prevent insect escape.

SOURCES OF RESISTANCE

The following varieties have been identified as moderately resistant to the yellow stem borer *Scirpophaga incertulas* based on 1983 screenhouse and field screening at IRRI.

IRRI accession no.	Variety	Origin
6331	CO18	India
6365	MTU15	India
6396	CO21	India

IRRI accession no.	Variety	Origin
8763	DM27	Bangladesh
10191	Mainagiri	India
11055	WC1253	India
11057	WC1263	India
11261	Lepqu	Philippines
11355	IR20	Philippines
12890	Ratna (CR44-11)	India
25832	Aus Balam	Bangladesh
26952	Biplab	Bangladesh
30416	IR36	Philippines
32658	IR1820-52-2-4-1	Philippines
33136	IB 56-8	Burma
36958	IR40	Philippines
39270	IET2845	India
39355	IR3941-2-1-3	Philippines
39360	IR3941-9-2	Philippines
39415	IR5098-B-Jn-1B	Philippines
39453	IR5908-79-2	Philippines
39473	Kalinga 2	India
39486	Kn-1b-361-8-6-9-2-7	Indonesia
47688	IR50	Philippines
53435	IR54	Philippines

Based on field screening in India, Indonesia, Sri Lanka, and Thailand the following varieties have been identified as resistant to the yellow stem borer as reported in the literature.

Variety	Origin	Reference
ADT2	India	Velusamy et al 1975
ADT5	India	Velusamy et al 1975
ADT7	India	Velusamy et al 1975
ADT8	India	Velusamy et al 1975
ADT11	India	Velusamy et al 1975
ADT17	India	Velusamy et al 1975
ADT22	India	Velusamy et al 1975
ARC5920	India	Shastry et al 1971.
ARC6033	India	Shastry et al 1971
ARC6045	India	Shastry et al 1971
ARC6049	India	Shastry et al 1971
ARC6184	India	Shastry et al 1971
ARC7080	India	Shastry et al 1971
ARC7098	India	Shastry et al 1971
ARC7119	India	Shastry et al 1971
ARC7132	India	Shastry et al 1971
ARC7137	India	Shastry et al 1971
ARC7312	India	Shastry et al 1971
ARC7316	India	Shastry et al 1971
ARC10217	India	Shastry et al 1971
ARC10257	India	Khush 1977
ARC10346	India	Shastry et al 1971

Variety	Origin	Reference
ARC10379	India	Shastry et al 1971
ARC10386	India	Shastry et al 1971
ARC10443	India	Shastry et al 1971
ARC10528	India	Shastry et al 1971
ARC10598	India	Khush 1977
ARC10692	India	Khush 1977
ARC10958	India	Shastry et al 1971
ARC11261	India	Shastry et al 1971
ARC11313	India	Shastry et al 1971
ARC11332	India	Shastry et al 1971
ARC11537	India	Shastry et al 1971
ASD7	India	Pongprasert et al 1975
ASD10	India	Velusamy et al 1975
ASD12	India	Velusamy et al 1975
ASD13	India	Velusamy et al 1975
Beta repot	Bangladesh	Soeharjan and Leeuwangh 1972
CI 6002-1	Philippines	Pongprasert et al 1975
CO 1	India	Velusamy et al 1975
CO 3	India	Velusamy et al 1975
CO 19	India	Velusamy et al 1975
CO 22	India	Khush 1977 Soejitno 1974
CO 23	India	Velusamy et al 1975
CO 25	India	Velusamy et al 1975
CO 32	India	Velusamy et al 1975
Dendekkolon	Indonesia	Khush 1977
Dikwee	Nigeria	Pongprasert 1975
DNJ 97	Bangladesh	Pongprasert 1975
Gangala	India	Pongprasert 1975
GEB24	India	Velusamy et al 1975
H4	Sri Lanka	Velusamy et al 1975, Fernando 1967
H5	Sri Lanka	Fernando 1967
Hashikalmi	Bangladesh	Pongprasert 1975
Hathiel	Sri Lanka	Pongprasert 1975
IARI 6600	India	Velusamy et al 1975
IARI 6638	India	Velusamy et al 1975
IR305-4-20-3-3	Philippines	Velusamy et al 1975
W1263	India	Velusamy et al 1975
Jhingasail	India	Pongprasert 1975
Kalu Heenati	Sri Lanka	Fernando 1967
Kipusa	Rwanda Urundi	Prakasa Rao 1977
Kuruhondarawala	Sri Lanka	Pongprasert 1975
Leri	Indonesia	Khush 1977
Lunah	[a]	Soeharjan and Leeuwangh 1972
Mahsuri	Malaysia	Prakasa Rao 1977
Manglar	Indonesia	Soejitno 1974
Manovari	Indonesia	Soejitno 1974
MTU15	India	Prakasa Rao 1977
MTU3502	India	Velusamy et al 1975
MTU4246	India	Velusamy et al 1975
Muthumanikan	Sri Lanka	Pongprasert 1975
Padi	Indonesia	Soehardjan and Leeuwangh 1972

Variety	Origin	Reference
Pathong	Taiwan	Soejitno 1974
Pelopor	Indonesia	Khush 1977
Ptb 15	India	Velusamy et al 1975
Ptb 18	India	Pongprasert 1975
Ptb 21	India	Pongprasert 1975
PVR1	India	Velusamy et al 1975
Rathu Heenati	Sri Lanka	Pongprasert 1975
Rindu	Indonesia	Soejitno 1974
RP4-12	India	Velusamy et al 1975
Seri Jedah	—[a]	Soeharjan and Leeuwangh 1972
Si Jambe	Indonesia	Soejitno 1974
SLO 17	India	Prakasa Rao 1977
SR26B	India	Velusamy et al 1975
STB Res. 12708	—[a]	Velusamy et al 1975
Ta-poo-cho-z	China	Pongprasert 1975
TC37	—[a]	Prakasa Rao 1977
Tetep	Vietnam	Velusamy et al 1975
TKM2	India	Velusamy et al 1975
TKM6	India	Khush 1977
TNR1	India	Velusamy et al 1975
TNR2	India	Velusamy et al 1975
TR1	India	Velusamy et al 1975
W1251	India	Khush 1977
W1253	India	Khush 1977
W1257	India	Khush 1977
W1263	India	Khush 1977
Wild rice:		
Oryza ridleyi	India	Padhi and Prakasa Rao 1978
O. tisseranti	India	Padhi and Prakasa Rao 1978
O. perreiri	India	Padhi and Prakasa Rao 1978

[a]Information not available.

Accessions of wild rices resistant to yellow stem borer *Scirpophaga incertulas,* identified at IRRI, 1981.

IRRI accession no.	Species	Origin
100937	*O. punctata*	Ghana
100947	*O. officinalis*	India
100948	*O. officinalis*	India
100953	*O. officinalis*	India
100954	*O. punctata*	India
100966	*O. latifolia*	Panama
100973	*O. officinalis*	Philippines
101086	*O. minuta*	Philippines
101089	*O. minuta*	Philippines
101092	*O. minuta*	Philippines
101094	*O. minuta*	Philippines
101096	*O. minuta*	Philippines

IRRI accession no.	Species	Origin
101128	*O. minuta*	Philippines
101417	*O. punctata*	Kenya
101418	*O. eichingeri*	Uganda
101421	*O. eichingeri*	Uganda
101422	*O. eichingeri*	Uganda
101424	*O. eichingeri*	Uganda
101426	*O. eichingeri*	Uganda
101430	*O. punctata*	Uganda
101439	*O. punctata*	Ghana
101443	*O. latifolia*	Morocco
102382	*O. officinalis*	Indonesia
102481	*O. latifolia*	Nicaragua

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Chapter 14
SMALL MOTH BORER

The small moth borer *Diatraea saccharalis,* also known as the sugarcane borer, is a pest of rice, sorghum, sugarcane, maize, and wheat in the southern USA and in South America. Wild grasses act as hosts. Larvae, which bore in the stem, destroy the growing points and cause deadhearts, broken stalks, or stem damage. Rearing and greenhouse and field screening methods have been developed.

REARING METHODS

The small moth borer can be reared on the southwestern corn borer diet (Davis 1976). It has also been reared on an artificial diet described by Bowling (1967).

Preparing the diet

Steps	Key points
1. Preparing ingredients for the medium	• Prepare the following ingredients: Dried pinto beans 100.0 g Brewer's yeast 15.0 g Ascorbic acid 1.5 g Methyl parahydroxybenzoate 1.0 g Sorbic acid 0.5 g Formaldehyde 1.0 g Agar 6.0 g Water 375.0 g
2. Preparing rearing containers	• Use 15-ml plastic containers with paper disc caps or other suitable containers.
3. Preparing the medium	• Soak the dried pinto beans in water overnight. • Mix the other ingredients, except the agar, with 250 ml water in a food blender for 10 s. • Place the soaked beans in water and bring to boiling point. Drain off the water. • Add the beans to the mixture and blend for 45-60 s. • Dissolve the agar in the remaining 125 ml of water at 38° C. Immediately add it to the mixture and blend for another 30 s. • Pour the hot medium in a plastic squeeze bottle and dispense 5 ml into each rearing container.

Rearing the insect

Steps	Key points
1. Collecting egg masses	• Collect egg masses and place in 100-ml jars that have a screw-top lid (Gerber bottles). • Remove the metal center piece of the screw-top and replace it with 2 thicknesses of paper towel. Keep the jars (with the eggs) in a room at 26 ± 2° C and 85-90% RH.
2. Transferring larvae into the rearing container	• Transfer newly hatched larvae into the rearing container (2 larvae/container) using a camel hair brush. Cover the container with the paper disc caps.
3. Rearing the larvae	• Place the rearing containers in a cabinet with fluorescent lamps programmed for a photoperiod of 14 h light and 10 h dark at a temperature of 26 ± 2° C. • Mature larvae will prepare a pupation chamber by tunneling in the remaining food and fecal material, and pupate in the container. Leave the pupae in the container until adult emergence.
4. Transferring adults into oviposition jars	• When adults emerge, remove them from the container and place 5 pairs of adults in 3.8-liter glass jars containing 5 cm moist soil with a paper towel on top for oviposition, and a 5% sugar solution on cotton for food.
5. Collecting eggs	• Collect egg masses and place them in the 100-ml screw-top jars and continue the rearing cycle as described.

SCREENING METHODS

Field and greenhouse methods have been developed to screen rices for resistance to the small moth borer.

Field screening

The field screening method described is that used in the USA and is described by Oliver et al (1973).

Steps	Key points
1. Preparing seed	• Obtain seed of test varieties. Varieties Gulfrose and Saturn can be used as susceptible checks and TKM6 and PI 245718 as resistant checks.

Steps	Key points
2. Sowing seed	• Sow 5 g seed of each entry in a 2-m row plot; space rows 1 m apart. • Use a randomized complete block design with four replications.
3. Flooding the plots	• Flood the plots 4 weeks after planting. Three days before flooding, apply herbicide (propanil) and complete (NPK) fertilizer at recommended rates as needed.
4. Infesting the plants	• Infestation depends on the natural field population of *D. saccharalis.*
5. Evaluating	• Count damaged and undamaged plants in 1 m (row) in each plot 105 days after sowing (DAS). This is done by cutting the plants and examining each for signs of stalk borer damage. • Compute percentage of damaged tillers and compare results. $\%\ \text{damaged tillers} = \frac{\text{no. of damaged tillers}}{\text{total no. of tillers observed}} \times 100$

Greenhouse screening

Methods developed for greenhouse screening in Brazil (Martins et al 1977) are described.

Steps	Key points
1. Preparing seed	• Prepare seed of test entries.
2. Sowing seed	• Sow seed of test entries in rows in seedboxes.
3. Transplanting seedlings	• At 20 DAS, transplant seedlings in pots filled with water-saturated soil. Plant 3 seedlings/pot and 5 pots/variety. One pot represents a replication.
4. Infesting the plants	• When the plants are 65 days old (45 days after transplanting), record the number of tillers per pot. Infest the plants of each pot with 20 first-instar larvae of the borer.
5. Evaluating	• At 37 days after infestation (DI), gather the following data: — Number of damaged and undamaged tillers per pot;

Steps	Key points
	— Number and weight of surviving larvae and pupae in the plants. • Compare results among varieties: — % damaged tillers = $\frac{\text{no. of damaged tillers}}{\text{no. of damaged and undamaged tillers}} \times 100$ — % survival of larvae = $\frac{\text{no. of larvae and pupae recovered}}{\text{no. of larvae infested}} \times 100$ — Av weight of larvae = $\frac{\text{total weight of larvae recovered}}{\text{no. of larvae recovered}}$ — Tolerance is based on the ability of the plant to tiller despite attack. Tolerance = $\left[\text{no. of tillers at 37 DI}\right] - \left[\text{no. of tillers at time of larval infestation}\right]$ The higher the number, the greater the recuperation ability or tolerance of a variety.

SOURCES OF RESISTANCE

The literature identifies some sources of rice resistance to the small moth borer *Diatraea saccharalis*.

Designation	Reference
C-409	Martins et al 1977
Chiang an Tsao Pai Ku	Martins et al 1977
Su Yai 20	Martins et al 1977
Ti Jo Hung	Martins et al 1977
TKM6	Martins et al 1977
World collection 1541	Oliver and Gifford 1975, Oliver et al 1973
World collection 1584	Oliver and Gifford 1975, Oliver et al 1973
World collection 3669	Martins et al 1977

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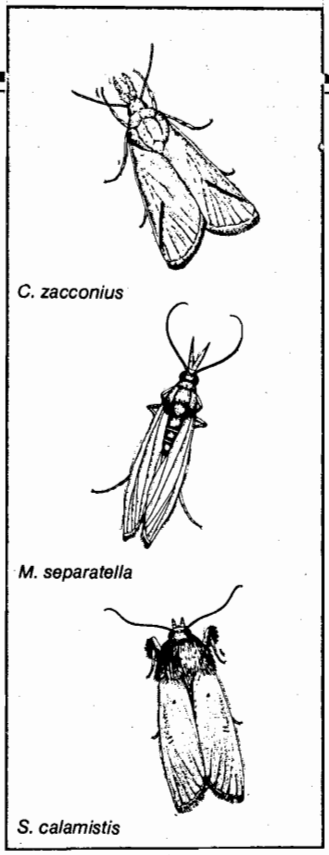
Chapter 15
AFRICAN STRIPED STEM BORER, WHITE STEM BORER, AND PINK STEM BORER



C. zacconius

M. separatella

S. calamistis

The African striped stem borer *Chilo zacconius* Blez. is a serious rice pest in West Africa. The African white stem borer *Maliarpha separatella* Rag. occurs throughout West and East Africa and Malagasy Republic but is restricted to rice. Damage is less severe in areas where only one crop a year is grown and where there is a distinct dry season than in areas where double-cropping is practiced and the wet season is long as in Malagasy Republic. The African pink stem borer *Sesamia calamistis* (Hamps.) is distributed throughout Africa south of the Sahara and in Malagasy Republic where it is polyphagous, feeding on rice, sugarcane, millet, sorghum, and maize (Bordat and Pichot 1980). The striped and pink stem borers cause deadheart and whitehead symptoms typical of other lepidopterous stem borers, but the larvae of the African white stem borer often feed on the lower internodes without developing deadhearts or whiteheads.

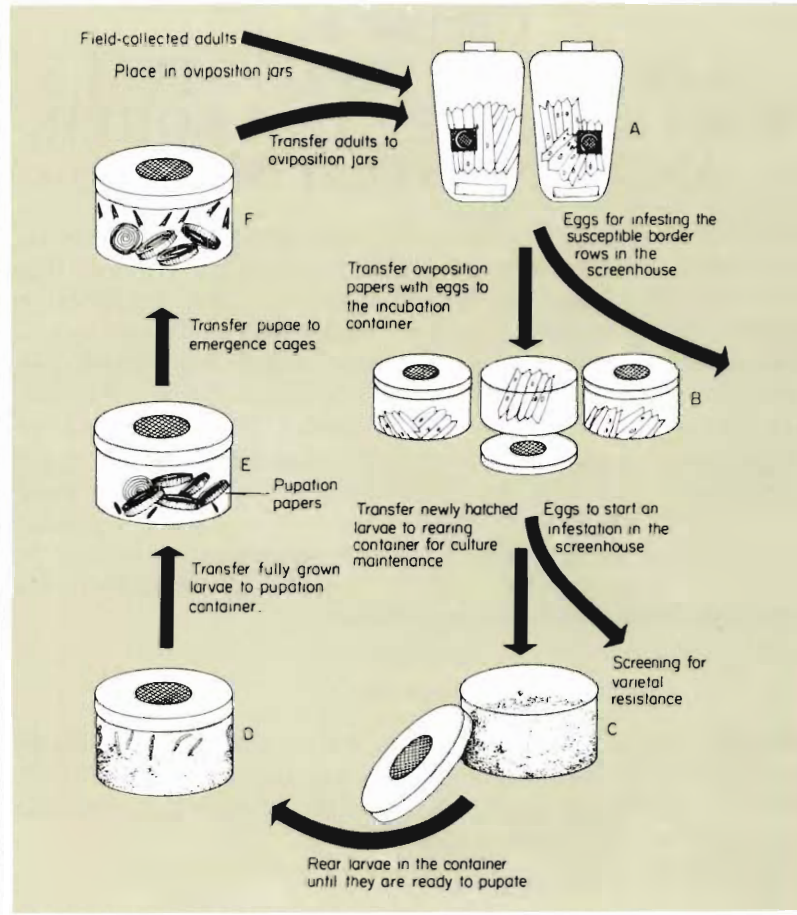
REARING

The African striped stem borer and the African pink stem borer can be reared on the same artificial diet described by Bordat and Pichot (1978, 1980). No artificial diet has been developed for the white stem borer. The rearing method is illustrated in Figure 1.

Steps	Key points
1. Preparing the medium	• Prepare the following ingredients:

Water	600.0 ml
Powdered agar	14.0 g
Corn meal flour	112.0 g
Wheat germ	28.0 g
Dry yeast	30.0 g
Ascorbic acid	10.0 g
Benzoic acid	1.2 g
Nipagine (methyl hydroxy 4-Benzoate)	1.0 g
Bactrim (sulfamide antibiotic)	0.25 g

- Mix the ingredients, except the agar, with 300 ml water in a food blender for 10 s.
- Dissolve the agar in the remaining 300 ml water at 38° C. Immediately add it to the mixture and blend for another 30 s.



1. Procedure for mass-rearing the African striped stem borer *Chilo zacconius*, and African pink stem borer *Sesamia calamistis* on an artificial diet

Steps	Key points
2. Putting the prepared diet in rearing containers	• Fill plastic containers (8-cm-diam and 5-cm-high) with the medium to about 1-cm-deep for the striped borer and 2-cm-deep for the pink borer (Bordat and Pichot 1978, 1980). These amounts are sufficient to rear 50 striped borer larvae and 20 pink borer larvae from the first instar to the pupal stage. The containers with the medium can be stored for 1 month in a refrigerator at 5-10° C.
3. Starting the insect culture	• Collect adults of the stem borers in the field and allow them to lay eggs in oviposition jars in the laboratory. The oviposition jars are cylindrical transparent polystyrene, 22 cm high and 12 cm in diam, and have 2 side openings (2 cm in diam) with fine wire mesh for ventilation (Fig. 1A).

Steps	Key points
	• Before putting the adults in the oviposition jar, place a piece of synthetic sponge saturated with water at the bottom of the jar to serve as a water source. • For the striped borer use rolls of corrugated or accordion folded paper (oviposition papers) 3 cm wide and 20 cm long for the adults to lay their eggs on. Writing paper formed into a cone is used as an oviposition substrate for the pink borer. • Put six females and eight males of the striped borer adults, or three females and four males of the pink borer adults in the oviposition jar.
4. Collecting eggs	• Remove the oviposition papers from the jar the following day and replace with new oviposition papers if additional eggs are required.
5. Incubating the eggs	• Incubate eggs in a round plastic container (incubation container) 5 cm high and 8 cm in diam (Fig. 1B). The lid or cover should have fine wire mesh openings. • Place a piece of water-saturated filter paper to maintain high atmospheric humidity in the container. • Cut the corrugated papers (oviposition papers) with eggs into pieces and place them over the moistened paper. • Keep the eggs in the container until they hatch. • Some eggs may be laid on the sides of the oviposition jar. If this happens, drench the eggs with 10% chlorinated water, rinse with plain water, and place them in the incubation container together with the eggs on the oviposition papers.
6. Transferring larvae to the rearing container	• Newly hatched larvae are positively phototaxic, so they gather on the top cover of the incubation container. • Make scratch marks on the surface of the medium in the rearing containers (from Step 2). • Transfer the newly hatched larvae into the rearing containers (Fig. 1C) by gently tapping the lid or cover of the incubation container over the medium to allow the larvae to drop onto the medium. • Place 50 striped borer larvae or 20 pink borer larvae in each rearing container. • Rear the insects in a room with the temperature maintained at 25 ± 1°C, relative humidity of 50-60%, photoperiod of 18 h light and 6 h dark, and light intensity of 3000-4000 lux.

Steps	Key points
7. Transferring fully grown larvae to the pupation container	• The larvae are ready to pupate when they start moving on the upper surface of the medium and spinning silken threads, attaching themselves to the sides of the rearing container (Fig. 1D). • Prepare pupation containers 8 cm high and 10 cm in diam with fine wire mesh windows on the cover (Fig. 1E). • Place a roll of pupation papers (corrugated papers) 2 cm wide for the larvae to pupate on. • After 24 h, remove the pupation papers and replace with new ones. Continue removing and replacing the pupation papers every 24 h until pupation is completed.
8. Transferring pupae to the emergence containers	• Adult emergence containers are similar to the rearing containers (Fig. 1F). • Place 30-50 pupae on pupation papers in each container. Keep them in the container until adults emerge.
9. Transferring adults to the oviposition containers	• Transfer newly emerged adults (6 striped borer females and 8 males and 3 pink borer females and 4 males) to the oviposition jars. • Allow the adults to lay eggs following procedures in Step 3. • Repeat the process to produce a continuous supply of insects for screening and to maintain the culture.

SCREENING METHODS

Screenhouse screening and field screening have been conducted at IITA (Soto and Siddiqi 1976).

Screenhouse screening

Screenhouse methods for screening entries against the borers have been developed at IITA.

Steps	Key points
1. Preparing seed of test varieties	• Obtain and prepare seed of the test entries. Use the following resistant and susceptible checks. — Striped borer Susceptible check: OS6 Resistant check: Taitung 16, SML 81B, or TKM6

Steps	Key points
	— Pink borer Susceptible check: OS6 Resistant check: W1263 — White borer Susceptible check: OS6 Resistant check: ITA 6-4-2 or TKM6
2. Preparing plots in the screenhouse	• Prepare the soil for planting.
3. Transplanting the susceptible variety	• Transplant a susceptible variety 20 days after sowing in a 2-m band around the plots.
4. Transplanting test materials	• Transplant the remaining area of the screenhouse with the test varieties 20 days after planting the susceptible variety. • Plant 25 hills of each variety at 1 seedling/hill in a 4-m row with the susceptible and resistant check varieties planted after every 10th row.
5. Infesting the susceptible border rows	• Infest the susceptible border rows with eggs from the rearing program or from field collections by clipping the egg masses to the leaves 20 days after transplanting (DT) (at the time test materials are being planted). This will serve as the initial source of infestation for the test entries.
6. Evaluating	• Count deadhearts in each variety at 30 and/or 50 DT.
a. Deadhearts	— Calculate percentage of deadhearts for each entry: $\%\ \text{deadhearts} = \frac{\text{no. of deadhearts counted}}{\text{total no. of tillers observed}} \times 100$ — Convert percentage of deadhearts to a new figure (D) which corrects for the level of infestation. $D = \frac{\%\ \text{deadhearts in test entry}}{\%\ \text{deadhearts in susceptible check (av of checks on each side of the test entry)}} \times 100$

Steps	Key points
	— Transform the converted figure to a 0-9 scale.

Scale	*Percent deadhearts*
0	None
1	1-20
3	21-40
5	41-60
7	61-80
9	81-100

b. Whiteheads

- Count the whiteheads before harvest.
  — Compute percentage of whiteheads:

$$\% \text{ whiteheads} = \frac{\text{no. of whiteheads}}{\text{total productive tillers (those with whiteheads and those with normal panicles)}} \times 100$$

- Convert percentage of whiteheads using the formula for deadhearts and use the following scale:

Scale	*Percent whiteheads*
0	None
1	1-10
3	11-25
5	26-40
7	41-60
9	61-100

Field screening

Field screening of rice varieties for resistance to the striped, white, and pink stem borer has been conducted at IITA. The same methods can be used for the three species.

Steps	Key points
1. Planting the test entries	• Plant 20-day-old seedlings of each test entry in 4-m rows at 25 hills/row and 1 seedling/hill. • If enough seeds are available and space permits, replicate each entry three times.
2. Infesting the test entries	• Infestation depends on natural populations of the pest.
3. Evaluating	• Use the evaluation procedure of the screenhouse test.

SOURCES OF RESISTANCE

Sources of resistance to African stem borers have been reported only for screenhouse and field screening conducted at IITA (IITA 1974, 1976; Soto and Siddiqi 1976).

Variety	Origin
African striped stem borer	
TOS2513	Nigeria
Taitung 16	Taiwan, China
PR325	Puerto Rico
PR403	Puerto Rico
ITA6-20-1-Bp1	Nigeria
IR503-1-91-3-2-1	Philippines
Ratna	India
H8	Sri Lanka
SML 81B	Surinam
African white stem borer	
Resistant	
ITA 6-4-2	Nigeria
Moderately resistant	
IR1168-76	Philippines
IR1561-38-6-5	Philippines
ITA 7-7-2	Nigeria
TKM6	India
African pink stem borer	
W1263	India
Taitung 16	Taiwan, China
Sikasso	Niger
SML 81B	Surinam
DNJ146	Bangladesh
DNJ171	Bangladesh

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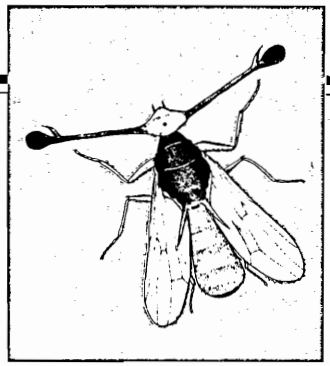


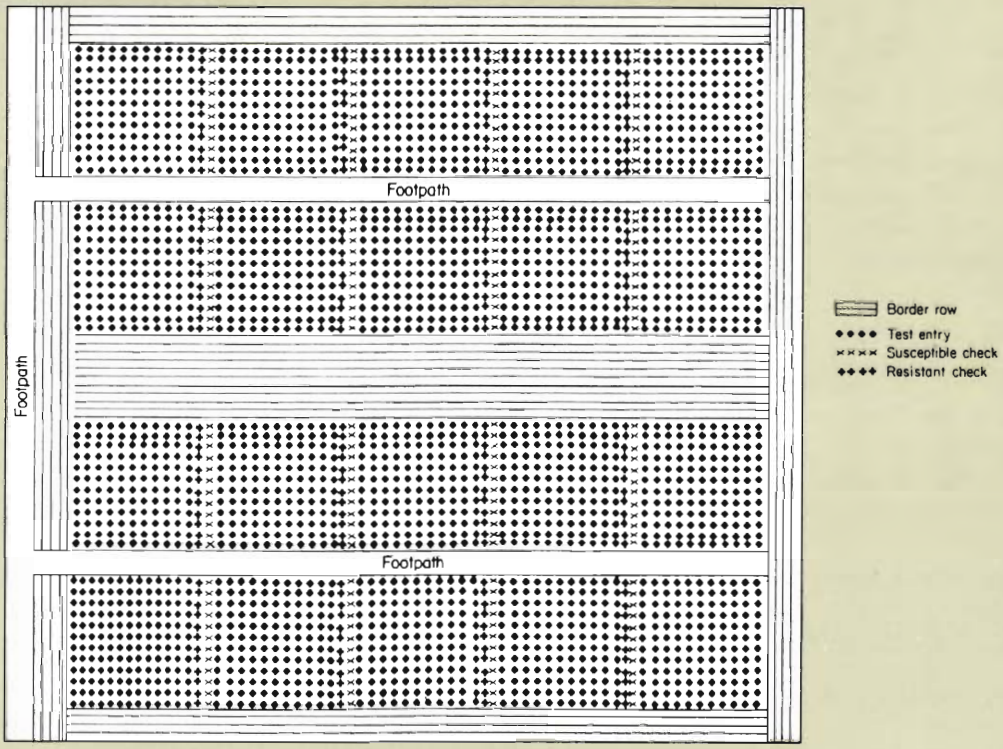
Chapter 16
STALK-EYED FLY

The stalk-eyed fly *Diopsis macrophthalmus* Westw. Dalman is a serious pest in West Africa. The maggots bore within the stem and cause deadhearts. The fly lays eggs on seedlings in the nursery or on newly transplanted seedlings. Most oviposition occurs on plants 20-40 days after transplanting (DT). Practically no eggs are laid on plants that have passed the tillering phase. A single maggot can kill up to 10 tillers (Brenière 1976).

SCREENING METHODS

Rearing methods have not been developed and no studies on mechanisms of resistance have been conducted. Greenhouse and field screening were conducted by Soto and Siddiqi (1976) at the IITA (International Institute of Tropical Agriculture). A screenhouse screening method recently developed by M. S. Alam (pers. comm., 1983) at IITA which has proven efficient is described. With the method, 150 entries replicated twice can be screened at one time within a 60-day period.

Steps	Key points
1. Constructing a screenhouse	• Make a screenhouse 24 × 21 × 2.5 m to accommodate 4 blocks of test entries, each 4.75 m wide, and 3 footpaths, each 0.75 m wide (Fig. 1).
2. Preparing seed	• Obtain seed of the test entries. Use Suakoko 8 for the border rows, TKM6 or OS6 as the susceptible check, and Leuang 28-1-64 or Iguape Cateto as the resistant check.
3. Planning the work schedule	• A typical work schedule from the initial sowing of seeds for the border rows to the planting of the second batch of test entries follows: Day 1. Sow seed of the border row variety. Day 22. Transplant border row seedlings. Day 41. Sow seed of the test entries and checks. Day 42. Release field-collected flies to infest border row plants. Broadcast fertilizer to border rows. Day 62. Transplant seedlings of test entries and checks. Day 67. Apply fertilizer to border rows. Day 74. Sow seed for the second planting of the border row variety. Day 77. Release second batch of field-collected flies to infest border row plants. Day 82. Apply fertilizer to test entries.



1. Layout for screenhouse screening of rice varieties or lines against stalk-eyed fly *Diopsis macrophthalmus*.

Steps	Key points
	Day 92. Make first deadheart count on test entries and checks. Remove first planting of border row plants. Day 95. Transplant second batch of border row seedlings. Day 99. Sow seed of second batch of test entries and checks. Day 107. Apply fertilizer to test entries. Day 112. Take second deadheart count on test entries and checks. Day 115. Apply fertilizer to border rows. Day 120. Transplant second batch of test entries and checks.
4. Sowing seed of the border row variety	• Border all blocks on 3 sides with at least 5 rows (1 m wide) of a susceptible variety (Fig. 1). Infest the border rows initially with field-collected flies to build up the insect population that will later infest the test entries.

Steps	Key points
	• Use Suakoko 8 for the border rows. Stalk-eyed flies are attracted to that variety for oviposition and the development of maggots is rapid — about 1 month.
5. Preparing the plant beds	• Prepare the plant beds a few days before planting the border rows (about 18 days after sowing [DAS] of the border row variety). Before the final harrowing broadcast 30 kg 15-15-15 NPK/ha. Later, make split applications of the same rate to the border rows and the test entries as topdressing 20 and 45 DT.
6. Transplanting border rows	• Transplant seedlings for the border rows 21 DAS. Transplant the border rows 40 days before transplanting the test entries and checks.
7. Sowing seed of test entries	• Sow seed of test entries 19 days after border rows are transplanted. The seedlings will be ready for transplanting at 21 DAS, or 40 DT the border rows.
8. Collecting flies for infesting the border rows	• Collect the flies in rice fields with a sweep net and release them into a 90- × 90- × 120-cm cage. *D. macrophthalmus* can easily be distinguished from other *Diopsis* species through its red abdomen, large body, and absence of black tint at the terminus of the fore wings.
9. Releasing flies on the border row plants	• Release initially 2,000-2,500 flies for the entire screenhouse 20 DT the border rows. It is important to time fly release in relation to crop age because *D. macrophthalmus* lays most of its eggs on plants at the active tillering stage 20-40 DT.
10. Transplanting test entries and checks	• Transplant 21-day-old seedlings of test entries 20 days after the initial release of flies on the border rows (40 DT the border rows). Transplant each entry in a row of 15 hills, 2 seedlings/hill, and at a 25- × 25-cm spacing. Replicate each entry twice. • After every 10 to 15 rows of the test entries, transplant a susceptible (TKM6 or OS6) and a resistant (Leuang 28-1-64 or Iguape Cateto) variety (Fig. 1).
11. Sowing seed for the second batch of border row plants	• At 12 DT the test entries, sow Suakoko 8 seed to produce plants that will replace the first set of border row plants.

Steps	Key points
12. Releasing flies into screenhouse (second release)	• At 15 DT the test entries, make a second and final release of about 1,500-2,500 flies into the screenhouse to ensure sufficient insects. No more releases will be needed after this because the population within the screenhouse will maintain itself. However, make additional releases if the population should decrease over time to a level that is insufficient for valid screening.
13. Counting the deadhearts (first count)	• At 30 DT the test entries, count the deadhearts on the test entries and the checks. — Compute the percentage of deadhearts for each test entry by the following formula: $\%\ \text{deadhearts} = \frac{\text{no. of deadhearts observed}}{\text{total no. of tillers observed}} \times 100$ — Convert the percentage of deadhearts to a 0-5 rating scale. *Scale* — *Percent deadhearts* 0 — None (highly resistant) 1 — 1-10 (resistant) 3 — 11-20 (moderately resistant) 5 — > 20 (susceptible)
14. Removing border row plants	• As soon as deadhearts have been counted, remove the border row plants. Plants at this time are 70 DT and are too old to serve as an insect infestation source.
15. Transplanting second set of border row seedlings	• Three days after removing the old border row plants, transplant 21-day-old seedlings of Suakoko 8 or IR5 to replace the first set of border row plants. Remove border row plants at 70 DT and replace them with fresh 21-day-old seedlings on a continual basis throughout the year. Adults emerging from the border rows will infest the test entries. Later, flies emerging from the test entries will infest the border rows, etc. Thus, flies will no longer have to be released after the second infestation.
16. Sowing seed of test entries for next test	• Five DT the second set of border row seedlings, sow seed of the second batch of test entries and the checks. The seedlings will be ready for transplanting in 21 days.

Steps	Key points
17. Counting deadhearts (second count)	• At 50 DT the test entries, make the second deadheart count. Compute the percentage of deadhearts as in the first deadheart count.
18. Transplanting test entries for second test	• After the second deadheart count, remove the plants and prepare the soil in the beds for planting the second batch of test entries. The seedlings of this second batch will be 21 days old 6 days after the second deadheart count and will be ready for transplanting.
19. Repeating steps 1-18	• Follow steps 1-18 throughout the year. Based on the schedule described, 1 batch of test entries can be screened every 60 days. Thus, in 1 year, 6 batches of 150 test entries (with 2 replications/entry) or 900 entries can be screened.

SOURCES OF RESISTANCE

Based on greenhouse and field screening at IITA, the following breeding lines and varieties have been reported as resistant or moderately resistant to the stalk-eyed fly *Diopsis macrophthalmus*.

Variety/line	Origin	Reference
	Resistant	
C5565	Philippines	IITA (1978)
Ctg 680	Bangladesh	IITA (1976), Soto and Siddiqi (1976)
DNJ171	Bangladesh	IITA (1976), Soto and Siddiqi (1976)
E. L. Golpher	USA	IITA (1976), Soto and Siddiqi (1976)
Huang Sengoo	China	IITA (1978)
Iguape Cateto	Brazil	IITA (1974), Soto and Siddiqi (1976)
IR523-1-218	Philippines	IITA (1974), Soto and Siddiqi (1976)
IR579-160-2	Philippines	IITA (1974), Soto and Siddiqi (1976)
IR589-53-2	Philippines	IITA (1976), Soto and Siddiqi (1976)
IR1561-38-6-5	Philippines	IITA (1976), Soto and Siddiqi (1976)
ITA6-16-7-Bp3	Nigeria	IITA (1976), Soto and Siddiqi (1976)
ITA6-22-2-Bp1	Nigeria	IITA (1976), Soto and Siddiqi (1976)
Leuang 28-1-64	Thailand	IITA (1976), Soto and Siddiqi (1976)
Magoti	East Africa	IITA (1978)
TD10A	Thailand	IITA (1978)
Tx 52-2-4	Nigeria	IITA (1974), Soto and Siddiqi (1976)
	Moderately resistant	
DL-5	Bangladesh	IITA (1976)
IR878-B2-83-3-3	Philippines	IITA (1976)
IR878-B2-143-2-2	Philippines	IITA (1976)
IR1749	Philippines	IITA (1976)
Milpal 18	Philippines	IITA (1976)
Pankaj	India	IITA (1976)
Sacondo Brasil TM 1377	El Salvador	IITA (1978)

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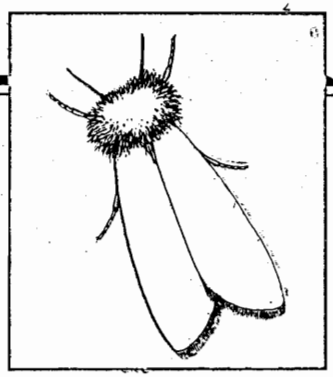

Chapter 17
SOUTH AMERICAN WHITE STEM BORER

The South American white stem borer *Rupela albinella* is a pest in Central and South America where it occurs only on rice. The larvae feed in the leaf sheath and tunnel into the nodes. Feeding damage is seen as yellow spots below the axis of leaf sheaths and as holes in the stem. Screening for resistance to the pest has been limited and methods of rearing and studies on mechanisms of resistance have not been published.

FIELD SCREENING

The field screening method described is that used in Panama (Navas 1965). Methods as described for the striped borer *Chilo suppressalis* and the yellow stem borer *Scirpophaga incertulas* can also be used.

Steps	Key points
1. Preparing seed	• Prepare seed of test entries.
2. Sowing seed	• Sow seed in rows spaced 25 cm in plots 1.25 × 8 m. Have four replications per entry with five rows per replication.
3. Infesting the plants	• Insect infestation depends on natural field populations.
4. Evaluating	• Pull plants from 8 points per plot at harvest. • Examine 150 tillers and record the number of damaged and undamaged tillers. Damaged tillers have holes in the stem caused by the boring of the larvae. • Compute the percentage of infested tillers and compare among entries: $\%\ \text{infested tillers} = \frac{\text{no. of damaged tillers counted}}{\text{total no. of tillers observed}} \times 100$

SOURCES OF RESISTANCE

Six sources of resistance to *Rupela albinella* have been identified in field screening in Guyana and Panama.

Variety	Reference
Camponi	Rambajan 1979
Chin chin	Navas 1965
"N"	Rambajan 1979
Nilo 1	Navas 1965
Nilo 2	Navas 1965
698-71	Rambajan 1979

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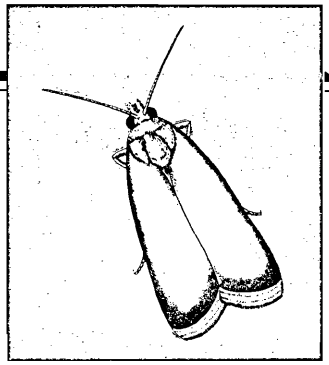
Rambajan, I. 1979. Screening for resistance to *Rupela albinella* in Guyana, South America. Int. Rice Res. Newsl. 4(5):11.

Navas, D. 1965. Susceptibilidad de 12 variedades de arroz al ataque del Barrenador del Tallo, *Rupela albinella* (Cramer). Pages 134-137 *in* Programa Cooperativo Centroamericano para el Mejoramento de Cultivos Alimenticos, Panama, 16-19 Marzo.



Chapter 18
LESSER CORN STALK BORER

The lesser corn stalk borer *Elasmopalpus lignosellus* is a pest of dryland rice in Brazil. The borer feeds within the stem and produces deadheart symptoms. Infestations are most severe during drought. Work on varietal resistance to this pest has been limited to field screening studies. Some sources of resistance have been identified. The mechanism of resistance is believed to be tolerance as related to the tillering capacity of the varieties.

FIELD SCREENING

Field screening has been done in southern Brazil (Ferreira et al 1976). No greenhouse methods have been developed.

Steps	Key points
1. Preparing seed of test materials	• Obtain seed of the test materials. Coletao can be used as the susceptible check variety.
2. Scheduling planting	• Plant toward the end of the wet season when there is still sufficient moisture to provide a good plant stand, followed by some moisture stress to favor development of the borer population.
3. Sowing seed	• Sow seed of each variety at 100 seeds/m² (about 1,000 seeds/variety) in 4 rows 5 m long, spaced at 0.5 m. • Sow four replications in a randomized complete block design. • Apply fertilizer 35 days after sowing (DAS), as needed.
4. Infesting the plants	• Natural infestation provides a source of insects.
5. Evaluating	• At 10 DAS mark 100 plants in the 2 center rows of each variety. • At 25, 50, and 75 DAS count the total number of plants with damaged tillers in the marked area. Count the total number of tillers and number of damaged tillers in the damaged plants (hills). Remove the damaged plants after each counting but add the previous counts of damaged plants to the next countings.

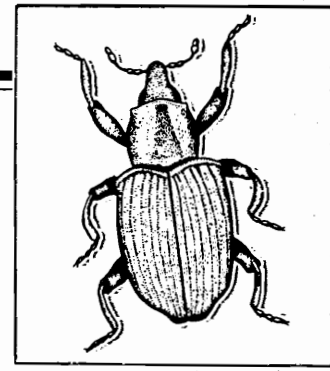
Steps	Key points
	• Compute the percentage of damaged plants for each counting using the following formula: $\%\ \text{incidence} = \frac{ab}{cd} \times 100$ where a = no. of damaged plants (hills) from d, b = total no. of damaged tillers in a, c = total no. of tillers in a, and d = total no. of plants (hills) marked.
6. Grading damage	• No rating system has been developed to classify the varieties into different categories of resistance, but varieties with the lowest percentage of plants killed are considered most resistant.

SOURCES OF RESISTANCE

Four sources of resistance to the lesser corn stalk borer are known (Ferreira et al 1976): BKN6652-249-5-1, CICA-4, IAC25, and IAC47.

REFERENCE CITED

Ferreira, E., J. F. S. Martins, and F. J. P. Zimmerman. 1976. Resistencia de cultivares e linhagens de arroz a broca do colo, *Elasmopalpus lignosellus.* (mimeo.)



Chapter 19
RICE WATER WEEVIL

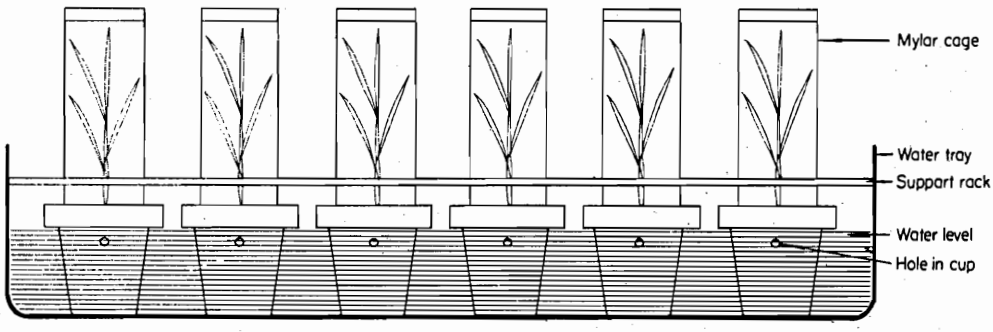
The rice water weevil *Lissorhoptrus oryzophilus* is a rice pest in the southern United States — Arkansas, California, Louisiana, and Texas. Adults feeding on the leaves make longitudinal slit-like scars, but leaf feeding damage is of minor importance. Serious damage is caused by the larvae that prune the roots, thus causing stunting of young plants and lodging and yield reduction in mature plants. Methods for screening and studying the mechanisms of resistance have been developed and sources of resistance have been identified.

SCREENING METHODS

Laboratory or greenhouse screening

The method used by Bowling (1973) is described.

Steps	Key points
1. Preparing materials and equipment	• Prepare clay pots or plastic drinking cups for growing the plants (Fig. 1). • Use water trays to hold the clay pots or cups. • Prepare test tubes and test tube racks. • The number of pots or cups and test tubes depends on the amount of materials to be screened and the number of replications.
2. Preparing seed of test entries	• Obtain seed of the test materials. Include a susceptible (Saturn) and a resistant (Bontoc, Finindoc, Nira, or Dawn) check in each replication.



1. Apparatus used for greenhouse screening of rice for resistance to the rice water weevil. The mylar cage which is embedded into the soil in the cup is held by the support rack.

Steps	Key points
3. Scheduling sowing	• Sow when there are actively ovipositing females in the field which can be collected and used for infestation.
4. Sowing seed and maintaining seedlings	• Fill 7-cm-diam clay pots or plastic cups with dry fertile soil. • Sow 2-4 seeds in a pot or cup. Have at least 10 replications per entry, 1 cup representing 1 replication. • Moisten the soil with water. • Transfer the seeded pots or cups to a germinator at 30° C. If a germinator is not available, keep the pots or cups in a water tray in the greenhouse. • As the seeds germinate (about 3 days), move the pots or cups from the germinator to a growth chamber timed for a photoperiod of 14 h light-10 h dark. • Program the temperature to vary from 15° C minimum at night to 33° C maximum in the daytime. • If a growth chamber is not available keep pots or cups in the water tray in the greenhouse. • One week after sowing (WAS), thin the seedlings to one per pot or cup.
5. Flooding the tray	• Two WAS, flood the tray to just above the top of the pots. If a plastic cup is used, make a hole through the cup below the water level but above the soil level to allow water to enter (Fig. 1).
6. Caging the plants	• Cage each plant with a mylar film cage. Support the cage with a wooden rack (Fig. 1).
7. Infesting the plants	• Collect adults in the field with sweep nets. • Infest the plants by introducing one or two pairs (male and female) of adults into each cage for oviposition.
8. Removing the plants from the soil	• After 1 week of exposure to the adults, remove the plants from the pots or cups.
9. Clipping leaves and roots	• Clip the leaves to 20 cm long and the roots to 2 cm long.
10. Cleaning the roots	• Wash off the soil attached to the roots.
11. Transferring plants to test tubes	• Transfer the clean plants to 20- × 150-mm test tubes containing 20 ml water. Arrange them in a

Steps	Key points
	test tube rack and put the rack in the growth chamber or maintain in a laboratory room.
12. Evaluating	• Base resistance on the number of larvae counted per plant. • First-instar larvae will emerge from the leaf sheath, drift downward, and settle at the bottom of the tubes. • Start counting the larvae 1 week after placing the plants in the test tubes. Place the test tube over a test tube agglutination viewer and count the larvae in each tube. • Some considerations: — If some plants are suspected to be resistant (low larval counts), keep the plants in the tubes longer and make additional countings. — This procedure can be modified, eg, varying temperature, photoperiod, age of plants, and other conditions, to suit available facilities.

Advantages and disadvantages. Greenhouse or laboratory screening for rice water weevil resistance has several advantages:

- It provides for a definite detection of plant resistance to rice water weevil.
- It requires a minimum number of seeds, amount of space, and time.
- Screening studies can be conducted continuously if ovipositing females are available.
- It is possible to test individual progeny of backcrosses between resistant and susceptible varieties.

A disadvantage is that the procedure is limited by the availability of actively ovipositing females for infestation if the insect is not being reared.

Field screening

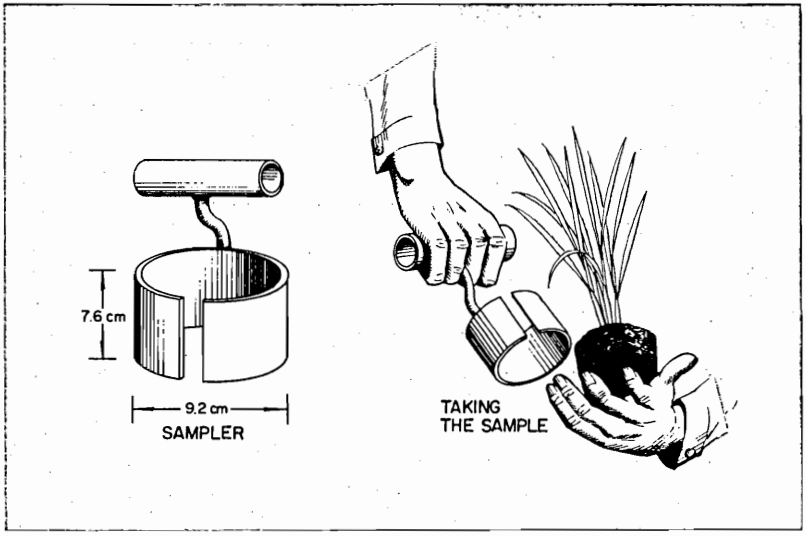
A field screening method used by Smith and Robinson (1982) in Louisiana, USA, is described.

Steps	Key points
1. Scheduling sowing	• Sow when rice water weevil population in the field is high.
2. Preparing test materials	• Obtain and prepare seed for testing.
3. Preparing the field layout	• Use nonreplicated trials for mass screening but for retesting or for advanced variety trials, have four to five replications in a randomized complete block design.

Steps	Key points
	• Plant a resistant and a susceptible check after each set of 25 test entries.
4. Sowing the test materials	• Sow entries in single rows 2 m long with 50 cm spacing between the rows. Sow about 2.5 g (or more depending on the germination rate) per single row. • Thin plants to 1 plant/15-cm distance within the rows 1 WAS.
5. Fertilizing the field	• Before flooding, which is usually done about 7 days after sowing (DAS), apply fertilizer and herbicide at recommended rates.
6. Flooding the field	• Flood the field 7 DAS for the first flooding and about 21-28 DAS for the permanent flooding.
7. Evaluating by larval or pupal counts	• Make larval or pupal counts 21-30 days after permanent flooding. • Sample individual plants using a core sampler (9.2-cm-diam and 7.6-cm-deep) in removing the entire plant, including the roots and soil (Fig. 2). • Take 3 samples/entry per replication. • Collect larvae and pupae from the soil-root core samples by washing with water over a 30-40 mesh sieve. • Allow the larvae and pupae collected on the sieve to float in a saturated salt solution and count them. • Entries with 10 or more larvae or pupae/plant (sample) are considered susceptible. In samples where larvae and pupae are less than 10 per plant, take second and third samples.
8. Evaluating for adult feeding	• Evaluate adult feeding ratings by grading plants about 10-15 and/or 40-50 days after flooding, using a 1-5 scale: 1 = no feeding, 2 = 1-3 leaf-feeding scars over the entire surface of all leaves, 3 = 4-5 feeding scars on leaf tips, 4 = 6-10 feeding scars over entire leaf, and 5 = more than 10 feeding scars over entire leaf surface.

MECHANISMS OF RESISTANCE

Laboratory or greenhouse and field methods for determining the levels of antixenosis, antibiosis, and tolerance of selected entries include the following.



2. A core sampler for sampling individual plants in the field screening for rice water weevil resistance. Larvae are removed from the soil-root core samples and counted.

Antixenosis for adult feeding (greenhouse or laboratory)

Steps	Key points
1. Preparing test materials	• Select varieties with different levels of resistance based on results of the screening tests. • Obtain and prepare seed for testing. • Include a susceptible and a resistant check.
2. Sowing seed and maintaining seedlings	• Fill pots or plastic cups with fertile soil. Place them in a water tray. • Sow 2-4 seeds/pot or cup. • Moisten the soil with water. • Replicate at least five times. One pot or cup represents one replication. • One week after sowing thin seedlings to one per pot or cup.
3. Flooding the plants	• Two weeks after sowing, flood the water tray to just below the rim of the pots or cups. If a plastic cup is used, make a hole in the cup below the water level but above the soil level to allow water to enter.
4. Caging the plants	• Cage all of the cups of each replication with a fine nylon, fiberglass, wire mesh, or mylar film cage with a nylon mesh top for ventilation.
5. Infesting the plants	• Introduce adult water weevils into each cage at a rate of 4 adults/plant (pot or cup).

Steps	Key points
6. Evaluating	• Determine adult feeding ratings at 8-10 days after infestation (DI) using the 1-5 scale in *Field screening*.

Antixenosis for adult feeding (field)

Steps	Key points
1. Scheduling sowing and infestation	• Select varieties on the basis of previous screening tests. Sow when rice water weevil population is high.
2. Preparing the layout	• Prepare the field layout using a randomized complete block design. Include a susceptible and a resistant check in each replication.
3. Sowing the test materials	• Sow each variety in 2 0.7-m rows with 30 cm spacing between rows and 15 cm spacing between plants in a row.
4. Preparing cages	• Cover each replication with a cage to prevent escape of weevils to be used for infestation and to prevent entrance of unwanted weevils and other pests. Cages can be made of fine mesh fiberglass or wire screen. • The number of cages required depends on the number of replications.
5. Caging the plants	• Place cages over the plants 2-3 WAS or just before flooding.
6. Flooding the field	• Flood the field 3-4 WAS.
7. Infesting the plants	• As soon as the field is flooded, introduce rice water weevil adults into the cages at the rate of 4 adults/plant.
8. Evaluating	• Determine adult feeding ratings at 8-10 DI using the 1-5 scale described under *Field screening*. Compare results among varieties.

Antixenosis for oviposition (greenhouse or laboratory)

Steps	Key points
1. Steps 1-5 of	• Use a similar experimental setup as in *Antixenosis*

Steps	Key points
Antixenosis for adult feeding (greenhouse or laboratory)	*for adult feeding (greenhouse or laboratory)* and follow the same steps.
2. Cleaning the plants	• About 6 days after initial exposure to the adults, remove the plants from the cups and wash the soil from the roots.
3. Clipping the leaves and the roots	• Clip the leaves to about 15-20 cm long and the roots to 2 cm long.
4. Blanching and bleaching the plants (Gifford and Trahan 1969)	• Blanch the plants in hot water (60-70° C) for 5 min. • Transfer the blanched plants to heated alcohol (70%) and leave them there for 1 day. • Transfer the plants again to fresh alcohol for further bleaching and preservation. This treatment removes the chlorophyll from the plant but leaves the eggs stained green. • Soak plants in a cold solution of 1 part phenol, lactic acid, and distilled water plus 2 parts glycerine (add acid fuchsin until the solution is deep violet) for 24 h. Wash soaked plants in tap water to remove excess stain, clear in 3% KOH for 24 h, and rinse in tap water.
5. Counting the eggs	• Remove leaves and leaf sheaths one at a time and examine them under a dissecting microscope. Eggs will be visible through the plant tissues as dark red objects. • Remove eggs from the leaf tissue as they are counted. • Examine the proximal 2 cm of the root system for eggs. • Compare the number of eggs laid per variety (per plant basis).

Antibiosis on larval survival (laboratory)

Steps	Key points
1. Scheduling sowing and infestation	• Select entries to be tested and obtain seed. Include a susceptible and a resistant check. • Sow seed when insects for infestation are available.
2. Sowing the test materials	• Double the number of pots or cups to be sown. One-half will be infested with adults and the other half with larvae.

Steps	Key points
	• Fill the pots or cups with dry fertile soil. • Sow 2-4 seeds/cup or pot. • Replicate each entry at least five times.
3. Infesting with adults	• With one-half of the plants, follow steps 4-12 in *Laboratory or greenhouse screening.* • When the first-instar larvae emerge from the leaf sheath, they settle at the bottom of the test tubes. Count the larvae from each plant as described in *Laboratory or greenhouse screening*.
4. Infesting with larvae	• Flood the remaining uninfested plants from Step 2 above. • Cage the flooded plants and infest them with the counted larvae coming from the corresponding varieties in Step 3. For instance, larvae coming from one plant of a variety should be used to infest another plant of the same variety. • Record the number of larvae used for infesting on each plant.
5. Maintaining the infested plants from Step 4	• Keep the plants in a growth chamber or in a water tray in the greenhouse.
6. Evaluating	• Count the surviving larvae from each plant at 15 or 20 DI, using the procedure for counting the first-instar larvae. • Compute survival percentage of larvae: $\%\ \text{survival} = \frac{\text{no. of surviving larvae}}{\text{no. of first-instar larvae used to infest plants}} \times 100$

Antibiosis on larval size and weight (field test)

Steps	Key points
1. Scheduling sowing of test materials	• Select entries to be tested and obtain seed for sowing. Include a susceptible and a resistant check. • Sow seeds when weevil populations in the field are high.
2. Preparing the field layout	• Lay out the field in a randomized complete block design.

Steps	Key points
3. Sowing the test materials	• Sow each entry in two 0.7-m rows with a 30-cm spacing between rows and 15 cm between plants within a row. Have 5 replications/entry.
4. Flooding the field	• Flood the field 3-4 WAS.
5. Evaluating	• At 20-25 days after flooding, determine the number, size, and stage of larvae found on each plant of each entry. • Sample individual plants using a 9.2-cm-diam by 7.6-cm-deep core sampler to remove the entire plant and the soil around the roots (Fig. 2). Take at least 5 samples/entry per replication. • Remove the larvae and pupae from the soil and roots by washing with water over a 30-40 mesh sieve. • Allow the larvae and pupae collected on the sieve to float in a saturated salt solution. Separate larvae from pupae. • Classify the larvae into small (<3 mm), medium (3-6 mm), and large (>6 mm) and count the larvae under each classification on a per plant basis for each entry (Smith and Robinson 1982). • Obtain the weights of the larvae under each classification and determine percentage of each classification in each replication. $\%\ \text{small larvae} = \frac{\text{no. of small larvae}}{\text{total larvae counted}} \times 100$ $\%\ \text{medium larvae} = \frac{\text{no. of medium larvae}}{\text{total larvae counted}} \times 100$ $\%\ \text{large larvae} = \frac{\text{no. of large larvae}}{\text{total larvae counted}} \times 100$ • If pupae are already present at time of sampling compute % pupae: $\%\ \text{pupae} = \frac{\text{no. of pupae}}{\text{total larvae and pupae counted}} \times 100$ • Entries with a high percentage of small larvae are considered more resistant than those with mostly large larvae. • Some considerations: — The above procedure depends on natural infestation. Where populations are low, artificial infestation with adults is necessary.

Steps	Key points
	— Instead of conducting the study in the field, a screenhouse can be used and plants artificially infested with adults.

Antibiosis on population growth (laboratory)

Steps	Key points
1. Scheduling sowing and infestation	• Select varieties to be tested and obtain seed for sowing. • Sow seed when adult weevils for infestation are available.
2. Sowing	• Sow 2-4 seeds in clay pots or plastic cups (10-cm-diam) filled with dry fertile soil. Arrange the pots or cups in a randomized complete block design with at least five replications. • Moisten the soil in the pots or cups with water.
3. Maintaining the plants	• Follow Step 4 of *Laboratory or greenhouse screening.* Thinning of plants is not necessary.
4. Preparing cages	• While waiting for the plants to reach the correct age for infestation, prepare cylindrical mylar film cages (9-cm-diam and 90-cm-high). The cage diameter should fit the inside rim of the pots or cups in which the plants are grown. Cover the top of the cages with fine nylon mesh for ventilation. • The number of cages to be prepared depends on the number of test varieties and number of replications.
5. Flooding the plants	• Flood the plants 2 WAS. Maintain the water at just above the soil level in the pots or cups throughout the experiment.
6. Caging the plants	• Cage the plants (per pot or cup) with the mylar cages in Step 4. Support the cages with a rack.
7. Infesting the plants	• Introduce 2 pairs (male and female) of adult weevils into each cage.
8. Evaluating	• Count the adults that emerge in each cage 40-50 DI. • Look for immature stages of the insect. Pull out the plants and individually wash the soil off the roots over a 30-40 mesh sieve. Allow the larvae or

Steps	Key points
	pupae collected on the sieve to float in a saturated salt solution. Count the immature stages and record on a per cage basis. • Compare the number of adults and immature stages among varieties.

Antibiosis on population growth (field test)

Steps	Key points
1. Scheduling sowing and infestation	• Select varieties to be tested and obtain seed for the test. Include a susceptible and a resistant check. Sow when the rice water weevil population is high.
2. Preparing the field layout	• Prepare the layout in a randomized complete block design with at least five replications.
3. Sowing the test materials	• Sow each entry in two 0.7-m rows in the field with 30-cm spacing between rows and 15 cm between plants within a row. Leave 45 cm between rows of each entry.
4. Preparing cages	• Prepare about 1.3-m-high cages long and wide enough to cover each entry. • Make cages of fine mesh wire or fiberglass screen to prevent escape of the weevils for infestation and prevent entrance of weevils or other insects occurring naturally in the field. • Compute the number of cages required: no. of test entries × no. of replications
5. Caging the test plants	• Cage each replication of every entry 2-3 WAS .
6. Flooding the field	• Flood the field 3-4 WAS.
7. Infesting the plants	• As soon as the field is flooded, introduce adults of the water weevil at the rate of 2 pairs (male and female)/plant.
8. Evaluating	• Count the number of emerging adults in each cage 40-50 DI and compare results among the entries.

Tolerance (field test)

The methods of Gifford and Trahan (1975) are described.

Steps	Key points
1. Scheduling sowing and preparing seed	• Sowing should coincide with an abundance of water weevil adults in the field. • Select the varieties and obtain seed.
2. Preparing the field layout	• Prepare the layout in a randomized complete block design with at least five replications.
3. Sowing the test materials	• Sow each variety in two 1-2 m rows in the field with 30-cm spacing between rows within a replication and 1 m between replications. Thin seedlings within a row to maintain a 15-cm spacing between seedlings.
4. Flooding the field	• Flood the field 3-4 WAS.
5. Sampling for larvae	• At 25 days after flooding sample 10 individual plants selected at random from each variety (5/row) by taking 9.2-cm-diam core samples. • Remove the larvae from the soil and roots of each plant by washing with water over a 30-40 mesh sieve. • Allow the larvae to float in a saturated salt solution and count them on a per plant basis.
6. Evaluating for root damage	• After removing the larvae from each core sample, evaluate the roots for weevil damage using a rating scale of 0-6. *Scale* — *Damage* 0 — No root pruning, no root discoloration 1 — About 1/3 of root system pruned, roots regrowing and white 2 — About 1/3 of root system pruned, little root regrowth, roots gray 3 — About 2/3 of root system pruned, roots regrowing and white 4 — About 2/3 of root system pruned, blackened, and no regrowth 5 — Entire root system pruned, some regrowth of new white roots 6 — Entire root system pruned, roots black and no regrowth • Classify roots that had been heavily pruned but had regrowth into Class 1 and those that had been

Steps	Key points
	heavily pruned with little or no indication of regrowth into Class 2. • Cut off the roots of each sampled plant and determine the volume of roots per plant by measuring the water displaced by the roots. Place the excised roots in a 1,000-ml graduated cylinder or beaker Then pour 500 ml water into the cylinder or beaker and record the volume of water displaced. • Next place the roots in bags for drying in an oven at 50° C for 72 h or in the sun. Weigh the roots and record the weight.
7. Comparing varieties	• Compare the results among varieties based on the larval population, root damage ratings, root volume, and root weight. Varieties that support large larval populations but have low root damage ratings and high root volumes and root weight are considered tolerant.

SOURCES OF RESISTANCE

Sources of resistance to the rice water weevil based on screening in the USA are as follows:

Variety, line	Origin	Reference
	Low resistance	
Bontoc	Philippines	Smith and Robinson (1982)
Carangiang	Philippines	Smith and Robinson (1982)
Dawn	USA	Smith and Robinson (1982)
Finindoc	Philippines	Smith and Robinson (1982)
Nira	USA	Smith and Robinson (1982)
	Moderate resistance	
Ae Guk Ai Koku	South Korea	Gifford and Trahan (1975)
Iljin	South Korea	Robinson et al (1981)
IR269-1-1-3	Philippines	Robinson et al (1981)
IR404-1-3-1-1	Philippines	Robinson et al (1981)
IR404-3-2-7	Philippines	Robinson et al (1981)
IR404-6-3-10-1	Philippines	Robinson et al (1981)
IR455-5-5-1-2	Philippines	Robinson et al (1981)
Mit Dari	South Korea	Gifford and Trahan (1975)
Toyokuni	Japan	Gifford and Trahan (1975)

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Chapter 20
ARMYWORMS

Several Noctuidae species of the genera *Mythimna* and *Spodoptera* feed on rice as well as on other grasses. The larvae cut rice stems off at ground level, feed on the leaves, and cut off panicles. Feeding occurs at night and thus young larvae often go undetected. In the later stages the larvae are voracious feeders and plants may virtually be stripped of foliage and panicles overnight. Swarms of larvae move from destroyed fields to adjacent fields like a marching army, thus the term *armyworm.*

A rearing and greenhouse screening technique was developed by Wilde and Apostol (1983) for the ear cutting caterpillar *Mythimna separata* and the swarming caterpillar *Spodoptera mauritia.* Methods for field screening and determining mechanisms of resistance have not been developed. No sources with high levels of resistance have been identified although minor differences among IR varieties were observed in the greenhouse screening of seedlings by Wilde and Apostol (1983).

M. separata

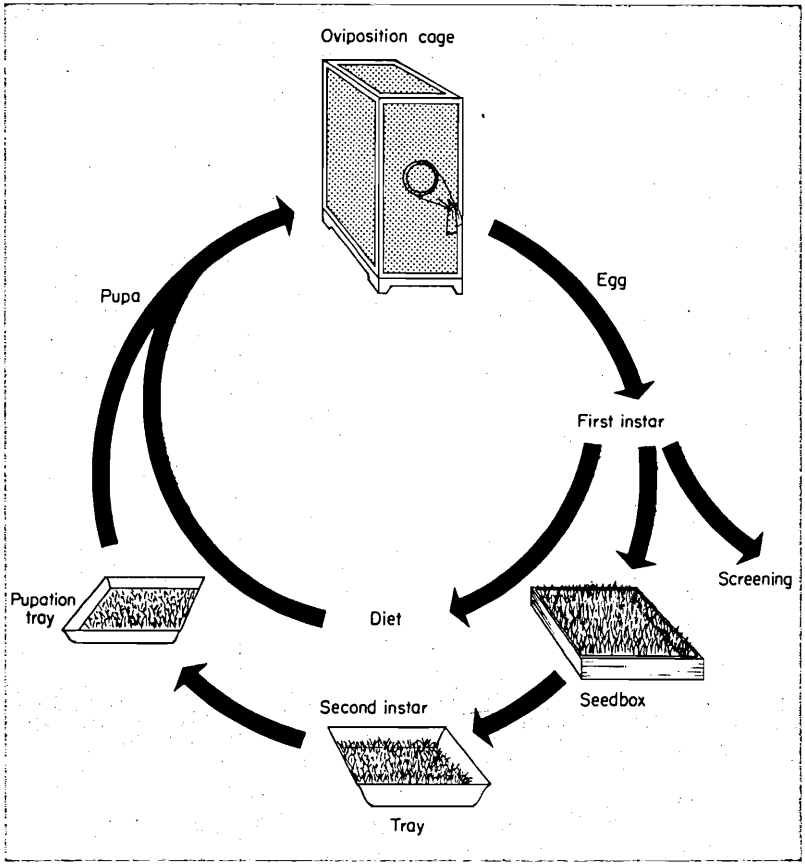
S. mauritia

REARING

The rearing procedure is illustrated in Figure 1.

Steps	Key points
1. Constructing cages	• Cages are needed to enclose adults and obtain eggs. Make cages about 50 cm long, 50 cm wide, and 90 cm high. Cage size can vary. Cover the cages with wire mesh small enough to enclose the moths.
2. Preparing seedboxes	• Seedboxes are used in both rearing and screening, although clay pots can be substituted. • Size of seedboxes varies, but boxes 69 × 114 × 7 cm are common.
3. Preparing food plants for larvae	• Maintain a continuous supply of 7- to 10-day-old seedlings of a susceptible variety such as IR20 as food plants for 1st-instar larvae. • Plant seed thickly in the seedbox to obtain growth like that in a seedbed. • Maintain 30- to 60-day-old plants of rice or various grasses for food for 2d- to 5th-instar larvae.
4. Starting a culture	• Collect armyworms in the field or obtain adults from light traps or other sources.

Steps	Key points
5. Rearing the insects	• Cage adults and place cotton wads soaked in 25% honey solution for the adults to feed on. Hang crushed paper bags from the top of the cage. The adults will lay eggs in the crevices or folds of the bags. • Place eggs in a container. When they hatch, distribute the larvae on 7- to 10-day-old seedlings. • After 3-5 days, transfer the larvae to mesh-covered plastic trays containing cut leaves of rice or other grasses for food. Tray size can vary. A tray 30 × 36 × 10 cm will accommodate 50-100 5th-instar larvae. • Change food every other day. • When larvae reach the 5th instar, add 0.5 cm of fine soil to the tray bottom as a pupation site. • When changing food, examine the soil for pupae and transfer those found to a dish.



1. Procedure for rearing armyworms.

Steps	Key points
	• Place the pupal tray in the cage for adult emergence.

SCREENING METHODS

Screening can start when a supply of larvae is assured.

Greenhouse screening

Steps	Key points
1. Sowing test materials	• Sow seed in a seedbox with 1 cm between rows, or in 3.5-cm² compartments. Plant check varieties in border rows.
2. Infesting test entries	• At 7-10 days after sowing, infest plants with newly hatched 1st-instar larvae by mixing larvae with ground maize cob grits and applying with the Davis inoculator. If an inoculator is not available, distribute larvae by hand. About 1 larva/2 seedlings is desired.
3. Maintaining infested plants	• Keep infested plants in a greenhouse and cover with cages to prevent predation.
4. Evaluating	• Three to five days after infesting, check plants daily for damage. When the susceptible check has been consumed, grade all entries using the following scale:

Scale	*Damage (% defoliation)*
0	None
1	1-20
3	21-40
5	41-60
7	61-80
9	81-100

Field screening

No field tests have been made in screening for armyworm resistance, but boot-stage plants were easily infested in the greenhouse by placing a cone made of filter paper among the tillers (Fig. 2). Plants were infested by placing 25 1st-instar larvae/plant into the cone by grits inoculation or placing 10 3d-instar larvae in the same cones by hand. Damage ratings for % defoliation are made 7-14 days later.

2. Cone made of filter paper used for inoculating plants with armyworm larvae.

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Wilde, G., and R. Apostol. 1983. Armyworm (Lepidoptera:Noctuidae) resistance in rice. Environ. Entomol. 12:376-379.

Chapter 21
HISPA

The rice hispa *Dicladispa armigera* is frequently a severe pest in Bangladesh, India, Nepal, and Pakistan. Hispa larvae and adults feed on rice leaves. Studies on varietal resistance have been limited to field screening. No rearing methods have been developed and there has been no study on mechanisms of resistance.

SCREENING METHODS

Field screening methods described here are a modification of those used in India (Dhaliwal 1980).

Steps	Key points
1. Preparing seed	• Prepare seed of test entries.
2. Planting test entries	• Transplant 20-day-old seedlings in two 2.5-m-long rows per entry at a spacing of 20 × 15 cm. Replicate each entry four times.
3. Obtaining insects	• Insects are provided through natural infestations.
4. Evaluating	• Count hispa-damaged and undamaged leaves on 10 hills randomly selected from each variety per replication 40 days after transplanting. • Compute percentage of damaged leaves of each variety using the following formula: $\%\ \text{damaged leaves} = \frac{\text{no. of damaged leaves}}{\text{total no. of leaves}} \times 100$ • Convert percentage of damaged leaves to a new figure based on the following formula: $\frac{\%\ \text{damaged leaves in test entry}}{\%\ \text{damaged leaves in susceptible check}} \times 100$ • Transform the converted figures to a 0-9 scale:

Scale	*Damaged leaves (%)*
0	No damage
1	1-10
3	11-20
5	21-35
7	36-50
9	51-100

SOURCES OF RESISTANCE

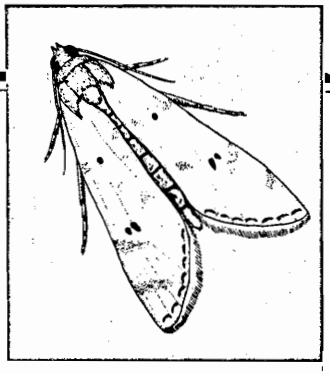
A few varieties have been identified as sources of resistance to hispa.

Variety	Reference
Asphata	Prakasa Rao et al (1971)
IET4109	Dhaliwal (1980)
IET6251	Dhaliwal (1980)
Monkompu 1	Prakasa Rao et al (1971)
Patnai 23	Prakasa Rao et al (1971)
PR285	Dhaliwal (1980)
PR299A	Dhaliwal (1980)
PR385	Dhaliwal (1980)
PR409	Dhaliwal (1980)
PR440	Dhaliwal (1980)
PR506	Dhaliwal (1980)
PR515	Dhaliwal (1980)
PR520	Dhaliwal (1980)
T23	Prakasa Rao et al (1971)

REFERENCES CITED

Dhaliwal, G. S. 1980. Field reaction of rice cultivars to hispa and leaf folder. Int. Rice Res. Newsl. 5(1):7.

Prakasa Rao, P. S., P. Israel, and Y. S. Rao. 1971. Epidemiology and control of the rice hispa, *Dicladispa armigera* Olivier. Oryza 8(2 suppl.):345-359.



Chapter 22
CASEWORM

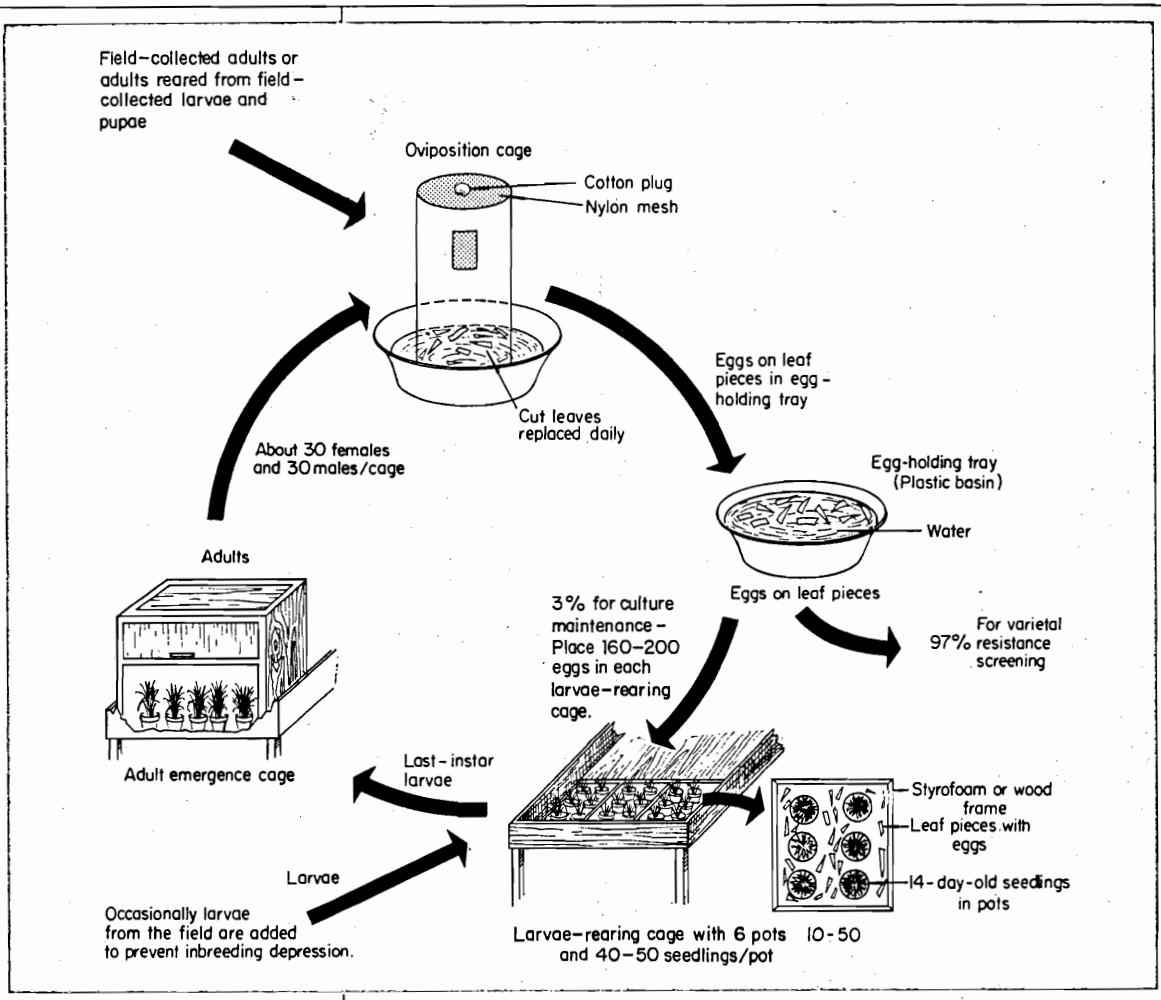
Because of the aquatic habits of its larva, the rice caseworm *Nymphula depunctalis* is a pest only in rice fields where there is standing water. It is most serious on young plants. It is widely distributed, occurring in South and Southeast Asia, China, Japan, Australia, South America, and Africa. The caseworm feeds on rice and also on grasses and millets. Few studies have been conducted on the resistance of rice varieties to the caseworm. Results of limited field screening have been reported in Africa (Soto and Siddiqi 1976, Virmani 1980) and India (Velusamy et al 1976). Greenhouse rearing and screening techniques developed at IRRI are described.

REARING METHODS

The caseworm is reared on rice plants in the greenhouse. It takes about 32 days to complete the rearing cycle in the greenhouse in the Philippines. One set of cages consisting of 1 oviposition cage, 1 larvae-rearing cage, and an adult emergence cage yields about 5,000 larvae/cycle. About 3% of the eggs are used to maintain the culture; the rest are for screening.

Construction of equipment

Steps	Key points
1. Constructing cages	• Three types of cages are used for rearing: 1) oviposition, 2) larvae-rearing, and 3) moth emergence. — The oviposition cage (Fig. 1) consists of a mylar film cylinder 25 cm in diam and 54 cm high. It has a nylon mesh top with a hole for insect release. The hole is plugged with cotton. The cage sits in a plastic basin called egg-holding tray, which contains water 6 cm deep. — The larvae-rearing cage (Fig. 1) consists of a 42- × 42-cm styrofoam or wooden frame enclosing six 13-cm-diam pots with plants. The frame serves as a barrier to prevent larvae from leaving the potted plants. — The adult emergence cage (Fig. 1) is made of a 102-cm-long, 68-cm-wide, and 90-cm-high wooden frame with nylon or fiberglass mesh walls. It has a swinging door for collecting adults. The cage holds 35 13-cm-diam pots.
2. Setting the cages in water pan trays	• The larvae-rearing cage and the adult emergence cage are placed in a water pan tray measuring 1.5 m × 6 m and 15 cm deep.



1. Procedure for greenhouse screening of caseworm *N. depunctalis*.

Growing of food plants

Plants provide cut leaf pieces that serve as a substrate for the moths to lay their eggs on in the oviposition cage, as food for larvae in the larvae-rearing cage, and as a substrate on which the larvae pupate in the adult emergence cage.

Steps	Key points
1. Cutting leaf pieces for the oviposition cage	• Choose plants that are at least 30 days after sowing (DAS). Leaves from younger plants are too narrow to float and to support the ovipositing moths. Plants are grown in a concrete bed in the screenhouse, in the greenhouse, or in the field.

Steps	Key points
2. Growing plants for the larvae-rearing cage	• Sow 50 seeds in 13-cm-diam pots. Use seedlings 14 DAS as food for the larvae.
3. Growing plants for the adult emergence cage	• Transplant 7-10 seedlings in each 13-cm-diam pot at 14 DAS. At 21 days after transplanting the plants are ready for use in the adult emergence cage.

Rearing procedure

The rearing procedure is illustrated in Figure 1.

Steps	Key points
1. Collecting adult insects in the field to start the culture	• Use small cylindrical mylar cages or test tubes to collect adults in the field. • Larvae and pupae can also be collected in the field and reared to adults in the greenhouse.
2. Placing leaf pieces in the oviposition cage	• Place about 15 leaf pieces (5 to 8 cm) on the water in the oviposition cage.
3. Placing adults in the oviposition cage	• Place 30 pairs (male and female) of adults in the oviposition cage on a Monday (Fig. 1). Thirty females ovipositing over a 5-day period will provide enough eggs (more than 3,000) to infest the 252 test entries and checks in 1 compartment-type seedbox.
4. Transferring the oviposition cage	• Moths lay eggs on the underside (side in water) of the leaf pieces. Allow the moths to oviposit for 24 h. Remove the oviposition cage from the egg-holding tray and transfer it to another egg-holding tray containing fresh leaf pieces. • Repeat the procedure daily for 5 days after which put fresh moths in the oviposition cage.
5. Transferring egg-infested leaf pieces to the larvae-rearing cage	• Allow the eggs to remain and develop in the egg-holding tray for 3 days. Make sure the leaf pieces do not overlap so that the eggs on the underside of the leaf pieces remain in water. Count the eggs on the leaf pieces that you intend to use in the resistance screening studies. • Transfer the leaf pieces with a total of about 160 eggs onto the water in the larvae-rearing cage which contains 6 pots with about 50 14-day-old seedlings in a pot (Fig. 1). Water should be 1-2 cm above the level of the pots.

Steps	Key points
	• As the eggs hatch, the larvae first feed on the underside of the cut leaf pieces where the eggs are laid. Then they move on to the potted plants and cut the leaf tip out of which they construct a case. The young larvae find it easier to make cases out of leaves of young (about 14 days old) seedlings than of older plants.
6. Placing fresh seedlings in the larvae-rearing cage	• After the larvae have fed for 3 days on the seedlings, replace the eaten potted plants with fresh 14-day-old seedlings. When changing the pots, lift and invert them and tap the plants over the water within the cage to dislodge the attached larvae-containing cases. • After the larvae have fed for another 3 days on the remaining seedlings, gradually replace the potted plants with fresh seedlings 21-30 days old. These older seedlings are needed for pupal case construction and should not be damaged severely by larval feeding before pupation occurs (in about 10-13 days).
7. Transferring potted plants to the adult emergence cage	• Larvae pupate on the leaf sheath near the plant base about 17-20 days after hatching. When pupation occurs, remove the pots from the larvae-rearing cage, cut the leaves leaving only the lower stem portion of the plants, and transfer the pots to the moth emergence cage (Fig. 1). Leaves are removed so that the emerging moths will rest on the cage sides rather than on the plants, thus facilitating moth collection.
8. Collecting moths and transferring them to the oviposition cage	• Collect moths daily using a cylindrical mylar cage or test tube. • Transfer the moths (30 pairs) to the oviposition cage and repeat the rearing cycle.

SCREENING METHODS

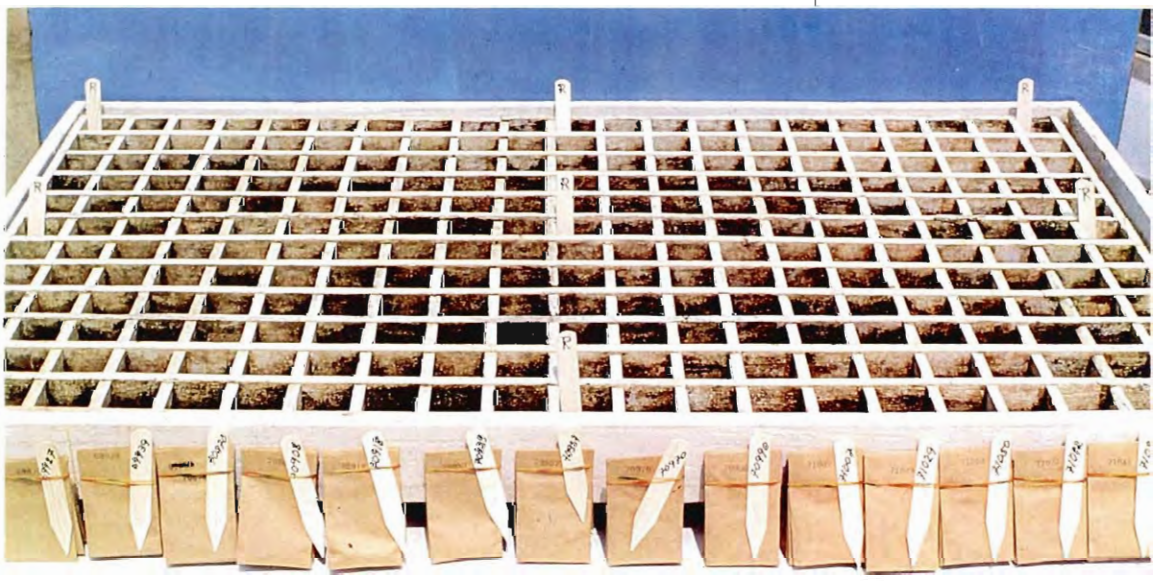
Field screening in India and Africa has been conducted. The methodology used is not reported. Greenhouse screening done at IRRI is described.

Greenhouse screening

Seedlings are screened in seedboxes. The damage rating is based on the extent of leaf scraping by the larvae.

Steps	Key points
1. Constructing the seedbox, sowing	• For screening entries, use a wooden seedbox 114 × 69 × 7 cm (Fig. 2).

Steps	Key points
guide, and seedbox cage	• Construct a sowing guide, consisting of 252 compartments to fit in the seedbox (Fig. 2). • Construct a cage with polyethylene sheet or mylar film sides and an open top (Fig. 3).
2. Sowing test entries	• Place soil containing fertilizer (40 kg N/ha) in the seedbox. • Place sowing guide over the soil and sow seed every Monday (Fig. 2). • Sow seed of test entries in the compartments, five seeds of each entry in a compartment. • Sow a susceptible check such as IR36 into the compartments (in one compartment of each of the four sides and in one compartment in the middle). No resistant check has yet been identified. • Cover seed with fine soil. • Keep soil moist by keeping the seedbox in the shade and spraying water in a fine mist so as not to disturb the seed.
3. Thinning seedlings	• Thin seedlings to 3/compartment every Tuesday at 8 DAS. • Remove the sowing guide from the seedbox after thinning. • Place the seedbox in a water pan tray with 1-2 cm water above the soil level.



2. Compartment seedbox with sowing guide.

Steps	Key points
4. Infesting test entries	• Test entries are ready for infestation at 11 DAS. • Place a cage (Fig. 3) on the seedbox to prevent uneven distribution of the larvae due to wind or water current. • Distribute cut leaf pieces containing eggs at the blackhead stage, which have been previously counted and removed from the egg-holding tray. Distribute the eggs evenly throughout the seedbox so that there are about 3-4 eggs/seedling (about 3,000 eggs/seedbox).

3. Seedbox in a topless polyethylene cage.

Steps	Key points
	• The larvae begin cutting leaf tips for case construction 2 to 3 days after infestation (DI). The cases may be clumped in 1 area and should be randomly redistributed every 2-3 days by hand for even distribution of the larvae on all test entries.
5. Grading damage	• The larvae begin to scrape the green portion of the leaves 8 DI. • Scraping is severe 14-16 DI (Fig. 4) and entries are ready for grading. Leaf cutting damage is not considered in grading because it is done to produce cases for protection and not for feeding. The grading scale is based on a visual estimation of the percentage of the leaf area scraped.

Scale	*Leaf scraping (%)*
0	None
1	Less than 1
3	1-10
5	11-25
7	26-50
9	51-100

SOURCES OF RESISTANCE

Screening for resistance to the caseworm has only recently begun at IRRI. No resistant varieties have thus far been identified. However, field observations in India (Velusamy et al 1976) and Africa (Virmani 1980) have identified some varieties as promising. About 700 varieties were screened at IITA (Soto and Siddiqi 1976); 4 with less than 8% damaged tillers were classified as resistant, compared with susceptible varieties which had as much as 71% damaged tillers.

IRRI accession no.	Variety	Reference
20427	ARC6626	Soto and Siddiqi (1976)
12556	ARC10651	Soto and Siddiqi (1976)
12580	ARC10696	Soto and Siddiqi (1976)
–	BKN6323	Virmani (1980)
17328	Brengut	Virmani (1980)
26915	BW78	Virmani (1980)
26844	CO 28	Velusamy et al (1976)
6621	Laki 396	Soto and Siddiqi (1976)
30351	ROK2	Virmani (1980)

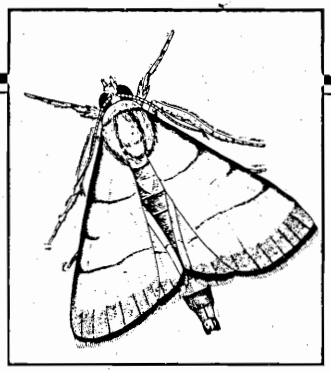
4. Caseworm scraping is severe at 15 days after infestation and plants are ready to be graded for damage.

REFERENCES CITED

Soto, P. E., and Z. Siddiqi. 1976. Screening for resistance to African rice insect pests. Paper presented at the WARDA Varietal Improvement Seminar, 13-17 September, Bouake, Ivory Coast.

Velusamy, R., I. D. Jomahi, P. Shanmugasunlaram, and S. S. Subramaniam. 1976. A preliminary note on the incidence of rice caseworm *Nymphula depunctalis* Guenée in rice varieties. Madras Agric. J. 63:180-181.

Virmani, S. S. 1980. Varietal resistance to rice diseases and insects in Liberia. Int. Rice Res. Newsl. 5(2):3-4.



Chapter 23
LEAFFOLDER

The leaffolder *Cnaphalocrocis medinalis* is distributed throughout South and Southeast Asia where its importance appears to have increased in recent years. Populations are often highest in fields that have received high rates of nitrogen fertilizer and at field edges near trees. The larva rolls the leaf by tying silk to the margins and feeds within the rolled leaf, removing the green layer. Heavily damaged fields turn brown and appear hopper-burned. Damage usually occurs during the reproductive stage of plant growth.

Methods of rearing the insect, screening entries for resistance, and determining the mechanisms of resistance have been developed at IRRI. Several resistant varieties have been developed in screening at IRRI and in national programs.

REARING

Leaffolders are reared in the greenhouse and headhouse, with most of the process being conducted in galvanized iron trays in the greenhouse. A modification of the method developed by Waldbauer and Marciano (1979a,b) is described. By this method, it takes about 5 h/day of a 5-day work week to rear sufficient insects to screen 1,000 entries a month.

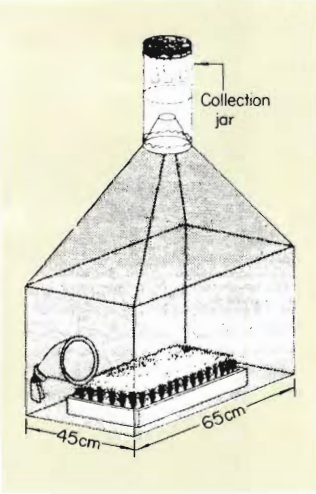
Construction of equipment

Seedboxes, rearing cages, moth collection cages, and oviposition cages are required in rearing the leaffolder.

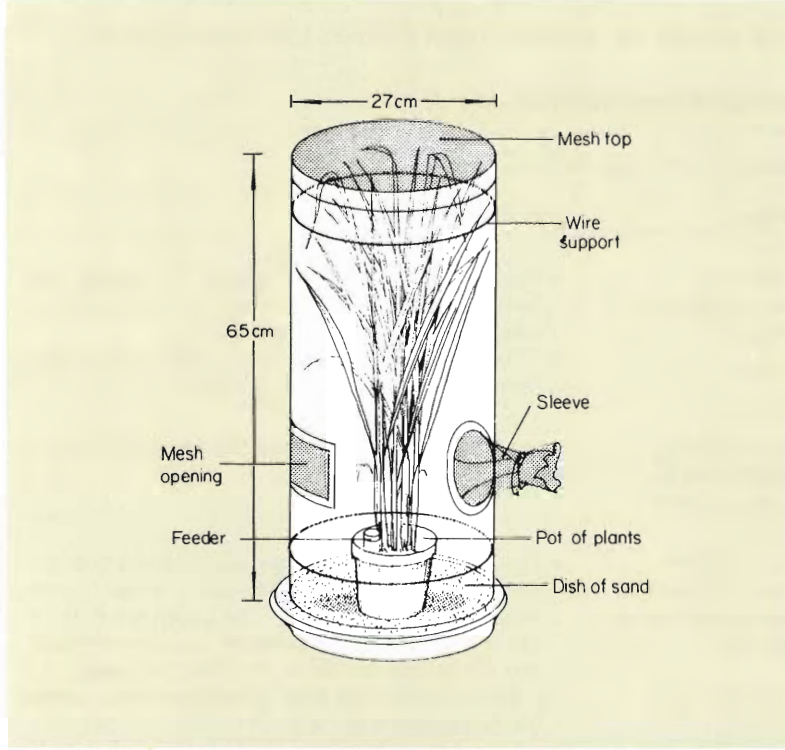
Steps	Key points
1. Installing galvanized iron trays	• Place open water trays made of thick galvanized iron sheets on flat wooden or iron benches inside a screened room in the greenhouse. • The trays are usually 1.5 m wide; the length depends on the size of the room.
2. Constructing seedboxes for insect rearing	• Prepare 24 units of wooden seedboxes (60 × 40 × 10 cm) for rearing (Fig. 1).
3. Constructing cages for rearing and collecting of adults	• Construct 24 rearing cages with open bottoms and tops designed to facilitate collection of emerging moths (Fig. 2). The cages are 65 × 45 cm or any appropriate size that can accommodate the 60- × 40- × 10-cm seedboxes. Cages are made of fiberglass netting with a heavy gauge galvanized wire frame.

Steps	Key points
4. Constructing oviposition cages (Note special cage for dry areas.)	• Because humidity affects oviposition, oviposition cages must provide the correct humidity for ovipositing females. For humid areas the oviposition cage should be made of a cylinder of stiff mylar film (Fig. 3).

1. Standard wooden seedbox.

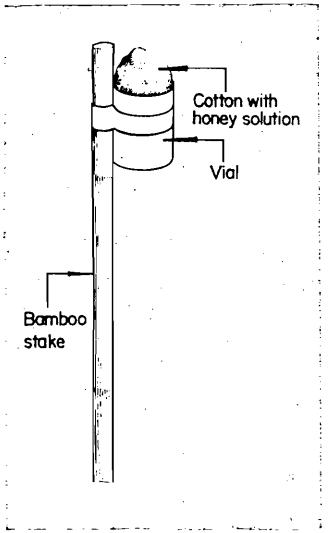


2. Bottomless leaffolder rearing cage over a seedbox with seedlings.



3. Oviposition cage used in rearing the leaffolder. This model is recommended for humid conditions as it has a mesh top.

Steps	Key points
	• The cage should be 65 cm high and 27 cm in diam. Roll the mylar plastic sheet and glue the overlapping edges together with contact cement. Reinforce the cylinder with two heavy gauge wire rings, which are held in place by contact cement. • Set a sleeve 14.5 cm in diam and 26 cm long 10 cm from the bottom of the cage. The sleeve and the cage roof are made of nylon mesh or fiberglass net and attached to the cage with masking or waterproof tape. • Close the open bottom of the cage by pressing it into an earthenware dish (5-cm-high, 30-cm-diam) almost full of dry sand. • For dry areas, have a similar cage but use a flexible plastic sheet with a circular mesh-covered hole 5 cm in diam for the top. • For the sleeve, use a flexible plastic bag with 20-25 holes (about 0.5-cm-diam) cut into the upper base. • Set the dry area cage into a dish of moist sand.
5. Preparing honey feeders	• For maximum egg production, the leaffolder moths require a source of sugar such as a 25% honey solution. • Make a feeder from a plastic vial 6.5 cm high and 3.5 cm in diam. With a hacksaw, cut 1 cm from the bottom of the vial and then glue it on top of the vial to form a shallow dish (Fig. 4).



4. Honey feeder from which leaffolder moths obtain nourishment required for oviposition.

Growing food plants

Steps	Key points
1. Growing seedlings for insect rearing	• Place 4 inverted clay pots inside each of the 24 rearing cages. Place a seedbox (60 × 40 × 10 cm) filled with fine fertile soil 8 cm deep on the inverted pots. The pots keep the seedbox out of the water in the water tray. • Uniformly broadcast about 4,000 pregerminated seeds of CR94-13, IR36, or any other susceptible variety in each of 3 seedboxes (A, B, and C; see Table 1) at weekly intervals. • Water the seedlings regularly with a sprinkler. When they have grown to about 4-5 cm remove the inverted clay pots under the seedbox. This will immerse the seedbox in water and watering with a sprinkler is no longer necessary. • The seedlings are ready for infestation with leaffolder larvae at 21 days after sowing (DAS).

Table 1. Schedule of activities to be conducted in succeeding weeks in insect rearing and screening for resistance to the leaffolder.[a]

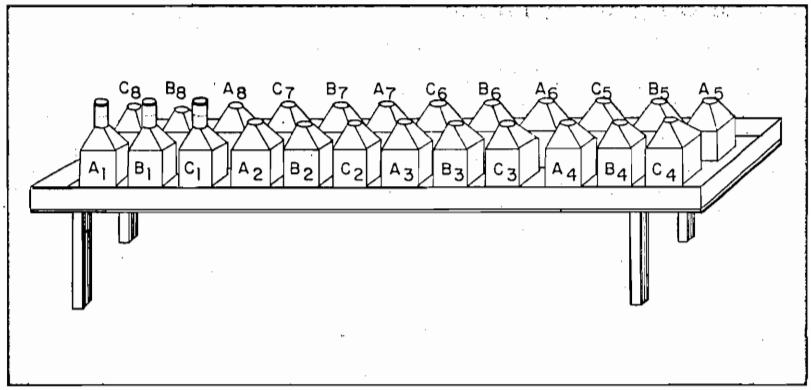
Week in sequence	Mass rearing			Screening test	
	Sowing food plants in seedboxes for the following rearing cages	Infesting seedlings of food plants in the following cages	Collecting adults from the following cages	Sowing entries to be infested with adults from the following cage	Infesting entries with adults from the following cages
1st	A_1 B_1 C_1	–	–	–	–
2d	A_2 B_2 C_2	–	–	–	–
3d	A_3 B_3 C_3	–	–	–	–
4th	A_4 B_4 C_4	A_1 B_1 C_1	–	–	–
5th	A_5 B_5 C_5	A_2 B_2 C_2	–	–	–
6th	A_6 B_6 C_6	A_3 B_3 C_3	–	B_1	–
7th	A_7 B_7 C_7	A_4 B_4 C_4	–	B_2	–
8th	A_8 B_8 C_8	A_5 B_5 C_5	A_1 B_1 C_1	B_3	B_1
9th	A_1 B_1 C_1	A_6 B_6 C_6	A_2 B_2 C_2	B_4	B_2
10th	A_2 B_2 C_2	A_7 B_7 C_7	A_3 B_3 C_3	B_5	B_3

[a]Activities from 1st to 8th week consist of starting the insect culture and screening test entries after which the cycle of routinary activities proceeds. A, B, and C are cages in eight groups, each of which is treated with the same rearing activities at weekly intervals.

Steps	Key points
2. Growing plants for oviposition	• Plants on which moths can oviposit are produced by sowing seeds in seedboxes and transplanting 5 14-day-old seedlings in each pot (9-cm-high, 12.5-cm-diam). • Fill 140 pots with fertile soil to about 1.5 cm from the top. Arrange pots in a water pan tray. • Enclose the 140 pots in a single fiberglass net cage, and plant 20 pots at weekly intervals. • Apply nitrogen fertilizer (about 2 g/pot) to improve the growth of the potted plants whenever they become yellowish. • Inspect plants regularly and remove any arthropods that may infest the potted plants. • These potted plants are ready for use in the oviposition cage about 60 DAS.

Starting the insect colony

Steps	Key points
1. Allocating rearing cages for specific functions	• Divide the 24 cages into 8 groups. Label the three cages in each group A, B, and C (Fig. 5). • Use the *A* cages to maintain the culture, *B* cages to rear adults for mass-screening tests, and *C* cages to rear insects to any desired growth stage for other experiments.



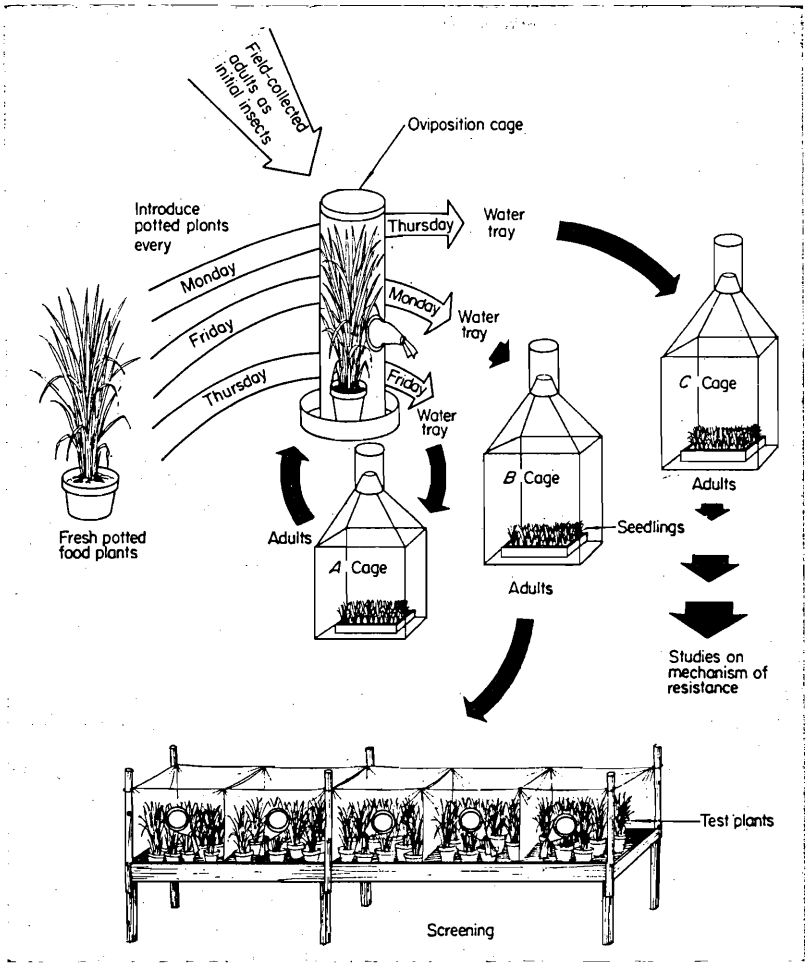
5. Leaffolder rearing cages in a galvanized iron water tray. The A-cages are used to rear insects for the oviposition cage to maintain the culture, the B cages to provide moths for screening, and the C cages to provide moths for studies on mechanisms of resistance. The three cages at the left front have moth collection jars.

Steps	Key points
	• Identify the eight groups of cages by placing under each letter a numerical subscript common to the three cages in a group: A_1 B_1 C_1; A_2 B_2 C_2... A_8 B_8 C_8. The labels serve as guide to the activities that occur in the cages in the succeeding weeks (Table 1).
2. Collecting adults in the field	• Collect adults in the field four times at weekly intervals. Adults are easily collected with an insect net. If no adults are present in the field, collect partly grown larvae of about the same age and rear them to adults in the greenhouse. Repeat field collections four times at weekly intervals to start a complete cycle of the culture. • Eggs of the adults that are collected during the first week will become adults in the fifth week. Thus, field collection may be stopped after the fourth week. • If leaffolders are not available in the vicinity, obtain a culture from another institution. Transporting newly laid eggs on leaf pieces is more convenient than transporting larvae. Place the leaf pieces on a moist paper towel in a plastic petri dish or in a vial with moist cotton at the bottom.

Mass rearing

Steps	Key points
1. Preparing food for adults	• Adults require a source of sugar for egg production. Prepare a 25% (by volume) solution of honey in water.

Steps	Key points
	• Tie the honey feeder 10-20 cm from the tip of a bamboo stick. Put the other end of the bamboo stick in the soil in a pot containing rice plants. Place loose cotton wool saturated with the honey solution in the feeder (Fig. 4). • Place fresh cotton wool and honey solution in the feeder every other day.
2. Collecting eggs	• Maintain adult insects (12 males and 11 females) in the oviposition cage containing 60-day-old plants on which the females oviposit. • If the plants are short, place them on inverted clay pots so that the tips of their leaves bend over as they touch the top covering of the oviposition cage. Adults prefer to lay eggs under the bent leaves. • The schedule of introducing and removing the potted plants from the oviposition cages is indicated in the following steps.
3. Rearing insects in *A* cages to maintain the culture	• Place the potted plants in the oviposition cage on Thursday, for example, and remove them on Friday (the following day) (Fig. 6). • Place the potted plants containing eggs in a water tray in the greenhouse. When the eggs hatch, cut the leaves containing the larvae into small pieces and uniformly distribute about 250 larvae over the 21-day-old seedlings growing in a seedbox inside the *A* cages. • The larvae will feed on the seedlings and develop into adults. • Place the collection jar (Fig. 2) on top of the rearing cage 2 weeks after infestation; by this time the larvae are about to pupate. Place tissue paper in the jar so that the moths have substrate on which to sit. • The adults are easily collected by placing a black cloth over the cage, gently tapping the seedlings, and gradually driving the insects up into the collection cage. • When the adults are trapped in the upper section of the collection jar, detach the jar and transfer the adults to the oviposition cage.
4. Rearing test insects in *B* cages for mass screening	• The eggs laid on potted plants placed in the oviposition cage on Friday and moved out on Monday are reared in *B* cages (Fig. 6). The adults that develop are used to infest plants in the mass screening test. • Follow other procedures in step 3 above.



6. Leaf folder rearing-screening procedure. Potted plants after exposure to ovipositing moths in the oviposition cage are transferred to a galvanized iron water tray. When eggs reach the blackhead stage, leaves on which they are laid are cut and the leaf pieces transferred to the A, B, or C cages.

Steps	Key points
5. Rearing test insects in *C* cages for studies on mechanisms of resistance	• The eggs that are laid on potted plants placed in the oviposition cage on Monday and moved out on Thursday are reared in *C* cages (Fig. 6). • If larvae of uniform age are needed for a certain experiment, such as studies on the mechanisms of resistance, cut the tips of leaves containing eggs and place them on moist filter paper in a petri dish. • The larvae that hatch on the same day provide test insects of uniform age. • If adult test insects are required, follow Step 3. The adults that emerge on the same day are considered of uniform age.

Steps	Key points
6. Adding field-collected insects to maintain quality of greenhouse culture	• Collect field populations and introduce them into the greenhouse culture at least once a year to minimize possible inbreeding depression. If immature stages are collected, rear them first in separate cages until adult emergence. This practice prevents introduction of parasites to the greenhouse culture.

SCREENING METHODS

Greenhouse screening

In initial greenhouse screening, seedlings are grown in pots and infested with moths which lay eggs on the seedlings. Seedlings should be at least 14 days old at time of moth infestation. The plants are graded for leaffolder damage at 21 days after infestation (DI). In initial tests, entries are not replicated. Entries with a rating of 0-5 in initial screening are retested using 10 replications/entry.

Steps	Key points
1. Setting the schedule of sowing and infesting	• Once the rearing program is established and moths are available on a weekly basis, the screening program can be started. • Schedule sowing of seed of the test entries and resistant and susceptible checks 14 days before infestation.
2. Listing test entries	• List entry numbers of the materials on the data sheet (Fig. 7). List 118 entries plus one susceptible (TN1) and one resistant (TKM6) check variety. These materials can be accommodated in each 1.7- × 1.4-m division of a water pan tray.
3. Preparing pots for sowing	• Use 11-cm-diam clay pots. • Fill pots with lowland paddy soil to within 6 cm of the top.
4. Sowing and maintaining test materials	• Sow 10 seed/entry into each pot. Cover seed with fine soil. • Place the pots in a metal tray with water. • Thin seedlings to 5/pot at 10 DAS. Then broadcast N fertilizer on the soil.
5. Infesting test seedlings	• At 14 DAS, enclose each group of 120 pots of seedlings with a 1.7- × 1.4- × 0.75-m fiberglass net cage (Fig. 8) and introduce 10 pairs (male and female) of 3-day-old adults for oviposition. Install

Entry No.	IRRI Acc. no.	Rep.	Leaves (no.)		Leaves (no.) with damage grades of:			Damage rating (R)	Adjusted damage rating (D)	Scale
			Total	Damaged	1	2	3			
TN1 (SeK)	00 105		26	23	3	2	18	39.1	-	-
TKM6 (SeK)	00 237		24	10	10	0	0	6.9	17.6	3
437	46688		16	5	4	1	0	6.2	15.9	3
438	46671		24	7	4	2	1	7.6	19.4	3
439	36282		10	7	2	2	3	23.0	63.9	7

7. Data sheet showing results of nonreplicated initial leaffolder screening tests.

Steps	Key points
	one honey feeder in each of the four corners of the cage (See step 1, *Mass rearing*). • Seven DI remove the honey feeders. When the moths are dead, remove the cage to allow more sunlight to reach the seedlings.
6. Grading damage on test seedlings	• At 21 DI evaluate the damage caused by the larvae. — For the test to be valid, at least 60% of the leaves of the susceptible check should be damaged. — For greenhouse screening consider the extent of damage on each leaf. For each entry first examine all the leaves and rate each one 0-3 based on the extent of damage.

Grade *Damage*

0 None
1 Up to 1/3 of leaf area scraped
2 > 1/3 to 1/2 of leaf area scraped
3 > 1/2 of leaf area scraped

— Based on the number of leaves with each damage grade, compute the damage rating (R) as follows:

$$R = \left[\frac{\frac{(\text{no. of leaves with damage grade of } 1 \times 100)\ 1}{\text{total no. of leaves observed}} + \frac{(\text{no. of leaves with damage grade of } 2 \times 100)\ 2}{\text{total no. of leaves observed}} + \frac{(\text{no. of leaves with damage grade of } 3 \times 100)\ 3}{\text{total no. of leaves observed}} \right] \div 6$$

8. Fiberglass mesh cage placed over potted plants in a water tray. Moths are released within the cage for oviposition on the plants.

Steps	Key points
	— Calculate damage rating (R) for each test entry and the susceptible check. Then determine the adjusted damage rating (D) for each test entry as based on extent of damage in the susceptible check by:

$$D = \frac{\text{R of test entry}}{\text{R of susceptible check}} \times 100$$

— Convert D to a 0-9 scale.

Scale	*% adjusted damage rating (D)*
0	0
1	1-10
3	11-30
5	31-50
7	51-75
9	more than 75

Steps	Key points
7. Retesting	• Entries with ratings of 0-5 in initial screening are retested. Entries are replicated 10 times. Seedlings are thinned to 5/pot and tillers pruned to 1/seedling at 21 DAS. Two 1st-instar larvae are placed on each tiller with a camel hair brush. • At 17 DAS entries are evaluated for damage and damage ratings determined as described in step 6. In determining D, the R of each replicate of the test entry is divided by the R of the susceptible check in the corresponding replication. Record data on data sheet (Fig. 9).

Field screening

One major problem commonly encountered in field screening is insufficient leaffolder populations. A test is considered valid when damaged leaves in the susceptible check average at least 50%. Several steps can be taken to increase the probability of obtaining sufficient populations. The method used by Velusamy and Chelliah at Tamil Nadu Agricultural University, Coimbatore, India, is described.

Entry No.	IRRI Acc. no.	Rep.	Leaves (no.)		Leaves (no.) with damage grades of:			Damage rating	Adjusted damage rating (D)	Scale
			Total	Damaged	1	2	3	(R)		
TN1 (SeK)	00105	1	29	29	0	1	28	49.4	-	-
		2	30	27	5	1	21	38.9	-	-
		3	22	22	1	0	21	48.5	-	-
		4	25	23	2	0	21	43.3	-	-
		5	20	18	0	0	18	45.0	-	-
		6	25	21	0	0	21	42.0	-	-
		7	27	26	1	0	25	46.9	-	-
		8	26	23	3	2	18	39.1	-	-
		9	23	22	2	2	18	43.5	-	-
		10	29	26	0	0	26	44.8	-	-
		$\bar{x}$	25.6	23.7	1.4	0.6	21.7	44.1	-	-
442	48069	1	15	4	4	0	0	4.5	8.9	1
		2	6	0	0	0	0	0	0	0
		3	16	4	4	0	0	4.2	8.6	1
		4	8	2	2	0	0	4.2	9.7	1
		5	10	2	2	0	0	3.3	7.3	1
		6	11	1	1	0	0	1.5	3.6	1
		7	11	3	3	0	0	4.6	9.8	1
		8	18	2	2	0	0	1.9	4.9	1
		9	12	4	4	0	0	5.6	12.9	3
		10	11	1	1	0	0	1.5	3.3	1
		$\bar{x}$	11.8	2.3	2.3	0	0	3.1	6.9	1.1

9. Data sheet showing the results of retesting entries with 10 replications.

Steps	Key points
1. Ensuring leaffolder populations	• Select the proper planting date and area (hot spot). Previous experience and light trap data can be used to select an area where the leaffolder population will be high at maximum tillering of the crop. Fields bordered by trees and partially shaded have higher populations than open fields. • Plant border rows of a susceptible variety. • Double the nitrogen recommendation. High nitrogen fertilizer rates increase leaffolder populations. • Apply phorate to the susceptible border row plants. This apparently decreases the natural enemy population and increases leaffolder damage.
2. Preparing the field for transplanting	• Apply complete fertilizer providing 70 kg N/ha basally and uniformly incorporate into the paddy before transplanting.
3. Transplanting the susceptible border rows	• Transplant at a 20- × 10-cm spacing 5 border rows of a susceptible variety such as CO 36, CR94-13, or IR36 around each replication.
4. Applying insecticide	• At 15 days after transplanting (DT) apply 1.0 kg ai granular phorate/ha to the susceptible border plants.
5. Transplanting test entries	• Thirty DT the susceptible border plants, transplant one 5-m row/test entry in each replication at 10-cm spacing between plants within the row. Alternate each row with a row of a susceptible check (CO 36, CR94-13, or IR36) at 20-cm spacing from the test entry row. Include one row of a resistant check (TKM6) per replication. Arrange entries in a randomized complete block design and replicate three times.
6. Releasing leaffolder moths	• After transplanting the test entries, release laboratory-reared or field-collected moths on the susceptible border plants to increase the level of infestation.
7. Applying fertilizer	• Apply nitrogen fertilizer in split doses of 60 kg/ha each at 30 and 50 DT the test entries.
8. Evaluating	• When the susceptible check has at least 50% of the leaves folded make the first damage grading. If damage increases, make another grading later.

Steps	Key points
	• Count the damaged and undamaged leaves in 20 hills selected at random in each entry. Sum up the total leaves damaged and undamaged in each entry and compute the percentage of damaged leaves.

$$\% \text{ damaged leaves} = \frac{\text{no. of damaged leaves}}{\text{total no. of leaves observed}} \times 100$$

- Convert the percentage of damaged leaves to another figure based on *D:*

$$D = \frac{\% \text{ damaged leaves in test entry}}{\% \text{ damaged leaves in the susceptible check (av of two nearest susceptible checks on each side of the test entry)}} \times 100$$

- Convert *D* to a 0-9 scale:

Scale	*D*
0	No damage
1	1-20%
3	21-40%
5	41-60%
7	61-80%
9	81-100%

MECHANISMS OF RESISTANCE

Entries having ratings of 0-5 in either the greenhouse or field mass screening test are retested in the greenhouse or laboratory to confirm the level and nature of resistance.

Antixenosis for oviposition

Steps	Key points
1. Selecting entries	• Select entries that have shown an average rating of 5 and below in the mass screening test. • Plant 1 seedling/pot (10-cm-high, 12-cm-diam) in 10 pots for each entry. Each pot represents a replication. • Grow the potted plants inside a cage if necessary to protect them from other arthropods. Apply fertilizer for good plant growth.

Steps	Key points
	• When the plants are 50-60 days old, arrange the potted plants in a randomized complete block design in a water tray. Place the potted plants about 30 cm apart so their leaves do not touch. Enclose each replication with a fiberglass net. Keep the top of the net at least 15 cm above the tip of the tallest test plants.
3. Infesting plants with moths	• Release 3-day-old adults at the rate of 5/plant. Do not provide a honey feeder in the cage because it may influence the adults to lay eggs on plants adjacent to it.
4. Evaluating	• After 2 nights of exposure to ovipositing moths, remove the potted plants and count the number of eggs laid on each entry. Compare results among entries. • Collect the moths that are still alive and place them in the oviposition cage for use in the rearing program.

Antixenosis for feeding

Steps	Key points
1. Growing plants	• Plant a 7- to 10-day-old seedling in clay pots using 10 pots/variety. Each pot represents a replication.
2. Infesting plants with larvae	• When the plants are 50-60 days old, individually infest each with a fourth-instar larva and enclose it with a mylar film cage.
3. Evaluating	• Two DI remove the larvae and evaluate the extent of leaf damage on each potted plant. • To measure leaf area damaged, place the leaf on the table and cover it with a piece of mylar film having mm^2 grids. • Count the number of squares (mm^2) that occupy the leaf area scraped by the larva: Use these figures to differentiate varieties on the basis of damage caused by the larvae.

Antibiosis on survival and insect weight

Steps	Key points
1. Growing test plants	• Grow test plants up to 50-60 days old as described in *Antixenosis for oviposition.* Maintain 3 plants/pot.

Steps	Key points
2. Infesting plants with larvae	• Five days before the scheduled date of infestation, place gravid females in the oviposition cage overnight. Collect leaf pieces with eggs and place in a petri dish with moist filter paper at the bottom. The larvae that hatch at the same time will be used as test insects. • Arrange the test plants in a randomized complete block design, then infest them with 10 newly hatched larvae/pot. Place the larvae on the auricles with a camel hair brush. Enclose the three plants of each pot with a mylar cage (Fig.10).
3. Evaluating	• When 95-100% of the insects on the susceptible plants have emerged to adults, collect, count, and weigh all insect stages from the individual cages. Identify the sex of the adults that emerge.

10. Mylar film cage (13 · 50 cm) used for studies on the levels of antibiosis in rice having leaffolder resistance.

SOURCES OF RESISTANCE

The following varieties are resistant or moderately resistant (ratings of 0-5) in greenhouse screening at IRRI (Heinrichs et al 1983, unpubl.).

Varieties resistant or moderately resistant to the leaffolder *Cnaphalocrocis medinalis*.

IRRI accession no.	Name	Origin
	Resistant[a]	
237	TKM6	India
5909	GEB24	India
6041	CO 7	India
15327	Muthumanikam	Sri Lanka
19325	Ptb 33	India
21166	ARC10982	India
36408	Yakadayan	Sri Lanka
39433	IR5685-26-1	Philippines
39558	BR116-3B-19	Bangladesh
45493	Darukasail	India
46020	Kumalbhog	India
46048	Kapurkanti	India
46077	Kataribhog	India
46671	Shete	India
47166	Calixto	Philippines
47774	Khao Gaw Diaw	Thailand
47852	Khao Mah Khaek	Thailand
48069	Khao Rad	Thailand
48078	Khao Sa Ahd	Thailand
49020	Balam	Bangladesh
49081	Dolachikon	Bangladesh
49086	Gora	Bangladesh
49088	Gorsa	Bangladesh

IRRI accession no.	Name	Origin
49099	Kalachikon	Bangladesh
49154	Biron	Bangladesh
49157	Bora	Bangladesh
49175	Coti	Bangladesh
49235	Madhu Madub	Bangladesh
49378	Anaikomban CO. 4 BK	India
49456	Bir-Me-Fen	India
49529	Choorapundy	India
50332	Cygalon	Italy
50362	Kaohsiung Sen Yu 169	China (Taiwan)
51275	Ching-Gan Tsan	China
54440	Hema	Malaysia
	Moderately resistant[b]	
6303	ASD7	India
11057	W1263	India
12119	ARC5752	India
20293	ARC5968	India
21167	ARC10985	India
36282	Kalu Mahatmaya	Sri Lanka
36342	Noorti Onru	Sri Lanka
36355	Pukuru Samba	Sri Lanka
36362	Ratha Thavalu	Sri Lanka
36379	Sri Murugan	Sri Lanka
40804	Hata Pandaru	Sri Lanka
40855	Nivudu Samba	Sri Lanka
43034	ARC14960	India
45446	CSC1	India
45460	Daharnagra	India
45484	Dandikasail	India
45490	Dapu	India
45756	Hanskal	India
45813	HS19/Taichu 65 (VKM 12)	India
45848	IB Rose/Latisail	India
45880	Jatadhanya	India
45890	Jathakatke	India
45893	Jessoa	India
45895	Jessoa	India
45905	Jhinga	India
45906	Jhingasail	India
45955	Kakowa	India
46049	Karpurkanti	India
46050	Karpursail	India
46051	Kartikbad	India
46052	Karticgeti	India
46053	Kartic Kalma	India
46061	Kartik Sail	India
46075	Katalgaria	India
46076	Kataribhog	India
46078	Kataribhog	India
46082	Kathibadal	India
46086	Katke	India
46091	Katki	India
46092	Kaurdhan	India
46688	Solepona	India
46693	Sonamukhi	India
46697	Sudarash	India

IRRI accession no.	Name	Origin
46765	T21	India
46873	Unnamed	India
46875	Unnamed	India
47239	Kabug-at	Philippines
47762	Hahng Mah	Thailand
47769	Jek Chuey Pw 55-161-272	Thailand
47795	Leuang Pra 3	Thailand
47859	Leuang Khamin	Thailand
48042	Khao Tawng Kam	Thailand
48073	Khao Rai Ton Tahn	Thailand
48360	Nahng Guang	Thailand
48612	Sihao	Indonesia
48933	Tatabo	Indonesia
48939	Tingga Loko	Indonesia
48940	Tobada	Indonesia
48944	Tokasumba	Indonesia
48954	Topembangu	Indonesia
49058	Asina	Bangladesh
49070	Boilam	Bangladesh
49173	Chin-Kartir	Bangladesh
49214	Katiringari	Bangladesh
49244	Moynasail	Bangladesh
49280	Kirun Dhan	Bangladesh
49593	Dau-Laung 88	Thailand
49603	Dodda Kamboothi	India
49739	Kalar Samba	India
49837	Kuthi Kondappan	India
49917	Mudugada Hariyance Vanna Bhata	India
49934	M 81	India
50005	Panja Cheera	India
50035	Pisini	India
50703	KMP 38	India
50986	Hondera Wala	Sri Lanka
51373	Ma-Tsan 1	China
54369	Acheh 62	Malaysia
54444	Itan Empati	Malaysia
55059	ADT 8	India
55111	Kizhuvela	India

[a]Damage ratings of 0-3. [b]Damage rating of 5.

Wild rice species resistant to the rice leaffolder *Cnaphalocrocis medinalis* at IRRI, 1983.

IRRI accession no.	Name	Origin
100581	*O. sativa* × *O. rufipogon*	Taiwan (China)
100115	*O. brachyantha*	Guinea
101231	*O. brachyantha*	Sierra Leone
101232	*O. brachyantha*	Sierra Leone
101233	*O. brachyantha*	Sierra Leone
101234	*O. brachyantha*	Sierra Leone
101236	*O. brachyantha*	Mali Republic
103622	*O. glaberrima* 'Diakao Mango'	Mali

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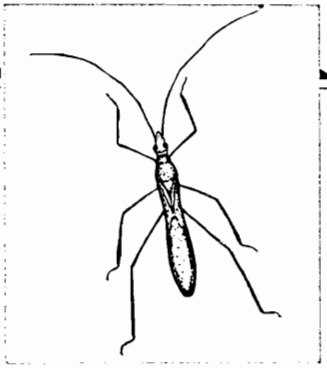
Waldbauer, G. P., and A. P. Marciano. 1979a. Mass rearing of the rice leaffolder. Int. Rice Res. Newsl. 4(2):7.

Waldbauer, G. P., and A. P. Marciano. 1979b. Rice leaffolder: mass rearing and a proposal for screening for varietal resistance in the greenhouse. IRRI Res. Pap. Ser. 27. 17 p.



Chapter 24
RICE BUGS

Several species of stink bugs in the families Alydidae and Pentatomidae attack the developing grains of rice. In Asia several species of *Leptocorisa* (Alydidae) are the most important and in the USA, the rice stink bug *Oebalus pugnax* is of major importance. The bugs feed on the developing grain mostly early in the morning and at dusk. The stylets of *Leptocorisa* spp. enter between the lemma and the palea and the removal of the milk results in empty grains. Damage at the dough stage results in spots visible on milled rice. Some studies on screening for resistance have been conducted on Pentatomidae attacking rice in the USA. In Asia only field screening has been done until recently. The methods described have recently been developed at IRRI for *L. oratorius*. A few sources of resistance to *O. pugnax* have been identified.

REARING

Plants with grains in the milk stage are used as food source in the rearing of the rice bug.

Construction of equipment

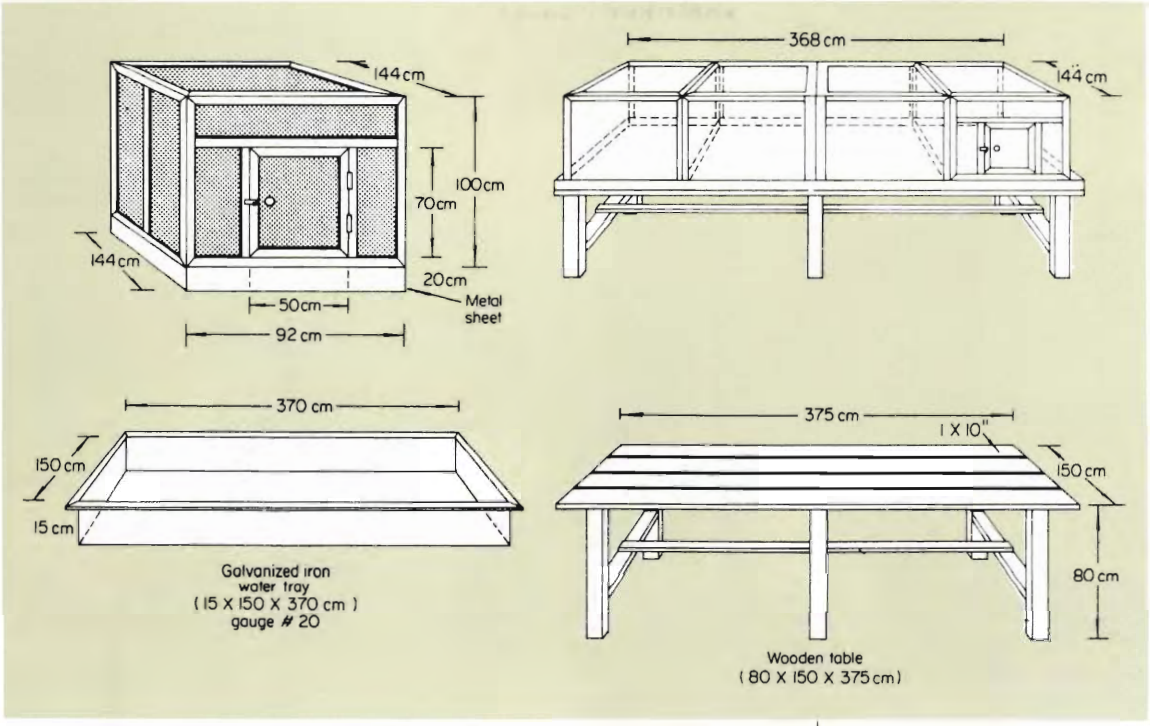
Steps	Key points
1. Constructing a greenhouse and a screenhouse	• The greenhouse and the screenhouse are similar to those used for leafhopper and planthopper screening.
2. Constructing benches and water trays	• Benches are made of wood or tubular and angular iron bars (Fig. 1). • Pan trays set on the benches are made of thick-gauge galvanized iron (GI) sheets (Fig. 1). • The bench and the tray are 1.5 m wide. The length depends on the room length. • Arrange the benches side by side with a 0.6-m space between them to serve as passageway.
3. Constructing rearing cages	• If the rearing cage must be placed outdoors, make bottomless cages of water-resistant wood with a thick-gauge GI sheet at its base (Fig. 2). • If the cages will be kept in a greenhouse where they are protected from wind and rain, a simple cage constructed like a mosquito net can be used (Fig. 3). Make the cage about 1.5-2.0 m high depending on the variety used for rearing. For IR36, 1.5 m is sufficient. A cage with a 1.3-m^2 floor

1. Water tray on bench with a cage for rearing rice bugs.

Steps	Key points
	area can maintain 1,000 rice bugs from eggs to adults. • A minimum of 10 cages are needed to produce a regular supply of insects for rearing and screening.
4. Constructing plant boxes	• A 32- × 24- × 12-cm plastic basin can hold 6 hills. • If a plastic basin is not available construct a plant box out of water-resistant wood.

Growing of food plants

Steps	Key points
1. Selecting the appropriate variety as a food source	• The ideal variety to use as food source is one that is early maturing, photoperiod insensitive, short-statured, stiff stemmed, and resistant to major insect pests and diseases but susceptible to the rice bug. At IRRI, IR36 and IR50 are used.



2. Detailed sketch of a bottomless rearing cage, water tray, and table for rearing rice bugs.

Steps	Key points
2. Growing food plants	• Screenhouse-grown plants require less protection from pests.
a. Screenhouse	• Construct a 30-m-long, 10-m-wide, and 0.5-m-deep soil bed surrounded by concrete walls. Fill the bed with soil to a depth of 0.3 m. • Enclose the bed with iron screen mesh. • Divide the bed into 10 plots, each plot to be planted at weekly intervals with 21-day-old seedlings of an early-maturing variety that reaches the milk stage about 70 days after transplanting. • Transplant seedlings at a 20- × 20-cm spacing during the dry season and at 25- × 25-cm during the rainy season. • By the time the 10th plot is planted, the plants in the first plot are available as food plants. • Apply the right kind and amount of fertilizers in each plot.
b. Field	• Follow the procedure of planting in the screenhouse with the following modifications: — Construct levees between the 10 plots. This will allow irrigation of individual plots of different

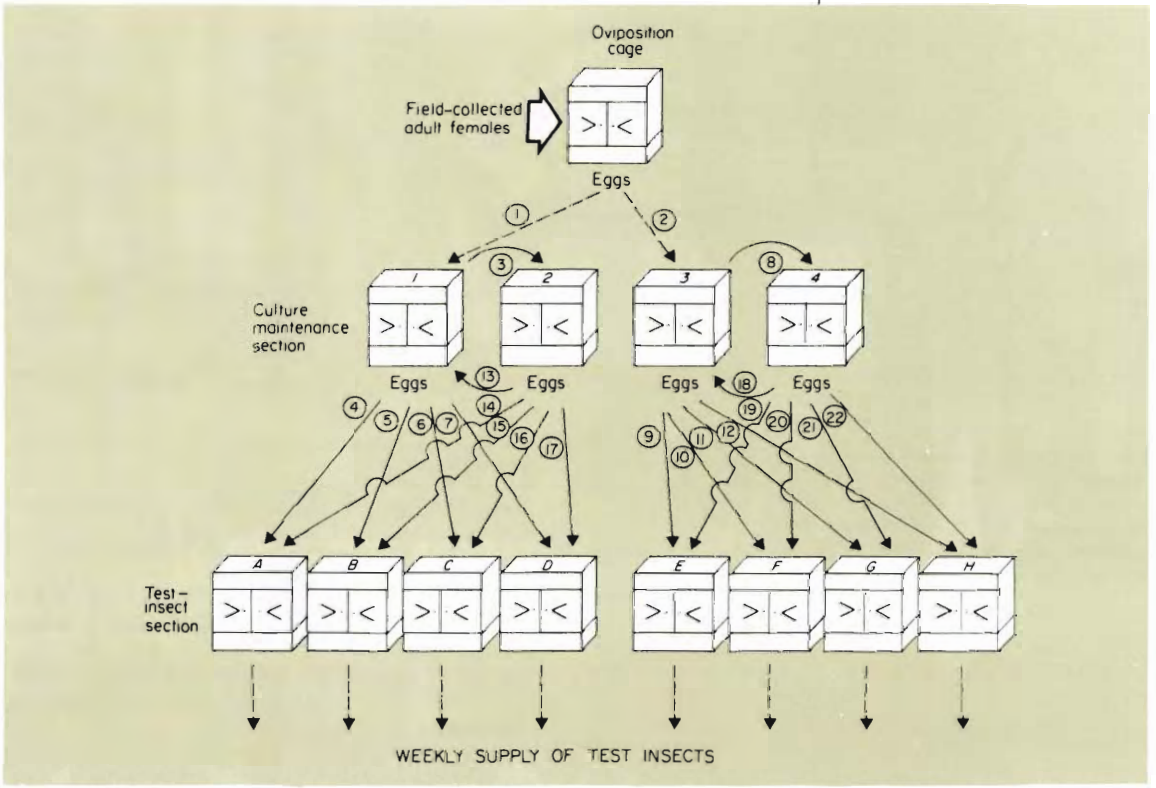
3. Fiberglass mesh cage for rearing the rice bug.

Steps	Key points
	plant ages and prevent irrigation water from carrying insecticide from one plot to another. — Construct an irrigation canal perpendicular to all the plots. Make a 10-cm opening in the levee through which water in the canal will enter the plot. — Apply 0.75 kg ai of carbofuran granules/ha before transplanting. This will protect the plants from most insect pests during the seedling stage and part of the tillering stage. — If rice stem borers or rice bugs are potentially serious pests, enclose each plot with a fiberglass net cage at the start of panicle initiation. — The panicles are ready as food sources when the spikelets at the midsection of the panicles are in the milk stage. This usually occurs about 10 days after anthesis when the pollen sacs appear outside the lemma and palea.

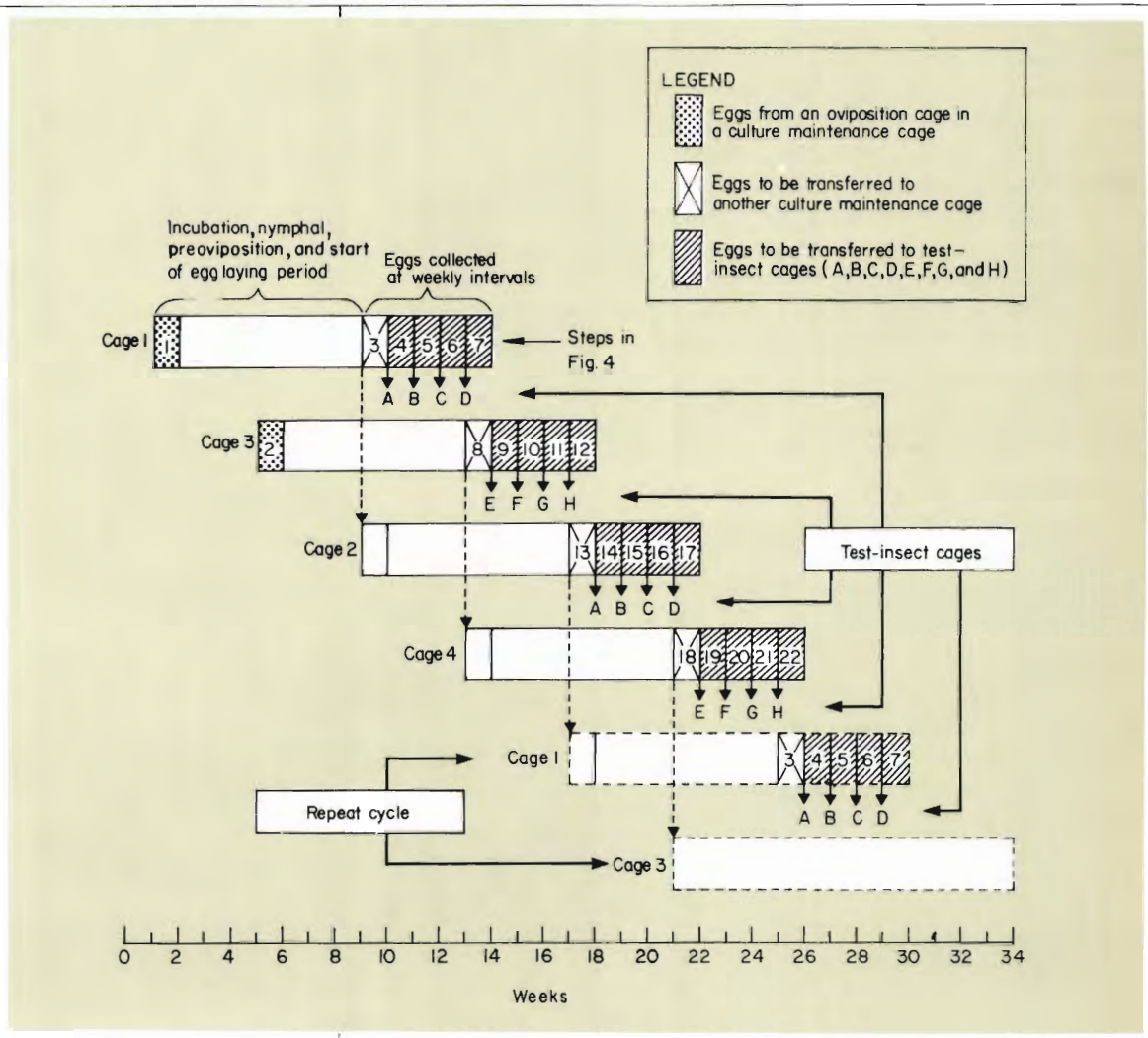
Rearing the rice bug

Three groups of cages are used: oviposition, culture maintenance, and test insect (Fig. 4). To start the culture, transfer potted plants with eggs from field-collected insects from the oviposition cage to the culture maintenance cage where the eggs will hatch and nymphs will be reared to the adult stage. The adults will also lay eggs within the same cage to supply another culture maintenance cage. Next, transfer the potted plants to the test-insect culture cages to rear bugs to ages desired for use in screening studies and studies on the mechanisms of resistance. The weekly schedule of the rice bug rearing is illustrated in Figure 5. In the rice bug rearing procedure, the movement of eggs from one rearing cage to another (A) and the changing of food plants and plants on which the bugs oviposit (B) are described.

Steps	Key points
A. *Movement of eggs*	
1. Collecting field insects	• Collect about 300 gravid *Leptocorisa oratorius* females from the field and enclose them in an oviposition cage containing food plants at the milk



4. Schematic representation of the rice bug rearing program showing the movement of eggs from the oviposition cage to the culture maintenance and test-insect rearing cages. Circled numbers indicate the order in which the steps are taken.



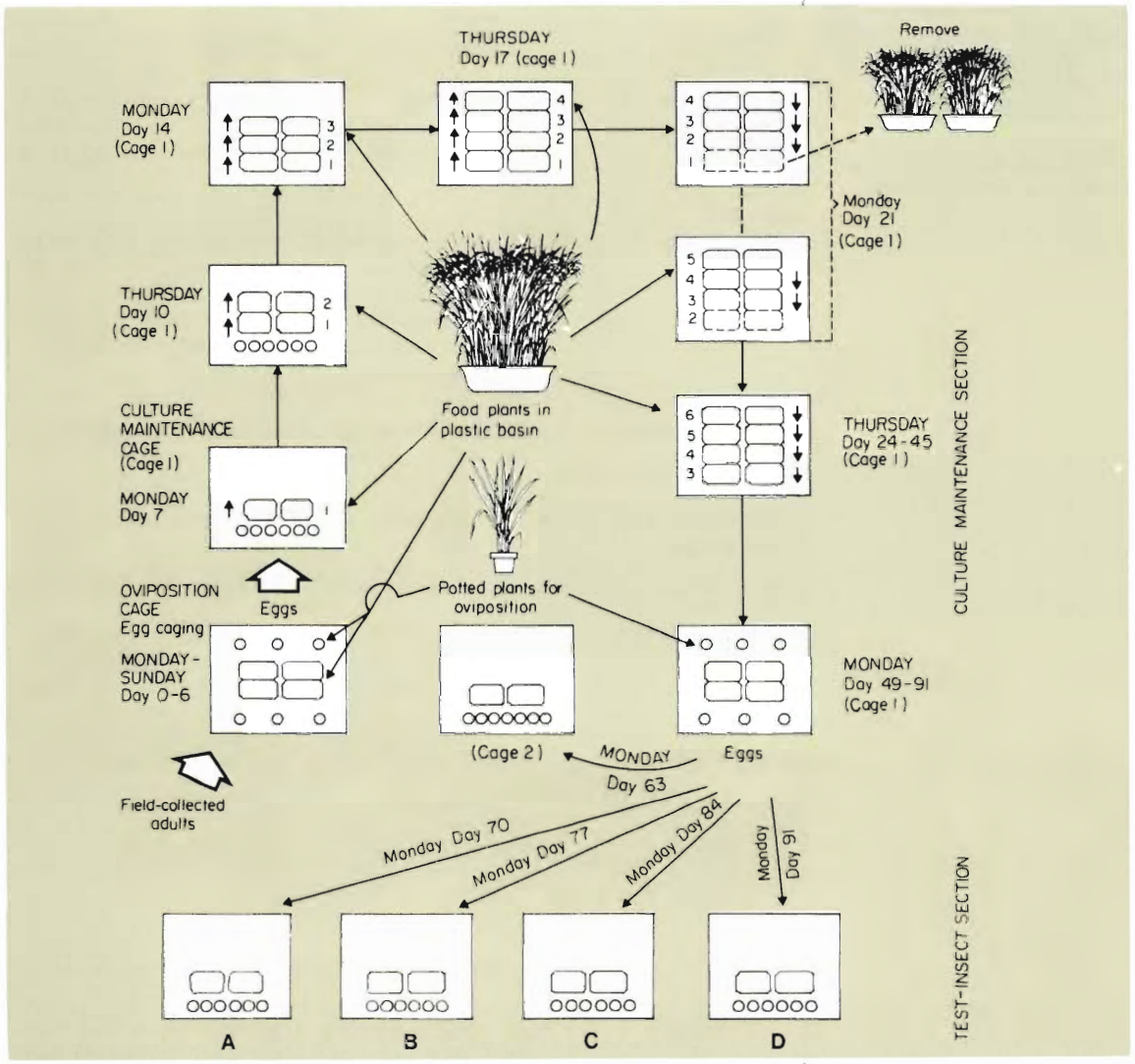
5. Schedule for the movement of rice bug eggs in culture maintenance cages 1, 2, 3, and 4 and test-insect rearing cages A, B, C, D, E, F, G, and H. Circled numbers indicate steps 3 to 22 as shown in Figure 4.

Steps	Key points
	stage and 6 potted plants at the booting stage on which the bugs will lay eggs (Fig. 4).
2. Filling cage 1 with eggs	• After 1 week transfer the potted plants at the booting stage containing eggs to cage 1 in the culture maintenance section (Fig. 4, step 1). Continue feeding the insects in the oviposition cage, but their eggs during the 3 succeeding weeks may be discarded. • Maintain at least 300 females in the oviposition cage. If the number is less than 300 at the end of

Steps	Key points
	the fourth week, collect additional females from the field.
3. Filling cage 3 with eggs	• At the beginning of the 5th week introduce 6 potted plants at the booting stage in the oviposition cage. After 7 days, transfer the potted plants containing eggs to cage 3 (Fig. 4, step 2). • The field-collected insects can now be discarded and the oviposition cage can be used as culture maintenance cage 4.
4. Filling cage 2 and the test-insect cages with eggs	• The eggs in cage 1 will hatch, develop to adults, and start laying eggs after about 7 weeks. At the beginning of the 9th week, introduce 6 potted booting plants into cage 1. Seven days later transfer the plants with eggs to cage 2 (Fig. 4 , step 3). • Fill test-insect cages A, B, C, and D with eggs from cage 1 at weekly intervals for the next 4 weeks (Fig. 4, steps 4-7).
5. Filling cage 4 and the test-insect cages with eggs	• At the beginning of the 13th week introduce booting plants into cage 3 and 7 days later transfer them to cage 4 (Fig. 4, step 8). • For the next 4 weeks, fill test-insect cages E, F, G, and H with eggs from cage 3 at weekly intervals (Fig. 4, steps 9-12). When cage E is filled with egg-infested potted plants, the bugs in Cage A become newly emerged adults (Fig. 4). Use the bugs from Cage A as test insects for screening or studies on the mechanisms of resistance. Females become gravid 2 weeks after becoming adults. If gravid females are to be used in experiments they will reach the gravid stage in cage A when cage G is filled with eggs. The following week, fill cage H with egg-infested potted plants and clean cage A. The next week, fill cage A with egg-infested potted plants and repeat the cycle as described in the following steps.
6. Refilling cage 1 and the test-insect cages with eggs	• Collect eggs from cage 2 and transfer them to cage 1 at the beginning of the 18th week (Fig. 4, step 13). • In the following 4 weeks transfer eggs to test-insect cages A, B, C, and D (Fig. 4, steps 14-17).
7. Refilling cage 3 and the test-insect cages with eggs	• At the beginning of the 22nd week move eggs from cage 4 to cage 3 (Fig. 4, step 18). Transfer eggs to the test-insect cages E, F, G, and H in the next 4 weeks (Fig. 4, steps 19-22). The cycle is now complete.

Steps	Key points
B. *Changing food plants and plants for oviposition*	
1. Oviposition cage	• The oviposition cage contains four plastic basins with food plants at the milk stage to serve as food for the bugs (Fig. 6). Food plants in a plastic basin are shown in Figure 7. — For food plants, select plants whose grains at the midsection of the panicles are at the milk stage. — Carefully uproot individual hills that are planted in a concrete bed or in the field. Leave a small amount of soil on the roots. — Trim the lower leaves and place the plants in the plastic basin. • The oviposition cage also contains six 12-cm-diam pots with plants in the booting stage, which serve as a substrate for oviposition. • On Monday (day 0) place four plastic basins with food plants at the milk stage and six potted plants at the booting stage in the oviposition cage. Place the booting plants about 30 cm away from the food plants (Fig. 6). The cage should contain about 300 gravid females. • Protect the eggs from ants by placing the legs of the benches on which the cages are set in a water pan tray, or placing a sticky material around the legs of the benches to trap the ants. • On Thursday (day 3) remove the four plastic containers with old food plants from the oviposition cage and replace with fresh plants. Repeat this process every Monday and Thursday. • On Monday (day 7), after a 7-day oviposition period, transfer the 6 pots with egg-infested plants from the oviposition cage to a culture maintenance cage, ie cage 1 (Fig. 6). Replace the potted plants in the oviposition cage with fresh plants. Leave the adults in the oviposition cage.
2. Culture maintenance cage	• On the same day (day 7), arrange the six potted plants with eggs at one side of the cage. Place the first two plastic basins close to the potted plants with eggs so that the newly hatched nymphs can easily transfer to the panicle of the food plants (Fig. 6). • On Monday, day 14, remove the 6 potted plants as eggs will have hatched and nymphs are now feeding on the plants in the plastic basins. • Every Monday and Thursday place two more basins with food plants in the cage (Fig. 6). When the 5th row of 2 plant basins is introduced on day 21, remove the first 2 plant basins (row 1) from the cage and move the second, third, and fourth rows

Steps	Key points
	down 1 row each. Repeat this process until the insects begin to lay eggs and cage 1 is used to supply eggs to cage 2 on Monday (day 63) and to test-insect cages A, B, C, and D (Mondays, days 70, 77, 84, and 91) (Fig. 6).
3. Test-insect cage	• Change food plants on Mondays and Thursdays as described for the culture maintenance cage in step 2.



6. Movement of food plants for rice bug feeding and potted plants as oviposition substrates into and out of the oviposition, culture maintenance, and test-insect cages

7. Plastic basin with food plants at the milk stage for rice bug feeding.

FIELD SCREENING

Screening for resistance to stink bugs attacking rice panicles has been limited to field screening, but techniques have not yet been well established. Methods described are based on those of Bowling (1979a, b) and Nilakhe (1976). Field screening can be conducted either by using a natural population or by caging bugs on panicles.

Natural bug populations

If natural populations of stink bugs are high, this method is the most practical to use.

Steps	Key points
1. Selecting a hot spot	• Select a site and time of the year when the rice bug population is high and consists primarily of the species toward which the screening program is directed.
2. Scheduling planting	• Schedule planting to coincide with peak abundance of bugs. The plants should be at the reproductive stage when maximum populations occur. Previous experience regarding the life history of the bug in a given area is necessary to select a proper planting date.
3. Preparing seed of test entries	• Select varieties that flower at about the same time. No resistant check has yet been identified.
4. Sowing seed of test entries	• For transplanting prepare raised seedbeds (wet-bed method) in the field. Sow about 3 g of each entry in a 30-cm row with 10 cm between rows.
5. Transplanting or sowing	• Before transplanting, apply fertilizer to the field. • Transplant three 21-day-old seedlings/hill in 3-m rows (12 hills/row) with 25 cm between hills and 50 cm between rows. Transplant one row of each entry per replication and replicate four times. If seed is sown with a planter, sow about 10 g seed/3-m row.
6. Labeling the entries	• Label the entries to facilitate identification.
7. Infesting	• Infestation is by naturally occurring populations.
8. Evaluating	• At maturity, harvest 10 panicles at random from each replication and place them in a paper bag.

Steps	Key points
	Bring panicles to the laboratory for evaluation of bug feeding activity. Three methods can be used to evaluate for bug damage.
a. Counting stylet sheaths	• Stylet sheaths on grains are counted to determine feeding activity. Stylet sheaths can be seen with the naked eye but are more easily counted when stained and viewed under a microscope. • The staining solution consists of 1 part each of phenol, lactic acid, and distilled water, and 2 parts glycerine. Add enough acid fuchsin to produce a dark red color. • Immerse panicles in the staining solution for about 1 h then remove and rinse in tap water. Panicles may be stored for counting at a convenient time. • Examine the grains and determine: — Percentage of grains with one or more stylet sheaths. — Av number of stylet sheaths/grain. Entries with the lowest percentage of grains with stylet sheaths and lowest number of stylet sheaths/grain are considered the most resistant but further studies are needed to prove this. No grading scale has been established.
b. Unfilled grains: seed cleaner technique	• Oven-dry harvested panicles at 50° C for 1 week. Thresh panicles in a panicle thresher. • Run filled and unfilled grains through a seed cleaner. • Unfilled (chaffy) grains, some of which are unfilled because of stink bug damage, will collect in the upper containers of the seed cleaner. • Compare percentage of unfilled grains among entries. Entries with the least unfilled grains are considered most resistant. No grading scale has yet been established.
c. Unfilled grains: flotation technique	• Oven-dry harvested panicles at 50° C for 1 week. Thresh each panicle by hand and collect all grains including those that are empty. • Prepare a saturated salt solution. • Place the grains from one panicle in a graduated cylinder containing the salt solution. Shake the contents vigorously. Bug-damaged empty grains float and filled undamaged grains sink to the bottom of the cylinder. • Estimate the percentage of floating grains. Compare entries. Those with the lowest percentage of floating grains are considered the most resistant. No grading scale has yet been established. • Compare results among entries using Duncan's Multiple Range Test.

8. Cage (19-cm-diam and 50-cm-long) used to study the survival and development of the rice bug.

Bugs on caged panicles

Field populations are unpredictable and often too low for valid screening. To overcome this problem, artificially reared bugs can be put on caged panicles. Plants for testing can be grown in a screenhouse or field. Methods used to evaluate damage by natural populations can also be used in the cage test.

Steps	Key points
1. Steps 3-6 under *Natural bug populations*	• Seed preparation and planting are as described in *Screening against natural populations.*
2. Preparing cages	• Prepare wire frame cages 9 cm in diam and 50 cm long with 10-cm fiberglass mesh sleeves at both ends (Fig. 8).
3. Caging panicles	• Because maturity of the varieties will vary, not all varieties will be caged on the same day. When the panicles of a variety emerge from the boot, select eight panicles at random in each of the four replications and cage them. Support the cage with a wood or bamboo stick (Fig. 8).
4. Placing bugs in cages	• When about one-fourth of the grains reach the milk stage, place two 3- to 5-day-old adult bugs in each of 4 cages of each replication. Leave the remaining four cages uninfested to serve as controls.
5. Evaluating	• When grains mature, cut the base of the panicle and take the cage containing the panicle to the laboratory. • Oven-dry panicles individually in paper bags at 50° C for 1 week. • Thresh by hand.
a. Percentage of grain weight loss	• Determine the difference in weight per grain between grains from the control and infested panicles. $\%\ \text{grain weight loss} = \dfrac{\text{Av wt (mg) of grains on control panicles} - \text{Av wt (mg) of grain on infested panicles}}{\text{Av wt (mg) of grains on control panicles}} \times 100$ Compare percentage of grain weight loss among entries using Duncan's Multiple Range Test. The same grains can be used for one or two additional tests.
b. Stylet sheaths	• Follow the procedure given in Step 8a, *Natural bug populations.*

Steps	Key points
c. Unfilled grains: flotation technique	• Follow the procedure given in Step 8c, *Natural bug populations.*

MECHANISMS OF RESISTANCE

Tests for levels of antixenosis, antibiosis, and tolerance have been developed.

Antixenosis for oviposition on plants at the booting stage

This test can be conducted in the greenhouse, screenhouse, or field. If a large number of entries are to be tested, a field study is most convenient.

Steps	Key points
1. Growing test plants	• If the maturity of selected test materials varies, adjust sowing so that the booting stage of all entries occurs at the same time. • Transplant 21-day-old seedlings at 3 seedlings/hill and 40- × 40-cm spacing for the screenhouse or field study. Plant one hill per entry in each replication. Arrange treatments in a randomized complete block design with four replications. For a greenhouse test, transplant three 21-day-old seedlings in 13-cm-diam clay pots. One pot represents one replication. • Place a screen cage over each replication. • When plants are at the booting stage, tie the tillers loosely to straighten the plants and prevent leaves of adjacent hills from touching. Fasten a bamboo stick at the base of the plants, if necessary.
2. Infesting test plants with bugs	• Release gravid females at the rate of five per hill or per potted plant.
3. Counting the eggs	• After 2 days count and remove the eggs laid. Express number of eggs on a hill basis.
4. Allowing plants to develop panicles	• Frequently inspect and remove nymphs that hatch from any eggs left on the plants. Allow the plants to develop panicles and use the plants as test materials in other tests where panicles are required.

Antixenosis for oviposition on cut tillers at the booting stage

This test should be conducted in a shady place such as a laboratory or headhouse room where the temperature is about 25-30° C. The greenhouse may be too hot and cause rapid drying of the panicles.

Steps	Key points
1. Growing test plants	• Follow step 1 under *Antixenosis for oviposition on plants at the booting stage.*
2. Preparing cut tillers	• Cut tillers about 40 cm below the base of the flag leaf. Use two tillers per entry. • Insert the tillers in soft drink bottles containing water and distribute at random in a circular pattern on a bench. In the center of the circle place one susceptible (IR36) potted plant at the milk stage from which the leaves have been removed. This plant serves as a food source.
3. Infesting the test entries with bugs	• Replicate four times. Enclose each replication with a net cage and use five gravid females per bottle of test entries.
4. Evaluating	• Count the number of eggs at 24 h after infestation with the bugs.

Antixenosis for probing on spikelets at the milk stage

Steps	Key points
Panicles **1.** Setting up the experiment	• Use as test plants three plants from the test *Antixenosis for oviposition on plants at the booting stage.* Set up the experiment inside a laboratory or headhouse room. • Cut panicles at the milk stage, 40 cm below the lowest rachis. Each entry should consist of a panicle with similar number of grains. If the number of spikelets per panicle is small, two or more panicles per entry in each replication may be required. • Insert the peduncle of the panicles in a vial, test tube, or bottle containing water. Arrange the bottles at random in a circular pattern. Replicate each entry four times.
2. Infesting test panicles with bugs	• Enclose each replication with a circular mylar cage and infest the plants with 5- to 10-day-old adults at the rate of 10 insects/panicle.
3. Evaluating	• Two days after infestation, collect and stain the panicle with an acid fuchsin solution for 1 h (see *Field screening*). Rinse the grains with water. Count the stained stylet sheaths on the grains with the aid of a binocular microscope.

Steps	Key points
	• Compare the entries based on the number of stylet sheaths per panicle.
Individual grains	
1. Removing grains	• Instead of using whole panicles, individual grains can be tested. • Cut panicles containing grains at the milk stage and immediately submerge panicle base in tap water in a container to prevent drying. • In the laboratory make sure the grains for testing are all at milk stage by pressing adjacent grains with the nail tip of the thumb and a finger. • Remove and place 20-grain samples per entry on moist filter paper or paper towel in petri dishes. Arrange entries at random, 15 cm apart in a circular pattern. • Replicate four times.
2. Infesting with bugs	• Enclose each replication with a circular mylar film cage and release 10 bugs/entry.
3. Evaluating	• After 24 h of caging, collect and stain the grains as previously described. Count the stylet sheaths on the grains.

Antixenosis for feeding activity

Steps	Key points
1. Preparing the materials	• Treat filter paper by dipping in bromocresol green solution made by mixing 2 parts bromocresol green powder and 1 part ethanol. Allow the filter paper to dry for 1 h. Repeat the dipping process and allow filter paper to dry for 1 h or more. • Use panicles with grains at the milk stage. Cut the panicle from its peduncle and trim off grains that are not at the milk stage. • Push the peduncle through an inverted cup, filter paper treated with bromocresol green solution, and the cover of a water container until about 10-15 grains are in the cup (Fig. 9).
2. Infesting test entries	• Collect uniform-age test insects which have been starved overnight and place them in a container with moist cotton or paper towel. • Infest the panicles with two female bugs per cage.
3. Evaluating	• At 20-24 h after infestation, remove the filter paper. Measure the area of the spots produced by the

9. Feeding chamber with bromocresol green-treated filter paper to study the feeding activity of the rice bug on grains of rice varieties.

Steps	Key points
	excreta of the bug on the filter paper by tracing the spots on tracing paper and measuring them over graph paper with mm^2 grids. • Express feeding activity in mm^2 area of spots.

Antibiosis on survival and development, and weight

Steps	Key points
1. Preparing the plants	• When about 25% of the spikelets in a hill reach the milk stage, enclose the panicle with a mylar cage 13 cm in diam and 50 cm long with 10-cm nylon mesh sleeves at both ends. Support the mylar cage with a bamboo stick (Fig. 8). • Arrange the entries in a randomized complete block design replicated 10 times, each cage representing a replication. • The number of spikelets per panicle varies significantly among varieties. Estimate and enclose about 1,500 spikelets/cage, irrespective of the number of panicles.
2. Preparing the insects	• One-day-old nymphs are needed to infest the panicles. Panicles are infested the same day mylar cages are placed over them. To have insects of the right age, collect newly laid eggs the day the panicles emerge. Cut the portion of the leaves on which the eggs were laid and place it on moist filter paper in a petri dish.

Steps	Key points
3. Placing bugs on the panicles	• One day after the nymphs hatch, put 10 bugs in each cage with a mouth aspirator or camel hair brush.
4. Evaluating the insect reaction	• Count insects 5, 10, 15, and 20 days after infestation (DI) to determine percentage of survival. • When the insects on the susceptible check have become adults record the number and growth stage of the insects in all cages. • Collect and oven-dry insects, then record their weight and compare among entries.

Antibiosis on insect weight

This test is used for preliminary information on antibiosis. It is less time-consuming than the test on survival and development.

Steps	Key points
1. Preparing the plants	• Follow step 1, *Antibiosis on survival and development, and weight.*
2. Preparing the insect materials	• Rear the test insects so that sufficient insects of the same age are available.
3. Infesting the panicles with bugs	• Introduce 10 newly hatched nymphs/cage with a mouth aspirator. • After 5 days, collect and oven-dry the insects at 50° C. Weigh the insects. • Compare weight of insects among entries.

Tolerance in panicles of the same plant

Steps	Key points
1. Preparing the plants	• If test plants are field-grown, protect the panicles from insect damage, especially by rice bugs, by placing a cage over the test entries.
2. Infesting panicles at the milk stage with bugs	• When the panicles reach the milk stage select 100 uniform grains within the same panicle. Trim off extra grains of the panicles using small scissors. Enclose with a mylar cage (5-cm-diam and 30-cm-long) with 5-cm sleeves at both ends (Fig. 10). • Install five cages per plant. • Introduce 4, 8, 12, 16, and 20 3-day-old to 5-day-old female adults in the cages. Replace dead insects daily with insects from the same source.

Steps	Key points
	• A plant with 5 cages, each containing 100 grains and infested at different insect densities, represents a replication. Conduct six replications per variety. Arrange in a randomized complete block design.

10. Cages (5-cm-diam and 30-cm-long) used to study tolerance of rice varieties for the rice bug.

Steps	Key points
	• Ten days after insect infestation, cut the base of the panicles and take them to the laboratory together with the insects in the cages.
3. Determining the weight of the insects	• Kill the insects with a knockdown insecticide and place them in paper triangles. Oven-dry them at 50°C and weigh after drying.
4. Determining the weight of grains	• Place individual panicles in a small paper bag and place them in an oven at 50°C for drying to 14% moisture content. • Remove the grains carefully from the rachis and weigh.
5. Evaluating damage using weight of insects vs grain weight	• Conduct a simple linear regression analysis using the weight of the insects *X*, as the independent factor and the weight of the grains *Y*, as the dependent factor. • Compare the regression coefficient among varieties. Significantly lower values of a regression coefficient indicate tolerance of the variety for the insect.
6. Evaluating damage using the number of insects vs grain weight	• Conduct a simple linear regression analysis using the number of insects *X*, as the independent factor and the weight of the grains *Y*, as the dependent factor. A low regression coefficient indicates tolerance.
7. Evaluating damage using the number of stylet sheaths vs grain weight	• Place the dried grains in a glass vial whose bottom has been removed and replaced with a nylon mesh. • Soak the grains in acid fuchsin staining solution for an hour and rinse in water. • Select as many grains as possible that have one, two, three, or more stylet sheaths among panicles treated at different levels of infestation from each variety. • Redry samples in the oven and weigh individual grains having different numbers of stylet sheaths. Conduct a simple linear regression analysis using the number of stylet sheaths *X*, as independent factor and the weight of grains *Y*, as the dependent factor. • A significantly lower regression coefficient value indicates tolerance.

Tolerance in spikelets on the same panicle

Steps	Key points
1. Protecting panicles at the booting stage	• Protect panicles from rice bugs by placing cages over them.
2. Infesting spikelets at the milk stage	• Select 100 uniform grains at the milk stage in a panicle. In each of 5 cages enclose 20 grains. The mylar cage is 3 cm in diam, 5 cm long, and has 3-cm nylon mesh sleeves at both ends (Fig. 11). • A panicle with 5 cages each containing 20 grains and different insect densities represents a replication. • In each cage, introduce 2, 4, 6, 8, or 10 5-day-old nymphs. Replace dead insects daily with insects from where the original test insects were obtained. • Conduct six replications per variety arranged at random in a completely randomized design. • At 5 DI cut the base of the panicles and bring them to the laboratory for evaluation.
3. Evaluating and analyzing damage	• Follow steps 3, 4, 5, 6, and 7, *Tolerance in panicles of the same plant.*

SOURCES OF RESISTANCE

Although screening for resistance to rice bugs has been limited, there appears to be moderate levels of resistance to the rice stink bug *Oebalus pugnax* in tests conducted in the USA. Nilakhe (1976) listed several breeding lines, plant introductions, and varieties with low amounts of grain weight loss due to bugs feeding on caged panicles. Bowling (1979a) listed Bluebelle, Nortai, P19810, and RU7603069 as having resistant reactions to *O. pugnax* in field and laboratory tests.

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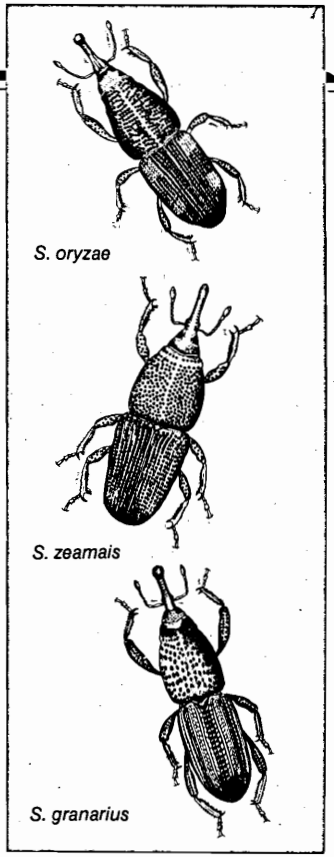
11. Cages (3-cm-diam and 5-cm-long) over spikelets used in test of tolerance for rice bugs.

Chapter 25
RICE WEEVIL, MAIZE WEEVIL, AND GRANARY WEEVIL



Stored grain insects are serious pests in the rice-producing countries of the tropics because temperature and humidity in most are suitable for insect growth and reproduction. Of the various stored grain pests that attack rice, the rice weevil *Sitophilus oryzae,* the maize weevil *S. zeamais,* and the granary weevil *S. granarius* are the most damaging. They are serious pests of rough and milled rice. The larvae and adults feed within the grain and cause major damage.

Methods for rearing the weevils are well established. Screening of rice for resistance to the weevils has been limited to a few laboratories, and few sources of resistance have been found.

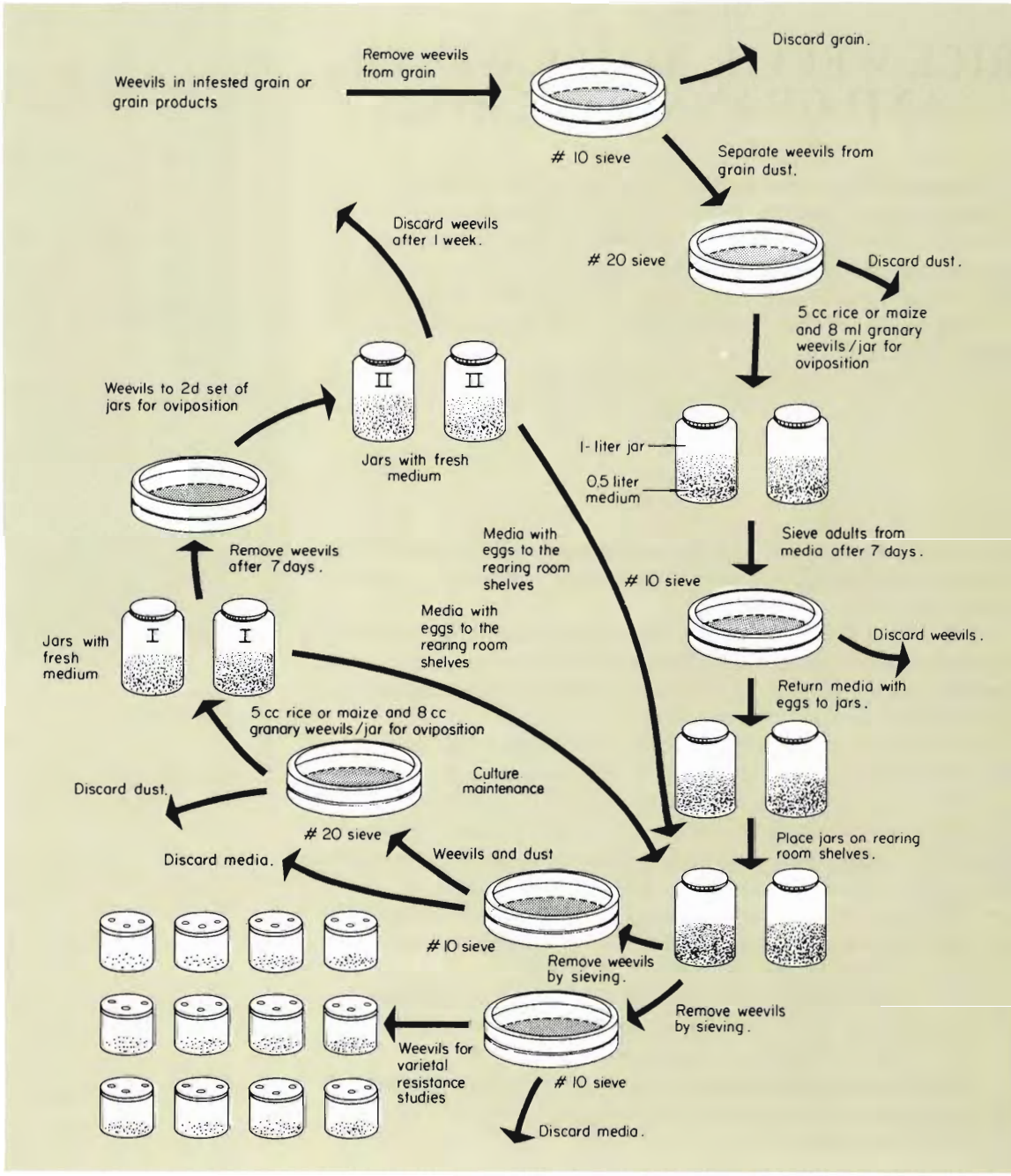
REARING

Weevils are easy to rear and large populations can be maintained in a relatively small rearing room space. The weevil rearing program described is illustrated in Figure 1.

In the tropics rice storage insects can be easily cultured in a laboratory without controlled temperature and humidity environment. It is, however, desirable to use standardized methods to ensure sufficient insects of a known age for varietal resistance studies.

Rearing procedures described here are adopted from those used by the Stored Products Insects Laboratory at Kansas State University (Pedersen et al 1977) and those used by Strong et al (1967) at the University of California. They have listed several general procedures and suggestions for a successful rearing program. These procedures apply not only to the weevils but to all stored-grain insects included in this manual.

- Test insects should always be reared on the grain that is going to be tested for several generations before use. For internal feeders, brown rice is an excellent medium.
- Freeze the medium for cultures at -15° C for at least 1 week before use to ensure that it is free of any live insects that might have infested it. Be sure to maintain sufficient conditioned food on hand.
- After the medium is removed from the freezer, seal the containers with masking tape or store them in a cold room at 5° C to protect them from further insect infestation.
- Widemouthed 1-liter or 4-liter jars covered with metal lids provided with a 5-cm-diam 60-mesh brass screen for ventilation are generally used in rearing most of the species. The container size and amount of medium per culture jar depend, however, on the insect species and



1. Procedure for rearing the rice, maize, and granary weevils for varietal resistance studies.

quantity of insects required for the screening program. Label each jar with the name of insect species, date of infestation, and other pertinent information.

- Rear the insects in multiples of 2 or more jars/week to ensure an abundant supply of the insects. In case a culture jar breaks, another is available.
- Rotate the cultures on a systematic basis to maintain insects of a known age.
- Maintain cultures in a rearing room held at 27 ± 2° C and 65 ± 5% RH with a 12:12 h light and dark period.
- When rearing more than one species, prevent cross contamination. As a precaution remove cultures from the rearing room when placing insects in or removing them from the culture jars. Transfer insects and change the medium of different species on different days.
- Never smoke in the culture room.
- Never bring insecticides into the culture room.
- Keep order in the rearing room by removing old cultures promptly, keeping the cultures in correct sequence on the shelf according to infestation date, and cleaning the rearing room weekly by use of vacuum cleaner, brushes, and hot water with detergent.
- To discard old cultures, place them in a freezer for 4 days or longer, throw ingredients in a trash can, and wash and store jars for future use.
- Mites, especially *Pymotes* spp., attack weevils, lesser grain borers, and Angoumois grain moths and can completely destroy cultures. Prevent mite infestation of cultures by following these procedures:
  1. Use metal, glass, or plastic shelves in the culture room. Do not use wooden shelves.
  2. Arrange culture jars so that they do not touch each other during the culture period. This prevents migration of mites from one jar to the other.
  3. Coat all shelving surfaces with a light sheen of mineral oil. Mites get stuck in the oil when they try to migrate between jars.
  4. Take the old cultures out of the culture room. Mites tend to build up in cultures as they age. Insects that have passed an optimum age are of little use in testing. Use some of the insects to start new cultures, freeze the old ones, carry off the old media, and clean the glassware as soon as possible.

Preparation of the culture medium

Weevils are cultured on brown rice.

Steps	Key points
1. Preparing the culture medium	• Place 0.5 liter of brown rice in a 1-liter jar.

Steps	Key points
2. Equilibrating temperature	• Bring media which have been in cold storage to a room temperature of 22-28° C.
3. Adjusting moisture	• Check moisture with a moisture tester and, if necessary, temper medium to 12.5 to 13% moisture content. If moisture is too high, dry in an oven. • If moisture is too low, temper by adding water. To determine the amount of water required, compute as follows: — Problem: Weight of rice = 20 kg Present moisture = 11% Desired moisture = 13% — Formula: $\text{Wt of water needed} = \frac{\text{Desired \% moisture} - \text{present \% moisture}}{100 - \text{desired \% moisture}} \times \text{wt of rice}$ $= \frac{13 - 11}{100 - 13} \times 20 \text{ kg}$ $= 0.460 \text{ kg}$ $= 460 \text{ g}$ — Mix half of 460 ml distilled water with half of the rice. Then add the rest of the rice and mix with the remaining water. — Seal the container and shake to mix thoroughly. — Store sealed container at room temperature for at least 7 days.

Starting the stock weevil culture

Collect weevils and place them on the medium to produce progeny to serve as stock cultures to start the rearing program.

Steps	Key points
1. Collecting weevils	• Collect weevil adults from infested grains or grain products in storage by sieving the infested grains through a series of #10 and #20 US standard sieves (Fig. 1). An aspirator made of a shell vial, rubber stopper, and bent glass tubing connected to a small vacuum pump can also be used for handling adults in rearing and screening. Use the #10 sieve to separate the insects from the old medium and #20 to separate them from the fine dust.

Steps	Key points
2. Infesting media	• Place 5 cc of the rice or maize weevils or 8 cc of the granary weevils in each of 2 jars with fresh medium for oviposition. Sieve and measure the adults quickly because the weevils are active and move rapidly. • Clean sieves between use to avoid contamination. • After 1 week of oviposition, remove the adults from the medium with the use of a #10 sieve. Discard the adults, return the medium to the respective jars, and keep in the rearing room until adult weevils emerge. • Emergence occurs about 6 weeks after oviposition for the rice and maize weevil and 7 weeks for the granary weevil. Remove adults on separate days for each species to avoid cross contamination of species. • Use the new adults as the stock culture in the mass rearing program.

Starting the rearing program

Adult weevils obtained from the stock culture are used to begin the rearing program.

Steps	Key points
1. Preparing culture jars	• Place about 500 cc of medium in each of the desired number of culture jars (1-liter jar) for each species. • The number of culture jars depends on the quantity of insects needed. — Computations are based on the reproductive potential of each species:

Species	*Quantity of parent stock*	*Reproductive potential (no. of progeny)*
Rice weevil	1 cc (about 110 pairs)	400-500
Maize weevil	1 cc (about 85 pairs)	300-450
Granary weevil	1 cc (about 85 pairs)	225-275

2. Collecting adults	• Collect adults from the stock culture jars by sieving through #10 and #20 US standard sieves.

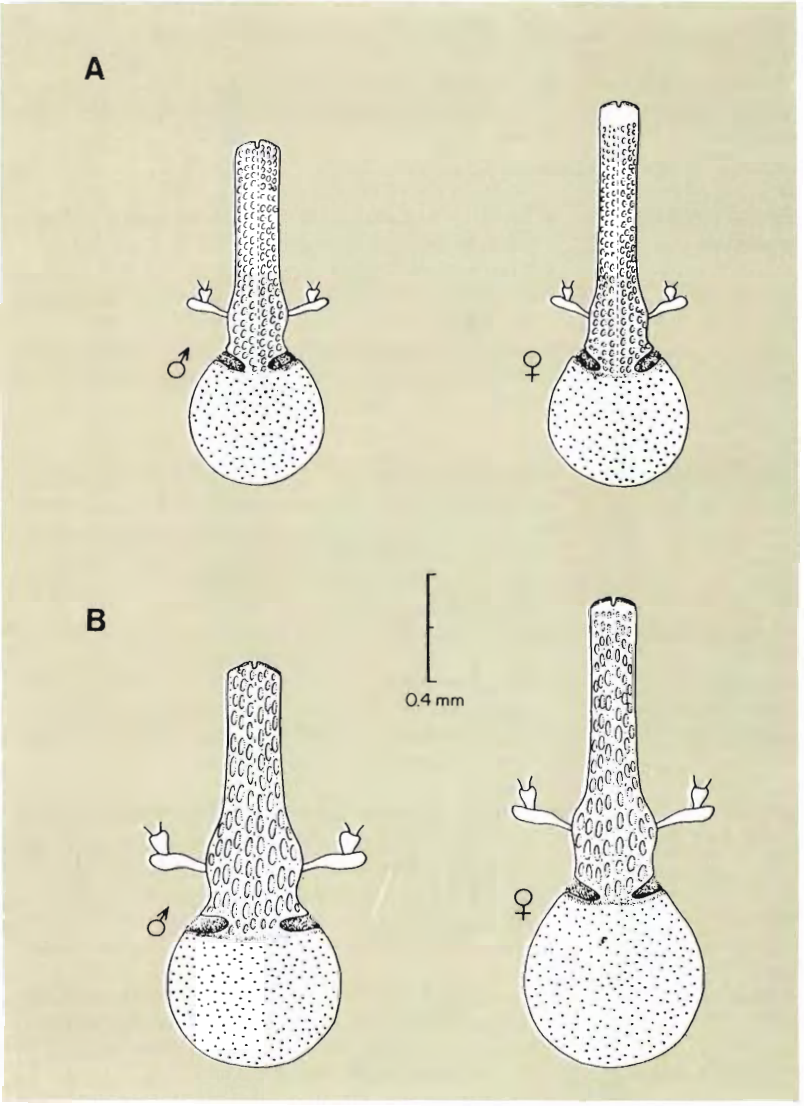
Steps	Key points
3. Infesting the culture medium and placing it in the rearing room	• Place 5 cc of the rice or maize weevil adults or 8 cc of the granary weevil in each of the desired number of culture jars with fresh medium intended for each species. Close the jars with their 60-mesh screened metal lids. Label these jars I and place them in the rearing room.-
4. Transferring adults to fresh medium	• After allowing weevils to oviposit for 1 week, remove the adults from jar I using a #10 sieve. Return the medium to the respective jars. • Transfer the adults to a second set of jars with fresh medium. Label these jars II indicating that the adults are used to set up two series of cultures. If more insects are needed, after allowing the adults to oviposit for 1 week in jar II, transfer the adults from jar II to a third set of jars and label these jars III. Otherwise, discard the adults after removing from jar II.
5. Keeping the cultures in the rearing room	• Arrange the culture jars on rearing shelves according to infestation date. • Adult rice and maize weevils emerge 6 weeks after oviposition, adult granary weevils emerge 7 weeks after oviposition.
6. Transferring new adults to fresh medium to maintain the culture or to use it as a source of test insects	• Transfer the new adults to fresh medium and repeat the process to maintain the culture. Weevils not used to maintain cultures are used in the screening program and for studies on mechanisms of resistance.

SCREENING METHODS

Rice varieties are screened for weevil resistance in a rearing room. The relative levels of resistance are assessed using degree of grain damage and number of weevils produced as criteria. The screening process consists of six steps as described by Cogburn (1974).

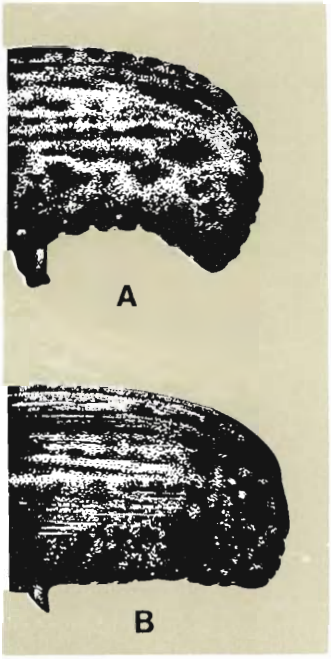
Steps	Key points
1. Collecting test materials and check varieties	• Obtain seed of the test materials. Include a susceptible check and CI 12273 (available from USDA World Collection) as the resistant check. • Freeze the seed for at least 7 days to eliminate mites and insects.

Steps	Key points
2. Preparing jars containing test materials	• Use small plastic containers provided with screen-covered air holes in the lid to evaluate test entries (Fig. 1).
3. Preparing test materials	• Remove test materials from the freezer and bring them to a temperature of 27 ± 2°C for 7 days. • Check moisture content and bring to 12.5-13% as described in step 3 in *Preparation of the culture medium.* • Weigh 30-g samples of each test variety and place in the plastic container. Replicate at least four times. Label each container.
4. Infesting test materials	• Infest samples with 15 pairs (male and female) of 2-week-old adults obtained from the laboratory culture. The sex of the rice and granary weevils can be determined by the length of their rostra and distinctness of pits on the rostra (Fig. 2) and shape of the abdominal tip (Fig. 3).
5. Maintaining test materials in the rearing room	• Place infested seed samples on shelves in a rearing room held at preferably 27 ± 2°C and 65 ± 5% RH and 12:12 h light and dark periods. Arrange them on shelves in a randomized complete block design. • One week after infestation, remove the old adults from the seed samples with an aspirator or by sieving the medium through a #10 and #20 sieve as described in *Rearing*. Discard the adults and put the samples in their respective jars.
6. Evaluating	• Count and remove new adults in each container every day from the start of adult emergence until it stops. Compare the varieties for the number of insects produced on each. • Determine weight and number of damaged and undamaged grains in each sample. Compute percentage of grain weight loss (Adams and Schulten 1978). $\%\ \text{net loss} = \frac{(UNd) - (DNu)}{U(Nd + Nu)} \times 100$ where: U = weight of undamaged grains, Nu = no. of undamaged grains, D = weight of damaged grains, and Nd = no. of damaged grains.



2. Rostra of male and female weevils. A: the rice weevil *Sitophilus oryzae*. B: the granary weevil *S. granarius* (Reddy 1951).

3. Sexual differences in the granary weevil *Sitophilus granarius*. Compare the lateral aspect of the abdominal tip of the male (A) with that of the female (B). Sexual differences in the rice weevil *S. oryzae* are similar (Qureshi 1963).



MECHANISMS OF RESISTANCE

Varieties selected as resistant in the screening test can be further evaluated by determining the mechanisms of resistance involved. There are methods for determining the relative importance of antixenosis and antibiosis in resistance of rough rice to weevils. The methods involve studies on antixenosis for feeding and oviposition and antibiosis studies based on population development on test entries. These studies confirm the resistance of test entries as recorded in the screening tests.

Antixenosis for feeding and oviposition

Steps	Key points
1. Preparing test materials	• Obtain seed of the test varieties. Include a susceptible and a resistant check if available. • Place the seed in a freezer for 7 days to destroy mites and insects. • Remove the seed from the freezer, seal the containers, and bring them to room temperature. • Temper the seed samples to test conditions. Keep them in the room where the test is to be conducted for at least 2 weeks to equilibrate moisture content of the seeds before testing.
2. Preparing test containers and cages	• Prepare shallow containers like petri dishes to hold the seed samples. Screen cages (exposure cages) are also needed when exposing the samples to the insects. • The number of petri dishes and exposure cages depends on the number of test varieties and the replications to be used.
3. Setting up the test	• Place 30 g of the tempered seed samples of each of the test varieties in the petri dishes. Have at least six replications per variety. Label each dish. • Arrange the dishes containing the samples randomly and equidistantly in a circle inside the screen cage.
4. Infesting the samples	• For every 10 entries, release about 1,000 2- to 3-week-old adults, which have been starved overnight. Release them at the center of the circle in the cage to give them free access to the test samples.
5. Evaluating	• Seven days after releasing the insects, cover the petri dishes to prevent escape of insects. Remove the samples from the exposure cage. • Count the insects in each sample and remove them from the samples. Record the sex of the insects as they are counted. This will indicate the number of weevils attracted to the samples either for feeding or oviposition or both. — To estimate the oviposition rate, culture the egg-infested grain samples and count the progeny produced on each entry.

Antibiosis

Relative levels of antibiosis are determined by measuring the weight of weevils and number of weevils (population growth) produced and the index of susceptibility on the test entries.

Steps	Key points
1. Preparing test materials	• Prepare test materials as in the antixenosis test.
2. Setting up the test	• Place 250 g of the tempered samples in each 1-liter jar with fine mesh brass or nylon screen covers. • Replicate each entry six times.
3. Infesting the samples	• Introduce 50 pairs of 2- to 3-week-old adults from laboratory cultures into each jar.
4. Discarding old adults	• Seven days after infestation, remove the adults from the samples. • Store the samples in tightly covered jars until the new adults emerge.
5. Evaluating	• Count and remove emerging adults from the samples daily from start of emergence until it ceases. • Weigh the insects at every counting time and when emergence has been completed compare total weight of insects among varieties. • Determine the population growth on each test entry and compare entries. • Determine the index of susceptibility (Dobie 1977): $\frac{\text{Natural log F}}{\text{D}} \times 100$ where: F = total no. of insects counted, and D = mean development period in days. • Compare the index of susceptibility among entries. The higher the index the more susceptible the variety.

SOURCES OF RESISTANCE

No sources of resistance have been reported for the maize or granary weevil. The following sources reportedly have low levels of resistance to the rice weevil but the resistance, according to Cogburn (USDA, Beaumont, Texas, pers. comm., 1982), is not sufficient to alleviate infestation problems.

Variety	Reference
63-83	Virmani (1980)
CI 12273	Cogburn and Bollich (1979)
Dawn	Cogburn (1977)

Variety	Reference
Gissi 27	Virmani (1980)
IR5	Virmani (1980)
IR1754-55-B-23	Virmani (1980)
IRAT13	Virmani (1980)
Lebonnet	Cogburn (1977)
Lac 23	Virmani (1980)
M55	Virmani (1980)
Nato	Cogburn (1977)
OS6	Virmani (1980)
Suakoko	Virmani (1980)

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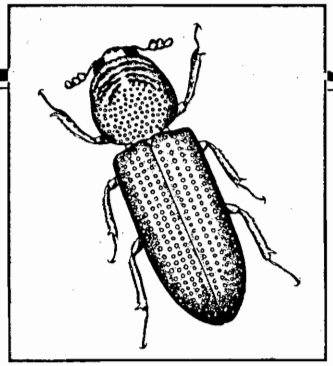


Chapter 26
LESSER GRAIN BORER

The lesser grain borer *Rhyzopertha dominica* attacks both rough and milled rice. Both the larvae and the adults bore into grains. Rearing and screening techniques are illustrated in Figure 1.

REARING

Rearing procedures as described by Pedersen et al (1977) and Strong et al (1967) are given.

Preparation of the culture medium

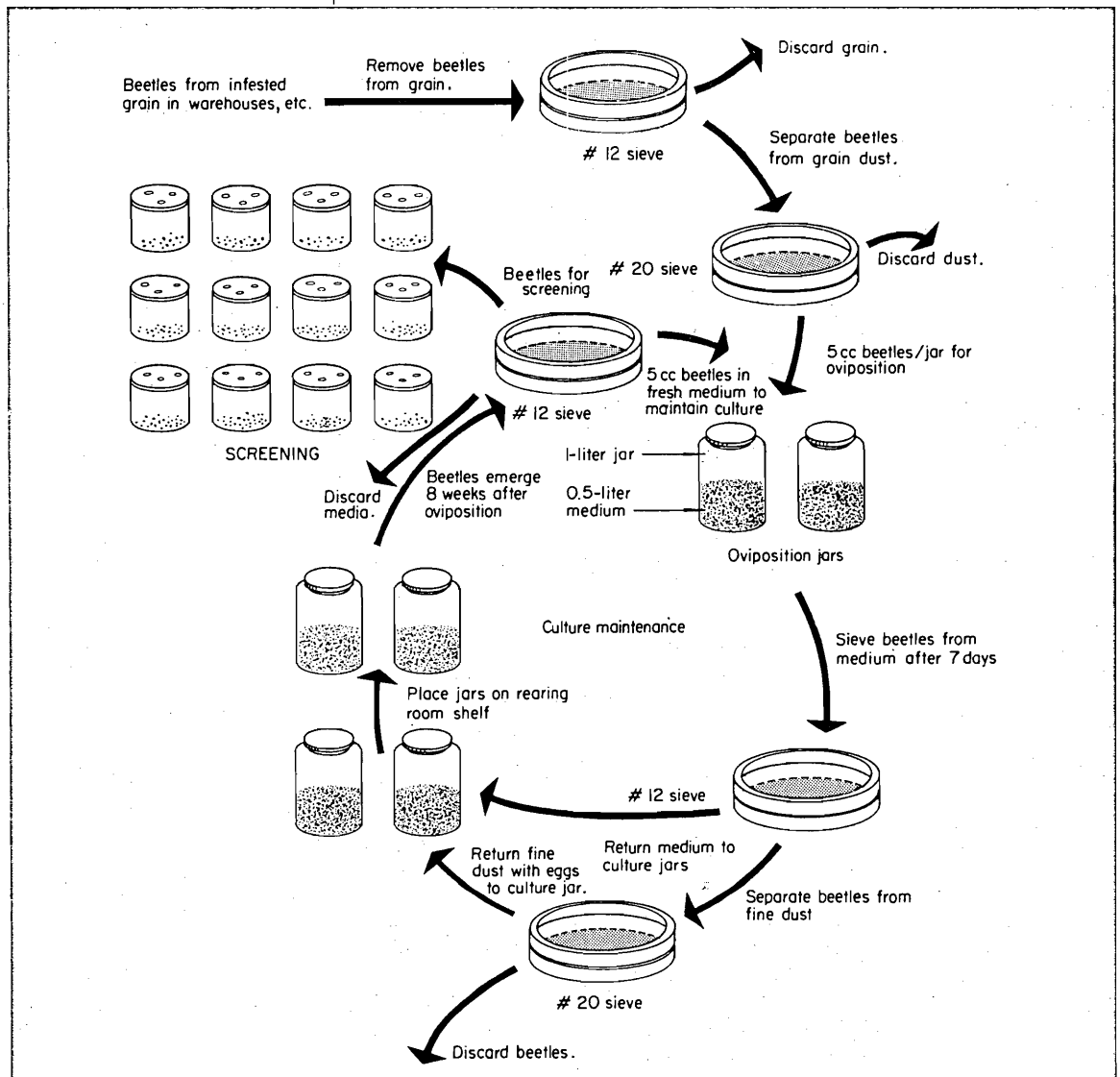
Steps	Key points
1. Placing the culture medium in jars	• Place 0.5 liter of brown rice in a 1-liter jar.
2. Equilibrating temperature	• Bring the medium, which has been in cold storage, to room temperature of 22 to 28°C.
3. Adjusting moisture	• Check moisture with a moisture tester and, if necessary, temper medium to 12.5 to 13% moisture content. — If moisture is too high, dry in an oven. — If moisture is too low, temper by adding water. $\text{Wt of water} = \frac{\text{desired \% moisture} - \text{present \% moisture}}{100 - \text{desired \% moisture}} \times \text{wt of rice}$ — Mix half of the water with half of the rice. Then add the rest of the water and mix with the remaining rice. — Seal the container and mix thoroughly. — Store sealed container at room temperature for at least 7 days.

Starting the stock beetle culture

Steps	Key points
1. Collecting beetles	• Collect adults of the lesser grain borer from infested grains or grain products in storage. Sieve infested grains through a series of #12 and #20 US standard sieves (Fig. 1) to separate adults from

1. Procedure for rearing the lesser grain borer for varietal resistance studies.

Steps	Key points
	the grains, or collect with an aspirator. Use the #12 sieve to separate the insects from the old medium and #20 to separate them from the fine dust.
2. Infesting the medium	• Place 5 cc of the collected adults in a 1-liter jar with 0.5 liter of fresh medium. • Allow the female adults to oviposit for 1 week, then



Steps	Key points
	remove and discard the adults with the #12 and #20 sieves. • Return the medium to the culture jar. Include any fine dust because the eggs are deposited loosely outside the grains. Close each jar with a 60-mesh wire screen lid. • Keep the cultures in the rearing room. • Eight weeks after the initial infestation, new adults emerge. Use these adults as the stock culture for the mass rearing program.

Beginning the rearing program

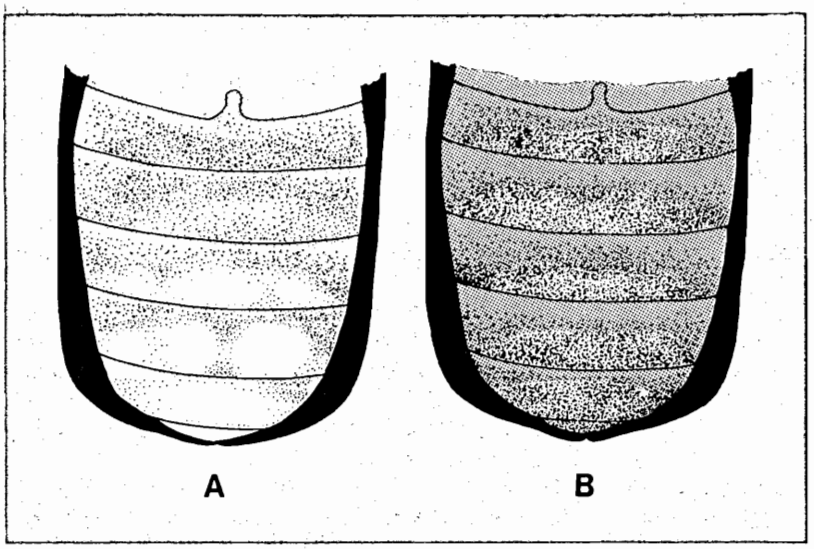
Adult beetles obtained from the stock culture are used to begin the rearing program.

Steps	Key points
1. Preparing the culture jars	• Place 0.5 liter of the medium in each of the desired number of culture jars (1-liter jar). The number of culture jars needed is calculated based on the reproductive potential of 1,000 progeny from 1 cc (about 150 pairs) of parent stock.
2. Collecting adults for oviposition	• Collect adults from the stock culture by sieving or with an aspirator.
3. Infesting the culture medium	• Place 5 cc of adults in each culture jar for oviposition.
4. Discarding the adults	• After 1 week of oviposition, remove the adults by sieving. Return any fine dust or particles to the medium in the culture jar because these may contain insect eggs.
5. Keeping cultures in the rearing room	• Keep the culture jars in the rearing room. • Eight weeks after oviposition, new adults emerge.
6. Transferring new adults to fresh medium to maintain the culture or for use as test insects	• Adults intended for maintaining the culture are transferred to fresh medium and the same rearing process as already described is repeated. Beetles not used to maintain the culture are used as test insects in the screening program and for studies on mechanisms of resistance. • To ensure use of healthy and active adults for tests, place the adults at the center of a petri dish. Cover the dish and select only adults moving to the edge of the dish.

SCREENING METHODS

Screening of rice varieties for lesser grain borer resistance is conducted in a rearing room. The relative levels of resistance are assessed using degree of grain damage and number of beetles produced. The screening process consists of six steps as described by Cogburn (1974).

Steps	Key points
1. Collecting test materials and check varieties	• Obtain seed of the test materials. Include a susceptible check and CI 12273 (available from the USDA World Collection) as the resistant check. • Freeze the seed for at least 7 days to eliminate mites and insects.
2. Preparing jars for test materials	• Use small plastic containers provided with air holes in the lid in screening test entries (Fig. 1).
3. Preparing test materials	• Remove test materials from the freezer and bring them to a temperature of 27 ± 2° C and keep them at that temperature for 7 days. • Check moisture content and bring it to 12.5-13% as described in Step 3, *Preparation of the culture medium.* • Weigh 30-g samples of each test entry and place them in plastic containers. Replicate at least four times. Label each container.
4. Infesting test materials	• Infest seed of each entry with 15 pairs (male and female) of 2-week-old adults obtained from the laboratory culture. Sexes can usually be separated based on the color of the last (5th) ventral abdominal segment. This segment is usually pale yellow in the female (Fig. 2A) and uniformly brown in the male (Fig. 2B).
5. Maintaining test materials in the rearing room	• Place infested seeds on shelves in a rearing room held at preferably 27 ± 2° C and 65 ± 5% RH and 12:12 h light and dark periods. Arrange them on shelves in a randomized complete block design. • One week after infestation, remove the adults from the samples by sieving the medium through a #12 and #20 sieve as described in *Rearing.* Discard the adults and return seed and fine dust with eggs to the jars.
6. Evaluating	• Count and remove adults in each sample every day from the start of adult emergence until it stops. Compare the varieties for the number of insects produced in each.



2. Sexual differences in the lesser grain borer. Venter of the female (A) has pale mottlings on the 3rd and 4th segments and a uniformly pale 5th segment. The male (B) has uniformly darker coloration on all segments (Stemley and Wilbur 1966)

Steps	Key points
	• Determine weight and number of damaged and undamaged grains in each sample (Adams and Schulten 1978). Compute percentage of grain weight loss:

$$\% \text{ net loss} = \frac{(UNd) - (DNu)}{U(Nd + Nu)} \times 100$$

where: U = weight of undamaged grains,
Nu = no. of undamaged grains,
D = weight of damaged grains, and
Nd = no. of damaged grains.

SOURCES OF RESISTANCE

Only limited screening has been conducted and only a few varieties with resistance have been selected. The level of resistance, however, is too low to alleviate infestation problems (R. R. Cogburn, USDA, Beaumont, Texas, pers. comm., 1982).

Variety	Reference
CI 12273	Cogburn and Bollich (1979)
Dawn	Cogburn (1974), Cogburn (1977), McGaughey (1973)
Labelle	McGaughey (1973)
Nato	Cogburn (1977)
PI 160849	Cogburn and Bollich (1979)

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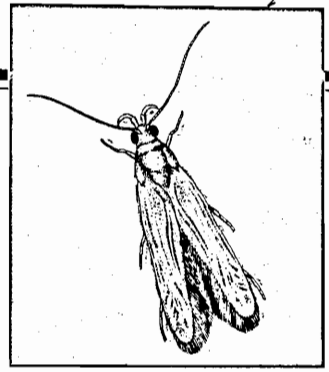


Chapter 27
ANGOUMOIS GRAIN MOTH

The Angoumois grain moth *Sitotroga cerealella* is a major pest of stored rough rice and commonly infests rice in the field before harvest. Eggs are laid on or near grains and the larva bores into the grain and feeds on the contents. The active larval and pupal periods are spent inside the grain.

REARING

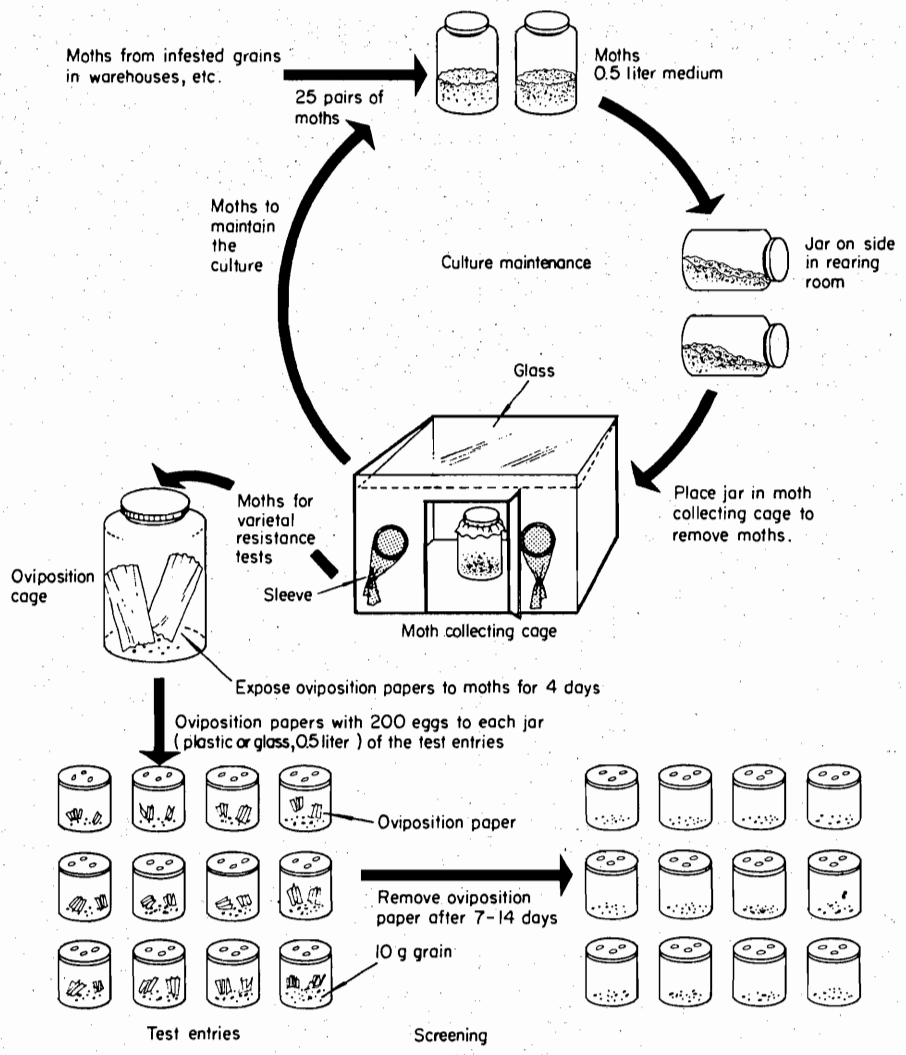
Most of the general methods and procedures used in the rearing program for weevils also apply to the Angoumois grain moth. However, specific procedures differ. The methods used at Kansas State University (Pedersen et al 1977) and the University of California (Strong et al 1967) are given (see Fig. 1).

Preparing the culture medium

Steps	Key points
1. Placing the culture medium in jars	• Place 0.5 liter of rough rice, wheat, or maize kernels in a 1-liter jar.
2. Equilibrating temperature	• Bring the medium, which has been in cold storage, to a room temperature of 22 to 28°C.
3. Adjusting moisture	• Check moisture with a moisture tester and, if necessary, temper medium to 12.5 to 13% moisture content. — If moisture is too high, dry in an oven. — If moisture is too low, temper by adding water. $\text{Wt of water needed} = \frac{\text{Desired \% moisture} - \text{present \% moisture}}{100 - \text{desired \% moisture}} \times \text{wt of rice}$ — Mix half of the water with half of the rice. Then add the rest of the rice and mix with the remaining water. — Seal the container and mix thoroughly. — Store sealed container at room temperature for at least 7 days.

Starting the stock culture

Collect moths and place them on the medium to oviposit and produce progeny for the stock culture used to start the rearing program.



1. Rearing Angoumois grain moths for varietal resistance studies.

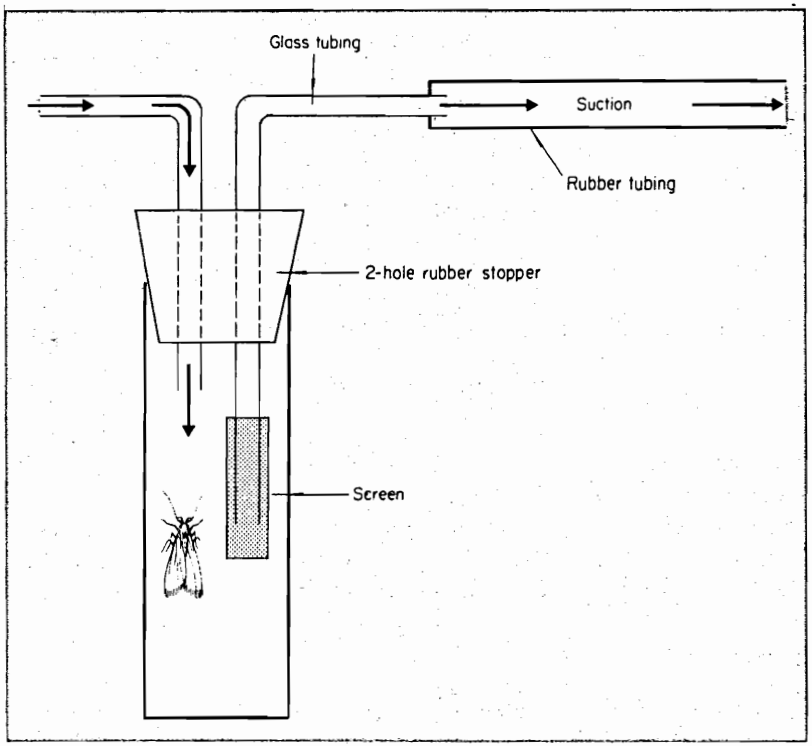
Steps	Key points
1. Collecting moths	• Collect moths from infested grains using an aspirator (Fig. 2) or test tube.
2. Infesting the medium	• Place 25 pairs (male and female) of moths in a 1-liter culture jar containing 0.5-liter of the medium. Females are larger and have a lighter colored

Steps	Key points
	abdomen than the males. Select gravid females with large abdomens. Close the jars with a lid provided with a screen. • Put culture jars in the rearing room. Lay the jars carefully on their sides to expose a larger surface area for oviposition. After 1 week in this position, set the jars upright. The moths die in a few days and are left in the jars. • New moths emerge about 6 weeks after oviposition. Use these moths to start the rearing program.

Beginning the rearing program

Moths obtained from the stock culture are used to begin the rearing program.

Steps	Key points
1. Preparing culture jars	• Place 0.5 liter of the medium in each of the desired number of culture jars. Calculate the number of



2. Aspirator used for collecting Angoumois grain moths. It can also be used for weevils and the lesser grain borer.

Steps	Key points
	jars on this basis: 25 pairs of moths from the stock culture will produce about 400-500 adults/jar.
2. Collecting adults from the stock culture	• Collect newly emerged adults from the stock culture by placing the stock culture jars in a moth-collecting cage or sleeved cage. Before placing the jars in the sleeved cage, tilt the jars to disturb the moths. • Collect moths capable of flying from the jar to the collecting cage walls or top with an aspirator (Fig. 2) or test tube. In collecting females, select those that have a large abdomen and have not yet laid their eggs. • Adults within the culture jar may be anaesthetized with CO_2 (if available) before collection. • Discard the old medium after collecting all adults.
3. Infesting the culture medium and placing it in the rearing room	• Place 25 pairs of moths in each of the culture jars with fresh medium. Close the jars with a metal lid provided with a screen. • Lay the jars on their sides during the first week after initial infestation with parent moths to allow maximum surface area for oviposition. After a week, set the jars upright until new adults emerge. New adults emerge in about 6 weeks.
4. Transferring new adults	• Collect and transfer new moths to fresh medium and repeat the process to maintain the culture. • Moths intended for experimental purposes should be removed from the jars at a designated time so that all will be within the desired age limit. Eggs obtained from the moths are used to infest varieties in the screening program. Both moths and eggs are used in the various studies on mechanisms of resistance.

SCREENING

Resistance to the Angoumois grain moth is evaluated using degree of grain damage and number of progeny produced as criteria. The screening procedure given is based on that described by Cogburn (1977). Many accessions from the USDA world collection with low levels of resistance to the Angoumois grain moth have been identified.

Infesting test materials with eggs

Infesting test materials with eggs consists of seven steps. This method is illustrated in Figure 1.

Steps	Key points
1. Collecting test materials	• Obtain seed of the test entries to be screened. Include Calrose or Belle Patna or any available susceptible check and CI 12273 as the resistant check.
2. Preparing containers for test materials	• Prepare plastic containers (8-cm-diam and 7-cm-high) or 0.5-liter glass jars with screen covers for each sample. The number of jars depends on the number of test varieties and replications.
3. Preparing test materials	• Remove test materials from the freezer and bring to a temperature of 27 ± 2°C. Maintain at that temperature for 7 days. • Check moisture content and bring to 12.5-13% as described in Step 3, *Preparing the culture medium.* • Weigh 10 g of seed of each test entry and place it in the containers. Replicate each variety at least four times.
4. Collecting eggs for infestation	• Expose oviposition papers folded like an accordion to a large number of moths in a glass container for 4 days. • Unfold the papers and cut areas with adhering egg clusters. • Count eggs on each paper with a binocular microscope and record the numbers.
5. Infesting the samples with eggs	• Place pieces of oviposition paper containing 200 eggs in each of the 10-g samples of the test varieties. • Remove the oviposition paper after 7-14 days. Examine eggs and count eggs that hatched. Record the data obtained.
6. Maintaining test materials in the rearing room	• Store the samples in constant environment, preferably at 27 ± 2°C and 65 ± 5% RH and 12:12 h light and dark period, until adult emergence begins.
7. Evaluating	• Count and remove adults daily from start of adult emergence until it ends. • Determine total number of adults that emerge from each sample. • Compute survival percentage: $\%\ \text{survival} = \dfrac{\text{total adults that emerged}}{\text{total larvae that hatched from the 200 eggs placed on each sample}} \times 100$

Steps	Key points
	Determine the susceptibility of varieties using the following scale: *Scale* — *Survival (%)* R — Less than 10 MR — 10-20 S — More than 20 • Determine the weight and number of damaged and undamaged grains in each sample. Calculate percentage of grain weight loss (Adams and Schulten 1978): $\%\ \text{net loss} = \frac{(UNd) - (DNu)}{U(Nd + Nu)} \times 100$ where U = weight of undamaged grains, Nu = no. of undamaged grains, D = weight of damaged grains, and Nd = no. of damaged grains.

MECHANISMS OF RESISTANCE

Entries selected as resistant or moderately resistant in the screening study can be studied further to determine the mechanisms of resistance. An antixenosis test and two antibiosis tests — on survival and population — are described.

Antixenosis for oviposition

Determining antixenosis for oviposition consists of four steps.

Steps	Key points
1. Preparing test materials	• Obtain seed of the test varieties. Include a susceptible and a resistant check, if available.
2. Setting up the test	• Select, at random, 50 tempered grains (see Step 3, *Preparing the culture medium*) of each test variety and place them in plastic petri dishes. Prepare at least six replications. Place the petri dishes inside a glass desiccator in a circular manner, with one replication per desiccator. If only one desiccator is available, replicate the experiment in time. In the absence of a desiccator, use a glass or plastic cage.

Steps	Key points
3. Infesting the samples	• Release 20 pairs (male and female) of moths in the desiccator and leave them overnight (16 h).
4. Evaluating	• The morning after infestation, remove the sample from the desiccator. • Count the eggs on the grains. Check the area where the lemma and palea join because moths frequently deposit clusters of eggs there. • Compare the number of eggs laid among the varieties tested.

Antixenosis for feeding and antibiosis

Russell and Cogburn's method (1977) measures the combined effect of antixenosis for feeding and antibiosis. Antixenosis is caused by the mechanical prevention of entry into the grain because of the hardness of the hulls. Antibiosis is measured by the developmental period of those that do penetrate the grain and survive to become adults.

Steps	Key points
1. Preparing test materials	• Prepare test materials as in the *Antixenosis for oviposition* test.
2. Setting up the test	• Place 25 g tempered grain samples of each variety in 0.5-liter jars. Have at least six replications. Label each jar.
3. Collecting eggs for infestation	• Collect eggs as described in the screening test.
4. Infesting the samples	• Place pieces of oviposition paper containing 200 eggs in each sample jar.
5. Evaluating	• About 7-14 days after infestation, collect the papers from each jar and count the eggs that have hatched to determine the exact number of larvae that entered the samples. The count is used to determine survival and population growth. • When moths begin to emerge, count and record emerging moths daily from each jar until emergence stops. These data are used to determine the mean developmental period. • Determine insect survival percentage: $\%\ \text{survival} = \dfrac{\text{no. of moths counted}}{\text{total number of larvae per sample (those that hatched from the 200 eggs)}} \times 100$

Steps	Key points
	• Calculate the index of susceptibility (Dobie 1977):
	$\text{Index of susceptibility} = \frac{\text{natural log F}}{\text{D}} \times 100$
	where: F = total no. of moths counted, and D = mean developmental period in days.
	• Compare results among varieties.

SOURCES OF RESISTANCE

More sources of resistance in rice to the Angoumois grain moth have been identified than for other rice storage insects. Level of resistance in the varieties, however, is too low to alleviate infestation problems (Cogburn, USDA, Beaumont, Texas, pers. comm., 1982).

Variety	Reference
Balilla, Cesariot	Russell (1976)
CI 7097, CI 12273	Cogburn and Bollich (1979)
Dawn, IR5, Labelle, Peta	Russell (1976)
PI 160849	Cogburn and Bollich (1979)
USDA World Collection no.	
1543-1, 1711-1, 5486, 7024	Russell and Cogburn (1977)
7097, 7098, 7102	Russell and Cogburn (1977), Cogburn (1977)
7103	Russell and Cogburn (1977)
7108, 7111	Cogburn (1977)
7129	Russell and Cogburn (1977), Cogburn (1977)
7367	Russell and Cogburn (1977)
7375	Cogburn (1977)
7389, 7392, 7763-3, 7811	Russell and Cogburn (1977)
8328	Russell and Cogburn (1977), Cogburn (1977)
8329-1	Russell and Cogburn (1977)
8330-1	Russell and Cogburn (1977), Cogburn (1977)
9173, 9233, 9443, 9481, 9482, 9484, 9518, 9521, 9524, 9553	Russell and Cogburn (1977)
12177	Cogburn and Bollich (1979)
12272, 12273, 160412-1	Cogburn (1977)
160432	Russell and Cogburn (1977), Cogburn (1977)
160443, 160448, 160461	Russell and Cogburn (1977)
160469	Russell and Cogburn (1977), Cogburn (1977)
160481, 160516, 160548, 160554	Russell and Cogburn (1977)
160557	Russell and Cogburn (1977), Cogburn (1977)

Variety	Reference
160573, 160577, 160583, 160584, 160585, 160589, 160592, 160601, 160602, 160609, 160611, 160613, 160618, 160620, 160621	Russell and Cogburn (1977)
160622	Russell and Cogburn (1977), Cogburn (1977)
160623, 160630, 160636	Russell and Cogburn (1977)
160638, 160639	Russell and Cogburn (1977), Cogburn (1977)
160640	Russell and Cogburn (1977)
160641, 160642	Russell and Cogburn (1977), Cogburn (1977)
160645	Russell and Cogburn (1977)
160662	Russell and Cogburn (1977), Cogburn (1977)
160663, 160665, 160675, 160667 160683, 160699, 160714, 160716 160724, 160725, 160726, 160729 160730, 166731, 160735	Russell and Cogburn (1977)
160739	Russell and Cogburn (1977), Cogburn (1977)
160746, 160764, 160764-3, 160783	Russell and Cogburn (1977)
160784	Russell and Cogburn (1977), Cogburn (1977)
160788, 160791, 160800	Russell and Cogburn (1977)
160849	Russell and Cogburn (1977), Cogburn (1977)
160974, 160975, 161003, 161005, 161018	Russell and Cogburn (1977)
161019	Russell and Cogburn (1977), Cogburn (1977)
161023, 161035, 161041, 161042, 338956	Russell and Cogburn (1977)

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APPENDICES

Appendix 1. National or international centers with rice germplasm collections.[a]

Brazil	Centro Nacional de Recursos Geneticos (CENARGEN) C. P. 10.2372, CEP 70.000 Brasilia, D. F.
Bangladesh	Bangladesh Rice Research Institute (BRRI) Joydebpur Dhaka-2
Burma	Agricultural Research Institute (ARI) Gyogon, Insein Road Rangoon
China	1) Crop Germplasm Resources Institute (CGRI) Chinese Academy of Agricultural Sciences Beijing 2) Taiwan Agricultural Research Institute (TARI) 189 Chung-Cheng Road Wan-feng, Wufung Taichung, Taiwan-431
France	IRAT - Département d'Amélioration des Plantes Avenue du Val de Montferrand GERDAT - B. P. 5035 34032 Montpellier Cedex
India	1) National Bureau of Plant Genetic Resources (NBPGR) IARI Campus New Delhi - 110012 2) Central Rice Research Institute (CRRI) Cuttack, Orissa-753006 3) All-India Coordinated Rice Improvement Project (AICRIP), Rajendranagar Hyderabad, A. P. - 500 030
Indonesia	Central Research Institute for Food Crops (CRIFC) Jalan Merdeka 99 Bogor
Japan	National Institute of Agricultural Sciences (NIAS) 3-1-1 Kannondai, Yatabe-cho Tsukuba-gun, Ibaraki-Pref. - 305
Korea (Rep. of)	Office of Rural Development (ORD) Suweon
Malaysia	Malaysian Agricultural Research and Development Institute (MARDI) Rice Research Center Bumbong Lima, Seberang Perai
Nepal	National Rice Improvement Program (NRIP) Parwanipur Agriculture Station Birganj, Narayani Zone
Pakistan	Pakistan Agricultural Research Council (PARC) P. O. Box 1031 Islamabad
Philippines	International Rice Research Institute (IRRI) P. O. Box 933, Manila
Sri Lanka	Department of Agriculture Peradeniya

Thailand	Rice Research Institute (RRI) Department of Agriculture Bangkhen, Bangkok-9
USA	Plant Genetics & Germplasm Institute, USDA Bldg. 046, BARC-West Beltsville, Maryland 20705
USSR	All-Union Rice Research Institute (AURRI) P. O. Box 353204 P. O. Belozernoe Krasnodar
Vietnam	Vietnam Central Institute for Agricultural Sciences Ministry of Agriculture Hanoi
West Africa	1) West Africa Rice Development Association (WARDA) P. O. Box 1019 Monrovia, Liberia
	2) International Institute of Tropical Agriculture (IITA) P. O. Box 5320 Ibadan, Nigeria
	3) Institut des Savanes (IDESSA) Department Cultures Vivrieres B. P. 635, Bouake, Ivory Coast

[a]Prepared by Dr. T. T. Chang, head, International Rice Germplasm Center, IRRI.

Appendix 2. Breeding codes used at various rice breeding stations.

Code	Meaning	Station where code is used
A	Araure	Estacion Experimental Araure, Apdo. Postal 102, Araure, Estado Portuguesa, Venezuela
AC	Accession	Central Rice Research Institute Cuttack, India
ADR	Adoor	Tamil Nadu Agricultural University Coimbatore, India
ADT	Aduthurai	Tamil Nadu Rice Research Institute, Aduthurai, Tamil Nadu, India
AKP	Anakapalli	Anakapalli Rice Experiment Station Anakapalli, Andhra Pradesh, India
ARC	Assam Rice Collection	Indian Council for Agricultural Research for Hilly Regions, Shillong, India
ASD	Ambasamudram	Paddy Experiment Station Ambasamudram, Paddy Tamil Nadu, India
AT	Amballantota	Amballantota Rice Research Station Amballantota, Sri Lanka
B	Bogor	BORIF, Bogor, Indonesia
BAM	Berhampur	Berhampur Rice Research Station Berhampur, Orissa, India
BCP	Buchireddipalem	Buchireddipalem Rice Experiment Station Andhra Pradesh, India
BG	Batalagoda	Central Rice Breeding Station, Ibbagamuwa, Batalagoda, Sri Lanka
BJ	Bhogsira	Rice Research Station Chinsurah, West Bengal, India
BKN	Bangkhen	Department of Agriculture, Rice Division Bangkhen, Bangkok, Thailand
Bmt	Beaumont	USDA Plant Genetics and Germplasm Institute Beltsville, Maryland, USA
BPI	Bureau of Plant Industry	Bureau of Plant Industry Philippines

Code	Meaning	Station where code is used
BR	Bangladesh Rice	Bangladesh Rice Research Institute Joydebpur, Dhaka
BRB	Bangladesh Rice Barisal	Bangladesh Rice Research Institute Barisal Station
BRC	Bangladesh Rice Comilla	Bangladesh Rice Research Institute, Comilla
BW	Bombuwela	Bombuwela Rice Research Station Bombuwela, Sri Lanka
C	College	University of the Philippines at Los Baños, Laguna, Philippines
CI	Cereal Introduction	USDA Plant Genetics and Germplasm Institute Beltsville, Maryland, USA
CIAT	Centro Internacional de Agricultura Tropical	CIAT, Apartado Aereo 67-13, Cali, Colombia
CICA	Centro Internacional de Agricultura Tropical-Instituto Colombiano Agropecuaria	CIAT, Apartado Aereo 67-13, Cali, Colombia
CG	Casamance Glaberima	Institut de Recherches Agronomiques Tropicales et des Cultures Vivrières Station de Bouaké, Ivory Coast
CNT	Chainat	Rice Experiment Station, Chainat, Thailand
CO	Coimbatore	Paddy Breeding Station, Tamil Nadu Agricultural University, Coimbatore, India
CP	Cuba Los Palacios	Rice Research Station, Los Palacios Pinal de Rio Province, Cuba
CR	Central Rice	Central Rice Research Institute Cuttack, India
CSC	Central Salinity Center	Central Salinity Center Karnal, Haryana, India
Ctg	Chittagong	Bangladesh Rice Research Institute Chittagong, Bangladesh
DA	Department of Agriculture	Instituto Agronomico, Campinas São Paulo, Brazil
DA	Dacca	A term used during British rule for lines from the Dhaka Research Station, Dhaka, Bangladesh
DI	Dalua Introduction	Andhra Pradesh, India
DR	Dokri	Dokri Rice Research Institute Dokri, Sind, Pakistan
DW	Deepwater	Chinsurah Rice Research Station West Bengal, India
ECIA	Estacion Central Investigaciones del Arroz	Estacion Central Investigaciones del Arroz Apartado No. 1, Bauta, Havana, Cuba
FR	Flood resistant	Orissa, India
GEB	Government Economic Botanist	Tamil Nadu Agricultural University Coimbatore, India
GZ	Giza	Agricultural Research Center, Giza, Egypt
H	Hybrid	Central Research Institute Batalagoda, Sri Lanka
HBJ	Habiganj	Habiganj Substation, Bangladesh Rice Research Institute
HPU	Himachal Pradesh University	Himachal Pradesh University Palampur, India
HR	Himayatsagar	Agricultural Research Institute Himayatsagar, Hyderabad, Andhra Pradesh, India
HR	Honam Rice	Honam Crop Experiment Station, Iri, Korea
IAC	Instituto Agronomico Campinas	Instituto Agronomico, Campinas, São Paulo, Brazil

Code	Meaning	Station where code is used
IARI	Indian Agricultural Research Institute	IARI, New Delhi, India
ICA	Instituto Colombiano Agropecuaria	Instituto Colombiano Agropecuaria, La Libertad Villavicencio, Colombia
IET	Initial Evaluation Trial	AICRIP, Hyderabad, India
IG	Ivory Coast Glaberima	Bouaké, Ivory Coast
IR	International Rice	International Rice Research Institute Los Baños, Laguna, Philippines
IRAM	Instituto Reserche Agricultura Madagascar	Instituto Reserche Agricultura Tananarive, Madagascar
IRAT	Institut de Recherches Agronomiques Tropicales	Institut des Gavanes/Gerdat, IRAT Research Station, B. P. No. 365 Bouaké, Ivory Coast
ITA	Institute Tropical Agriculture	International Institute of Tropical Agriculture, Ibadan, Nigeria
J	Jeypore	Rice Research Station (OUAT) Jeypore, Orissa, India
JBS	Jeypore Botanical Survey	Collection from Jeypore, India
JKW	Jhona Kasar Wala	Rice Research Station Kala Shah Kaku, Pakistan
K	Karjut	Karjut, Maharastra, India
K	Kashmir	Khudwani, Kashmir, India
KMP	Karnataka Mandya Paddy	University of Agricultural Sciences Mandya, Karnataka, India
Kn	Kuningan	Kuningan, Branch Station of Sukamandi Rice Research Station Cirebom, Indonesia
L	Lua (paddy)	Can Tho University Can Tho, Vietnam
LAC	Liberian Agricultural Company	Liberian Agricultural Company Central Agricultural Research Institute Suakoko, Bong Country, Liberia
LS	Liberia Suakoko	Central Agricultural Research Institute Suakoko, Bong Country, Liberia
M	Maros	Maros Agricultural Research Institute Maros, Sulawesi, Indonesia
MCM	Muchilipatnam	Muchilipatnam Andhra Pradesh, India
MG	Mali Glaberima	Rice Research Station Mopti, Mali
MGL	Mangalore	Agricultural Research Station Mangalore, Karnataka, India
MI	Mahaillupalama	Agricultural Research Station Mahaillupalama, Sri Lanka
MR	Mandya Rice	University of Agricultural Sciences Mandya, Karnataka, India
MR	MARDI Rice	MARDI, Kuala Lumpur, Malaysia
MRC	Maligaya Research Center	Maligaya Rice Research and Training Center Muños, Nueva Ecija, Philippines
MTL	Mien Tay Lua	Can Tho University Can Tho, Vietnam
MTU	Maruteru	Agricultural Research Station (APAU) Maruteru, Andhra Pradesh, India
N	Nagina	Nagina Rice Research Station Nagina, Uttar Pradesh, India
NC	New Collection	Chinsurah Rice Research Station Chinsurah West Bengal, India

Code	Meaning	Station where code is used
NCS	New Chinsurah Collection	Chinsurah Rice Research Station Chinsurah West Bengal, India
OB	Observation (cross from India given as OB codes in Sri Lanka)	Central Rice Breeding Station Ibbagamuwa, Batalagoda, Sri Lanka
OC	Old Collection	Chinsurah Rice Research Station Chinsurah, West Bengal, India
OM	Ô Môn	Cui Long Delta Agricultural Technology Center Vietnam
OR	Orissa Rice	Orissa University of Agriculture and Technology Bhubaneswar, Orissa, India
OS	*Oryza sativa*	Federal Department of Agriculture Nigeria
P	Pakistan	Agricultural Research Council Islamabad, Pakistan
P	Palmira	Palmira, Colombia
PARC	Philippine Atomic Research Center	Philippine Atomic Research Center Manila, Philippines
PD	Peradeniya	Central Agricultural Research Institute Peradeniya, Sri Lanka
PDR	Pakistan Dokri	Dokri Rice Research Institute. Dokri, Sind, Pakistan
PI	Plant Introduction	Plant Genetics and Germplasm Institute USDA, Beltsville, Maryland, USA
PK	Pakistan	Kala Shah Kaku Rice Research Institute Kalashakaku, Punjab, Pakistan
PL	Pussallawa	Pussallawa Rice Research Station Pussallawa, Sri Lanka
PNAP	Programa Nacional del Arroz de Peru	Programa Nacional del Arroz de Peru, Centro de Investigacion Promocion Agraria, Visto Florida, Chiclayo, Peru
PR	Punjab Rice	Regional Rice Research Station Kapurthala, Punjab, India
Ptb	Pattambi	Central Rice Research Station Pattambi, Kerala, India
PVR	Peravurani	Rice Research Station Peravurani, Tamil Nadu, India
PY	Pondicherry	Farm Science Center, Pondicherry
R	Raipur	Rice Research Station Raipur, Madhya Pradesh, India
RAU	Rajendra Agricultural University	Rajendra Agricultural University Pusa, Bihar, India
RDR	Rudrur	Regional Agricultural Research Station (APAU), Rudrur, Andhra Pradesh, India
RP	Rice Project	All India Coordinated Rice Improvement Project Hyderabad, Andhra Pradesh, India
RS	Rio Grande do Sul	Instituto Rio Grandense do Arroz Porto Alegre, RGS, Brazil
SML	Stichting Mechanische Landbouw	Stichting Mechanische Landbouw Agriculture Foundation New Nicherie, Surinam
SLO	Samalkot	Rice Research Station Samalkot, Andhra Pradesh, India
SR	Suweon Rice	Suweon Crop Experiment Station Suweon, Korea

Code	Meaning	Station where code is used
T	Type	Cuttack Rice Research Station Cuttack, Orissa, India (breeding lines developed before foundation of CRRI)
TCA	Tirhut College of Agriculture	Tirhut College of Agriculture Pusa, Bihar, India
TKM	Tirurkuppam	Paddy Experiment Station Tirurkuppam, Tamil Nadu, India
TNAU	Tamil Nadu Agricultural University	Tamil Nadu Agricultural University Coimbatore, India
TNR	Thalaignayar Rice	Thalaignayar, Tamil Nadu, India
TOS	Tropical Oryza Selection	International Institute of Tropical Agriculture, Ibadan, Nigeria
TOX	Tropical Oryza Cross	International Institute of Tropical Agriculture, Ibadan, Nigeria
TR	Trombay	Bhabha Atomic Energy Research Center Trombay, Maharashtra, India
UPL	University of the Philippines at Los Baños	University of the Philippines at Los Baños, Laguna, Philippines
UPR	Uttar Pradesh Rice	G. B. Pant University of Agriculture and Technology Pantnagar, Uttar Pradesh, India
W	Warangal	Agricultural Research Station APAU, Warangal, Andhra Pradesh, India
WC	Warangal culture	Agricultural Research Station APAU, Warangal, Andhra Pradesh, India
YN	Yezin	Agricultural Research Institute Yezin, Burma
YR	Yungnam rice	Yungnam Crop Experiment Station Milyang, Korea

Appendix 3. Suggested references on insect resistance.

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GLOSSARY

Accession — a variety or strain or a bulk population registered at a natural center and worth conservation. Two or more morphologically or ecologically different accessions may have the same name.

Adult — the mature stage. It comes after the nymphal stage in insects that have incomplete or no metamorphosis, and after the pupal stage in insects that have complete metamorphosis.

Adult plant resistance — resistance manifested mainly in maturing plants and less apparent in the seedling stage.

Anaesthetize — to immobilize an insect by exposing it to carbon dioxide.

Antibiosis — a component of varietal resistance: insects do not grow, survive, or reproduce well because of a toxic or other direct detrimental effect of the host plant.

Antixenosis — a term proposed by Kogan and Ortman to replace *nonpreference*. It conveys the idea that the plant is avoided as a *bad host*.

Armyworm — the larva of the family Noctuidae which often travels in large populations from field to field.

Arthropod — animals in the Arthropoda, a phylum consisting of animals with jointed limbs, eg, the Insecta (insects) and Arachnida (mites and spiders).

Artificial diet — food source, other than the natural host plant, consisting of amounts of various components which are mixed and provided to insects in the rearing program. The diet may be holidic (chemically pure), meridic (one or more chemically undefined materials such as wheatgerm), or xenic (host plant materials plus supplemental nutrients).

Artificial inoculation — infesting plants with insects by placing the insects on or near the plant, with an instrument such as the Davis inoculator.

Auricle — a pair of small ear-like appendages borne at the base of the leaf blade and usually arising at the sides where the ligule and the base of the collar are joined.

Autoclave — an airtight chamber filled with superheated steam under pressure, which is used for sterilization.

Avirulent gene — a gene in an insect which is unable to break down the gene for insect resistance in a plant.

Backcross — a breeding method in which a desired character such as insect resistance is transferred into an improved variety by the repeated use of the variety carrying it as a recurrent parent to reinforce or increase the gene frequency of the character.

Basal application — broadcast application of fertilizer and pesticides to the paddy before the last harrowing during land preparation.

Basic studies — studies dealing with the mechanisms of resistance in contrast to screening studies to identify resistant sources.

Batch — a quantity of material destined for one test.

Biotype — a population of insects that are capable of surviving and damaging varieties that are resistant to other populations of the same insect species.

Blackhead stage — stage in the development of insect eggs wherein the head can be observed as a black spot through the chorion.

Blanch — to plunge into boiling water and then into cold water to remove the color.

Blend — to mix thoroughly.

Bloodworm — the red aquatic larva of dipterous flies of the genus *Chironomus*.

Booting stage — the reproductive phase of rice growth and development when the developing panicle causes a swelling of the culm. The swollen area is referred to as the *boot*.

Border row — a row of plants around test plots which protect test entries from a *border row effect*, such as greater than normal growth due to lack of competition for nutrients and light, lower insect populations than on plants surrounded by other plants, etc.

Bore — to make a hole or tunnel by the feeding action of an insect.

Breakdown of resistance — the inability to maintain resistance when attacked by a newly selected insect biotype that has a gene for virulence at every locus corresponding to a gene for resistance in the host.

Breeding line — a genetic line bred in a crossing program, before it is named and officially released for commercial cultivation.

Broadcast — to apply insecticide granules by hand or machine over a surface area. To spread randomly.

Bromocresol green — a brominated dye which acts as a pH indicator. In studies to determine the feeding activity of hoppers, bromocresol green-treated filter paper reacts by changing color when it comes in contact with honeydew.
Brood — a generation of insects from the egg to the adult stage and back to the egg stage.
Bug — an insect of the order Hemiptera.
Cage — an enclosure for confining plants and insects.
Calibrate — to set the capacity of an instrument such as the Davis inoculator so that it delivers a specific number of insects per discharge.
Camel hair brush — a slender, pointed, soft-haired brush used for handling small lepidopterous larvae.
Caseworm — insect of the species *Nymphula depunctalis* whose larva lives in a case consisting of a piece of rolled rice leaf.
Clip leaves — to cut leaves with scissors or shears.
Concrete bed — a plant bed, usually in a screenhouse, which has concrete sides.
Core sampler — a cylindrical device open at both ends, used to remove a core of soil when sampling for soil insects such as rice water weevil larvae.
Correlation — a mutual relationship between two things such that an increase or decrease of one is generally associated with an increase or decrease of the other. Linear correlation is measured by the correlation coefficient (r), which may range from -1 to $+1$.
Corrugated paper — a thick, coarse paper with a ridged or furrowed surface, such as that used in cardboard box construction.
Cross contamination — in insect rearing, a situation where a culture of one insect species is infested with insects of another species.
Culture — the rearing of organisms such as insects to serve as a source for varietal resistance studies.
Culture maintenance — in an insect rearing program, the activities involved in maintaining the insect culture in contrast to those activities involved in rearing test insects for use in the screening program.
Culture medium — in insect rearing, a food source for the insects.
Damage (plant) — destruction or loss in value caused by feeding activity of insects and rats, or by disease infection.
Davis inoculator — a mechanical device developed by Frank Davis, Mississippi State University, USA, used to dispense insect larvae in artificial inoculation of plants.
Deadheart — dead rice tiller, the result of the attack of stem borer which cuts the tiller's base.
Defoliator — any chewing insect that feeds on the leaves of plants and removes foliage.
Desiccator — a glass jar, fitted with an airtight cover, containing some desiccating (drying) agent such as calcium chloride at the bottom.
Direct seeding — a rice planting system in which seed (either pregerminated or dry) is sown directly in the field.
Dislodge — to remove from or drive out of a plant.
Dissect — to separate and expose the parts of a plant for examination, such as to count insects.
Dissolve — to cause to pass into solution, such as to dissolve sugar in water.
Donor — in plant breeding, a variety that serves as a source of a characteristic such as insect resistance.
Double-cropping — the planting of 2 crops in 1 year in a field.
Dough stage — the stage in rice development which occurs during the ripening phase when the milky caryopsis inside the developing grain turns into soft dough and later into hard dough.
Drench — to wet thoroughly by covering with water or another liquid.
Dryland rice — nonbunded, direct-sown rice grown in a manner similar to wheat, and which depends on rainfall for moisture.
Egg — in insects, the reproductive body in which the embryo develops and from which the nymph or larva hatches.
Egg mass — a group of eggs deposited by the female insect, which are adjacent to each other as in the rice bug or overlapping as in the striped stem borer.
Egg plug — a gelatinous fluid which the rice weevil places over the egg, located in the egg cavity in the plant.
Emergence — when the adult insect leaves the pupal case or last nymphal skin.
Equilibrate — to balance.
Erlenmeyer flask — a thin glass flask, flat-bottomed and cone-shaped allowing its contents to be shaken laterally without danger of spilling.

Escape — a type of pseudoresistance in which there is lack of insect infestation.
Escapes — individual susceptible plants that show no infestation damage because pests did not attack or feed on them.
Evaluate — to examine and estimate the amount and degree of a character and to express it numerically.
Excise — to remove by cutting.
Feeding lesion — a damaged area on a plant part resulting from the feeding of an insect.
Field-collected — in varietal resistance studies, insects are collected from plants growing in the field in contrast to rearing of insects in the laboratory as a source of test insects.
Field resistance — resistance observed in the field as distinguished from resistance observed in the laboratory or greenhouse. It may involve seedling resistance as well as adult plant resistance and often involves resistance to all locally occurring biotypes.
Field screening — evaluating varieties for resistance in the field in contrast to greenhouse and screenhouse evaluations.
Filter paper — porous paper used for filtering liquids.
Flag leaf — the uppermost leaf originating just below the panicle base.
Fold — to double upon itself.
FPLI (Functional Plant Loss Index) — a formula based on the plant weight loss and damage ratings due to insect feeding which is used in determining levels of tolerance when identifying components of resistance.
Gall — an abnormal plant growth, swelling, or tumor induced by another organism such as an insect.
Gall midge — a dipterous insect, *Orseolia oryzae,* whose presence at the growing point of a rice plant results in the formation of a plant gall.
Gene-for-gene resistance — *see* Vertical Resistance.
General resistance — *see* Horizontal Resistance.
Germination — growth of an embryo in a seed after sowing.
Germplasm — a) the material basis of heredity; b) the potential hereditary materials within a species, taken collectively.
Germplasm collection — a collection of genotypes of a particular species, from different sources and geographic sites, used as source materials in plant breeding.
Grade — to evaluate for insect damage in which the damage level is based on a numerical rating system.
Grain (syn. rough rice, paddy, palay, padi, caryopsis, seed) — the ripened ovary and its associated structures such as the lemma, palea, rachilla, sterile lemmas, and the awn if present.
Gramineous — belonging to the grass family.
Gravid — an insect containing fertilized eggs.
Greenhouse screening — the process of evaluating varieties for resistance in the greenhouse, in contrast to field and screenhouse screening.
Grind — to reduce to powder by friction.
Handpicking — removing insects from plants by hand.
Hatch — in the nymph or larva of insects, to come out of the egg after the embryo has completed development.
Healthy tiller — a tiller that is free of pest damage.
Herbicide — a chemical used to kill weeds or prevent their growth.
Hill — a group of one or more rice plants directly adjacent to each other.
Hispa — common name for insects of the species *Dicladispa (Hispa) armigera.*
Honeydew — a sugary liquid excretion of plant-sucking hoppers which consists of a mixture of undigested plant material and excretory products.
Horizontal resistance — a type of resistance expressed equally against all biotypes of a pest species. It does not involve a gene-for-gene relationship.
Host evasion — a type of pseudoresistance where the plant evades insect injury by passing through the susceptible stage quickly or when insect numbers are low. An example is an early-maturing variety which is harvested before the insects reach damaging levels.
Host plant resistance — the relative genetic ability of a cultivar to produce a larger or higher quality crop compared with other cultivars exposed to the same infestation level.
Hot spot — site where the natural field infestation of a particular insect is high, providing sufficient pressure for reliable results in varietal resistance tests.
Humidity — the amount of water vapor in the air.

Hydrophilic — water-loving.

Inbreeding depression — the result of inbreeding as in the continued rearing of insects without the occasional introduction of field-collected insects where the continuous breeding of genetically related individuals affects the biology, ie, decrease in reproductive capacity.

Incubate — to maintain eggs under conditions favorable for development and hatching.

Index of susceptibility — index used in evaluating the levels of susceptibility of rice to grain storage insects. It is calculated as follows:

$$\frac{\text{Natural log of the total number of insects that emerge from a grain sample}}{\text{Mean insect development period, in days}} \times 100$$

Induced resistance — a type of pseudoresistance in which a plant temporarily acquires increased resistance from some conditions of plant or environment, such as change in soil fertility.

Infest — to place insects on a plant or grain; to inoculate.

Infestation — the presence of insects on a plant or on grain.

Ingredient — component part of a mixture.

Inoculate — *see* Infest.

Insect — members of the phylum Arthropoda ("jointed legs") which has six legs, three distinct body regions (head, thorax, and abdomen), one pair of antennae, and usually wings.

Insecticide — a chemical used for killing insects.

Instar — the stage of an insect between successive molts, the first instar being the stage between hatching and the first molt.

Jar — a deep, widemouthed glass container.

Larva (pl. larvae) — the immature stage between the egg and pupa of an insect that undergoes complete metamorphosis.

Layout — a detailed plan showing the arrangement of replications and treatments in an experiment.

Leaf-feeding scar — the leaf-feeding damage by the adult rice water weevil, which consists of small longitudinal streaks where the epidermal layer is removed; a type of feeding lesion.

Leafhopper — insects of the order Homoptera, family Cicadellidae, which feed on the leaf portion of the plant by sucking plant sap.

Leaf scraping — the removal of the epidermal portion of a leaf by the feeding of an insect.

Leaf sheath — the lower part of the leaf originating from a node and enclosing the culm above the node.

Lemma (syn. outer glume) — the hardened 5-nerved bract of the floret, partly enclosing the palea.

Lepidopterous — pertaining to the insects in the order Lepidoptera.

Levee (syn., bund) — a dike made of soil to retain water in rice fields.

Light trap — a device for collecting insects, consisting of a light source which attracts insects at night and a mechanism that traps the insects.

Linear regression — a type of regression where a straight line or linear relationship characterizes the amount of change in a dependent variable which is associated with a unit change in the independent variable.

Maggot — larval stage of the order Diptera (fly).

Maize cob grits — finely ground particles of maize cobs used as medium for dispensing larvae with the Davis inoculator.

Major gene resistance — genes show clear-cut and discrete segregation in the F_2 or later generations of crosses between resistant and susceptible parents. Effects are thus qualitative.

Mass rearing — rearing large numbers of insects, which serve as test organisms in varietal resistance studies.

Mass screening — screening a large number of varieties in preliminary studies. Varieties selected as resistant in the mass screening trial are retested to confirm their resistance.

Mechanisms of resistance — processes involved in the resistance of a plant to an insect, including nonpreference, antibiosis, and tolerance as proposed by Painter.

Mesh size — the size of the openings in a sieve, expressed as the number of such openings per linear inch.

Microplot — small plot 1-2 m^2 covered with a cage, used to simulate large field plot conditions.

Milk stage — stage in the ripening phase of rice growth and development when the contents of the caryopsis (starch portion of the grain) is at first watery, but later turns milky in consistency.

Milled rice — rough rice from which the hull and bran have been removed.
Mist chamber — an enclosure containing a device that produces a fine mist of water on plants, facilitating the hatching of gall midge eggs and the movement of first-instar larvae.
Mite — small arachnid of the order Acarina.
Moderate resistance — intermediate levels of resistance, between highly resistant and susceptible.
Modified seedbox screening — screening of seedlings in a seedbox where the number of insects per seedling is less, and where the plants at the time of infestation are older than those in the seedbox screening test.
Monogenic resistance — resistance governed by one gene. Major gene resistance.
Mortar and pestle — a device for grinding, consisting of a vessel (mortar) in which substances are ground with a club-shaped implement (pestle).
Moth — an adult of the insect order Lepidoptera, suborder Heterocera.
Multiline — mixing seeds of several resistant lines which differ only in the resistance genes they carry.
Multiple resistance — resistance to several stresses such as insects, nematodes, diseases, drought, and nutritional deficiences.
Mylar film — trade name for a stiff, plastic-like film used in making insect cages.
Natural population — a population of insects which naturally occur on test plants in the field in contrast to insects which are artificially put on the plants.
Ninhydrin — a chemical for the colorimetric determination of amino acids, which becomes violet to red when in contact with honeydew on filter paper. It is used to determine the feeding activity of the brown planthopper.
Node — the solid portion of the culm, panicle axis, and panicle branches. Leaves, tillers, and adventitious roots arise from nodes on the culm.
Nonpreference — the negative response of insects to plants that lack the characteristics to serve as hosts in their search for food, oviposition sites, and shelter.
Nursery — a place where seedlings are grown before transplanting in the field.
Nymph — the stage of development in certain insects immediately after hatching. It resembles the adult but lacks fully developed wings and sexual organs.
Oligogenic resistance — resistance governed by a few genes. Major gene resistance.
Origin — in referring to accessions in the germplasm collection, the site where the accession was collected.
Oviposition — the act of laying or depositing eggs.
Oviposition cage — a cage where an insect lays eggs on plants or other materials.
Pair of insects — a male and a female insect.
Palea — the hardened 3-nerved bract of the floret, which fits closely to the lemma. It is narrower than the lemma.
Panicle — the terminal shoot of a rice plant that produces grain.
Parasite — an insect whose larvae attack one prey species or closely related species and usually feed on a single host and destroy it.
Peduncle — main stalk of the panicle.
Permeate — to pass through.
Pest — an organism which competes with people for food and shelter, or threatens their health, comfort, or welfare.
Petri dish — a small shallow dish of thin glass or plastic, with a loosely fitting, overlapping cover.
Photoperiod — the duration of the light period during a given day, which fluctuates during the year and according to latitude.
Phototropic — moving toward (positively) or away from (negatively) a light source.
Planthopper — insect of the order Homoptera, family Delphacidae, which feeds primarily on the stem at the lower portion of the rice plant.
Polygenic resistance — resistance governed by many genes. Minor gene resistance.
Population growth — increase in number of an insect population over a given period due to reproduction. Used to measure the levels of varietal resistance.
Population peak — when a given insect population is at its highest level.
Predator — an animal that attacks and feeds on other animals, such as an insect, bird, or spider which feeds on many different species of insects and attacks a number of prey by quickly eating or sucking their body fluids.
Pregerminate — to germinate seed before sowing by exposing it to moist conditions.

Productive tiller — a tiller that produces a panicle.
Progeny — offspring or descendants produced by an insect.
Propagate — to increase by natural reproduction.
Prune — to cut off what is not needed such as to prune tillers so that the number of tillers in each entry is uniform.
Pseudoresistance — apparent resistance which results from transitory characters in potentially susceptible host plants. Types of pseudoresistance are host evasion, induced resistance, and escape.
Pupa (pl. pupae) — in insect development, a nonfeeding and usually inactive stage which occurs between the larval and adult stages.
Rachis — the elongated axis of a panicle.
Randomization — location of treatments by chance so as to provide unbiased estimates of treatment means and experimental error.
Randomized complete block design — an experimental design in which the experimental area is divided into blocks and all of the treatments are randomly arranged within each block.
Rat — rodent of the genus *Rattus* and other allied genera distinguished from mice by their larger size and difference in teeth and other structures.
Rating — classifying test entries based on degree of plant damage, number of insects, etc., by expressing on a numerical scale, usually 0-9. It is a relative estimate.
Ratoon — new tillers that grow from the stubble of harvested plants and constitute the ratoon crop.
Rear — to raise or multiply organisms such as insects.
Relative humidity — refers to the water vapor, exclusive of condensed water in the atmosphere. It is the ratio of e/em expressed as a percentage: e = vapor pressure, em = saturation vapor pressure at the existing temperature.
Remnant seed — seed that is in excess and remains after sowing for a given test.
Replicate — in an experiment, to repeat a treatment, such as a variety, two or more times.
Resistant check — *see* Standard check.
Resurgence — a significant increase in an insect population on insecticide-treated plants, as compared to untreated plants.
Retest — in screening for resistance, re-evaluating in subsequent tests an entry selected in the mass screening test.
Rice stalk — the stem or culm portion of the plant.
Rinse — to cleanse by dipping into or exposing to a stream of clean water.
Rough rice (syn. paddy rice) — unhulled or whole grain rice.
Row grading — evaluating for insect damage where the evaluator, while standing at one spot, rapidly observes all the plants in a row and mentally estimates the average damage rating for the row.
Scale — the range of damage ratings, based on a numerical system, usually 0-9 where 0 = no plant damage and 9 = severe damage (all plants killed).
Scrape — to remove the green layers from the leaf by the feeding of an insect.
Screenhouse — a structure consisting of wire mesh (screen) walls and roof in which plants are grown for varietal resistance studies. The materials used prevent the entrance of birds, rats, and insects.
Screening — evaluation of varieties or breeding lines for resistance where the resistant ones are selected for further studies and possible use as donors in the breeding program and the susceptible are eliminated.
Seed — the mature, fertilized egg including the seed coat, embryo, and endosperm. *Seed* refers to grain that is to be sown.
Seedbox — a wooden box in which rice seedlings are grown and infested with insects in greenhouse screening for resistance.
Seedbox screening — a term used primarily for leafhopper and planthopper screening where test entries are screened as seedlings in seedboxes in the greenhouse.
Seedling — the stage of the plant after seed germination until the development of the fifth leaf.
Seedling resistance — the characteristically vertical resistance of plants in the seedling stage.
Selected entries — varieties or breeding lines that are resistant in initial screening tests and are selected for further screening in replicated tests to confirm the resistance.
Sex ratio — the number of males in relation to the number of females.
Sieve — a meshed device through which material is strained to separate particles of different sizes.

Silvershoot — the gall produced by an abnormal growth of the leaf sheath in response to the attacks of the gall midge.
Sources of resistance — varieties or breeding lines that have genes for insect resistance.
Sow — to place seeds into a medium where they will germinate.
Sowing guide — a structure consisting of intersecting strips of wood which form a network of square compartments. It is placed over the soil in a seedbox and seeds of test entries are placed in the compartments.
Specific resistance — *see* Vertical resistance.
Staggered planting — planting different fields or plots in an area in succession with a time gap between each planting.
Standard check — a rice variety or breeding line whose response to a given insect species or biotype is known and which is repeatedly included in tests as basis for comparing the reactions of test entries. A resistant and a susceptible check are usually included.
Standard Evaluation System for Rice — an international system of classifying responses of rice to biological, chemical, and physical stresses, developed through cooperative efforts of rice scientists and published by the International Rice Research Institute in a booklet.
Stem borer — a larva of the Lepidoptera order that feeds within the stem of a rice plant.
Sterile — free of microorganisms.
Stock culture — an insect culture which serves as a source of insects for beginning a rearing program.
Straw — a narrow tube made of paper or plastic used to suck up a beverage. Cut sections of a straw serve as pupation sites in striped stem borer rearing.
Stubble — the lower portion of the stems remaining in the field after the rice has been harvested.
Stylets — bristle-like mandibles and maxillae in leafhoppers and planthoppers, which join to form a piercing-sucking mouth.
Stylet sheath — a sheath formed in the plant from the saliva released by the stylets of hoppers during feeding.
Susceptible — when the host plant is unable to suppress or retard an injurious insect.
Susceptible check — *see* Standard check.
Tap — to strike rice plants with the hand or some object to dislodge insects.
Test entries — varieties or breeding lines being evaluated.
Test insect — the insect species and biotype against which a test entry is being evaluated.
Thin — to remove surplus plants.
Tiller — shoot arising from the main culm (stem).
Tillering capacity — the genetic potential of a given variety to produce a certain number of tillers.
Tolerance — ability of a plant to withstand infestation and to support insect populations that would severely damage susceptible plants.
Trim — *see* Clip leaves.
Tunneling — making a passage through plant material by the feeding of larvae.
Valid screening — screening that produces convincing or justifiable results.
Variety — a group of similar plants which, by structural features and performance, may be identified from other varieties (groups) in the same species. It differs from a breeding line in that it has been named and made commercially available to farmers.
Vegetative stage — a stage in plant development from germination to panicle initiation.
Vertical genes — *see* Major gene resistance.
Vertical resistance — a type of resistance which is expressed against only some biotypes of a pest species and is governed by one or more genes in the host plant, each of which corresponds to a matching gene for parasitic ability in the pest species; sometimes called gene-for-gene resistance (*see also* Horizontal resistance).
Virulent gene — a gene in an insect which is able to break down the gene for insect resistance in the plant and thus allows the insect to use the plant as a host.
Virus — a submicroscopic infectious agent consisting of particles made up of DNA or RNA which are usually covered by protein and reproduce only in living cells.
Weed — any unwanted plant that is injurious to the rice crop.
Weevil — the adult form of certain coleopterous species.
Wet-bed method — method of raising seedlings in which the seedbed consists of a raised bed 5-10 cm high and about 1.5 m wide, which is irrigated.
Whitehead — white, empty spikelets resulting from the attack of a stem borer which cuts the lower portion of the culm stopping the flow of nutrients to the panicle.

Whorl — the arrangement of leaves in a circle.
Whorl maggot — *Hydrellia philippina,* the larva of which feeds on leaves in the whorl.
Wild rice — species of *Oryza* other then *O. sativa;* usually a noncultivated species.

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