



**Food and Agriculture Organization
of the United Nations**

Field Guide Exercises for IPM in COTTON

Vietnam IPM National Programme

**FAO - IPM Hanoi
Vietnam Cotton Company
Plant Protection Department, MARD
2009**

Introduction

The Field Guide is a compilation of the learning activities during TOF and FFS training. Based on a field guide originally developed for the Indonesian Rice IPM Programme, additional activities have been incorporated and some adapted for the implementation of IPM training activities in rice and vegetables under the Vietnam National IPM Programme from 1992 up to the present. Further revisions were made based on cotton exercises from India and Malaysia, among others, were made for the first cotton TOT in 1996 which was undertaken in cooperation with the Vietnam Cotton Company. Further revisions were made based on the results of an impact evaluation of cotton IPM farmers conducted in 1999. In year 2000, the Field Guide was updated to incorporated exercises on gender and soils. In preparation for the ToF that will take place in year 2001, the sections on health risks of pesticides and nuclear polyhedrosis virus have been upgraded.

The Field Guide Exercises is a set of activities designed to be carried out in the cotton field in order to learn basic skills and knowledge of cotton IPM. The approach of study is to understand the cotton-agro ecosystem by studying both the components and the interactions between components. The guide begins with exercises to learn about methodology for studying together in the field. There is a heavy emphasis on discovery and exploration in the field. Drawing collected specimens is commonly used to emphasize observation skills. Discussion and questioning are important two methods to be developed within study groups. Since the goal of IPM is for independent analysis and decision making, discussion and analysis are encouraged in the training to develop these skills. Lecturing or giving answers too quickly without discussion does not allow participants to be active and involved in decision making. The exercises in the field guide are so designed to support discovery learning, analysis, and critical thinking.

The Field Guide Exercises are divided into several sections to address the contents of the weekly training cycle. Each week, a part of each section should be studied to provide a complete view of the cotton ecosystem. Plant physiology, insects and their natural enemies, and diseases change weekly in the cotton field and therefore these topics should be covered each week. Each week, the field situation should be summarized by an Ecosystem Analysis Activity. Other IPM topics such as sampling methods, thresholds, pesticides, etc. can be studied to improve basic skills at the appropriate time during the season. A section on gender has been incorporated in the latest revision to raise awareness and for trainers and farmers to make plans to increase female participation in cotton IPM activities.

The Field Guide Exercises are intended to support and not limit training activities. Trainers are encouraged to create new exercises or adapt existing ones. By doing so, it is hoped that training quality will continue to be improved.

Table of contents

Introduction

Research methods

| | |
|-------------------------------------|---------|
| Introduction to research methods | 3 |
| Variability in the field and layout | 4 – 5 |
| Sampling and data recording | 6 – 10 |
| Designing studies for FFS | 11 |
| Simplified statistics | 12 – 14 |

Economic threshold levels

| | |
|---|---------|
| Background | 15 |
| What is the economic threshold level? | 16 – 17 |
| Between economic threshold levels and action: risk assessment | 18 – 19 |
| Variability of economic threshold level | 20 – 21 |

Ecosystem

| | |
|----------------------------------|---------|
| What is this? What is that? | 25 – 26 |
| Ecological function of organisms | 27 – 28 |
| The cotton ecosystem | 29 |
| Concept of agroecosystem | 30 |
| Agroecosystem analysis | 31 – 32 |
| Agroecosystem analysis table | 33 |

Ecosystem analysis questions

37 – 48

Crop development

51 – 54

Insect Zoo

| | |
|--|---------|
| Introduction | 57 |
| Life cycles and biology of pests and natural enemies | 58 |
| Life cycles and food webs | 59 – 60 |

Predation

| | |
|--|---------|
| Pests and predator densities in sprayed and unsprayed cotton | 63 |
| Predation on sucking insects in field cages | 64 |
| Direct observations of consumption rates of predators in the field | 65 |
| Predation on sucking insects in the laboratory | 66 |
| Spiders | 67 – 68 |

Parasitism

| | |
|--|----|
| Measuring the parasitism level of caterpillars in cotton | 71 |
|--|----|

American bollworm

| | |
|---|---------|
| Measuring natural mortality of eggs of American bollworm | 75 – 76 |
| Field parasitism of eggs of American bollworm | 77 |
| Discovering predators of eggs of American bollworm | 78 |
| Direct observations of predators of larvae of American bollworm | 79 |

Aphids and Jassids

| | |
|---|----|
| Determining predators of cotton aphids | 83 |
| Direct observations of predators of cotton aphids | 84 |
| Measuring field parasitism of cotton aphids | 85 |
| Determining predators of cotton jassids | 86 |

Insect collection

87

| | |
|--|-----------|
| <i>Bacillus thuringiensis</i> | |
| Assessment of viability of Bt | 91 – 92 |
| Inhibition of larval feeding by Bt | 93 |
| Sensitivity of Bt to sunlight | 94 |
| Effect of Bt on predators and parasitoids | 95 |
| Nuclear Polyhedrosis Virus | |
| Identification of virus-infected insects (classroom exercise) | 99 |
| Identification of virus-infected insects (field exercise) | 100 |
| Method of production and application of virus in managing insect pests on cotton | 101 – 102 |
| Spread of virus to surrounding fields | 103 |
| Assessment of viability of NPV | 104 – 105 |
| Inhibition of larval feeding by NPV | 106 – 107 |
| Sensitivity of NPV to sunlight | 108 |
| Effect of NPV on predators and parasitoids | 109 |
| Effect of fungicides on the viability of NPV | 110 – 111 |
| Pesticides | |
| Understanding farm pesticide labels | 115 – 116 |
| What is an LD ₅₀ ? | 117 – 118 |
| Pesticide calculations | 119 – 121 |
| Spraying | 122 – 124 |
| Self-survey of pesticide poisoning | 125 – 127 |
| Yearly pesticide liters of exposure | 128 |
| Household storage and disposal of pesticides | 129 |
| Pesticides and pest resistance | 130 – 133 |
| Effect of pesticides on predators | 134 |
| Effects of pesticides on natural enemies | 135 |
| Poison sprayer maintenance | 137 – 138 |
| Weather | |
| General introduction and effect on diseases | 141 |
| Effect on disease organisms | 142 |
| Effect on insects | 143 |
| Weather, insects and pathogens | |
| Measuring the micro-habitat | 144 |
| Weather and plants | 145 |
| Diseases | |
| Identification | |
| Identification of disease symptoms | 151 |
| Disease groups | 152 |
| Disease groups game | 153 |
| Study of symptom development of leaf spots: session room exercise | 154 |
| Study of symptom development of leaf spots: field exercise | 155 |
| Study of symptom development of bacterial wilt disease | 156 – 157 |
| Spread | |
| Factors that influence disease development – exercise 1 | 161 |
| Factors that influence disease development – exercise 2 | 162 – 163 |
| Demonstration of spread of disease | 164 |
| Virus and vectors | 165 – 166 |
| Study of spread of fungal leaf spot | 167 |
| Pot experiment to test whether root diseases are soil borne | 168 |
| Test of cotton seed quality | 169 |
| Effects of inundation of fields on incidence of wilt diseases | 170 |

| | |
|---|-----------|
| Management | |
| Disease management | 173 – 174 |
| Disease triangle to explain disease management | 175 – 176 |
| Management of different disease groups | 177 |
| Gender | |
| Gender awareness | |
| Background | 183 |
| Female and male characteristics and stereotypes | 184 |
| Optional | 185 |
| Women’s and men’s roles in cotton cultivation: two generations | 186 |
| Women’s and men’s roles and responsibilities in educating children in IPM | 187 |
| Background notes for the trainers | 188 |
| Gender division of labor | |
| Background | 191 |
| Gender division of labor in agriculture, household, and commune | 192 – 194 |
| Gender division of labor in cotton growing | 195 – 196 |
| Background notes for the trainer | 197 |
| Gender communication, facilitation and sensitivity skills | |
| Background | 201 |
| Preparing for a farmer field school focusing on communicating and negotiating participation of women with village leaders | 202 |
| Facilitating a farmer field school focusing on creating a constructive atmosphere for women to participate actively | 203 – 204 |
| Background notes for the trainer | 205 |
| Gender activity plan | |
| Ideas for elaborating the gender activity plan | 209 |
| Background note for the trainer | 210 |
| Soils | |
| Water infiltration rates | 213 |
| Existence of microorganisms | 214 |
| Organic content of local soils | 215 |
| Nutrient uptake | 216 |

Research Methods

Introduction to research methods

Why research methods?

Why should we use field studies to understand IPM in the field? And why should we learn more about Research Methodologies if we are not researchers?

First, it is clear that for us who work in the field, we must use the guide to look at problems in the field. We identify insects and diseases in the field by the way they move, fly, smell and develop over time. This is why slides, close up photos and other static materials are not used. Field identifications are different from identifications made by taxonomists that use dead insects. Field studies get us in the field where we can learn and help others learn in a realistic way.

Second is the question about research methods. Every field is slightly different and every farmer/extension worker must adapt to local conditions. This is especially true for varieties, management techniques and pest components. Simple proven methods of testing a new variety or improving fertilizer management use research methods. The goal of learning about these methods is to assist in understanding how to adapt global recommendations to local situations in a reliable way. As an IPM trainer, having these methods in your skill set will make you a valuable assistant to farmer groups interested in improving local methods.

How do you do research?

Actually research is very simple, especially compared to extension work. Researchers set up studies and record the results, good or poor. Extension workers and farmers, however, must give good answers because they are responsible for someone's food. For research we should follow some rules for setting up studies (Research methods part 1): Variability in the field and layout; sampling and recording data (Research methods part 2): Sampling and Data recording; and analyzing data.

Dealing with variability in the field is the key to useful results. The way a study is set up will determine whether the results will reflect the actual questions being answered. Randomized designs and sufficient repetitions of treatments can reduce underlying variation in the environment (e.g. field moisture or soil type). Field layout is very important and very basic to good studies.

Sampling and data recording are essential aspects of research. Sampling methods must be developed to reflect the characteristics of the organisms or parameters being sampled. Sample unit and sample size should be defined before sampling begins to assure precision and accuracy. The way data are recorded will make analysis easy or difficult. The key point is to set up data recording at the beginning of the season to reflect the type of data analysis at the end of the season.

Data analysis includes descriptive statistics (mean, standard deviation and variation) and graphing methods (bar charts, x-y graphs and frequency distribution graphs). Next is the process of determining if there are significant differences between treatments. There are several tests (t-test, And Wilcoxon/Mann-Whitney non-parametric) which will assist in determining if differences in treatment means indicate differences in treatments. An example of the last confusing statement is this: a group of male adults from Hanoi have a mean weight of 62.4 kg, and a group of male adults from Danang have a mean weight of 65.8 kg. Does this difference in average weight between the groups indicate that male adults in Hanoi weigh less than male adults in Danang? Probably not, because each group also has some variability.

Variability in the field and layout

The main problem which we must deal with in research is how to reduce variability in the field. Every field is different from adjacent fields, and even fields are different from one side to the other. Water moisture is a good example. One field may be very moist while adjacent fields are dry. If we are doing research on yield due to variety in these fields, the wet areas will have a different yield from the dry areas because of the water and not only due to differences in variety. For rice, the wetter area will probably have a high yield. For cabbage the wettest place will probably have low yields because of disease. No conclusion about the study can be made directly because of the underlying variability. At the end of the study, no useful statement can be made about the differences in variety if we do not try to control the variability. There are many ways to control variability by using the proper layout and by trying to maintain identical environmental situations (impossible, but as close to identical as possible).

Controlling variability by layout

Randomization and Repetition are important concepts. Randomization assures that one treatment is not placed in a special position. Using the above example, each variety would have equal chance to be placed in a moist or dry part of the field. Randomization can be done many ways. The easiest is to put numbers in a hat and pull out the numbers.

Repetition on the other hand means that each treatment is tried several times. The differences due to variability are reduced by averaging the treatment over several sites. In the above example, each variety would be grown in several places so that sometimes the variety is in a moist place and sometimes in a dry spot. The average of each variety will provide better indication of each variety's relative yield characteristics. If the results look like there are major differences due to moisture, then in another study, perhaps varieties could be tested with several repetitions in moist places, and several repetitions in dry places. How you randomize and repeat treatment is partially determined by the question being asked.

Layout of studies refers to how many times a treatment is going to be tested and where the repetitions are going to be placed on the land available. The size of the land often determines these numbers. On research stations, usually 4 to 10 repetitions of each treatment are made. The size of each repetition (plot size) should be determined by the question being asked. Differences in varieties can be tested in relatively small plots but differences in management need a much bigger area in order to reduce the influences of adjacent plots (consider spray and no-spray field with natural enemy sampling!). In our studies, there are usually 3 repetitions per treatment. More repetitions would be desirable if we had more land and more time.

In this exercise, we will begin with laying out the studies for this season based on our curriculum and land available. This exercise will concentrate on Layout.

Objectives:

- Explain how different factors contribute to variability in fields
- Layout field studies for the coming season (soil moisture depth and soil type)

Materials:

Paper, pencil, ruler (30 cm), ruler (50 cm)
Newspaper
Strong, clear plastic bags

Method:

(Note: begin the soil type activity first, then begin the soil moisture depth activity)

I. Field Variability - Soil type

1. Go to the study area. Collect soil from 5 different sites and place them in strong clear plastic bags. (It is preferable to use two plastic bags, i.e., put one bag inside the other.) Place the label between the two bags so that it does not get wet and at the same time each of the samples are identified. Use pencil or waterproof marker to write on the label.
2. Add enough water to cover the soil samples. Next mix and shake the soil in water in the plastic bag. Place the bags somewhere to let the soil settle. Do not disturb the bags for at least 30 minutes.
3. When the soil has settled and the water above the soil is clear, measure the different layers in the soil:

Component:

| | | |
|------------------------|----------|-------|
| a. bottom layer (sand) | cm |% |
| b. middle layer (loam) | cm |% |
| c. top layer (clay) | cm |% |
| Total height | cm | |

(note the smallest particles are closest to the top)

% component = 100 x component height (cm)/total height (cm)

4. Record the data and compare sites. Can you relate the differences to any aspect of the site topography? Is soil moisture related to the soil type? How could you test these questions further?

II. Field Variability - Soil moisture depth

1. This activity is to measure soil moisture over a large area to be used for studies.
2. Go to the field area. Make a rough map of the area if you do not already have a map. Choose 25 places in the field over the entire area and mark on the map.
3. To measure soil moisture, we will use the paper absorption method. Cut soil at 90° angle with surface. Place newspaper against soil so that there is good contact. Remove newspaper after 30 seconds. Measure the depth where the paper is dark from being moist. Record the data for each of 25 sites.
4. Describe the variability of soil moisture depth over the entire area. Try to relate the differences to the slope, cropping pattern, irrigation system, etc. Are there major differences in the soil moisture depth? How will these difference influence studies on the site? How can you layout one study so that most of the study is done on similar soil moisture depth or reduces the influence of variation?

III. Field Layout

1. Use your data to specify areas of the field that seem to have similar characteristics. Note the size of the area needed for your studies.
2. Now over the area, measure the size needed to give equal size plots for each repetition. Remember that the total number of plots needed is equal to the number of treatment multiplied by the number of repetitions. The plots do not have to have the same shape, but it is better to have a similar shape. Add a 30 cm row between plots. Make a map of the area.
3. After the plots have been drawn or laid out in the field, then it is time to assign treatments to each plot. There are many ways of doing this. The best is to use a random assignments do the following:
 - a. on small pieces of paper write numbers from 1 to the number of treatments.
 - b. place the numbers upside down so that the numbers can not be seen.
 - c. choose a number for the first plot and write the number on the layout plan.
 - d. return the number to the other numbers and continue choosing numbers. Repeat for each treatment number only the number of repetitions. For example for a study with four repetitions, return the number if it has not been chosen four times.
4. Layout the field now using the randomized treatments for each study. Place signs that indicate the numbers of repetitions and treatments.
5. Why is it important to randomize treatments? Why does each treatment have more than one repetition? Do the numbers give a good randomization of the treatments? Why are maps and signs important? How would you do this with a study that has five treatments and ten repetitions? Why are more repetitions better?

Sampling and data recording

Why Sampling?

Sampling is an important part of doing studies and an important part of doing IPM. But there are differences which are important to consider for making sampling plans. In an IPM programme, the goal of sampling is to decide if the field population densities of pest insects and their natural enemies is such that some control is necessary. Usually this means that the sampler is trying to decide if the pest is above the action threshold, and if it is above, then whether the natural enemy density is sufficient to control the population.

The second type of sampling is for studies. The actual population level is important to know so that the results of the study can be explained in terms of as many parameters as possible. In studies, it is important to know the development of populations during the season and to have an idea what the effect of these populations have on the study. Also, during this training, we are trying to understand the population dynamics of components of the agroecosystem.

Recording data

Recording data can be done in many ways. The most important aspect of data recording is that it is done in the same way throughout the season, and that the data are clear for later analysis. Data statistics will be computed from the original field data, so there should be space on the form for both means and standard deviations. The date, treatment, repetition, and other information should also be placed on each form.

Objectives:

- Define "population density"
- Make a form for data recording.

Materials (per group of 5):

100 stones (or seeds), 1 crayon, 1 big paper, 5 sheets of paper, rulers, pencils and pens

Method:

I. What is the meaning of "population density"?

1. Each person in the group first writes down on a piece of paper their own definition of population density. Save this for after the activity.
2. Now, on the large piece of paper, draw a grid (5cm x 5cm) over the entire sheet.
3. Take the stones and spread the stones over the sheet.
4. Fill in attached form by counting the number of stones in 20 randomly selected boxes:

| | box = 5x5 cm | box = 10x10 cm | box = 15x15 cm |
|--------|--------------|----------------|----------------|
| Box 1 | | | |
| Box 2 | | | |
| Box 3 | | | |
| Box 4 | | | |
| Box 5 | | | |
| Box 6 | | | |
| Box 7 | | | |
| Box 8 | | | |
| Box 9 | | | |
| Box 10 | | | |
| Box 11 | | | |
| Box 12 | | | |
| Box 13 | | | |
| Box 14 | | | |
| Box 15 | | | |
| Box 16 | | | |
| Box 17 | | | |
| Box 18 | | | |
| Box 19 | | | |
| Box 20 | | | |

| | | | |
|--------------------|--|--|--|
| Mean no. stones | | | |
| Standard deviation | | | |

5. Now convert the number of stones per box to number of stones per square meter: (average number of stones/box size)*10,000 = number of stones/m². Example: (3 stones/25cm²)*10,000 = 1200 stones/m².
6. Measure the total area of the grid. Now convert this number of 100 stones for the actual area to number of stones for 1 square meter. Example: total size of the sheet is 50*100 cm² = 5000 cm²; (100 stones/5000 cm²)*10,000 = 200 stones/m².
7. Compare the actual density and the sampled densities. Which of the box size sampling have the best approximation of the actual population density? Why is it important to measure density? Why is it important to be able to convert all measures into numbers/m²? Why does percentage not make any sense for measuring population density?

II. Densities and thresholds

Actual density and sampled density is important and useful to know for studies, but farmers only need to know if the population density is above or below a certain density. Let's make some examples.

1. Assume that the economic threshold level is 50 stones per square meter.
2. Now fill out the form below. Spread different numbers of stones on the floor for several samples. Count 10 random boxes each time to make a decision.

Use 10 stones

Total stones in 10 random boxes:

Average number of stones/m²:

Above or below ETL:

Use 25 stones

Total stones in 10 random boxes:

Average number of stones/m²:

Above or below ETL:

Use 50 stones

Total stones in 10 random boxes:

Average number of stones/m²:

Above or below ETL:

Use 75 stones

Total stones in 10 random boxes:

Average number of stones/m²:

Above or below ETL:

Use 100 stones

Total stones in 10 random boxes:

Average number of stones/m²:

Above or below ETL:

3. When the actual density of stones is above the economic threshold, does the sampling give the same result? if you did not make an exact count, could you estimate if the density is above or below the economic threshold? Is it important for farmers to know the density exactly?

III. Making forms

It is important to make forms that reflect the questions being asked for each study. We are especially interested in insect and natural enemy population densities in many studies. Development of the plant, disease and other aspects are important in other studies.

1. To make a form, first list the data to be collected. Include information about the study, dates, and names of the sampling team.
2. Now decide which information is to be collected once during the season and which is collected weekly.
3. For weekly collected data, ten random plants per collection should be selected, so at least ten columns should be made. Two more columns are made for average and standard deviation (SD). Example:

Study:

Sampling date:

Sampling team:

Remarks: (ex. weather, etc.)

| | sampl. unit 1 | samp. unit 2 | samp. unit 3..... | | samp. unit 10 | average | SD |
|---------------------------|---------------|--------------|-------------------|--|---------------|---------|----|
| Plant height | | | | | | | |
| # leaves | | | | | | | |
| <i>H. Armigera</i> larvae | | | | | | | |
| Spiders | | | | | | | |
| <i>Trichogramma</i> | | | | | | | |
| Leafspot | | | | | | | |

Indicate what unit you use, for example plant height in cm, insects in numbers per m², etc.

4. Prepare forms for summarizing the data for the season. The summary should include the average and standard deviation. Example:

Study:

Summary of seasonal data

Sampling team:

Remarks: (ex. table on pesticide use, weather conditions)

| | week 1 | | week 2 | | week 3 | | week 4..... | | | | | | |
|---------------------|--------|----|--------|----|--------|----|-------------|----|--|--|--|--|--|
| | av. | SD | av. | SD | av. | SD | av. | SD | | | | | |
| plant height | | | | | | | | | | | | | |
| # leaves | | | | | | | | | | | | | |
| H. armigera larvae | | | | | | | | | | | | | |
| Spiders | | | | | | | | | | | | | |
| <i>Trichogramma</i> | | | | | | | | | | | | | |
| Leafspot | | | | | | | | | | | | | |

Indicate what unit you use, example plant height in cm, insects in numbers per m², etc.

5. Prepare enough forms for the season (the trainers will assist in giving the forms to people who will help type and print and copy them). Do not change forms during the season if possible.

6. Use a dark pencil or black pen to record data so the forms can be photocopied.

Note: In the TOT, introduce the simplified statistics method from the Indonesia National Programme. The method may be used to analyze data from TOT field studies instead of mean standard deviation. The method may be also be used for data analysis for Farmer Field Schools. Details about the simplified statistics methods and are on p. 11.

Designing studies for FFS

Field conditions can be very variable and unpredictable, owing mainly to erratic water availability, and the erratic occurrence of different pests. Therefore, it is impractical to have a fixed programme with exercises to be conducted in pre-set FFS sessions (for example study 1 to be conducted in session 1, study 2 in session 2, etc.). Much better is to remain flexible and respond to current field conditions. Only then will we really be able to help farmers with their direct problems.

So far, we have conducted a number of studies as part of the training programme. These studies can be classified in the following categories:

1. Identification / lifecycles
2. Agroecosystem analysis
3. Influence of natural enemies on pests
4. Plant physiology
5. Risk of using insecticides
6. Applying biological control (e.g. releases)
7. Cultural/agronomic practices

In this exercise we learn how to select studies and exercises to be conducted with farmers, and how to adapt the studies to local conditions. It will help to use our creativity in preparing suitable studies, and it will help thinking about how farmers will perceive a study.

Method:

1. Each group will visit a field (fields of different stages and different conditions for each group)
2. Visit the field and prepare a summary agro-ecosystem analysis, from ten sampled plants in the field, with information on the condition of the crop, the soil, the weather, water availability, agronomic practices, and pests, diseases and natural enemies.
3. Each group designs two studies, that are relevant and feasible under the local conditions found. The studies should be from two different categories (see above). Show creativity, the methods and numbers (number of pests/defenders, number of replicates) you propose do not have to be exactly as conducted in earlier training, and you could think of totally new studies on topics not yet covered in the training.
4. Discuss whether a feasibility study is required. Field leaders should always have experience with each study they teach to farmers; this will avoid unexpected problems. If the proposed study is new, a trial - or feasibility study by the trainer himself is required before doing the study with farmers.
5. Prepare a report on charts; one chart for the agro-ecosystem analysis, and one chart for each study. Present the objectives of each study, the materials needed and methods of the study.
6. Present the report with charts for everybody to discuss.

Questions:

1. Is the proposed study an important one, when we consider the current field situation?
2. Is the study likely to give simple and clear results, from which we can learn?
3. Will farmers be able to conduct the study?
4. Are farmers likely to understand the aim of the study?
5. What will farmers learn from this study?
6. At what times do observations have to be made and how does that fit into FFS sessions (for example, if observations have to be made over some period of time, how would you propose that farmers do it?)

Simplified statistics

Adopted from the Indonesia IPM National Programme

Background

In pilot studies on weeding in soybeans in Indonesia, farmers commonly evaluated the averages per treatment without observing the variation among the three replicates. Consequently, faulty or premature conclusions were drawn in several cases. To help farmers evaluate whether differences between treatments were clear or not, a simple test was developed that encouraged farmers to evaluate the variability of their data. These “Simplified Statistics” consist of three steps (see below). Step 1 considers only averages per treatment; this is the only step farmers took in analyzing results during the pilot study. Steps 2 and 3 consider variation between replicates. In step 2 farmers determine for each replicate which treatment wins and which loses; if a treatment consistently wins in all replicates it is clearly better than the treatments. In step 3, farmers draw the minimum-maximum range of values for each treatment. If treatments have no values in common they are clearly different. Step 3 is more rigid than step 2; if the difference between treatments is consistent, their minimum-maximum values may still overlap. This test works satisfactory for studies with 3 - 4 replicates, but the chance of overlap between treatments increases with the number of replicates.

The test was applied to the results of the 29 farmers’ experiments. The outcome of the “Simplified Statistics” was identical to Tukey’s honestly significant difference ($P < 0.05$) in 80% of the experiments. In five experiments (or 17%), the two tests differed in their level of differentiation, with Tukey’s HSD test being most sensitive. In one experiment (or 3%), a significant effect was found with the Simplified Statistics but not with Tukey’s HSD. Thus, for studies with three replications, the Simplified Statistics test is comparable to Tukey’s HSD. Simplified Statistics will help farmers making better conclusions about their results. We observed in various occasions that farmers were able to use the test on their own data.

Guidelines for simplified statistics for farmers

Three steps of Simplified Statistics for farmers:

Step 1: Is the difference large? To compare the average yields or outcomes of each treatment: Is the difference between the averages large ($> 10\%$) or not? Normally farmers only use this first step.

Step 2: Is the difference consistent? In this step we look at the individual replicates or blocks (per row) to determine which treatment has the highest value (this treatment “wins”) and which the lowest (this treatment “loses”). If the same treatment “wins” in each replicate (for studies with 3 - 4 replicates), it is a consistent winner. If a treatment loses in each replicate it is a consistent loser.

Step 3: Is there overlap between the minimum and maximum? This is the most important step. Now, we look at individual treatments (per column) to find out the replicate with the minimum and the one with the maximum value. We do this for each treatment and draw the range of values for each treatment (as indicated in the drawing). Is there an overlap between treatments or not? (even if treatments have only one value in common, there is still an overlap). If there is no overlap between 2 treatments, those treatments are clearly different. If there is an overlap, the difference is not clear.

Step 2 is not really necessary, but increases understanding of variation. Step 3 determines the rigor of the test. This test works properly for studies with 3 - 4 replicates (as blocks); the results are comparable to traditional statistics.

If treatment values are consistently different but there is an overlap, the difference is not clear.

Examples:

Suppose a simple farmers' study on weeding in soybean, with only two treatments: local weeding practice and intensive weeding. Each treatment is replicated three times.

| | | |
|---------------|-----------|-----------|
| Replication 1 | Local | Intensive |
| Replication 2 | Intensive | Local |
| Replication 3 | Local | Intensive |

At harvest, the farmers take yield samples from each plot. Together with their field school facilitator, they prepare the following table of results (in t/ha):

| | Local | Intensive |
|---------------|--------|-----------|
| Replication 1 | 2.0 | 2.4 |
| Replication 2 | 2.3 | 2.2 |
| Replication 3 | 2.2 | 2.3 |
| (Average) | (2.17) | (2.30) |

The farmers are a bit confused with these data. Apparently, the intensive weeding treatment has a higher average yield, but is this difference really clear? The facilitator helps farmers using Simplified Statistics to draw a conclusion.

Step 1: Is the difference large? Yield in the intensive treatment is one-fifth higher than in the local treatment. This is at least a moderate difference.

Step 2: Is the difference consistent? In replicate 1, the intensive treatment wins (indicated with underlining); in replicate 2, the local treatment wins; in replicate 3, the intensive treatment wins. Therefore, no treatment is a consistent winner (or the difference between the treatments is not clear).

| | Local | Intensive |
|---------------|------------|------------|
| Replication 1 | 2.0 | <u>2.4</u> |
| Replication 2 | <u>2.3</u> | 2.2 |
| Replication 3 | 2.2 | <u>2.3</u> |
| (Average) | (2.17) | (2.30) |

Step 3: Is there an overlap between minimum and maximum between the treatments? (after step 2 has shown that the difference is not consistent, step 3 will automatically show that there is also no overlap between the treatments; but let us conduct this step anyway).

First we draw the range of values for each treatment, from minimum to maximum value.

Local 2.0 -----2.3

Intensive 2.2 -----2.4

The minimum value in the local treatment is 2.0, and the maximum is 2.3. In the intensive treatment the minimum is 2.2 and the maximum 2.4. If we draw these ranges, it appears that there is an overlap of values!! Therefore, the treatments are not clearly different.

THE CONCLUSION IS THAT, ALTHOUGH THE AVERAGES DIFFER, THIS DIFFERENCE DOES NOT CONVINCE US (YET) THAT INTENSIFIED WEEDING INCREASES YIELD OF SOYBEAN. FARMERS COULD STICK TO THEIR LOCAL PRACTICE, OR REPEAT THE STUDY.

Ask groups to try out the following exercise on the simplified statistics method

Yield results of a study on urea fertilizers in soybean are shown below.

1. Analyze with Simplified Statistics.

| | 1 No urea | 2 Urea 50 kg | 3 Urea 100 kg (Local) |
|----------------|----------------------|-------------------------|--------------------------------------|
| Replication 1 | 4.71 kg | 3.82 kg | 3.41 kg |
| Replication 2 | 6.15 kg | 4.61 kg | 3.10 kg |
| Replication 3 | 5.34 kg | 5.11 kg | 3.74 kg |
| Average | 5.40 kg | 4.51 kg | 3.42 kg |

2. What is your conclusion?

3. Now, we look back at observations made during the season in those three treatments, in order to find out WHY differences in yield occurred. Try to relate the results of observations to the yield results.

| Main observations | Treatment 1 | Treatment 2 | Treatment 3 |
|--------------------------|---|--------------------|--------------------|
| Number of leaves | 130 | 156 | 198 |
| Plant height | 42 cm | 47 cm | 49 cm |
| Number of pods per plant | 26.7 | 20.6 | 20.2 |
| Plant weight | 19.6 | 20 | 19.0 |
| Pests | more aphids, but levels are not serious; other pests are not affected | | |
| Natural enemies | same in all treatments | | |
| Weeds | few | normal | normal |

Economic Threshold Levels

Economic threshold levels

Background

The goal of IPM training is to empower farmers to make their own decisions. These decisions are usually economic decisions about pest control - if I don't spray, will I lose some yield that is worth more than the cost of the spray? The decision requires knowledge of the ecosystem: recognition of pests and natural enemies, understanding of the interaction of pests and natural enemies, cotton plant and its ability to compensate. The decision also requires knowledge of the effect of pests on yields and the effect of pesticides on natural enemies.

We have seen that sampling is the first step in making decisions. This is the step of getting information. Using Economic Threshold Levels is the second step. Assessing the risk of pest populations is part of the economic step that begins the third step of the analysis. In the next section we will explore the meaning of Economic Threshold Levels and look at risk assessment when pest populations have surpassed their ETL.

WE WILL ANALYZE WHY FIXED ETLs ARE NOT USEFUL AND WHY FARMERS' UNDERSTANDING OF THEIR AGROECOSYSTEMS IS MORE USEFUL FOR DECISION MAKING.

What is the economic threshold level?

The Economic Threshold Level (ETL) is an attempt to improve decision making practices by using partial economic analysis on the impact of a control practice, such as spraying a pesticide. The ETL is computed usually based on three parameters using the following equation:

$$\text{ETL} = \frac{\text{cost of control (Bgd taka/ha)}}{\text{commodity value at harvest (Bgd taka/ha) x damage coefficient(kg/ha/#pest/ha)}}$$

At the ETL the benefits of spraying are equal to the losses caused by the insects in the field. There are many ways of making this definition, but they are usually based on the same parameters. What is the use of the ETL? Traditionally, when the ETL was surpassed (field populations are sampled and found to be higher than the ETL) the farmer was advised to spray. IPM now includes a larger analysis of the ecosystem (like the IPM being taught in the field!). Other factors including levels of natural enemies, plant health and ability to compensate for damage, other investment opportunities, personal health, and weather are involved in the decision making process. The ETL is still a useful part of the analysis, but the ETL is not the only analysis. In this activity we will explore the behaviour of the ETL given many scenarios. In the following exercises we will further explore other types of thresholds, decision making, and ecosystem analysis.

Objectives:

- Define ETL and parameters for computation of ETLs
- demonstrate how ETL changes with changes in at least two of the parameters (commodity value and cost of control)
- Discuss why the traditional fixed ETLs are not useful when one considers other factors in the ecosystem like natural enemies, crop stage, etc.

Time required: 120 minutes

Materials:

Paper and pencil, bar charts

Method (for groups of 5):

1. Prepare one bar chart per person in the group.
2. Each group should sit together in a circle. One person will be the secretary to record discussions.
3. Each person in the group should now adjust the two bars of the chart to reflect their definition of the ETL. The left bar reflects the cost of controlling pest (taka/ha) (usually cost of one spray), and the left bar the loss due to pests present (taka/ha).
4. Each person should use the chart to define their idea of the ETL. Go around the circle and let each person say their definition aloud.
5. Now go through the following questions and movements of the bars:
 - a. The total costs of application include labour, chemical, transport, equipment, etc.. How does the chart change if the cost of pesticides and labour increase? Does the other bar change position to maintain the definition of the ETL? What is the effect of increase of control cost to the ETL?
 - b. If you invest Taka 2,000 how much profit do you expect from your investment? If you spend Taka 3,000 for pesticide control, how much profit do you expect to receive? Would you ever spend Taka 3,000 today to receive Taka 3,000 two months from now? Is this good business? Is this what the ETL means?
 - c. On your chart, if your losses due to pests are going to be Taka 4,000 how much are you willing to spend for pesticides to protect from that loss? What if that pesticide money is invested for chickens, or in the bank, how much profit should you receive?
 - d. Do farmers have other economic activities besides cotton? Do they have to use their money for maximum production of cotton or for maximum profit from their money resources? What is the best management for a farmer with only small amounts of cash?
 - e. A problem: You have a pest infestation at the ETL in your field, and have to pay for your children's schooling. Unfortunately you have only enough money to pay for control the pests or for the school. Which one will you choose? What if the extension worker tells you have to spray because the ETL is reached in your field?

6. Now go to step 4 and repeat defining the ETL. Is it different? What are the parameters for computing the ETL? How does the ETL change with increase in pesticide cost? Since you are an extension worker, have the pesticide prices changed? Have "official" ETLs changed?
7. How is the ETL useful? Why do some farmers ignore recommendations to control pests when the pest population equals the ETL?

Find the Appropriate Economic Threshold Level

| | | | |
|----------------|----------------------------|---|-----------|
| Taka 3,850 /ha | | | 50 /plant |
| Taka 3,700 /ha | | | 48 /plant |
| Taka 3,550 /ha | | | 46 /plant |
| Taka 3,400 /ha | | | 44 /plant |
| Taka 3,250 /ha | | | 42 /plant |
| Taka 3,100 /ha | | | 40 /plant |
| Taka 2,950 /ha | | | 38 /plant |
| Taka 2,800 /ha | | | 36 /plant |
| Taka 2,650 /ha | | | 34 /plant |
| Taka 2,500 /ha | | | 32 /plant |
| Taka 2,350 /ha | | | 30 /plant |
| Taka 2,200 /ha | | | 28 /plant |
| Taka 2,050 /ha | | | 26 /plant |
| Taka 1,900 /ha | | | 24 /plant |
| Taka 1,750 /ha | | | 22 /plant |
| Taka 1,600 /ha | | | 20 /plant |
| Taka 1,350 /ha | | | 18 /plant |
| Taka 1,200 /ha | | | 16 /plant |
| Taka 1,050 /ha | | | 14 /plant |
| Taka 900 /ha | | | 12 /plant |
| Taka 750 /ha | | | 10 /plant |
| Taka 600 /ha | | | 8 /plant |
| Taka 450 /ha | | | 6 /plant |
| Taka 300 /ha | | | 4 /plant |
| Taka 150 /ha | | | 2 /plant |
| Taka 0 /ha | | | 0 /plant |
| | Cost of application per ha | Crop loss (Taka/ha) due to given population | |

Directions: Cut out two bars. Cut chart along dotted lines so that bars can be inserted into the chart like a bar graph. Place shaded bar on left, and population bar on right. Follow instruction in text.

Between economic threshold levels and action: risk assessment

The economic threshold is the population density (no. pests / unit area) which is causing as much economic damage as would be needed to control the pest. For example, the "official" ETL suggests that an average population density of one pink bollworm will reduce yield (Taka/ha) by the amount of money that it will cost to control the insects (Taka/ha). The threshold however is only a partial guide for decision making. You have seen in an earlier activity that the economic threshold changes with changes in prices of commodities and controls. Also you have discussed that other factors are included in decision making (natural enemies, investment opportunities, weather, etc.). Indeed, farmers usually have a shortage of money and are looking for the best opportunities for investing their money for maximum return on the money.

So what is the action that should be taken when the economic threshold is reached? This is an important question to answer since the academic answer ("spray when at the threshold") is not appropriate for the reality of farmers.

Risk assessment is part of making a decision after the ETL is reached. What will happen if I don't spray? What will happen if I do spray? Are natural enemies sufficient in the field? Will the weather change this week? Are there opportunities for controlling the problem?

The answers to these questions and for determining what you will do next begins with an understanding of the ecosystem. Understanding the interactions between weather, plants, herbivores, and natural enemies allows you to be able to predict which outcomes are most likely. For an experienced person, the ability to guess the outcome is due to both an understanding of the ecosystem and experience with similar situations.

In this activity we will develop scenarios and futures. These will be used to use your knowledge of interactions in the ecosystem to assess risk and outcomes in the future. Analyzing systems will improve your skills at risk assessment.

Objective:

Discuss the relative risk to a field given a set of ecosystem factor and assuming an economic threshold has been reached

Time required: 120 minutes

Materials:

Large paper, markers

Method (for groups of five persons):

1. Copy the following chart on your large piece of paper:

| FACTOR | ODD | EVEN |
|--------------------|-----------|-------------|
| 1. natural enemies | many | none |
| 2. variety | resistant | susceptible |
| 3. weather | sunny | cloudy |
| 4. immigrants | few | many |
| 5. age of insects | young | old |
| 6. disease | few | many |
| 7. rats | few | many |

2. The next step is to choose one insect per group, which occurs in cotton. Each group chooses a different insect.
3. Each person should now pick two numbers which have seven digits. Write the numbers on a piece of paper.
4. Now you are going to create futures! Take your number to describe the future by using each digit to choose the condition of each factor. If the first digit is odd, then in your future you have many natural enemies. If it is even then you will not have any natural enemies. If the second digit is odd,

then you will have a resistant variety. If the second digit is even, then your variety is susceptible. Use your number to determine the future.

5. Now each person should analyze what he/she will do about a pest population that is above the ETL and have the future given above.
6. What other information is useful for making a decision? Is "do nothing" a kind of action? Or is "observe again in one week" a better action? What combinations of odds and evens are possible to say that "observe again in a week" is a good action?

You can use this exercise for risk assessment of diseases.

Method (for groups of five persons):

1. Copy the following chart on your large piece of paper:

| FACTOR | ODD | EVEN |
|---------------------|-----------------|-------------|
| 1. variety | resistant | susceptible |
| 2. weather | sunny | rainy |
| 3. humidity | low | high |
| 4. wind | weak | strong |
| 5.% diseased plants | low | high |
| 6. lesion type | not sporulating | sporulating |
| 7. pest | few | many |
| 8. natural enemies | many | few |
| 7. rats | few | many |

2. The next step is to choose one disease per group, which occurs in cotton.
3. Each person should now pick two numbers that have seven digits. Write the numbers on a piece of paper.
4. Now you are going to create futures! Take your number to describe the future by using each digit to choose the condition of each factor. If the first digit is odd, then in your future you have many natural enemies. If it is even then you will not have any natural enemies. If the second digit is odd, then you will have a resistant variety. If the second digit is even, then your variety is susceptible. Use your number to determine the future.
5. Now each person should analyze what he/she will do about a pest population that is above the ETL and have the future given above.
6. What other information is useful for making a decision? Is "do nothing" a kind of action? Or is "observe again in one week" a better action? What combinations of odds and evens are possible to say that "observe again in a week" is a good action?

Variability of economic threshold level

The Economic Threshold Level (ETL) is an attempt to improve decision making practices by using partial economic analysis on the impact of a control practice, such as spraying a pesticide. The ETL is computed usually based on three parameters using the following equation:

$$\text{ETL} = \frac{\text{management costs (Bgd taka/ha)}}{\text{commodity price (Taka/kg)} \times \text{damage coefficient(kg/ha/#pest/ha)}}$$

At the ETL the benefits of spraying are equal to the losses caused by the insects in the field. There are many ways of making this definition, but they are usually based on the same parameters. What is the use of the ETL? Traditionally, when the ETL was surpassed (field populations are sampled and found to be higher than the ETL) the farmer was advised to spray. IPM now includes a larger analysis of the ecosystem (like the IPM being taught in the field!). Other factors including levels of natural enemies, plant health and ability to compensate for damage, other investment opportunities, personal health, and weather are involved in the decision making process. The ETL is still a useful part of the analysis, but the ETL is not the only analysis. In this activity we will explore the behaviour of the ETL given many scenarios. In the following exercises we will further explore other types of thresholds, decision making, and ecosystem analysis.

Objectives:

- Define ETL
- Discuss the variability of each factor of the ETL
- Explain why fixed ETLs are not useful

Time required: 120 minutes

Materials:

Newsprints and markers

Method:

(for the big group):

1. Present the equation for ETL:

$$\text{ETL} = \frac{\text{management costs (Bgd taka/ha)}}{\text{commodity price (Taka/kg)} \times \text{damage coefficient(kg/ha/#pest/ha)}}$$

2. Go through each factor. Ask participants to explain what they know about each factor.

Note:

Management costs: depend on the type of management used (cheap or expensive), access to tools (owned or rented), labor costs (own or hired; time of the year), differences between provinces (near cities or far from cities), other conditions.

Commodity price: may change each year depending on markets, quality of produce, etc.

Damage coefficient: varies according to the variety, water availability, natural enemy populations, weediness of the field, nutrient levels, weather, farmer skillfulness in growing the crop, disease infection, stage of the plant, plant spacing, etc. Not all damage leads to yield loss.

(for small groups):

- Now each small group will calculate ETLs for different situations for two farmers. Management costs for each of the farmers should be different: own vs. hired labour, cheap or expensive pesticides, close to or far from shop, own sprayer or rented, etc. List down all the management costs for Farmer A and Farmer B.

You can use the following table.

| Management costs (Taka/ha) | Farmer A | Farmer B |
|----------------------------|----------|----------|
| Labour | | |
| Pesticides | | |
| Transportation | | |
| Sprayer rental | | |
| Environmental costs | | |
| Health | | |
| Others | | |
| | | |
| | Cost: | Cost: |

Also list down the commodity price for cotton in different months of the year. The damage coefficient used in the exercise is: $0.1 \text{ kg/m}^2/\text{insect/m}^2$.

| Month | Commodity price (Taka/kg) | | Computed ETL Given: $1 \text{ kg/ha/1 insect/m}^2$ | |
|----------|---------------------------|----------|---|----------|
| | Farmer A | Farmer B | Farmer A | Farmer B |
| December | Price: | Price: | Loss: | Loss: |
| January | Price: | Price: | Loss: | Loss: |
| February | Price: | Price: | Loss: | Loss: |
| March | Price: | Price: | Loss: | Loss: |

Calculate ETLs (# insects/ha) for each farmer for the different months.

For example:

$$\text{Farmer A, ETL (\# insects/ha)} = \frac{\text{Management costs Farmer A}}{\text{Commodity price Jan.} \times \text{damage coefficient}}$$

Make graphics of the ETLs for each of the farmers. On the X axis write the months. On the Y axis write the ETL.

- Present your findings to the group and discuss.

Discussions:

- Is the ETL fixed for the whole season? Why does it vary? If a farmer has higher management costs, what happens to the ETL? If the commodity price is lower, what happens to the ETL?
- Do you think that a single ETL is useful for farmers? What factors do farmers have to worry about as well? On what factors will a farmer base his management decision?
- For the damage coefficient, do you think that it can be a fixed number? Why or why not? What other factors should be included? Is it possible to determine a single ETL for many locations? Why or why not?

Ecosystem

What is this? What is that?

The goal of training is to provide an educational opportunity for participants. The methodology of training is very important for achieving the goal of education. One important method of training is to ask questions that allow the participants to develop their own analysis and understanding. You are stealing an opportunity for education if you reply directly with an answer. Ask questions. Lead the participant to the answer by asking questions. In a field a common question is: “What is this?”

There are many ways to answer the question “What is this?” For most of us, the natural response is to give the name of the object often in a foreign language (English or Latin). The question is often answered by saying “Oh that is *Lycosa pseudoannulata*” or “This is *Xanthomonas campestris*”? The result of this answer is that an educational process has been stopped.

A better way to answer the question is to ask a question; “Where did you find it? What was it doing? Were there many of them? Have you seen this before?” The idea is to promote learning by discovery and to lead persons toward their own analysis.

Objective:

Give several responses none of which should be the name of the object

Time Required: 60 minutes

Materials:

Cotton field, plastic bags

Method:

1. Go into a cotton field in groups of two or three persons per group.
2. In the group, take turns in the following roles:

The “farmer” should take anything in the cotton ecosystem (pests, natural enemies, weeds, others) and ask “What is this?” The other member will act as a “recorder” and must write down questions and responses.

The “technician” should respond with one of the following type of responses:

- “That is a good question. Where did you find it? What was it doing?”
- “I don’t know. Where did you find it? What was it doing. Did you ever see it before? What do you think it is?. (keep asking questions)”.

Use this specially when you know what the specimen is. Try not to give the answer!

If the question is to be answered, the “technician” should avoid answers that give more emphasis to identification. Rather the function of the organism should be emphasized:

- “This is an insect that feeds on the plant. It is not really a problem insect until there are very many. There are many organisms which eat this insect including spiders and parasites.”
- OR “This is a spider that eats insects and is a friend. It happens to be called a hunter because it moves around the field searching for insects.”
- OR some other response that only give biology/ecological information.
- OR encourage the participant to do some small study or experiment to find out more

NEVER GIVE THE ANSWER WITH A NAME. THAT ONLY KILLS THE QUESTION. THE QUESTION IS A CHANCE TO LEARN!

3. After the members had took their turns, return to session hall/shade and process experiences.

Discussions:

1. How often do you usually give just a name for an answer? Do you think it is helpful in training to ask questions to assist in learning?
2. In your usual job, is helping farmers learn an important aspect in day to day work? Do you think it would be useful to answer questions with questions to help farmers?
3. Many field workers think they have to be smarter than farmers, even though the farmer is much older and more experienced. Do you think this method can help you in working with older farmers? Does this facilitate the educational process? Can you also learn from farmers by asking questions? Do farmers think respect, a desire to learn, or an instant answer is most important for a government worker?

Ecological function of organisms

In the activity "What is this?" learning to answer questions with questions was emphasized. The response could be any question about the specimen. In the cotton ecosystem, however, everything has a function, and the function is more important than the name. There are different levels of functions in all ecosystems.

The *first level* is the producer of organic materials: the plants. Plants include cotton intercrops and weeds. The weeds have an additional function in the field. Weeds are also competitors for water, nutrients, sunlight and space. "Weeds" are defined in many ways, but one good definition is "a producer that is not wanted by mankind at that time and place".

The *second level* are organisms that feed on the plant. These include insects, rats and diseases. These are usually referred to as "pests". But "pests" are defined by their populations, not by their function. For example, when a population of jassids reaches a high level that damages the cotton, then the jassid is a pest. If the population is low, then they are not pests. They are food for natural enemies in this case.

The *third level* are organisms that feed on the second level. These include spiders, insects (predators and parasites), virus that attack insects, plant fungi and bacteria, owls, cats and other predators of rats. These organisms are usually called "natural enemies" or "friend of the farmer" because they attack things that could become pests. Preserving these organisms is important to keep the second level from increasing.

The *fourth level* are the decomposers and scavengers. These include bacteria, fungi and insects that feed on the dead plants, insects, spiders, rats, etc. that are in the cotton ecosystem. These organisms cycle the nutrients in the system back into the soil. They can also serve as food for the natural enemies.

In this activity we will practice identifying the function of organisms found in the cotton ecosystem. This is a good introductory activity for the study of ecology by farmer groups or students.

Objective:

After this activity you should be able to give the function of specimens found in the cotton ecosystem.

Time required: 90 minutes

Materials:

Cotton field, plastic bags, alcohol, glue and large paper

Method:

1. Go into a cotton field in groups of two or three persons.
2. Each group should collect as many different types of organisms in the cotton ecosystem. Include plants, plants with disease, insects, spiders, rats, snakes, etc.
3. Go to a shady spot. Add alcohol to the plastic bag and shake the bag so that the insects and spiders die.
4. Discuss and separate the collected organisms by their function in the ecosystem. Place them in levels with plants at the bottom, plant feeders at level two, natural enemies at level three, and decomposers at level four. Glue them onto the paper. If uncertain of the function, ask the trainer, or glue on the paper and label "uncertain".
5. Were there many organisms of each level in the cotton ecosystem?
6. Could all plants be called "weeds"? Why or why not? Could all insects be called "pests"? Were there many level four decomposing insects in the fields?

7. Present the specimens to other groups, and describe the function and relationships between each level. Use descriptions of functions such as:
"This is an insect that feeds on the plant. It is not really a problem insect until there are very many. There are many things which eat this insect, including spiders and parasites." OR "This is a spider that eats insects and is a friend. It happens to be called a hunter because it moves around the field searching for insects".

The cotton ecosystem

Each week during the season, you will study the components of the cotton ecosystem. You will study the plant anatomy and agronomy, herbivores, and natural enemies of the herbivores. You will also look at diseases and weeds, the weather, water conditions.

Ecosystem Analysis is a way of assembling what we are studying and place it into a process useful for decision making based on many factors. Old IPM practices relied on economic threshold levels to make decisions. ETL's however, are extremely limiting and do not include the other factors in the ecosystem or farm management.

The following activities will lead you through weekly set of questions and drawing. In the beginning, the analysis will take a lot of time. By the end of the season, however, you should be able to do a complete analysis while standing in the field.

Concept of agroecosystem

IPM is based on ecological interactions between the environment, plants, herbivores (diseases and insects), and natural enemies of herbivores (spiders, parasites, etc.). The health of the plant is determined by the environment (weather, soil, nutrients) and the herbivores. The herbivores are balanced by their natural enemies.

The adoption of input intensive agriculture has greatly influenced the interactions of the different components of the ecosystem. For example the indiscriminate use of pesticides has led to resurgence of minor pests.

We need to begin looking at the cotton ecosystem from the viewpoint of maximizing profits without destroying the system. We need to understand the interactions and components. In this exercise we will look at the interactions of the different components of the ecosystem.

Objective:

Demonstrate the balance of the components of the cotton ecosystem

Time requirement: 120 minutes

Materials:

Markers, glue, crayons, scissors, newsprint, ruler, drawing board, plastic bags

Method:

1. Assign each group (groups of five) to different stages of the cotton crop (if available). If there is a newly plowed or harvested one assign a group to this field.
2. Let each group take an area of one square meter and record all kinds of plants, insects, and spiders. Let them collect the specimens in plastic bags. Repeat the procedure in another 1 m² plot.
3. If the farmer is present inquire about additional information as fertilizer use, pesticide use, and so on.
4. Return to the session hall/shade and group the things collected from the field according to similarities, e.g. weeds, different insects as pests and natural enemies. Let them draw the things they have seen in the field on newsprint. Things with similar functions must be drawn near each other.

Another procedure that can be followed is for participants to write the names of things seen in the field on a photocopy paper (size may be 2 cm X 5 cm). Add papers with names "sunshine", "rain", "high fertilizer", "low fertilizer".

5. Discuss with group members how the parts interact. Paste the names of ecosystem components on the newspaper, and draw lines between all components that interact. Explain what the lines mean.

Discussions:

1. What are the major components of the ecosystem?
2. What happens when pesticides are applied and natural enemies are killed?
3. The plant is resistant to all pests, so that there are no pest in the field. What happens?
4. What happens when there is high fertilizer and sunny conditions?
5. What happens to the plant when a high fertilizer dosage has been applied and the weather is rainy and cloudy?

Agroecosystem analysis

Decision making in IPM requires an analysis of the ecosystem. We have seen how sampling and thresholds are important parts of the analysis. We have also discussed how some parts of the ecosystem interact. Now we will begin to use a method of Ecosystem Analysis to facilitate discussion and decision making.

The Agroecosystem Analysis will be done weekly following monitoring activities and studies of components of the cotton system. The results of the field observations will be drawn on a large piece of paper using specific rules given below. The drawing will then be used for discussion. There are questions designed for discussion during each stage of the crop. After discussion it is important that the results are presented to other groups. Everyone should be involved in the observations, drawing, discussion, and presentation. Changing the person who gives the presentation each week is important to keep everyone involved.

Objective: (The goal of the activity is to analyze the field situation by observation, drawing, and discussion.)

Make decisions about any actions required in the field

Time required: 120 minutes

Materials: (per group)

Newsprint, pencil, crayons, marker pen, graphing paper, plastic bags

Method:

1. Go to the field. Walk diagonally across the field and randomly choose 20 plants (every 5 meters) on the diagonal. For each plant follow this examination process and record your observations. Each group should do this on their basic experiment plots.

2. Select three leaves from the plant, one taken from the top, one from the middle, and one from the bottom of the plant. Pick or turn the leaf and count the number of jassids (ignore the other sucking pests if not common).

3. Count the total number of fruiting parts.

4. Open the bracts of each individual fruiting part and record:

- number of fruiting parts with bollworm damage
- number of bollworm larvae
- any predators

5. Note also:

Disease: Notice the leaves and stems. Are there any discolorations due to diseases? (Ask the trainer if uncertain). Estimate the percent of the leaf/stem area infected. Record all observations.

Weeds: Note the type of weeds in the field and the corresponding density.

Other insect pests: Count the number of each type

Natural enemies: Count the number of each type of predator and parasites.

6. Uproot one cotton plant for drawing.

7. Find a shady place to sit as a group. Each group should sit together in a circle, with pencils, crayons, data collected and the drawing of the field ecosystem from the previous week.

8. Now make a drawing of the agroecosystem observed on newsprint. Everyone should be involved in the drawing. There are several rules for drawing as follows:

- a. Draw the plant with the correct number of branches. Draw shedding fruiting parts in yellow. If the plant is healthy, color the plant green. If the plant is diseased or lacking nutrients (low in fertilizer) then color the plant yellow. Draw dead or dying leaves in yellow.
 - b. For weeds, draw the approximate density and size of weeds in relation to the size of the cotton plant. Draw the kind of weeds in the field (broad-leaf or grass type) next to the plant.
 - c. For pest population intensity, draw the insect on the right side of the plant but in the area (top, middle or bottom of the plant) where usually found. Write the number of each next to the insect and then add to get their total and the average per plant. The data can also be summarized in a table on the right side.
 - d. For natural enemy population intensity, draw the insects and spiders as found in the field on the left side of the plant. Write the average number of natural enemies and their local names next to the drawing.
 - e. If the week was mostly sunny, add a sun. If the week was mostly sunny and cloudy all day for most of the week, put a sun partly covered by dark clouds. If the week was cloudy all day for most of the week, put just dark clouds.
 - f. If the field was fertilized, then place a picture of a hand throwing N's, P's or K's into the field depending on the type of fertilizer used.
 - g. If insecticides were used in the field, show sprays with a nozzle and write the type of chemical coming out of the nozzle.
9. Below the drawing, provide space for general information, observations and recommendations.
- a. General information includes the age of the plant, type of insecticide applied, variety planted, fertilizer used.
 - b. Observations include the general situation in the field such as water situation, density of weeds, presence of other pests and natural enemies seen but not found in the sample plant.
 - c. After the small group analysis, their recommendation for the week can be written.
10. Now discuss the questions listed below for each stage of the plant depending on the crop observed. One person in the group is designated as the questioner. (Change the person each week). This person will ask questions about the field. Write your answers on the paper and add a summary as recommendation.
11. Each group should make plot the number of pests and natural enemies on a graphing paper. This should be done weekly to come up with a pattern of the weekly dynamics between natural enemies and pests.
12. Each group should present their agroecosystem analysis to the big group. A different person should make the presentation each week.

Ecosystem Analysis

| <i>Steps</i> | <i>What to observe</i> | <i>What to ask - discuss</i> | <i>Actions</i> |
|--------------|----------------------------------|---|---|
| 1 | Weather | Rainy? Dry? Trends in weather Dry/wet season Effects on crop | 1. Do we need to water? 2. Preparation of beds? 3. Disease management? |
| 2 | Crop condition | Age of crop: Seedling Veg. growth Flowering Fruiting Colour of leaves Leafspots? Diseases? | 1. Needs fertilizer? 2. Needs watering? 3. Fungicides? 4. Special topic? |
| 3 | Field condition | Soil structure: Hard? Wet? Weeds present? | 1. Hand weeding? 2. Soil aeration? |
| 4 | Herbivores | What herbivores? Population? Are they damaging? | 1. Insect zoo 2. Special topic |
| 5 | Natural enemies (and neutrals) | What NE? Population? | 1. Insect zoo 2. Special topic |
| 6 | Activities in neighbours' fields | Do neighbours spray? What was sprayed? | Compare with FP plot |
| 7 | Decisions made last week | Was the decision made effective? Do we need to do similar action? | 1. Insect zoo 2. Special topic |

FINAL DECISION FOR THE WEEK BASED ON ALL 7 STEPS

Ecosystem Analysis Questions

I/ 5-15 days after seeding

1. How many days is it after seeding? Describe plant development. Are there new leaves? How do new roots develop? Are there any changes in plant height? Are there dead or yellow leaves? Is this normal?
2. What kind of herbivores and diseases did you see in the cotton field? What is their density? Where did they come from? What is the role of natural enemies? What kinds of natural enemies did you observe? What are their population densities?
3. What about weeds? Are there many in the field? What needs to be done?
4. What is the management decision for this week? What do you expect to happen in the field next week?

II/ 16 - 23 days after seeding

1. What was the effect of last week's decision?
2. Describe plant development. How many branches have developed? How many leaves have developed? Are some leaves yellow? Is this normal?
3. What is the weather condition?
4. Do the plants need fertilizer and/or water?
5. Are there weeds? How does it influence plant growth?
6. Did herbivore populations and diseases change in comparison with last week? (Did they increase or decrease? Are there any new herbivores?) Which one is the most important at this stage? How many are there? Are they causing damage? What should be done about them?
7. Compare densities of pests and natural enemies in FP and IPM plots. Which natural enemies did you see in the field? Are there any new natural enemies? How can you get information about their functions (suggestions for Insect Zoo)? What is their role in the ecosystem?
8. Which cultivation practices do farmers use? Why? Have you ever discussed with experienced farmers? Did you check by testing yourself?
9. What is the management decision for this week? What do you expect to happen next week?

III/ 24 - 31 days after seeding

1. What was the effect of last week's decision?
2. Are there new branches? Is the plant growth and development normal? Are the plants uniform throughout the whole field? Are there some plants that could not develop? Why? What is the leaf color? How does the bottom leaf develop? Are there new leaves? Is it normal?
3. How is the root development?
4. What is the weed situation? Do we need to remove weeds?
5. Is the soil wet enough? Does the field need water?
6. What are the fertilizer requirements during this stage (nitrogen and potassium)?
7. How is the weather affecting the plant? (sunshine, wet weather, rain, fog...)
8. Did the herbivore population change compared to last week? What is its significance at this stage? Which cultivation practices and weather conditions are affecting disease development?
9. What natural enemies (predators, parasites) are there in the field? How many are there? What are the results of insect zoo and field observations? What is their role in the ecosystem?
10. Compare natural enemies and pest populations in the Regular Spray, IPM and No Spray plots.
11. What do farmers do to prevent herbivores and disease from damaging the crop? Did you observe control fields or farmers' field in the area? How are they different from your field? Describe the difference. Why is there a difference?
12. What is the management decision for this week?

IV/ 32 - 38 days after seeding

1. What was the effect of last week's decision?
2. Describe the development of stems and leaves. How many leaves does the plant have? What is the stage of the plant development? What is the effect on the growth and development of the plant if 50% of the leaves are damaged? Why? How many branches developed?
3. How is the weather (sunny, rainy, windy)? How does it influence plant growth and diseases? How can you protect the crop from harsh weather conditions?
4. Does your crop need fertilizer? What kind, how much and what method of application will you use? What other cultivation practices are needed in this stage (water, etc.)? Why?
5. What kind of herbivores and diseases are present? What is the composition and density of pests in the three plots compared to last week? Which one is important?
6. What kind of natural enemies are present? What densities? What is the role of natural enemies? What are the results from insect zoos?
7. Compare your practices with farmers' practices in the area? Did you ask farmers why they practice that?
8. What is the management decision for this week? What do you think will happen in your fields next week?

V/ 39 - 45 days after seeding

1. What was the effect of last week's decision?
2. Describe plant development (height, leaf length, number of branches, number of leaves, buds)? Describe root development?
3. What kind of nutrient does the plant need in this stage?
4. What is the difference between composition and density of pests and natural enemies in the three plots? Which of these is important?
5. What natural enemy did you see in the field? What is the role of natural enemies in the ecosystem?
6. Describe the weather condition (hot sunny, dry cold, wet cold, etc.). Is the soil wet enough? How does it influence plant growth and development? How does it influence diseases?
7. What is your management decision for this week? Compare this with farmers' decision.

VI/ 46 - 52 days after seeding

1. What was the effect of last week's decision?
2. Describe plant growth and development (height, number of leaves, leaf length). What percentage of plants have flowers? Are new branches developing?
3. What is the significance of old and yellow leaves at the bottom?
4. What cultivation practices are needed at this stage (fertilizer, water, etc.)? What is the fertilizer requirement of the plant at this stage? What type of fertilizer does the plant need? How much? What method of application should be employed? What is the effect of water on plant in this stage? Does the plant need water?
5. How does the weather influence cotton development?
6. What are the main herbivores and diseases at this stage? Which is important? How many are there? Is there any damage?
7. What natural enemy did you see in the field? What is the role of the natural enemy? Collect for insect zoo observations.
8. What are farmers in neighbouring fields doing now?
9. What is the management decision for this week?

V/ 53 - 66 days after seeding

1. What was the effect of last week's decision?
2. Comment on plant development at this stage (stem, leaves)?
3. What is the significance of old and yellow leaves at the bottom? Why are there yellow leaves? What is your management decision?
4. What is the percentage of plants with flowers or bolls in comparison with last week? Are any flowers or bolls shedding? Why?
5. What is the influence of the weather on flowering and fruit development?
6. Which cultivation practice do we need to do at this stage? (fertilizer, water, maintaining beds)
7. What herbivores and diseases occurred this week? How do the densities compare with last week? Which one is important?
8. Which natural enemies occur in the field? What is their role? Compare their densities this week with last week?
9. How do you compare your fields with farmers' fields in the area?
10. What is the management decision for this week? What do you think will happen next week?

VI/ 67 - 73 days after seeding

1. What was the effect of last week's decision?
2. Comment on plant development at this stage. Compare this week's number of flower and bolls with last week? Did any flowers and bolls fall? What is the reason for this? What is your management decision? What factors influence development of flowers and bolls?
3. What cultivation practice should be paid attention to now (fertilizer, water...)?
4. What herbivores are present this week? Which factors will result in their increase or reduction next week? Which one is important? Describe disease development. What are the factors and how do they influence disease development? What is your management decision for these herbivores and diseases?
5. What natural enemy occurred in the field? What is their role?
6. Compare your management method with those of farmers in surrounding fields. What is your comment on the ecosystems in the three plots?
7. What is the management decision for this week?

VII/ 74 - 80 days after seeding

1. What was the effect of last week's decision?
2. Comment on plant height and number of leaves at this stage? How many bolls form from each flower set? Is there any difference between flower sets? What is the reason for this? Describe boll formation at this stage (quantity and fruit size). What conditions are affecting boll formation?
3. Is it necessary to fertilize and water at this stage? What about after each harvest?
4. What kind of pest do you have to pay attention to at this stage? Why? What management method will you use?
5. What diseases are present this week? Compare the disease situation this week with last week? What conditions favour their development? Have you seen any bollworms in the fields? How do the bollworms create damage at this stage? Compare the degree of bollworm damage between the three plots.
6. What natural enemies are found in the field? What is their role? What are their densities?
7. What is the management decision for this week?

VIII/ 81 - 90 days after seeding

1. Comment on flowering and boll formation this week. What factors affect flowering and boll formation?
2. Do you need to water or fertilize?
3. What kind of herbivores and diseases are present in the field this week? What are their densities? What is the degree of damage? What is your management decision?
4. What natural enemies did you observe? What is their role?
5. Compare your management decision with that of other farmers in surrounding fields.

VIII/ 91 - 100 days after seeding

1. Comment on flowering, boll formation, and boll opening this week. What factors affect flowering, boll formation, and boll opening?
2. Do you need to water or fertilize?
3. What kind of herbivores and diseases are present in the field this week? What are their densities? What is the degree of damage? What is your management decision?
4. What natural enemies did you observe? What is their role?
5. Compare your management decision with that of farmers' in surrounding fields.

VIII/ 101 - 120 days after seeding

1. Comment on flowering and boll formation this week. What factors affect flowering and boll formation?
2. Do you need to water or fertilize?
3. What kind of herbivores and diseases are present in the field this week? What are their densities? What is the degree of damage? What is your management decision?
4. What natural enemies did you observe? What is their role?
5. Compare your management decision with that of farmers in surrounding fields.
6. Comment on the yield of your plot in comparison with that of farmers' plots.
7. How many times have you harvested? How much time does it take between harvests?
8. Compare the yield per harvest between the three plots. Which plot has the highest yield? Why? Which plot has the highest economic benefits?

Crop Development

Plant emergence stage (1 - 15 DAS)

In the first 15 days after seeding the cotton seeds will germinate, forming roots and the cotyledons. After this the first true leaf will appear.

Objectives:

- Describe characteristics of germination and the development of roots in seedling stage
- Explain physiological characteristics, nutrient and water requirements in order to apply appropriate cultivation methods to obtain good establishment of the plants

Duration: 120 minutes

Materials:

Soaked seeds, incubated seeds and germinating seeds
Seedlings with 1 - 2 leaves (10 days old) and 3 - 4 leaves (15 days old)
Paper
Crayons
Magnifying glass

Method:

Observe, describe, and draw all the stages of the seedling

Discussions:

1. What part emerges first when cotton germinates? How many days after seeding does the cotyledon emerge?
2. How many roots are there at each stage? What are their functions for the growth and development of plants at each stage?
3. How many leaves are there at each stage? What is the size and thickness of leaves? What about presence or absence of hair on the leaves? Did you observe 'poison glands'? What is their function? At what stage did you start to observe them? What is the colour and the arrangement order of leaves in each stage? When were new leaves formed? Did you see old leaves that were dead? Is this normal?
4. What is the influence of weather and climate on the cotton plant at this stage?
5. What kinds of herbivores should be paid attention to during this stage? What is the best management method?
6. Is it necessary to fertilize at this stage? What is the water requirement for the seedling?

Plant establishment stage (15 DAS to 50 DAS)

In this stage the main branch and vegetative branches develop. No flowers or bolls will develop on these monopodial branches. The vegetative branches emerge from main stem nodes at the base of the plant. Fruits and flowers will develop on sympodial branches.

Objectives:

- Explain how the cotton plant develops in the establishment stage
- Compare the speed of root and leaf formation

Duration: 120 min.

Materials:

Plants of 15, 25, 35 and 55 days after seeding

Magnifying glass, large paper, crayon, pencil

Method:

1. Collect plants from the field.
2. Draw and describe different parts of the plants at different ages. Observe new root formation, places that roots occur, colour of roots, number of roots, length of roots, significance of root formation. Observe new leaf formation. Observe the formation of monopodial branches. Observe the formation of sympodial leaves (if any already).

Discussions:

1. Describe plant development (height, number of roots, number of leaves, number of branches) at this stage? How many vegetative branches have developed? At what place of the plant did they develop? What kind of (monopodial or sympodial) and how many branches develop at each stage? Did you observe 'nectar glands'? When did you begin to observe them? What is their function? What are squares? When do you start to observe them?
2. Why is it important to compare the speed between root and leaf formation?
3. What is the nutrient requirement of plants at this stage?
4. What cultivation practices can you use at this stage to obtain good plants?
5. How does the weather influence this stage?
6. What herbivores occur at this stage? What natural enemies occur? What happens if the leaf area is reduced by insects? What happens if the top of the plant gets damaged by insects?

Flowering and fruit formation stage (50 -90 DAS)

The cotton plants develop fruiting (sympodial) branches on the vegetative and main stem. No fruits occur directly on the main and vegetative parts. Squares that are formed on the first few nodes of the main stem and vegetative branches normally fail to develop. Fruiting branches are formed from the 5th or 6th node up. Flowers do not develop all at the same time. New flowers and fruits continue to be formed for a prolonged period of several weeks. It is normal that a high percentage of squares are shed before or shortly after flowering. The cotton plant only maintains as many bolls as it can supply with sufficient nutrients. If no bolls would be shed the cotton plant would overfruit and produce smaller fruits with inferior fiber quality.

Objectives:

- Describe characteristics of cotton plant from plant establishment to fruit formation stage
- Explain effect of different factors on the plant in this stage
- Explain mechanism of fruit shedding and how it influences cotton development

Duration: 120 minutes

Materials:

Large paper, pencil, crayons

Cotton plants at different stages:

- with squares
- with flowers
- with start of boll formation

Method:

1. Collect cotton plants of different stages from the field.
2. Observe, draw, describe morphological characteristics.

Discussions:

1. Describe plant development at this stage? What is the significance of the quality of plant development at this stage?
2. Comment on vegetative and reproductive growth at this stage? On what branches are flowers formed? What is the color of the flowers? Did you observe any change in their color? When and why does the color change?
3. Have fruits been shed? Is this normal? Why does it happen? What factors influence fruit shedding?
4. What is the order of fruit formation? Are all fruits formed at the same time? How many fruits are there on the plant?
5. What is the role of water and fertilizer at this stage? Why?
6. What is the impact of herbivores at this stage? What happens if herbivores attack the young fruits? What natural enemies are important at this stage?

Boll growth and maturation stage (90 - 130/140 DAS)

After a high percentage of fruits has been shed, the cotton plant retains the bolls that it can support with sufficient nutrients. The bolls that are maintained appear to be firmly affixed to the plant for the remainder of their growth. The bolls continue to mature and will finally open. Then they are ready for harvest. Since the flowering occurs for a prolonged period not all bolls ripen at the same time and cotton needs to be harvested several times.

Objectives:

- Describe characteristics of cotton plant in 50% maturity stage (110 DAS)
- Explain factors that affect this stage

Duration: 120 minutes

Materials:

Plant with bolls in different stages of ripening
Large paper, crayons, knife

Method:

1. Collect plants with fruits in different stages of ripening from the field.
2. Observe, draw, describe plant and fruit form (also dissect the fruit).

Discussions:

1. What are the plant characteristics at this stage?
2. What percentage of the fruits were economically ripe in your field? What percentage was not? Can you make an estimate of the final harvest already in this stage (50% boll maturity)?
3. Is it necessary to apply cultivation methods after each harvest?
4. What herbivores have to be paid attention to?
5. What is the role of natural enemies at this stage?

Insect Zoo

Introduction

Insect Zoos is done by participants in the training to help them learn about insects and natural enemies by direct observation and manipulation. Insects and spiders are more interesting when seen alive and active. A living organism is much more than what is seen in an alcohol filled jar. In fact some things can only be recognized when living.

The activity and behavior of insects and natural enemies can only be seen in live specimens. The Insect Zoo will give you many living specimens for demonstration that will keep farmers more involved and help them remember better that predators and parasites are friends in the field.

The Insect Zoo will also help you learn about the biology of the animal. Life cycles, egg laying, feeding, mating, growth and behavior can be learned directly through the process of rearing insects and natural enemies.

There are many ways to rear insects and natural enemies. Many parasites can be obtained directly from their host by collecting eggs, mature larvae, and pupae from the field and placing them in any plastic, glass or paper container. Place the collected specimens in the container and merely watch. If the specimens were parasitized, small wasps will emerge.

For parasites that are not collected from hosts, it is sometimes possible to put “sponge plant” in the field. This means that from reared insects you have plants in pots with egg masses or larvae. These plants with the host are placed in the field for up to four days to attract the parasites. The parasites will lay their eggs in or on the host. The “sponge” is then brought back to the pot and kept in a cage.

For other insects and spiders, collecting young nymphs, adult moths or spiders is the best way to begin rearing. However, for nymphs and for adult moths, you must have prepared plants ahead of time. For spiders, it is best to have lots of insect prey in a rearing cage before beginning to rear.

In this section on Insect Zoo, the following topics are included:

- Life cycles of main pests and natural enemies
- Life cycles and food webs
- Predation exercises
- Parasitism exercises
- Exercises on:
 - ⇒ American bollworm
 - ⇒ Aphids and jassids
 - Insect collection

Life cycles and biology of pests and natural enemies

During the cotton season we will rear pest insects and natural enemies to understand their life cycles. We can learn about the different stages of development of the insects and spiders, and how long it takes for them to complete their lifecycles.

Materials:

Cotton plants, small plastic bottles, cages, plastic bags, brushes

Method:

There are many ways for rearing insects and spiders. Below are some general methods and specific tips for specific insects

General rearing methods

1. Bottles and plastic bags are very useful rearing tools. Always carry a couple in your pocket or bag. If egg masses, larvae or nymphs are found in the field, collect and place in a bottle or plastic bag. The bottle should have a piece of netting over the mouth. Add plant material daily for herbivores. Transfer to larger cages if necessary. Try to collect older larvae that will pupate quickly. Parasites will also emerge from egg masses, larvae and pupae.
2. Simple cages can be made using waste materials such as transparent glass or plastic bottles. Place leaves and stems in the bottles with insects and cover with netting. For soft drink bottles, place a bouquet of stems and leaves in the bottle and cover with a large plastic bag. For seedlings, invert the plastic bottles which have one end open and the other end covered with netting material.
3. Field cages are useful to cover infestations of larger larvae and other insects. Make cages from large plastic bags, or netting materials. Use bamboo sticks to hold the cages above the plant.
4. Potted plants and cages are useful especially for demonstrations and exhibitions. Grow your own plant in the pot, or transplant from field grown plants. For cages use netting suspended strings or frames, or use plastic bags with netting glued over one end. Expensive thick stiff plastic is also useful.
5. Be creative! It is surprising where insects can be reared. Use discarded cans for pots, and transparent plastic bottles for cages. Clear glass jars and small plastic containers will suffice for most needs.

Rear the following insects and spiders:

American bollworm
Pink bollworm
Jassids
Aphids
Leaf worm
Other bollworms
Spiders
Parasites
Coccinellid beetles
Chrysopa

and more insects you can find in the field.

Observe regularly. Write down your observations in your notebook. Make drawings of different stages. At the end of the studies, summarize your findings on a big piece of paper. Draw the different stages of insect development.

Regularly present your results to the other participants in the training course.

Life cycles and food webs

Life cycles of plants, insects and natural enemies are well known to us. The development from egg or seed to adult insect, spider or plant has been seen in the field and in the Insect Zoo.

Food chains are the interactions between plants, herbivores and natural enemies of the herbivores. The energy from one level of the ecosystem (plants) moves to another level (herbivore) along a chain of interaction.

As a trainer working with farmers, you must begin to integrate these two motions together into a smooth acting dynamic ecosystem. Seeds germinate to be eaten by insects that lay eggs that are parasitized.

In this exercise, you will have to put the two systems together so that they are functional. This will help you to understand that interactions have a time frame. For example: the life cycle of caterpillars all begin with an egg stage. In the next stage, the larvae feed on the leaves by chewing. Finally adults mate and lay eggs on the same plant or migrate to other fields. During each stage, different natural enemies attack the caterpillars. During the egg stage, parasites complete their own egg/larva/pupa/adult in the eggs and kill the eggs. During the larval and adult stage, hunting spiders, lady beetles and other predators feed on them. Parasites and other natural enemies act the same.

This combination of interacting life-cycles of the plant, caterpillars and natural enemies is a good view of the dynamic system of the cotton field. It shows also that balance is needed in the system to make each life cycle possible; for example, a spider life cycle depends on aphids. If there are no aphids then there will be no spiders to protect the field. In this system, insects such as aphids at low population are actually very beneficial to the farmer because they are spider food; and spiders are what protect the beneficial insect from large population changes. Did you ever think that an aphid might be a beneficial insect to the farmer? It all depends on how many are in the field. This can be explained now by looking at how the system interacts.

Objectives:

- develop a concept of the food web and food chains
- discuss the importance of food web and food chains in relation to ecosystem and pest management

Time Requirement: 120 minutes

Materials:

Paper, pens, crayons

Method:

1. Each group should choose a guild to analyze: leaf feeding caterpillars, aphids, etc.
2. Draw a large circle and write in the general stages for insects of the guild around the circle.
3. On one side make a list of the stages of the insects in one column. In the next column, make a list of natural enemies (by guild) which attack each stage. (Show that at each life stage of a pest, there is a corresponding natural enemy with its own life cycle.)
4. On the drawing, draw a circle for each natural enemy which attacks a particular stage of the insect. On the natural enemy circle, write the stages of the natural enemy's life cycle. If there are natural enemies of the natural enemies (example, a spider that eats another spider) then make a third level of circles for these natural enemies. Follow the chains until the last organism dies and its nutrients return to the soil and is consumed by the plant.
5. After finishing the diagram, do a short role play on natural enemies and insect pests, if possible, working through whole life cycles and describe parts of predators that are important for their function as killers!

Discussions:

1. Explain life cycle, food chain and food web.
2. How does food web relate to biodiversity?
3. How do you group different organisms involved in a food web in relation to the amount of energy consumed?
4. What will happen to natural enemies if there are no insect pests?
5. What is the effect of pesticide application to the ecosystem?

Predation

Pests and predator densities in sprayed and unsprayed cotton

(This exercise can be carried out several times during the season.)

Both pests and predators are always present and play a very important role in the cotton ecosystem. Spraying pesticides disturbs this ecosystem. This exercise will demonstrate how spraying chemicals disturbs the cotton ecosystem as shown by densities of pests and predators in sprayed and unsprayed cotton fields.

Objectives:

- Observe the effect of spraying on populations of pests and predators
- Practice scouting methods for use by farmers

Materials:

Notebook, pen, vials

Method:

Field:

1. Group 1 and 2 take the sprayed treatment, group 3 and 4 take the unsprayed treatment.
2. Each group samples 10 plants across the diagonal of the field. To select a plant, walk across the diagonal of the field and choose a plant every 5 m.
3. Before touching the plant, carefully observe and record easily disturbed predators.
4. Then, select three leaves from the plant, one taken from the top, one from the middle, and one from the bottom of the plant. Carefully turn each leaf and record the number of sucking insects (jassids and aphids).
5. Then, check all vegetative plant parts for any predators (starting from the top leaf downwards); turn the leaves.
6. Then, count the total number of fruiting parts.
7. Then, open the bracts of each individual fruiting part and record the number of fruiting parts with bollworm damage (including the fruiting parts that started to show yellowing, but not those that have already dried up). Record bollworms found. When looking at the fruiting parts, also record whether there are any small predators like Orius hiding in the squares or flowers.
8. Then, check on the ground surface under the plant and record any predators found.
9. Collect predators encountered in plastic vials for use in the laboratory experiment.

Session room:

1. Calculate the average density of sucking insects per leaf
2. Calculate the percentage of fruiting parts with bollworm damage
3. Calculate the average density of predators per plant
4. Results of the four groups are compared, and the effect of spraying on pests and predators is evaluated in class

Predation on sucking insects in field cages

Objective:

Observe role of predators in reducing jassid or aphid populations in the field

Materials:

20 nylon field cages (0.6 x 0.6 x 1.2m) with bamboo supports

Labels to number the cages

Unsprayed field with plenty of sucking insects (jassid nymphs or aphids)

Method:

1. Each group select individual plants for caging. The selected plants should have similar numbers of sucking insects.
2. Construct a cage around each selected plant.
3. Carefully count the jassid nymphs and the aphids on all the leaves in each cage. This will take some time.
4. Label the cages with numbers 1-4.
5. Collect 30 predators (preferably chrysopid larvae or coccinellid larvae).
6. In cages 2 and 4: release 15 predators per cage.
7. In cages 1 and 3 (these are the control cages without predators, for comparison): remove all predators that were present inside the cages (coccinellids, chrysopids, ants, etc.).
8. Close every cage by burrowing the margins into the soil.
9. After four or five days, lift the bottom margin of the cage, and carefully count the jassid nymphs and the aphids in each cage. Check all plant parts systematically from the bottom of the plant to the top. Also record the number of predators recovered from each cage.
10. Calculate the average number of jassids and aphids in cages 1 and 3 (without predators) and in cages 2 and 4 (with predators).
11. Obtain the data from the other groups, and calculate the average number in cages with and without predators.

Discussions:

1. What was the average number of nymphs/pupae at the beginning of the experiment in the cages without and in the cages with predators?
2. What were the average numbers of the three pests at the end of the experiment in the predator treatments and in the predator-free treatment?
3. What was the role of the predators in reducing jassids populations? And aphids?
4. What other predator species in the field are likely to feed on sucking pests?
5. Besides predation, what other factors influence the population of sucking pests?

Direct observations of consumption rates of predators in the field

(This exercise can be repeated several times during the season.)

Some predators are not so easily disturbed so we can study their natural feeding behaviour by simply observing them for a while in the field and recording what and how many prey they eat during a certain period of time. Such observations take a lot of our patience but with a group of observers (for example at a Training of Trainers or at a Farmer Field School), we can obtain interesting results within a short period of time.

Objective:

Observe consumption rates of predators in the field

Materials:

Hand lens, watch, whistle

Method:

1. Early morning at 7 am, the trainees are briefed, and are divided into two groups that will each observe a particular predator species in an unsprayed field:
 - Paederus or chrysopid larvae
 - Coccinellid adult or large larvae
2. Other predators may be chosen depending on their availability in the field, but this exercise is not suitable for easily disturbed predators, such as spiders.
3. Each member of the group is required to find a predator of the appropriate species. When everyone has found a predator, a field leader gives a whistle to start the 10 minutes observation and everyone follows his predator and counts the number and sizes of the prey it eats within ten minutes. The predators should not be disturbed and should not be given prey, because we want to observe the natural feeding behaviour.
4. After 10 minutes, the field leader gives a second whistle to end the observation. Everyone gathers and the results of everyone's observations are compiled on a board directly in the field. The average predation rates (per hour) are calculated for each predator.
5. The same activity is repeated at 9.30 am and (if possible) at 6.30 pm, in order to compare the activity of predators at different times of the day.
6. After each observation, results are compiled and discussed in the "field class".

Discussions:

1. How many prey can each predator feed on/eat per hour?
2. What is the preferred prey of each predator species?
3. Are there differences in feeding rates at different times of the day?
4. Which predator is the most active searcher?
5. When the pests are less common, would the predators eat the same numbers of prey or less? Why?

Predation on sucking insects in the laboratory

A number of predators may feed on sucking insects in the field. To evaluate their consumption rate we conduct a simple study in the laboratory. This study could be conducted on jassid nymphs or apterous aphids.

Objective:

Observe consumption rates of predators in the laboratory

Materials:

20 clear plastic or glass tubes (60 ml)
Some tissue paper
Scissors
Fine brush
Hand-lens
Tube labels

Method: (per group)

1. In the unsprayed field, collect sufficient leaves with moderate levels of one of the following sucking insects:
young jassid nymphs, or
apterous aphids
2. Also collect three species of predators, with 5 individuals of each of the species; in total 15 predators. Each group could choose their own predator species, for example, chrysopid larvae, Paederus adults, larvae or adults of coccinellids, or Orius.
3. Cut twenty 1' x 1' square fragments with 20 sucking insects (aphids) on each leaf fragment.
 - a. In case of aphids, remove the winged adults with a fine brush, and remove the surplus of pests so that exactly 20 pests are present per leaf fragment. (If field densities are low, 10 pests per fragment would suffice).
 - b. In case of jassids, cut the leaf fragments and with a fine, wet brush, collect small jassid nymphs from the leaves and put 20 nymphs per leaf fragment inside each tube. Be careful not to damage the nymphs.
4. Add one predator in each of the tubes numbered 1 to 15 (1-5: predator species 1, 6-10 predator species 2, 11-15 predator 3); the remaining 5 tubes (number 16-20) are the control, without predator. Always keep the predators away from direct sunlight.
5. Observe the predators for a while, to see if any is found feeding on the pests.
6. After 24 hrs, carefully count and record the number of remaining pests (that are alive) inside the tube or on the leaf fragment of each tube. Check whether you can retrieve any remains of pests that have been killed. The study now has ended.
7. Calculate the average number of pests that disappeared (were eaten) in each treatment (predator 1, predator 2, predator 3, and the control).
8. Compare the results with those of other groups.

Discussions:

1. How many prey did each predator consume in 24 hrs?
2. Which predator species ate most and which ate least?
3. What is the value of having a control without a predator?
4. Do predators behave differently in tubes than when free-living in the field? When would the predators feed more: in tubes in the laboratory, or in the field? Explain why.

Spiders

There are many insects and spiders found on the cotton plant and on the beds and irrigation ditches. Most of the insects are not pests or even potential pests. In fact they are beneficial to the cotton farmer because natural enemies such as spiders feed on these non-pest insects. This is how spiders can survive even when pest populations are low.

In this activity, we will search for spiders and their prey. You should be able to explain where spiders are living in and around the cotton field and what kind of spiders can be found.

Objective:

Describe spiders in and around cotton fields

Time Requirement: 1 hour and 30 minutes

Materials:

Newsprint, pentel pens, test tubes and spiders

Method:

1. Each group counts spider populations in a square meter area in/on the:
 - plot
 - a. seedling
 - b. vegetative
 - c. flowering
 - d. ripening
 - side of the plot
 - grassy area near the cotton field (2 meters from the cotton field)
 - newly plowed field
2. Identify the kinds of spider species seen.
3. Consolidate and present data to the big group. Use the matrix below:

| Spider species | Plot | | | | Side plot | of Grassy area | Newly plowed | Total |
|----------------|----------|------------|-----------|----------|-----------|----------------|--------------|-------|
| | Seedling | Vegetative | Flowering | Maturing | | | | |
| Total | | | | | | | | |

Discussions:

1. Where can you find the highest spider population in the four areas and why? The lowest, why?
2. What are the kinds of spiders found in the different areas?
3. What will happen to spiders when there are no pests present?
4. In what part of a cotton plant are spiders commonly found?
5. How many insects does a spider eat in one day?
6. What are the characteristics of spiders?
7. Differentiate spiders from insects?
8. How does a spider eat insect pests? Do a role play.
9. What is a pest? If at low populations, spiders survive on some insects, are these insects pests? Does "pest" refer to an insect, a damage or an intensity of insect?
10. Get the average number of spiders in the different ecosystems surveyed and extrapolate population into per hectare basis. How many spiders are there in a hectare? If one spider can eat 5 - 10 pests in one day, how many pests will they eat in one day? For FFS activities seeds could be used to determine the dynamics of spider population using the following assumptions:

| | | |
|--------------------------|---|----------------|
| Ratio of male and female | = | 50:50 |
| Birthrate | = | 30 spiderlings |
| Survival rates: | | |
| Group 1 | = | 0.1 percent |
| Group 2 | = | 0.3 percent |
| Group 3 | = | 0.5 percent |
| Group 4 | = | 0.7 percent |
| Group 5 | = | 0.9 percent |

Compute for three generations. How many pests are consumed by spiders in each generation. Make a graph of the different survival rate data from each group.

Parasitism

Measuring the parasitism level of caterpillars in cotton

(This exercise can be done several times during the season.)

Certain larval growth stages act as host for some parasites. The best way to learn about larval parasites is to observe what emerges from larvae collected from the field.

Objectives:

- Observe parasitoids of caterpillars (bollworms or leafworms)
- Explain the importance of parasitism
- Explain aspects of the biology of parasitoid species, e.g., are they restricted to certain larval growth stages on the host

Materials: (per group)

30 plastic tubes with labels

Tissue paper

Fresh plant material for larval feeding

Method:

1. This exercise could be conducted at any time that caterpillars are common in the field.
2. Each group select two species of larva that are common in the field (for example, *Plusia*, *Helicoverpa* or *Spodoptera*), and collect five small (<0.8 cm), five medium (0.8 - 1.5 cm) and five large (>1.5 cm) larvae of each species from an unsprayed field. If plenty of tubes are available, each group could collect more larvae.
3. Put larvae individually inside plastic tubes, and label the tubes with the date of collection, host species, and size of the host at collection. Add some fresh leaves as food, and secure a piece of tissue paper between the lid and tube to prevent condensation.
4. Observe each tube daily and replace food. For small caterpillars, food can be replaced once every two days.
5. Observe carefully whether parasites emerge from the caterpillar, whether the caterpillar has pupated, or whether the adult has emerged. If parasites emerged, count them and keep them for identification.
6. Continue these observations until the end of the course, until parasites or adult moths have emerged, or until the host has died due to other causes. Calculate the intensity of parasitism for each stage (small, medium, large) of the host as follows:

$$\% \text{ parasitism} = (\text{parasitized larvae} / \text{total larvae}) \times 100\%$$

7. Collections could be repeated weekly or every 14 days, to study how parasitism levels change during the season. Make calculations for every sampling occasion, and evaluate how parasitism fluctuated during the season.

Discussions:

1. What were the parasitism levels of each pest species?
2. Did you find different parasite species in the small, medium and large stages of the host? Could you explain why?
3. Describe the parasite species found in each pest; which parasites occurred in both pest species? Which occurred in only one pest species?
4. Describe how each parasite species developed in the tubes (e.g. was development mostly inside or outside the host; how many parasites emerged per host; from which host stage did the parasite emerge).

American Bollworm

Measuring natural mortality of eggs of American bollworm

When one considers the number of eggs that are laid by pests in the field and if one imagines that all these eggs will hatch it presents a very scary picture. However, because of natural mortality not all eggs hatch. Natural mortality is the destruction of the eggs by force of nature. Predation and parasitism, among others, contribute to this.

Objective:

Observe how predation and parasitism reduce the egg stage of American bollworm in unsprayed cotton fields

Materials: (per group)

2-4 nylon cages (L:0.7m X W:1m X H:1.5m , with 0.5 mm diameter nylon mesh) and supported at each corner with a stick, each covered a 1 meter row (i.e. 3 plants) of cotton.

10 plants labels

20 plastic tubes (60 ml)

Marker (water-proof felt-tip pen) to write on the leaf surface

Scissors

Hand-lens or microscope

Method:

1. Collect *Helicoverpa* moths from light traps or from a culture in the laboratory.
2. Place the cages, supported by sticks, over selected plants in the field (one or more plants per cage)
3. Check all parts of the plants in each cage and remove any young, white eggs which might confuse the study
4. Late afternoon before dusk, release 6-12 moths of mixed sexes inside each cage and burrow the margins of the net into the soil.
5. Allow *Helicoverpa* to oviposit for one night (*Helicoverpa* oviposits mainly in the evening hours from 19.00 to 23.00 hrs)
6. Early next morning, collect the moths in plastic tubes (for next trials), remove the cages, and systematically check all parts of the plants that were enclosed in the oviposition cage for any newly laid eggs. Put a small mark (using a water-proof felt-tip pen) just next to the egg (not on the egg) so that even if the egg disappears, the mark can be retrieved.
7. Make notes on how many eggs are marked on which plants, so that no marks are missed later on. On a data sheet, record how many eggs are found per plant.
8. When more than 10 eggs are laid per plant, remove the surplus so that the number of eggs per plant ranges from 1 to 10.
9. Tag the exposure plants so that they can be found back in the field.
10. Leave the plants with marked eggs for a 48 hour period of field exposure.
11. After 48 hours (which is before the eggs start emerging) retrieve the marks. If an egg is still present at the mark, examine the egg with a 10 fold hand-lens to check whether the egg was sucked empty (only a collapsed egg shell remains) by a sucking predator (such as small Orius species).
12. Record for each plant: the number of eggs remaining, how many of those were sucked, and the number of eggs lost.
13. To examine whether eggs are parasitized, the healthy-looking eggs that remain on the plants are collected by removing a small piece of plant tissue (3 x 3 mm) with the eggs using scissors. Put the eggs from each plant together inside one plastic tube. Keep the eggs of different plants separate.
14. Bring the tubes with the eggs to the session room and put a label on the tube with date and number of eggs. Place a piece of tissue paper between the tube and the lid to avoid condensation inside the tube. Keep the tubes out of direct sunlight.
15. On the second day after collection, all healthy unparasitized eggs will have emerged. Carefully check each egg, observe with a hand-lens or microscope whether the egg has hatched, and remove pieces of plant tissue with hatched eggs. (Note that the hatchlings usually consume their own egg shell.) Record the number of eggs hatched and the number remaining.
16. Three days after collection, check the eggs again to record parasitism. Eggs parasitized by *Trichogramma* turn black. Eggs that remain white/yellow are infertile eggs. Record the number of black, grey and white/yellow eggs.
17. The minute adult parasites emerge 9-14 days after collection, collect them for identification. *Trichogramma* is smaller but less stout than *Telenomus* spp. One host egg can contain 1-3 *Trichogrammas* but only 1 *Telenomus*.

18. When all data are collected, calculate the percentage mortality due to different factors as follows:

% disappearance = (number of eggs disappeared/total eggs at the start of exposure) X 100

% sucked = (number of eggs sucked/total eggs at the start of exposure) X 100

% parasitism = (number of eggs parasitized/total eggs collected after the exposure) X 100

% failed to hatch = (number of eggs not hatched nor parasitized/total eggs collected after the exposure) X 100

Optional:

1. Repeat the above procedure during different times of the season, preferably weekly, in order to follow the seasonal pattern of mortality levels from the vegetative stage until the maturing stage of cotton. Make calculations of the different mortality factors for each occasion.
2. It would be especially interesting to conduct this study in unsprayed plots, where natural enemies are conserved, and in sprayed plots. In this way, we could compare the direct effect of spraying on natural mortality factors.

Discussions:

1. What mortality factor(s) was most important?
2. What factors could have caused the disappearance of eggs?
3. Did you observe any sucking predators (for example Orius) in the field? Do their numbers reflect the percentage of sucked eggs?
4. Besides sucking predators, what other predators were found in the field? How could we find out the number of eggs consumed by these "chewing predators"?
5. Why did we restrict the number of eggs to 10 or less per plant? What could have happened if we had, say, 100 eggs per plant?

Field parasitism of eggs of American bollworm

Objectives:

- Observe if eggs of American bollworm are killed by parasitoids
- Identify kinds of parasitoids that attack eggs of American bollworm
- Estimate the level of parasitism caused by the egg parasitoids
- Observe seasonal fluctuations of host eggs and level of parasitism
- Compare level of parasitism between sprayed and no spray fields

Materials:

Sprayed and unsprayed cotton fields

Plastic/glass tubes

Scissors

Fine hair brush

Fine pointed forceps

Tissue paper

Labels

Method:

1. Exercise to begin soon after cotton plants are established in the field. During weekly visits to the field, examine 20-50 plants and collect yellow coloured eggs of the American bollworm. To collect eggs, use a pair of scissors and remove the plant tissue around the eggs and using the forceps place the plant tissue and egg(s) into a clean plastic/glass tube. The tube should be lined with a moist tissue paper (not wet or the eggs will drown). For each field visit the tube used should be labelled with field number, date and name of collector.
2. In the session room, using a wet fine hair brush, remove the eggs from the plant tissue and place them on clean moist tissue paper and place in another clean glass tube. Remember to label the tube accordingly. Keep the tube in a cool place away from direct sunlight. Observe for emergence either daily or once in two days. Record and remove larvae that emerged together with empty egg shells. Similarly record and remove egg parasitoids and keep these in 70% alcohol for preservation and later identification. If no parasitoid specimens are required, the study can be completed in 1 week. Parasitized eggs turn black and can be recorded within a week.
3. Calculate the level of parasitism using the following formula:
$$\% \text{ parasitism} = (\text{number of eggs parasitized} / \text{total number eggs collected}) \times 100\%$$
4. Calculations should be made for every sampling occasion and data for the whole season can be plotted using both number of eggs and % parasitism on the Y-axis and date on the X-axis.
5. After processing the data and preparing the graphs, discuss the results with farmers, extension workers and researchers. The points of discussion should follow the sequence given in the objectives above.

Discussions:

1. Are eggs of American bollworm parasitized?
2. Is it true that parasitized eggs turn black?
3. If this is so, why?
4. What level of parasitism did you observe in both fields?
5. Is the level of parasitism consistent throughout the season?
6. If the level is consistent, why? If the level is not consistent, why not?
7. Are there any differences in level of parasitism between sprayed and unsprayed fields?
8. Why is there a difference?
9. What have we learned from this exercise?
10. How can we improve this study?

Discovering predators of eggs of American bollworm

Objectives:

- Observe predators of American bollworm eggs
- Identify key predators of eggs of American bollworm
- Estimate level of predation on American bollworm eggs

Materials:

Unsprayed cotton fields

Plastic/glass tubes

Scissors

Fine hair brush

Fine pointed forceps

Tissue paper

Labels

Cages made of nylon material (25 x 25 x 25 cm) to collect eggs of American bollworm

Moths collected in light traps or from field

Sugar/honey solution

Sweep net

Method:

1. Collect American bollworm moths from light traps or from net sweeps around cotton fields. Potted cotton plants are placed inside the cage before the captured moths are released inside. Place some sugar/honey solution on wet cotton wool in a saucer or plate inside the cage. Release the moths and keep them inside the cage for 24 hours. Remove the plants when eggs of American bollworm are found. Count the number of eggs found and using a marker pen make a mark next to the egg.
2. If it is difficult to transport potted plants into the field, cut leaf pieces with fresh eggs and staple these onto leaves of plants in the field. Mark the plants where the eggs are stapled.
3. With 4-10 participants take the pots with eggs or eggs with leaf pieces to the field. Place them at a suitable site so that participants can watch predators attacking the eggs. Observations can be carried out over a staggered period in the morning or evening. After some predators have been identified, these could be collected for further evaluation in the session room.
4. After processing the data and preparing the graphs discuss the results with farmers, extension workers and researchers. The points of discussion should follow the sequence given in the objectives above.

Discussions:

1. How many types of predators attack eggs of American bollworm?
 2. How many eggs can each predator feed on?
 3. What are the main predators?
 4. What have we learned from this exercise?
- Is there any way we can improve this study?

Direct observations of predators of larvae of American bollworm

This exercise should be carried out after trainers/farmers are familiar with predators of American bollworm. It should be carried out in a field where populations of predators and bollworms are sampled regularly as the population information will be used in the discussion at the end of the exercise.

This exercise is described as a farmer field school activity. It should be evaluated first by trainers before being used in farmer field schools.

Objective:

Observe predators of American bollworm larvae in cotton fields

Materials:

Unsprayed cotton fields

Plastic/glass tubes

Sweep nets

Fine hair brush

Tissue paper

Method:

1. Start this exercise early in the day, about 8.00 hours. Farmers should gather at a no spray field that has been sampled regularly. The farmers are divided into three groups and each group is assigned a separate part of the field. Each member of the group is required to find a predator. When everyone has found a predator, the trainer/farmer leader blows a whistle to initiate a 10-minute observation of the activity of each predator found. The number and size of prey eaten will be recorded by members of the group. At the end of the 10 minutes the trainer/farmer leader will blow the whistle again to terminate this study. All the farmers will return to the field school and the results are compiled and discussed.
2. The exercise may be repeated at different times of the day, e.g., at 11.00 hrs just to compare the rate of feeding or even presence of predators.
3. After the information has been summarized discuss the results with farmers, trainers and researchers. The points of discussion should follow the sequence given in the objectives above.

Discussions:

1. What are the predators of bollworm larvae?
2. How many can each predator eat?
3. Which predator is the most important?
4. Which stage of larvae is preferred?
5. What have we learned from this exercise?
6. Is there any way we can improve this exercise?
7. Are there more predators when there are more larvae?

Aphids and Jassids

Determining predators of cotton aphids

This exercise should initially be carried out by trainers to gain experience in confirming the predators in the cotton fields. It should then be carried out jointly with farmers in the Farmer Field Schools.

In the field, there will be many arthropods but most of them will be friendly ones that feed on pests. In this exercise, we will determine if these friendly arthropods are predators of aphids.

Objectives:

- Observe predators in cotton fields that feed on cotton aphids
- Observe preferred stage of aphid eaten by each kind of predator
- Count the number of aphids eaten by each kind of predator per day

Materials:

Sprayed and unsprayed cotton fields
Plastic/glass tubes
Scissors
Fine hair brush
Fine pointed forceps
Tissue paper
Labels

Method:

1. Collect arthropods for this exercise like spiders, ladybird beetles (larvae and adults), carabid beetles, chrysopa larvae and staphylinid beetles. To collect these predators do not use your fingers. Try coaxing the arthropod into clear plastic/glass tubes by placing the tube over the arthropod and tapping on the other side of the leaf. Each tube should have a drop of water to maintain humidity and should be covered with a lid. There is no necessity to make large numbers of holes in the lid. A single small hole will suffice if the lid is tight. If the lid is a snap on there is no need to make any hole. Also collect some aphids on leaves and bring these back to the session room.
2. In the session room, use a wet fine hair brush, remove aphids from the plant and place them together with an arthropod. Count the number of aphids in the tube and count them again after 24 hours. Remember to label the tube accordingly. Keep the tube in a cool place away from direct sunlight. At the end of the study, return the predator into the field unless an identification is necessary.
3. Upon confirmation that an arthropod is a predator, the next stage is to determine which stage of the aphid is preferred. In this part of the exercise, allow 5 aphid nymphs, 5 wingless adults and 5 winged adults together with each predator. Keep them together for 24 hours and count the number of each stage of aphids left. It is necessary to keep a short cotton stem in the tube for the aphids to rest on during the study. Repeat for other predators.
4. The next stage of the exercise requires an understanding of how many aphids can be consumed in a day. This is carried out using a larger tube with a piece of cotton stem. Each end of the stem is covered with moist cotton wool. Place about 30 aphids (of the preferred stage) in each tube and find out the number eaten in 24 hours.
5. After the data have been collected and the information has been summarized discuss the results with farmers, trainers and researchers. The points of discussion should follow the sequence given in the objectives above.

Discussions:

1. Which are the major predators of aphids?
2. How many can each predator eat?
3. Which stage of the aphid is preferred?
4. What have we learned from this exercise?
5. Is there any way we can improve this study?

Direct observations of predators of cotton aphids

This exercise should be carried out after trainers/farmers are familiar with predators of cotton aphids. It should be carried out in a field where populations of predators and aphids are sampled regularly as the population information would be used in the discussion at the end of the exercise.

This exercise is described as a farmer field school activity. It should be evaluated by trainers first before being used in Farmer Field Schools.

Objectives:

- Observe predators of cotton aphids in cotton fields
- Establish the number of aphids eaten by each predator in the field at a specific time
- Explain the importance of predators in controlling aphids

Materials:

Unsprayed cotton fields
Plastic/glass tubes
Whistle for trainer/farmer leader
Watch

Method:

1. Start this exercise early in the day, about 8.00 hours. Farmers should gather at an unsprayed field that has been sampled regularly. The farmers are divided into three groups and each group is assigned a separate part of the field. Each member of the group is required to find a predator. When everyone has found a predator, the trainer/farmer leader blows a whistle to initiate a 10-minute observation of the activity of each predator found. The number and size of prey eaten will be recorded by members of the group. At the end of the 10 minutes the trainer/farmer leader will blow the whistle again to terminate this study. All the farmers will return to the field school and the results are compiled and discussed.
2. This exercise may be repeated at different times of the day, e.g., 11.00 hours just to compare the rate of feeding or even presence of predators.
3. After the data have been summarized discuss the results with farmers, trainers and researchers. The points of discussion should follow the sequence given in the objectives above.

Discussions:

1. Are there differences in feeding rates at different times of the day?
2. How many can each predator eat?
3. Which stage of the aphid is preferred?
4. What have we learned from this exercise?
5. Is there any way we can improve this study?
6. Are there more predators when there are more aphids?

Measuring field parasitism of cotton aphids

Objectives:

- Identify kinds of parasitoids that attack cotton aphids
- Calculate the level of parasitism caused by the aphid parasitoids
- Observe seasonal fluctuations of aphid populations and level of parasitism
- Compare level of parasitism between sprayed and unsprayed fields

Materials:

Sprayed and unsprayed cotton fields
Plastic/glass tubes
Scissors
Fine hair brush
Fine pointed forceps
Tissue paper
Labels

Method:

1. Exercise to begin soon after cotton plants are established in the field. During weekly/fortnightly visits to the field, examine 20-50 plants and collect dark brown coloured aphids. To collect mummified aphids, use a pair of scissors and remove the plant tissue around the dead aphid and use the forceps to place the plant tissue and aphids into a clean plastic/glass tube. Occasionally, use the forceps directly to remove dead aphids from the plants and transfer these into the tubes. The tube should be lined with a moist tissue paper (not wet). For each field visit, the tube used should be labelled with field number, date and name of collector.
2. In the session room, use a wet fine hair brush, remove the aphids from the plant tissue and place them on clean moist tissue paper and place in another clean glass tube. Remember to label the tube accurately. Keep the tube in a cool place away from direct sunlight. Observe for emergence either daily or once in two days. Record and remove parasitoids and keep these in 70% alcohol for preservation and later identification.
3. The number of parasitized aphids should be compared with the total aphid population sampled on the same day. Usually this involves examining young shoots and leaves of cotton plants and counting the aphids present.
4. Calculate the level of parasitism using the following formula:
$$\% \text{ parasitism} = (\text{number of parasitized aphids} / \text{total aphids counted}) \times 100\%$$
5. Calculations should be made for every sampling occasion and data for the whole season can be plotted using both aphid populations and % parasitism on the Y-axis and date on the X-axis.
6. After the data have been summarized discuss the results with farmers, trainers and researchers. The points of discussion should follow the sequence given in the objectives above.

Discussions:

1. Why do parasitized aphids turn brown?
2. What level of parasitism was found?
3. Is the level of parasitism consistent throughout the season?
4. If the level is consistent, why? If the level is not consistent, why?
5. Is there any difference in level of parasitism between sprayed and unsprayed fields?
6. Why is there a difference?
7. What have we learned from this exercise?
8. Is there any way we can improve this study?

Determining predators of cotton jassids

Objective:

Observe predators that attack cotton jassids

Materials:

Sprayed and unsprayed cotton fields

Plastic/glass tubes

Scissors

Fine hair brush

Fine pointed forceps

Tissue paper

Labels

Method:

1. When jassid populations in the field are high, visit the field in the morning with farmers and trainers. Divide the participants into three groups and suggest competition between groups that will find the most number of predators of jassids (in terms of number of specimens and number of species). Each group will be given half an hour to search for predation activity and to record these. In addition, each predator will be collected. Collect jassids separately for study in the session room. In the session room, keep each predator with a known number of jassids of specific age group for 24 hours.
2. Keep the tube in a cool place away from direct sunlight. Record number of jassids and stage eaten. For identification, keep specimens in 70% alcohol for preservation.
3. After collecting information about predators of jassids discuss the results with farmers, trainers and researchers.

Discussions:

1. Are there any predators of jassids?
2. How many jassids can be eaten in 24 hours?
3. Which is the most important predator?
4. What have we learned from this exercise?
5. Is there any way we can improve this exercise?

Insect collection

Insects and spiders can be collected many ways. The best way is to sit in a field and watch the insects and spiders to observe their activity and behaviour. Keep a record of what specimens are doing in the field. Collecting can be done by hand, with a sweep net or with an aspirator. Kill insects by placing the specimens in a bag with a small amount of alcohol or by placing the bag into a freezer for an hour.

Insects, especially parasites and adult moths, can also be collected by collecting larvae or eggs in the field and rearing the insects until adult parasites or other insects emerge.

Insects and spiders that are collected can be divided into two groups. First are the hard-bodied insects that are usually adults. Second are soft-bodied insects, which are usually immature nymphs and larvae, and soft-bodied spiders. Hard-bodied insects should be placed on pins, and soft-bodied insects and spiders should be placed in 70% alcohol.

All specimens should be correctly labeled with the following information where appropriate:

Common name (Vietnamese, or local language)

Host (plant or insect)

Ecological function (plant feeder, predator, parasite, detritus feeder, etc)

Name of collector

Date collected

Place collected

Latin name

You should use thick paper with writing in black ink for labels on pins. Thick paper and writing in pencil should be used for specimens in alcohol.

Mounting insects on pins should be done as follows:

- For small insects, the insect can be glued on a triangular piece of paper using some glue. Clear nail polish is commonly used as a glue for this kind of mounting.
- Large insects can be put on a pin directly, put the pin through the thorax, the middle part of the insect.

Keep the collection in a safe place away from ants and other insects. Dry the insects well using a lamp. Keeping moth balls in the collection will reduce insect damage.

Note that collections are only a process in which to learn functions, structures and the names of insects. The final product is nice to look at, and usually very impressive, but the goal of collection is the actual collecting process. Collecting and mounting are good ways to get to know the insect and spider communities in your area, and to understand the ecological relationships between organisms in an ecosystem.

Bacillus thuringiensis

Assessment of viability of Bt

This exercise will use living organisms to determine if Bt has maintained its toxicity in storage or whether the Bt purchased from a store is still useful for application in the field.

Introduction:

Cotton caterpillar pests such as *Helicoverpa armigera* have become resistant to a wide range of chemical insecticides. From different research studies around the world, Bt has been shown to effectively control *Helicoverpa armigera* and other caterpillar pests. However, since it is a sensitive biological agent, it is subject to rapid breakdown and loses its killing power. Use of Bt is part of an IPM programme that works with other natural enemies to control cotton pests. Chemical insecticides do not do this. Therefore, this exercise is to discover the toxicity of Bt as well as to determine if the Bt bought from the local shop is still useful. After this exercise, the farmer/trainer will be able to answer the common question: Did I buy a good Bt and will it be effective against the caterpillar pest in my field?

Objective: Answer the common question: Did I buy good Bt and will it be effective against the caterpillar pest in my field?

Materials:

- 1 unsprayed cotton plant
- 2 camel or fine hair brushes
- 1 pair of scissors
- 10 plastic cups with plastic/organdie sheets used to cover the cup with rubber bands
- Different kinds of Bt bought from a local shop (use a different brand of BT for each group)
- 1 litre of clean water
- 16 or more bollworm/*Helicoverpa armigera* larvae - preferably small ones
- 1 roll of tissue paper

Method:

1. Fill 2 plastic cups with water. Mix 1/4 teaspoon Bt into water in one cup. Label the cup "Bt" and the other cup "Water".
2. Collect fresh leaves from the unsprayed cotton plant. Cut the leaves into sections of 1" diameter.
3. Dip one leaf section into the "Bt" cup and continue for three other sections. Similarly, dip four leaf sections into "Water" cup.
4. After removing the leaf sections from the solution, place one in each cup and label according to the treatment used. There should be four sections treated with "Bt" and four more with "Water".
5. Each cup should be lined with tissue paper. The leaf sections should be allowed to dry in a cool, shaded place.
6. After the leaf sections are fairly dry, using the brush, transfer two caterpillars into each of the leaf sections. Avoid damaging the caterpillars. Quicker results are obtained if smaller caterpillars are used. Do not use too many caterpillars per leaf section as they may be cannibalistic.
7. Each cup should be covered with either the plastic or organdie sheet held securely with rubber bands.

Observations:

Observe after 12 hours, 24 hours, 48 hours, and 72 hours. Record observations on table suggested below taking note of the leaf damage, frass production (droppings of caterpillars), and the state of the larvae. Usually, obvious differences can be seen within 1.5 day.

Discussions:

1. What happened to the larvae in the different treatments?
2. Is there any difference in the amount of frass produced by the caterpillars? If yes, why so?
3. Why did we line the cup with tissue paper?
4. Why did we place the cups in the shade?
5. Why did we include a comparison with water?

| Items | Treatment | Bt A | | | Bt B | | | Bt C | | | Bt D | | | Control (water) | | |
|------------------|-----------|------|---|---|------|---|---|------|---|---|------|---|---|-----------------|---|---|
| | hours | 1 | 2 | 3 | 1 | 2 | 3 | 1 | 2 | 3 | 1 | 2 | 3 | 1 | 2 | 3 |
| Leaf damage | 12 | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | |
| Frass production | 12 | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | |
| State of larvae | 12 | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | |
| Total | 12 | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | |

Scoring system

Leaf damage

- 1: low**
- 2: moderate**
- 3: high**

Frass production

- 1: none**
- 2: little**
- 3: much**

State of larvae

- 1: dead**
- 2: moribound**
- 3: active**

Inhibition of larval feeding by Bt

This study will show how Bt inhibits larval feeding. Many farmers spray Bt without seeing immediate kill of the target insect. This is because Bt acts slower than conventional chemical insecticides but no less effective. Before actual death occurs, feeding by larvae stopped. This often causes farmers to think that Bt is not effective. However, the benefits of Bt (conservation of parasitoids and predators, overall minimal health risk to farmers and consumers, minimal adverse effects on the environment) far outweighs the speed of killing caterpillars using chemical insecticides. Moreover, resistance in the target caterpillars to chemical insecticides have rendered them less effective than Bt. After this exercise, the farmer/trainer should be able to understand how Bt kills the target caterpillar and realize that Bt makes the pest stop feeding hence, there is less damage caused.

Objective: Understand how fast Bt kills the target caterpillar and to realize that Bt makes the pest stop feeding hence there is less damage caused.

Materials:

- 1 unsprayed cotton plant
- 2 camel or fine hair brushes
- 1 pair of scissors
- 10 plastic cups with plastic/organodie sheets and rubber bands
- 1 roll of tissue paper
- 1 packet of Bt
- 2 litres of clean water
- 1 plastic pail/container
- 1 long wooden stirrer
- 1 set of paper and pencil

Method:

1. Collect fresh leaves from the upper part of the cotton plant. Cut leaves into 1" diameter sections.
2. Using a pail or containers, pour a litre of water and mix the recommended dose of Bt on the label. Mix well using a long stirrer.
3. Dip four leaf sections into the pail with Bt solution. Place one section per cup lined with tissue paper and label "Bt". Using another four leaf sections, dip into a cup with only water and place these into separate cups labeled "Water".
4. The next morning, check for feeding and/or larval death. Replace the leaf sections (Bt treated ones in the "Bt" cup and water treated ones in "Water" cup).
5. At noon, check again on feeding. Using paper and pencil, trace the area of the leaf section from the "Bt" cup and the "Water" cup. Replace the leaves removed for drawing. Compare the leaf tracings from both "Bt" and "Water" cup. Observe the amount of faecal matter in both sets of treatments.
6. Repeat the above observations in the late afternoon and continue to three days.

Discussions:

1. Were there any differences in feeding between Bt treated and water treated leaf sections?
2. When did these differences occur?
3. Were there any differences in amount of faecal matter produced?
4. What do these differences indicate?
5. Did you observe the larvae to stop feeding? What could have caused this to happen?

Sensitivity of Bt to sunlight

This study will show how sunlight breaks Bt down. Since Bt is a biological agent, it is sensitive to sunlight. In bright sunlight, it loses its effectiveness and strength to kill caterpillars. After this study, you should be able to appreciate the effect of sunlight on the effectiveness of Bt and to make appropriate decisions on how to apply Bt.

Objective: Make appropriate decisions on how to apply Bt

Materials:

- 2 rows of cotton plants (about 15-30 days after planting)
- 1 potted cotton plant (about 15-30 days after planting)
- 16 bollworm or similar caterpillars (small and of similar size)
- 1 hand sprayer (1 litre size will suffice)
- 1 packet of Bt
- 3 camel or fine hair brushes
- 1 pair of scissors
- 1 pail of clean water
- 12 plastic cups with plastic/organdie sheets and rubber bands

Method:

1. Mix Bt at recommended rate in a pail of water and spray one row of cotton plants at midday. Use about four plants.
2. In the evening, just before sunset, spray another row of cotton plants (another 4 plants).
3. An hour after the last spray, collect leaves from both rows and cut out leaf sections of 1" diameter and ensure that these are labeled. Similar sections are prepared from a potted plant free of insecticides.
4. Each of the leaf section is kept in a plastic cup lined with tissue paper and the cups labeled as "Bt-sunlight", "Bt-no sun" and "No Bt".
5. Caterpillars collected from the field (preferably smaller ones as these react faster than older caterpillars) were used for the study. Two caterpillars are dropped onto each leaf section and the cups stored in a cool, shade place.
6. Observe for signs of feeding (size of holes made in the leaf section as well as amount of faecal matter produced) as well as record number of living larvae. Continue the study for up to 3 days.

NOTE: This study may be repeated at two days interval to determine the effectiveness of Bt on cotton in the field.

Discussions:

1. Did the larvae feed on the leaves?
2. Did any of the larvae die? In which treatment?
3. What do you think was the effect of sunlight on Bt?
4. Why should we repeat the study?
5. When is the best time of the day to apply Bt?

Effect of Bt on predators and parasitoids

This study will attempt to show the impact of spraying Bt on both predators (insects or spiders that eat other insects, particularly pests) and parasitoid (insects that lay eggs in or on its host so that the host provides food for the young stages of the parasitoid). A danger in using chemical insecticides is that it kills friendly insects that help farmers control pest organisms. As Bt is applied as a spray, this exercise will help farmers to discover the impact of Bt on these beneficial insects. After this activity, you should be able to relate the action of Bt on a natural enemy and better appreciate its role in an IPM programme.

Objective: Describe the effect of Bt on a natural enemy and better appreciate the role of Bt in an IPM programme

Materials:

- 2 cotton plants (15-30 days after planting)
- 2 hand sprayers (1 litre)
- 1 pail of clean water
- 1 packet of Bt
- 4 large plastic cups with organdie cloth sheet and rubber bands
- 2 camel or fine hair brushes
- 10 parasitoid cocoons
- 10 common predators from cotton field
- 10 clear plastic film containers
- 1 small bottle of honey
- 1 roll of cotton wool
- 1 roll of tissue paper

Method:

1. Place one each of the parasitoid cocoons collected from the field into the film containers. Store the containers in a cool shaded place until adult parasitoids emerge. Feed the adult parasitoids with a diluted honey solution (on a moist cotton wool).
2. When there are sufficient adult parasitoids, mix Bt at the recommended rate and spray a cotton plant with it. Allow an hour to dry.
3. Collect leaves from the upper part of the plant and cut out leaf sections of 1" diameter size and place these into the large plastic cups with cover. Label each cup.
4. Leaves from an unsprayed plant should be collected and similarly prepared. Place a solution of diluted honey in each plastic cup and introduce a parasitoid into each of the cups and secure the cover with rubber bands.
5. Store the cups in a cool shaded place and observe every day. Record the number of dead parasitoids in each situation.
6. A similar study is conducted with field collected predators (e.g. spiders, syrphid larvae etc.). However, with predators there is no need for honey solution.

Discussions:

1. Why did we put a diluted honey solution in cups with parasitoids?
2. Was there any dead parasitoid or predator in the cups? Why?
3. Do you think that Bt kills parasitoids and predators?

Nuclear Polyhedrosis Virus

Identification of virus-infected insects (classroom exercise)

Introduction

Today, natural viruses are used to manage insect pests and to reduce the use of chemical insecticides that are harmful for man and the environment. Natural viruses as a biocontrol agent have become a component of the IPM system. In Vietnam, the Biological Control Research Center, National Institute of Plant Protection (NIPP) has done some work on methods of production and application of viruses in controlling insect pests. The methods have been simplified to allow trainers and farmers to work with the agent with minimum support from the research institute and trainers. This exercise will provide trainers and farmers first hand experience in the classroom in observing and comparing symptoms of virus-infected insect pests and insects which died because of other reasons, e.g., pesticides, etc.

Objective: Describe symptoms of virus-infected insects

Materials:

Specimen, i.e., virus-infected insects from the laboratory (enough material for all five small groups)
Paper and markers

Procedure:

Use this exercise at the start of the season, when infected insects or insects which have died because of virus are not yet readily seen in the field. Ask each small group to recall their experiences on seeing dead or infected insects in the field. Each group should then draw their observations on big paper for presentation.

After all groups have presented their outputs, introduce the specimens of virus-infected insects from the laboratory. Ask each group to describe symptoms of the specimens from the laboratory.

(Note: Insects infected by viruses become weak and activity is slowed down; the body color is changed; the cuticle becomes fragile and ruptures easily when touched, releasing the body content which has become liquefied. Dead larvae may be found hanging from or lying on leaf or plant surfaces with no filamentous structure on the cuticle.)

Discussions:

1. Based on experience, describe different appearance or symptoms exhibited by dead insects in the field? Describe the field conditions at the time the observations were made. Discuss about host populations, insecticide use, weather etc.
2. Describe appearance and characteristics of specimen from the laboratory. Have insects with such appearance and characteristics been observed in the field? What could have caused such symptoms? What does this mean for management of insect pests?

Identification of virus-infected insects (field exercise)

Introduction

Today, natural viruses are used to manage insect pests and to reduce the use of chemical insecticides harmful for man and the environment. Natural viruses as a biocontrol agent have become a component of the IPM system. The Biological Control Research Center, National Institute of Plant Protection (NIPP) has done some work on methods of production and application of viruses in controlling insect pests. The methods have been simplified to allow trainers and farmers to work with the agent with minimum support from the research institute and trainers. This exercise will provide trainers and farmers first hand experience in the field in observing and comparing symptoms of virus-infected insect pests and insects which died because of other reasons, e.g., pesticides, etc.

Objective: Describe symptoms of virus-infected insects

Materials:

Specimens collected from fields
Paper and markers

Procedure:

Do this activity once viruses are seen to spread in the FFS area. (Initially, inoculum may be introduced from the laboratory.) Ask each group of five to recall the classroom exercise done earlier, i.e., the discussion re: experiences on seeing dead or infected insects in the field and introduction of specimen from the laboratory.

Go to the field. Ask each subgroup to collect all dead insects that they see in the field.

In the classroom, groups should sort the dead insects based on appearance and symptoms exhibited. Then, using the group's drawing from the earlier exercise, groups present their field observations, i.e., their collection of insects from the field.

(Note: Insects infected by viruses become weak and activity is slowed down; the body color is changed; the cuticle becomes fragile and ruptures easily when touched, releasing the body content which has become liquefied. Dead larvae may be found hanging from or lying on leaf or plant surfaces with no filamentous structure on the cuticle.)

Discussions:

1. Based on experience, describe different appearance or symptoms exhibited by dead insects in the field? Describe the field conditions at the time the observations were made. Discuss about host populations, insecticide use, weather etc.
2. Were there insects with the same appearance and characteristics as the specimen from the laboratory? What does this mean for management of insect pests?

Method of production and application of virus in managing insect pests on cotton

Introduction

In the natural setting, insect pests are infected by many microorganisms like viruses, bacteria, fungi, protozoa, etc. In a number of cases, viruses have been recognized as a biocontrol agent in checking insect pest population. Every year, during months when the climatic condition is hot and sunny and the humidity is high, several insect pests on cotton, e.g., leaf roller (*Silepta derogata*) and armyworm (*Spodoptera litura*) are infected by viruses. This helps reduce the caterpillar population.

Today, natural viruses are used to manage insect pests and to reduce the use of chemical insecticides which are harmful for man and the environment. Natural viruses as a biocontrol agent have become a component of the IPM system. In Vietnam, the Biological Control Research Center, National Institute of Plant Protection has done some work on methods of production and application of viruses in controlling insect pests. The methods have been simplified to allow trainers and farmers to work with the agent with minimum support from the research institute and trainers.

Objective: Experience producing virus for use in testing whether they infect other insects

Materials:

Small rectangular plastic containers/penicillin vials and wood rack (depending on species of larvae)
Muslin cloth for filtering virus solutions
Paper to cover the plastic containers
Mortar and pestle
Glass bottle with cover 0.5 liter capacity
Pincers
Dark plastic can or glass bottle to keep virus suspension
A large number of small larvae
Natural diet: cotton leaves, etc.
Jaggery or vegetable oil
Boiled water (boil for 20-30 minutes and let it cool)

Procedure:

I. Producing the virus solution

There are two procedures for producing virus solutions. Virus products are prepared using infected larvae collected from the field or larvae infected in the laboratory.

Procedure A: Using infected larvae collected from the field

1. Look out for the time of appearance and the development of disease in natural conditions. As soon as they are observed, collect dead larvae from the field and store them in a covered glass bottle to produce the virus products.
2. Putrefy for two – three days.
3. Macerate in mortar.
4. Add a little boiled water at a time, stir and filter through muslin cloth to discard tissue debris. Repeat this step until the extract obtained is clear.
5. To extract from 500 – 1000 large-sized larvae, add one liter boiled water. Do not use unboiled water because it can ruin products.
6. After filtering, store the mixture in colored bottles or dark cans. Store in a cool dark place.
7. Add 0.5% jaggery before using to spray in the evening.

Procedure B-1: Using small-sized larvae infected in the laboratory

1. Mass rearing of insects
A large number of larvae is needed to produce viruses. Rear the larvae in clean plastic containers with natural diet, i.e., cotton leaves, etc. Replace leaves every day. Leaves should be fresh and

not too old. Cover the containers with cloth or paper to keep larvae from escaping. When the larvae have grown up to 10 - 15 mm long, they can be infected with the inoculum.

2. Preparing the inoculum

To create an initial disease source or inoculum, collect larvae from the field exhibiting symptoms of virus infection. Use one (1) rather big larva (about 30mm long) to 100ml clean boiled water. With the mortar and pestle, grind the diseased dead larvae. Add a little clean boiled water at a time, stir and filter through muslin cloth to discard tissue debris. Repeat this step until the extract obtained is clear. Do not use unboiled water because it can ruin products. Soak food plant leaves in the liquid, air dry them, and then feed them to the healthy larvae in the containers.

3. Preparation of virus product/solution

Observe the larvae every day. Three to four days after the symptoms of disease have appeared and larvae begin dying, use pincers to remove all the dead larvae and transfer them into a glass bottle with lid/cover. Discard all the dead larvae not infected by virus.

4. Putrefy for 2 - 3 days.

5. Macerate in mortar. Add a little clean boiled water at a time, stir and filter through muslin cloth to discard tissue debris. Repeat this step until the extract obtained is clear.

6. To extract from 500 – 1000 large-sized larvae, add one liter boiled water. Do not use unboiled water because it can ruin products.

7. After filtering, store the mixture in colored bottles or dark cans. Store in a cool dark place.

Procedure B-2: Using medium-sized larvae infected in the laboratory

1. Collect medium-sized, healthy larvae from the field.

2. Inoculate larvae by feeding virus treated leaves for two days. To prepare the leaves, soak them in the inoculum and air dry before feeding to the healthy larvae. (For details, refer to Procedure B-2 step 2.)

3. Infected larvae will turn white and die in seven days. Collect diseased larvae in clean boiled water.

4. Putrefy for 2 - 3 days.

5. Macerate in mortar. Add a little clean water at a time, stir and filter through muslin cloth to discard tissue debris. Repeat this step until the extract obtained is clear.

6. To extract from 500 – 1000 large-sized larvae, add one liter clean boiled water. Do not use unboiled water because it can ruin products.

7. After filtering, store products in colored bottles or dark cans. Store in a cool, dark place.

II. Applying the virus solution

Larvae in the final instar are resistant to the virus. However, the virus will efficiently control earlier instar larvae if applied as follows:

1. Use a dosage of extract from 500 – 1000 large-sized, diseased larvae to 600 - 800 liters of water per hectare.

2. Add 0.5% jaggery or vegetable oil.

3. Spray 2-3 times at intervals of 7-10 days.

4. Spray in the evening hours to prevent destruction of the virus by the UV fraction of sunlight.

Avoid:

1. Brackish water for storing as well as spraying the virus

2. Grown up caterpillars for virus inoculation

3. Spraying in hot, sunny conditions.

Discussions:

1. How are larvae infected by the virus?

2. Were viruses found in the FFS fields? Describe conditions in the field at the time when the viruses were observed. What factors influence whether or not viruses can spread? Discuss about host populations, insecticide use, weather etc.

3. What action should the farmer group take if it wants more farmers in the area to make use of viruses? How can this be done?

Spread of viruses to surrounding fields

Introduction

In the ToF field, viruses (NPV) have been sprayed and groups have made weekly observations in the study area. If you start finding diseased/deceased insects (insects that are sick or have died because of virus) regularly in the ToF area, it will be good to find out whether the viruses have also spread to surrounding fields. Carry this activity out as a special topic, once you start to see the virus spread in the ToF area. It can also be repeated several times during the season to determine the extent of spread of the virus.

Objective: Find out if the viruses can spread to other fields surrounding the ToF area

Materials:

Fields surrounding the FFS area
Paper and markers

Procedure:

Ask each small group of five to select three fields close to the ToF area. Each group will observe 30 plants in each field following the same methods as in the FFS area:

Spodoptera: number of larvae per plant
 number of pupae per plant
 if possible, the number of egg mass per plant (though it is quite difficult to see) If too difficult, do not observe.
 number of diseased/deceased larvae (by virus infection)

Heliothis: number of larvae per plant
 number of pupae per plant
 if possible, also the number of eggs per plant (though it is quite difficult to see) If too difficult, do not observe.
 number of diseased/deceased larvae (by virus infection)

After observations by each small group in the field, ask each group to summarize the following information:

number of larvae of *Spodoptera litura* per plant
number of larvae of *Heliothis* per plant
number of virus diseased/deceased larvae of *Spodoptera litura* per plant
number of virus diseased/deceased larvae of *Heliothis* per plant
 $\% \text{ of infection by NPV} = \frac{\# \text{ of virus diseased or deceased larvae}}{\text{Total number of larvae}}$

(Note: The total number of larvae = healthy larvae + virus diseased or deceased larvae + diseased or deceased larvae due to other factors)

(If parasites were observed, groups should also summarize % of parasitized eggs or larvae.)

Ask each group also to record the stage of crop development.

Make one map indicating the fields each group observed. Write down on the map the number of insect pests and how many of these are sick or died due to virus and the % of infection in each field.

Discussions:

1. Were viruses found in the surrounding fields?
2. What factors influence whether or not viruses can spread? Discuss about host species, host populations, insecticide use, weather etc.
3. If activity was repeated at a later stage: How does the situation of surrounding fields compare with the previous observation? Did the number of virus-infected insects increase or decrease?
4. What action should the farmer group take if it wants more farmers in the area to make use of viruses? How can this be done?

Assessment of viability of NPV

This exercise will use living organisms to determine if NPV has maintained its toxicity in storage or whether the NPV purchased from a store is still useful for application in the field.

Introduction

Cotton caterpillar pests such as *Helicoverpa armigera* have become resistant to a wide range of chemical insecticides. From different research studies around the world, NPV has been shown to effectively control *Helicoverpa armigera* and other caterpillar pests. However, since it is a sensitive biological agent, it is subject to rapid breakdown and loses its killing power. Use of NPV is part of an IPM programme that works with other natural enemies to control cotton pests. Chemical insecticides do not do this. Therefore, this exercise is to discover the toxicity of NPV as well as to determine if the NPV bought from the local shop is still useful. After this exercise, the farmer/trainer will be able to answer the common question: Did I buy a good NPV and will it be effective against the caterpillar pest in my field?

Objective: Answer the common question: Did I buy good NPV and will it be effective against the caterpillar pest in my field?

Materials:

1 teaspoon
Marker for labelling cup
Pincers for dipping leaf sections
1 unsprayed cotton plant
2 camel or fine hair brushes
1 pair of scissors
10 plastic cups with plastic/organdie sheets used to cover the cup, and rubber bands
Different kinds of NPV bought from a local shop (use a different brand of NPV for each group)
1 litre of clean water
16 or more *Spodoptera* or fewer *Heliothis* larvae - preferably small ones
1 roll of tissue paper

Procedure:

1. Fill two plastic cups with water. Mix 1/4 teaspoon NPV into water in one cup. Label the cup "NPV" and the other cup "Water".
2. Collect fresh leaves from the unsprayed cotton plant. Cut the leaves into sections of 1" diameter.
3. Dip one leaf section into the "NPV" cup and continue for three other sections. Similarly, dip four leaf sections into "Water" cup.
4. After removing the leaf sections from the solution, the leaf sections should be allowed to dry in a cool, shaded place.
5. Place one in each cup and label according to the treatment used. There should be four cups with leaf sections treated with "NPV" and four more with leaf sections treated with "Water". Each cup should be lined with tissue paper.
6. Using the brush, transfer two caterpillars onto each of the leaf sections. Avoid damaging the caterpillars. Quicker results are obtained if smaller caterpillars are used. If using *Heliothis*, use one caterpillar per leaf section as they may be cannibalistic.
7. Each cup should be covered with either the plastic or organdie sheet held securely with rubber bands.

Observations:

Observe after 12 hours, 24 hours, 48 hours, and 72 hours. Record observations on table suggested below taking note of the leaf damage, frass production (droppings of caterpillars), and the state of the larvae. Usually, obvious differences can be seen within 1.5 day.

Discussions:

1. What happened to the larvae in the two treatments?
2. Is there any difference in the amount of frass produced by the caterpillars? If yes, why so?
3. Why were cups lined with tissue paper?
4. Why were cups placed in the shade?
5. Why was a comparison with water included?
6. Did any of the NPV products perform better than others?
7. Did I buy good NPV?

| Items | hours | NPV A | | | NPV B | | | NPV C | | | NPV D | | | Control (water) | | |
|------------------|-------|-------|---|---|-------|---|---|-------|---|---|-------|---|---|-----------------|---|---|
| | | 1 | 2 | 3 | 1 | 2 | 3 | 1 | 2 | 3 | 1 | 2 | 3 | 1 | 2 | 3 |
| Leaf damage | 12 | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | |
| Frass production | 12 | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | |
| State of larvae | 12 | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | |
| <u>Total</u> | 12 | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | |

Scoring system

Leaf damage

- 1: low
- 2: moderate
- 3: high

Frass production

- 1: none
- 2: little
- 3: much

State of larvae

- 1: dead
- 2: moribound
- 3: active

Inhibition of larval feeding by NPV

Introduction

This study will show how NPV inhibits larval feeding. Many farmers spray NPV without seeing immediate kill of the target insect. This is because NPV acts more slowly than conventional chemical insecticides, though it is no less effective. Before actual death occurs, feeding by larvae stops. This slow action often causes farmers to think that NPV is not effective. However, the benefits of NPV (conservation of parasitoids and predators, overall minimal health risk to farmers and consumers, minimal adverse effects on the environment) far outweighs the benefit of speedy killing of caterpillars using chemical insecticides. Moreover, resistance (in the target caterpillars) to chemical insecticides has rendered insecticides less effective than NPV.

Objective: Understand how fast NPV kills the target caterpillar and to realize that NPV makes the pest stop feeding hence there is less damage caused.

Materials:

- 1 unsprayed cotton plant
- 2 camel or fine hair brushes
- 1 pair of scissors
- 8 plastic cups with plastic/organdie sheets and rubber bands
- 1 roll of tissue paper
- 1 packet of NPV
- 2 litres of clean water
- 1 plastic pail/container
- 1 long wooden stirrer
- Pincers for dipping leaves
- 1 set of paper and pencil
- Marker for labelling cups
- Caterpillars

Procedure:

1. Collect fresh leaves from the upper part of the cotton plant. Cut leaves into 1" diameter sections.
2. Using a pail or containers, pour a litre of water and mix the recommended dose of NPV on the label. Mix well using a long stirrer.
3. Dip four leaf sections into the pail with NPV solution.
4. Place one section in each of four cups lined with tissue paper and label "NPV".
5. Dip another four leaf sections into a cup with only water, and place each of these into a separate cup labelled "Water".
6. Place caterpillar in each cup with camel hairbrush.
7. The next morning, check for feeding and/or larval death.
8. Replace the leaf sections (NPV treated ones in the "NPV" cup and water treated ones in "Water" cup).
9. At noon, check again on feeding.
10. Using paper and pencil, trace the area of the leaf section from the "NPV" cup and the "Water" cup.
11. Replace the leaves removed for drawing.
12. Compare the leaf tracings from "NPV" and "Water" cups.
13. Observe the amount of frass, caterpillar droppings in both treatments.
14. Repeat the above observations in the late afternoon, and continue up to three days.

Discussions:

1. Were there any differences in feeding/and caterpillar deaths between NPV-treated and water-treated leaf sections?
2. When did these differences occur?
3. Were there any differences in amount of frass produced?
4. What do these differences indicate?
5. Why did the larvae stop feeding, if they did?
6. What does this mean for crop damage after NPV is applied?

Sensitivity of NPV to sunlight

Introduction

Since NPV is a biological agent, it is sensitive to sunlight. In bright sunlight, it loses its effectiveness and strength to kill caterpillars. This study will show that sunlight breaks NPV down.

Objective: Make appropriate decisions on how to apply NPV

Materials:

2 rows of cotton plants (about 15-30 days after planting), untreated with any pesticide
1 potted cotton plant (about 15-30 days after planting), untreated with any pesticide
16 *Spodoptera* or fewer *Heliothis* (or similar caterpillars) (small and of similar size)
1 hand sprayer (1 litre size will suffice)
1 packet of NPV
3 camel or fine hair brushes
1 pair of scissors
1 pail of clean water
12 plastic cups with plastic/organdie sheets and rubber bands

Procedure:

1. Mix NPV at recommended rate in a pail of water and spray one row of cotton plants at midday. Use about four plants.
2. In the evening just before sunset, spray another row of cotton plants (another 4 plants).
3. An hour after the last spray, collect leaves from the canopy of both rows, cut out 4 leaf sections of 1" diameter, and ensure that these are labelled.
4. Prepare four similar sections from a potted plant free of insecticides.
5. Keep each of the leaf sections in a separate plastic cup lined with tissue paper and label as "NPV-sunlight", "NPV-no sun" and "No NPV".
6. Collect caterpillars from the field (preferably smaller ones, as these react faster than older caterpillars) for the study.
7. Using a camel hairbrush, drop two caterpillars (one if using *Heliothis*) onto each leaf section.
8. Store the cups in a cool, shady place.
9. Observe signs of feeding (size of holes made in the leaf section, as well as amount of frass produced).
10. Record the number of living larvae.
11. Continue the study for up to three days.

NOTE: Use this same method at two-day intervals to determine the effectiveness of NPV on cotton in the field.

Discussions:

1. Did the larvae feed on the leaves?
2. Did any of the larvae die? In which treatment?
3. What was the effect of sunlight on NPV?
4. Why should the study be repeated?
5. When is the best time of the day to apply NPV?

Effect of NPV on predators and parasitoids

Introduction

This exercise will attempt to show the impact of spraying NPV on both predators (insects or spiders that eat other insects, particularly pests) and parasitoids (insects that lay eggs in or on its host so that the host provides food for the young stages of the parasitoid). A danger in using chemical insecticides is that it kills friendly insects that help farmers control pest organisms. Since NPV is applied as a spray, this exercise will help farmers to discover the impact of NPV on these beneficial insects.

Objective: Describe the effect of NPV on a natural enemy and better appreciate the role of NPV in an IPM programme

Materials:

2 cotton plants (15-30 days after planting)
2 hand sprayers (1 litre)
1 pail of clean water
1 packet of NPV
4 large plastic cups with organdie cloth sheet and rubber bands
2 camel or fine hair brushes
10 parasitoid cocoons
10 common predators from cotton field
10 clear plastic film containers
1 small bottle of honey
1 roll of cotton wool
1 roll of tissue paper

Procedure:

1. Place one parasitoid cocoon collected from the field into each of the film containers.
2. Store the containers in a cool shaded place until the adult parasitoids emerge.
3. Feed the adult parasitoids with a diluted honey solution (on a moist cotton wool).
4. When there are sufficient adult parasitoids, mix NPV at the recommended rate and spray a cotton plant with it. Allow the plant to dry for an hour.
5. Collect leaves from the upper part of the plant and cut out leaf sections of 1" diameter and place these into each of two large plastic cups with cover. Label each cup.
6. Two cups with leaves from an unsprayed plant should be similarly prepared.
7. Place a solution of diluted honey in each plastic cup.
8. Introduce parasitoids into each of the cups and secure the cover with rubber bands.
9. Store the cups in a cool shaded place and observe every day.
10. Record the number of dead parasitoids in each situation.
11. A similar study is conducted with field collected predators (e.g. spiders, syrphid larvae etc.). With predators there is no need for honey solution.

Discussions:

1. Why was diluted honey solution placed in cups with the parasitoids?
2. Was there any dead parasitoid or predator in the cups? Why?
3. Does NPV kills parasitoids and predators? Why or why not?
4. What role could NPV play in an IPM programme?

Effect of fungicides on the viability of NPV

Introduction

We have seen how insect pests can be infected by microorganisms like viruses that are now recognized as a biological control agent in checking insect pest populations. We also know from experience that farmers still have problems managing crop diseases, and use fungicides to control disease. NPV is a sensitive biological agent. Since NPV is a causal organism for disease in insect pests and farmers use fungicides to control disease in the crop, there is a danger that fungicides may inhibit the action of NPV. This exercise will attempt to show the effect of fungicide sprays on the viability of NPV.

Objective: Explain the effect of fungicides on the action of NPV

Materials:

Method A:

20 plastic cups covered with plastic/muslin cloth and secured with rubber bands
32 or more *Spodoptera* or 16 or more *Heliothis* larvae - preferably small ones

Method B:

20 plastic cups covered with plastic/muslin cloth and secured with rubber bands
32 or more *Spodoptera* or 16 or more *Heliothis* larvae - preferably small ones
Four small hand sprayers (0.5 liter capacity)

For both methods:

1 unsprayed cotton plant
2 camel or fine hair brushes
1 pair of scissors
NPV preparation
Fungicide
1 litre of clean water
1 roll of tissue paper
Labels
Paper and pen

Method A: Using leaf sections

1. Fill 4 plastic cups with water. Prepare solutions for 4 treatments:
 - Mix 1/4 teaspoon NPV into water in one cup and label the cup "NPV"
 - Mix fungicide based on recommended dose into water in one cup and label the cup "fungicide"
 - Mix 1/4 teaspoon NPV and fungicide based on recommended dose into water in one cup and label the cup "NPV + fungicide"
 - Keep one cup only with water, and label the cup "Control"
2. Collect fresh leaves from the unsprayed cotton plant. Cut a total of sixteen leaf-sections, each section measuring 1" in diameter.
3. Dip four leaf sections into the "NPV" cup. Similarly, dip four leaf sections into the "Fungicide" cup. Do the same for the "NPV + fungicide" and the "Water" cups.
4. After removing the leaf sections from the solutions, allow them to dry in a cool, shaded place.
5. Line each cup with tissue paper.
6. When the leaf sections are fairly dry, place one section in each cup. Label cups according to the treatment used. There should be four cups for each treatment.
7. Using the brush, transfer two caterpillars onto each of the leaf sections. Avoid damaging the caterpillars.
8. To obtain quicker results use smaller caterpillars. If using *Heliothis*, use one caterpillar per leaf section, as they may be cannibalistic.
9. Cover each cup with the plastic/muslin cloth and secure with rubber bands.

Method B: Spraying directly on insects

1. Prepare four hand sprayers before the practical. If a sprayer has been used before, wash it thoroughly with detergent.
2. Prepare solutions according to the four treatments below. Put one solution each of the hand sprayers and correspondingly. The four preparations are:
 - “NPV”: Mix 1/4 teaspoon NPV with one cup of water
 - “Fungicide”: Mix fungicide with one cup of water based on recommended dose
 - “NPV + Fungicide”: Mix 1/4 teaspoon NPV and fungicide based on recommended dose with one cup of water
 - "Control": Put only water
3. Spray four pieces of muslin cloth with one of each treatment and air-dry the pieces of cloth.
4. Collect fresh leaves from the unsprayed cotton plant. Cut a total of sixteen leaf-sections, each section measuring 1" in diameter.
5. Line each cup with tissue paper and place one leaf section in each.
6. Collect several caterpillars from the field. Avoid damaging the caterpillars. Quicker results are obtained if smaller caterpillars are used.
7. Using the brush, transfer two caterpillars onto each of the leaf sections. If using *Heliothis*, use one caterpillar per leaf section as they may be cannibalistic.
8. Cover each cup with the muslin cloth and secure with rubber bands.
9. Label cups according to the treatment used. There should be four cups for each treatment.
10. Check the cups every 10-12 hours and look for frass (droppings of caterpillars) and larval death. Usually, obvious differences can be seen within 3- 4 days.

Discussions:

1. What happened to the larvae in the four treatments?
2. Is there any difference in the amount of frass produced by the caterpillars? If yes, why so?
3. How many days did it take to observe symptoms of disease on the caterpillars? Describe the symptoms and time of occurrence of disease on caterpillars in the different treatments. Do caterpillars in one treatment exhibit more symptoms than those in other treatments? Did they get sick sooner? What are the possible reasons for these differences?
4. What do these observations mean for the use of NPV in managing insect pests?
5. What do these observations mean for the use of fungicides in managing crop diseases?

Pesticides

Understanding farm pesticide labels

Introduction

The main objectives of IPM training are helping farmers to increase their knowledge, enabling them to take maximum advantage of ecological relationships to control pests, and to reduce their pesticide use as much as possible in order to protect the environment and produce clean agricultural products. However, in some specific cases, pesticide use is necessary. How much do farmers understand about the kind of pesticide they want to buy? How can we help farmers to avoid making mistakes when choosing pesticides?

In Vietnam, all pesticide companies have their own logo and labels for their products. However, most Vietnamese farmers pay attention only to the effects of pesticides; few of them look at other information on the label before they buy a pesticide. This exercise will help farmers to understand in-depth the information that is printed on labels of pesticide containers/packets. They should then be able to make better decisions when choosing pesticides to protect public health and the environment.

Objective:

Explain the signs and information printed on pesticide labels, including safety precautions

Materials:

Pesticide labels (Ask farmers to bring labels from pesticides that they are using to raise awareness about the kinds that they are using, i.e., highly hazardous, etc. The trainers should also collect as many pesticide labels in case farmers do not bring any or there are not enough samples.
Papers and pens enough for five farmer groups

Time: 90 minutes

Method:

1. The trainer should divide the pesticide labels into different groups, for example: insecticides, fungicides, herbicides OR extremely hazardous, moderately hazardous, slightly hazardous groups, etc.
2. The trainer should ask farmers to work in their small groups. Give each group some pesticide labels and ask farmers to bring out the labels they brought for the session.
3. Ask everybody to observe, read, and discuss carefully about the information and signs printed on the pesticide labels.
4. Members in each group explain the signs and information printed on pesticide labels to each other.
5. Discuss in the big group.

Discussions:

1. How many kinds of information are printed on a pesticide label? What is the information about? Which are the most important parts of the label? Why?
2. What is the trade name? What is the common name? Which one – trade or common name – expresses the essence of a pesticide? Why does one common name have many trade names?
3. Are the colors of the bars at the bottom of pesticide labels similar? What does the color of the bar mean? Looking at pesticide labels, how do you know if one pesticide is more hazardous than others? Explain the signs printed on the indicative color bar. Have the farmers ever paid attention to these signs? Why? If the indicative color bar of the pesticide label in your hand is red, what do you do with this pesticide?
4. What do the labels say about how to avoid pesticide poisoning during mixing and spraying? What do they say about what to do first if you are exposed to pesticide? What do farmers

actually do? Why? According to your group, through which routes do pesticides enter human body?

5. In the past, if you needed to buy a pesticide, how did you choose which pesticide to buy? Why?
6. Were there information on the labels that were not true? Give examples.
7. In the opinion of your group, what information is missing on the labels?

What is an LD₅₀?

Introduction

Test on the dosage of insecticide which kills test animals are called Lethal Dosage tests. Basically the process is simple and depends on the fact that not all animals will die with the same dosage because some individuals are more sensitive than others. If a very low dose is applied to 100 individuals, only a few individuals will die. If a very high dose is given, then most of the 100 individuals will die. The dose at which 50 of the 100 (50%) die is called the 50% lethal dosage or LD₅₀. The dosage at which 90 of the 100 individuals (90%) die is called the LD₉₀. This is a moderately useful measure, except that even at low dosages there is still an LD₁₀ in which 10% die. What does this 10% probability mean in another example? It means that there is a probability that for every ten people that cross the road, one will die while crossing the road. In other words, 10% probability is still very high. Dosage for mammal is usually measured in mg/kg. This means that a LD₅₀ of 1 mg/kg for a person who weighs 50kg is about 50mg, which is a very small quantity. Lethal dosages are usually given in both oral (through the mouth) and dermal (exposure to skin) levels.

Objective:

Explain what is an LD₅₀ measure

Materials:

Graphing paper and pencils

Time: 60 minutes

Method:

1. The following are the results of several trials for different dosage levels.

| Trial Dosage | 100 test individuals | | Alive |
|--------------|----------------------|----|-------|
| | Dead | | |
| 0 ppm | 1 | 99 | |
| 30 ppm | 2 | 98 | |
| 60 ppm | 3 | 97 | |
| 90 ppm | 10 | 90 | |
| 120 ppm | 25 | 75 | |
| 150 ppm | 45 | 55 | |
| 180 ppm | 65 | 35 | |
| 250 ppm | 85 | 15 | |
| 300 ppm | 90 | 10 | |
| 400 ppm | 95 | 5 | |

- a. Graph the trials. Use dosage on x-axis, and % dead on the y-axis.
 - b. According to the data, what is the LD₁₀, LD₅₀, and LD₉₀ for this population of 100 individuals?
 - c. In a cotton field, if there are four cotton bollworm per plant and the planting distance is 1.2 m x 0.4 m, how many cotton bollworm are killed when a spray at the LD₅₀ is used? How many are alive? What do you think is the best dosage to use for field application? How about at the LD₉₀? What do you think farmers are doing?
 - d. Natural enemies are usually more susceptible to insecticides than pests because natural enemies usually do not build up resistance. What happens when a low dosage is applied to the field (i.e., a dosage that is LD₂₀ for cotton bollworm, but LD₉₅ for natural enemies)?
2. Define oral LD₅₀. Give an example. What are the LD₅₀ oral and dermal of legal compounds in cotton?

Discussions:

Present your results and definitions.

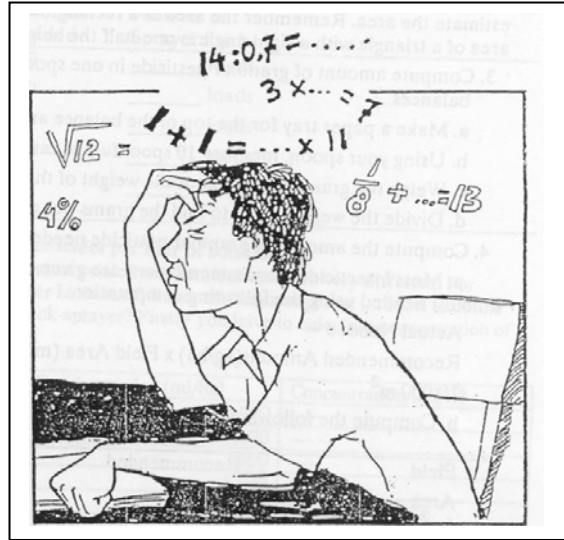
Pesticide calculations

Introduction

There are some who claim that farmers cannot do IPM because it is too complex. These same people claim that simple pesticide recommendations are “easier”.

In fact, IPM is not too complex for anyone to implement, and pesticides are not easy to use. Pesticide calculations are somewhat complicated for proper application based on plot size, dosage, and calibration (rate of spraying).

In this activity, we will investigate the typical calculations needed for recommended pesticide applications and how to provide farmers with useful measuring methods.



Objectives:

- Find the area of a field
- Compute the amount of poison needed to cover the field

Materials:

Meter stick
Paper
Pencils
Weighing scale
Spoons

Time: 120 minutes

Method:

(DO NOT USE A CALCULATOR; USE PENCIL AND PAPER)

1. To measure your foot step:
 - a. Lay the meter stick on the ground for about 12 meters.
 - b. Take ten steps next to the meter stick and measure the length of ten steps.
 - c. Divide the length by ten to get the average footstep length.
 - d. Try to make a step that is exactly 0.5m or 1.0m. This will make calculations of area easier.
2. Compute the area of fields (most farmers say they know, but few farmers actually know the area of fields):
 - a. Make a map of the field with the approximate shape.
 - b. Measure the sides of the fields by walking and counting the steps.
 - c. If the field is not a rectangle, then divide the field into rectangles and triangles to estimate the area. Remember the area of a rectangle is the height times the width. The area of a triangle with a right angle is one-half the height times the base.
3. Compute amount of granular pesticide in one spoonful; (for farmers without balances.)
 - a. Make a paper tray for the top of the balance and write down the weight.
 - b. Using your spoon, measure ten spoonfuls of Carbosulfan 5G.
 - c. Weigh the granules and minus the weight of the paper.

- d. Divide the weight by ten to find the grams per spoonful.
4. Compute the amount of granular pesticide needed for a field.
- a. Most insecticide recommendations are given based on 1 ha. Compute the actual amount needed using the following computation:

$$\text{Actual Needed} = \frac{\text{Recommended Amount (kg/ha)} \times \text{Field Area (m}^2\text{)}}{10,000\text{m}^2}$$

- b. Compute the following:

| Field Area (m ²) | Recommended Amount (kg/ha) | Actual Amount (kg) | Number Spoonfuls Needed |
|------------------------------|----------------------------|--------------------|-------------------------|
| 800m ² | 17kg/ha | _____kg | _____spoons |
| 1200m ² | 8.5kg/ha | _____kg | _____spoons |
| 750m ² | 17kg/ha | _____kg | _____spoons |
| 1050m ² | 12kg/ha | _____kg | _____spoons |
| 350m ² | 10kg/ha | _____kg | _____spoons |

5. Compute the amount of liquid in a spoonful.
Most liquid pesticide recommendations are given in ml. To find the number of ml. in a spoon, put ten spoonfuls of water in a measuring glass. Read the measurement and divide by ten to get the number of ml. in a spoonful.
6. Compute the number of sprayer loads and number of spoonfuls of pesticides needed for a field.
- a. To spray a field, usually 200 to 500 liters per hectare are required. In the early stages, 200 liters per hectare is sufficient. In the later stages, 400 liters is necessary because there is more foliage to cover. These are recommendations which are not usually implemented for good reasons (weight, time, access to clean water, etc.) To properly compute the number of tank loads, simply divide the amount required by the number of liters in the sprayer.
- b. Compute the following:

| Sprayer size | Amount needed | Number of sprayer loads |
|--------------|---------------|-------------------------|
| 111 | 300 | _____ loads |
| 131 | 500 | _____ loads |
| 151 | 300 | _____ loads |
| 91 | 400 | _____ loads |
| 131 | 500 | _____ loads |

- c. Define concentration of pesticides per liter of solution
In many cases, the application rates printed on pesticide labels are very different. For example: Use 1.0 liter of BT per ha or use 1.5 liter of Bassa per ha. How do you define the amount of pesticide per one back-sprayer? Firstly, you have to calculate the concentration of pesticide needed, as follows:

| Solution needed (liters/ha) | Recommended (ml/ha) | Concentration (ml/l) ? |
|-----------------------------|---------------------|------------------------|
| 300 | 1,000 | ????? |
| 400 | 1,200 | ????? |
| 500 | 800 | ????? |
| 300 | 1,500 | ????? |
| 500 | 2,000 | ????? |
| 400 | 500 | ????? |
| | | |

- d. Now for each sprayer load, some insecticide must be added. You have to multiply the usual recommendation of number of ml. per liter to get the total ml. needed for the sprayer. Then you must figure the number of spoonfuls enough for that number of ml. The number of spoonfuls is the total number of ml. divided by the number of ml. per spoonful.

| Sprayer size | Recommended ml/l | Total ml/load | Number of spoonfuls needed |
|--------------|------------------|---------------|----------------------------|
| 111 | 2 ml/l | _____ml | _____ spoons |
| 111 | 3 ml/l | _____ml | _____ spoons |
| 131 | 5 ml/l | _____ml | _____ spoons |
| 131 | 2 ml/l | _____ml | _____ spoons |
| 151 | 3 ml/l | _____ml | _____ spoons |
| 151 | 4 ml/l | _____ml | _____ spoons |

NOW YOU CAN SEE THAT PESTICIDES ARE POISONOUS AND THE COMPUTATIONS ALSO GIVE YOU A HEADACHE!

(Thank you to ideas and field studies from Dr. James Mangan.)

Spraying

Adapted from Helen Murphy's Guide for Farmer-to-Farmer IPM Health Studies

Introduction

Spraying pesticides is dangerous. The compounds used for spraying are in a concentrated form which makes them even more dangerous than usual exposure. Concentrated liquids direct from the bottle, and exposure to sprays in the field during application causes numerous symptoms such as skin rashes, dizziness, nausea, and headaches. The usual recommendation for gloves, boots, rain clothes, and respirator are impossible to implement for most farmers because of the costs. While some farmers use "protective" clothing they do not fully understand how pesticides enter the body and how so-called "protective" clothing does NOT guarantee that contamination will not occur.

There are many other precautionary measures that should be taken to reduce exposure to poisons when spraying. For example, the direction and velocity of the wind should be considered. If the wind is blowing hard, farmers should not spray. The chemical will never reach most of the plant. Never walk into the wind when spraying. Always walk at a 90 degree angle to the wind. This exercise will help participants understand is there is really "safe application" of pesticides.

Objective:

- Discuss that protective clothing is NO guarantee against exposure to pesticides
- Discuss if there is really "safe application" of pesticides

Time: 120 minutes

Materials:

Sprayer
Bucket
Red dye
White pants
Shirt
Gloves
Mask
Cigarette
Snacks to be eaten with the hands
Cup of water for drinking
Newprints and markers

Method:

1. The facilitator should mention that in real life, participants/farmers should observe precautionary measures to reduce exposure to poisons. These include the maintenance and preparation of the equipment, the preparation of the pesticide, wearing appropriate clothing, using appropriate spraying techniques, etc. (For details, see examples in table under section on Discussions.) However, in this exercise, participants should be able to observe farmers' common practices, mostly incorrect, practices in spraying. These will be the basis for later discussions. (More incorrect practices demonstrated and observed will lead to more discussions on what can be done to reduce exposure to poisons.) Stress that the exercise is intended to initiate discussions on whether or not there is really "safe application" of pesticides.
2. All participants go to the field. One person in the group will play the role of a "farmer". This person should put on the white pants, shirt, gloves, and mask – to make it easier to see the red dye ("pesticide") stains. The "farmer" will show common practices, MOSTLY INCORRECT, practices in spraying. The "farmer" may exaggerate for emphasis.
3. The other members of the group should make notes on what the "farmer" is doing. Also note how the "farmer" could have reduced exposure to the spray liquid.
4. The "farmer" should fill the tank with water and add red dye. Add a lot so that the water is very red. Close the tank and shake the tank to mix the water and the dye. (Farmers often mix pesticides with their bare hands.)
5. The "farmer" will spray 500 m² of the field with the tank of water and dye using 2-3 tanks (as farmers practice) and take a break between spraying to smoke, eat with hands and drink from a

- cup (without washing hands). The “farmer” sprays without checking the direction or velocity of the wind. Others should measure the time required and observe the spraying technique.
- After finishing spraying, the “farmer” empties the excess mixture from the tank. (Farmers normally empty tanks into irrigation canals.)
 - Now observe the sprayer. Is the red dye on the skin or clothing of the person who sprayed? Using a piece of newsprint, ask each group to draw the points of contamination. Use red color to show pesticide contamination.

Discussions:

- Process the activity by eliciting observations taken on the role play/demonstration. (More incorrect practices demonstrated and observed will lead to more discussions on what can be done to reduce exposure to poisons.) Use the following table as an example:

| What the “farmer” did | What should the “farmer” have done |
|--|--|
| The “farmer” did not clean the sprayer. | If the sprayer has been used before, wash it thoroughly with detergent. Use gloves when washing the sprayer. (Another possible answer: The owner of the sprayer should clean it before keeping to avoid corrosion and clogging.) |
| The “farmer” used his mouth (blew) to clear the clogged hose. | Check to see if the sprayer is working properly by pumping and spraying water. This will also clean the hose and nozzle of the sprayer. If necessary, use water or a soft probe like a weed stem to clean the hose and clear the holes of the sprayer. Take note of the size and type of the nozzle to see if this suits your requirements. (Farmers may not have very much choice about sizes and types as they might only have one nozzle!) |
| The “farmer” did not measure the red dye put just put it inside the sprayer. | Check the recommended dosage on the label or ask help from a neighbor or another family member if the farmer can not read and do calculations. (Higher doses do not produce better effects; lower doses will be less effective.) |
| The “farmer” used his bare hands for mixing the “chemical”. | Use a long disposable stirrer to mix the pesticide and properly dispose of the stirrer. |
| The “farmer” had red dye all over his back – the sprayer was leaking. | Check for leaks by carrying the tank and spraying with water. |
| The “farmer” sprayed against the wind. | Check for the direction and velocity of the wind. If the wind is blowing hard, do not spray. Never walk into the wind when spraying. Always walk at 90 degrees angle to the wind. |
| The “farmer” was smoking while spraying. | Do not smoke while spraying; use a mask while spraying. |
| The “farmer” ate without washing his hands. | Wash hands thoroughly with soap and water after handling pesticides and especially before eating. |
| The “farmer” emptied his tank into the irrigation canal. | Use all the pesticides in the field. (Depending on the product, surplus may or may not be used the following day.) |
| The “farmer” left the empty “pesticide bottle” (red dye container) in the field. | Etc. |
| Etc. | Etc. |

| | |
|------|------|
| Etc. | Etc. |
| Etc. | Etc. |
| Etc. | Etc. |

2. What signs and symptoms of poisoning can be caused by pesticides?
3. What are the experiences of the groups with spraying pesticides?
4. Discuss the easiest ways for pesticides to enter the body (SKIN, WET CLOTHES) and increase the risk of pesticide poisoning.
5. Discuss that the MOST important time when contamination leading to poisoning occurs is during mixing the pesticide concentrates. . .which is WORSE when using a pre-mixed pesticide cocktail!
6. Discuss that protective clothing is NO guarantee that contamination will not occur, but ask what low cost practical measures can be taken to reduce skin contamination.
7. Present the following situation: *A farmer sprays for two hours. He only changes clothes and takes a bath four hours after spraying.* (Note: The farmer's skin is not exposed just two hours but six hours because his skin has continued contact with pesticides for the extra four hours between finishing spraying and taking his bath.) Ask for ideas of everyone in the group. Discuss importance of bathing with SOAP immediately after spraying and always using freshly washed clothing for spraying.
8. How can farmers reduce the exposure to pesticides?
9. Discuss "Is there really 'safe application' of pesticides?"

Self-survey of pesticide poisoning

Adopted from Helen Murphy's Guide for Farmer-to-Farmer IPM Health Studies

Objectives:

- Recognize signs and symptoms of pesticide poisoning
- Discuss how through IPM experimentation with non-chemical alternatives to pest management, pesticide related illness can be eliminated

Materials:

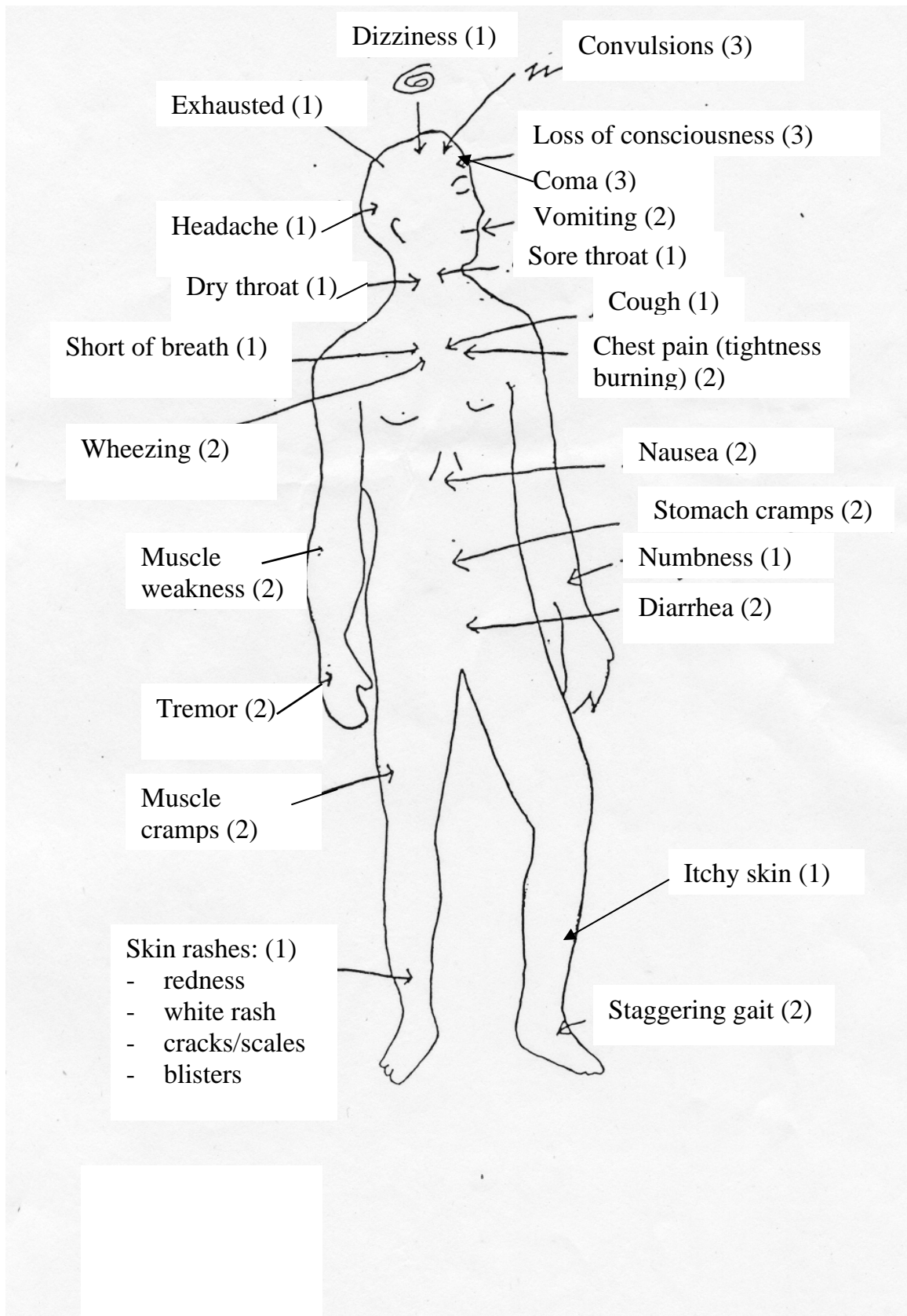
Signs and symptoms of pesticide poisoning body map (body and head)

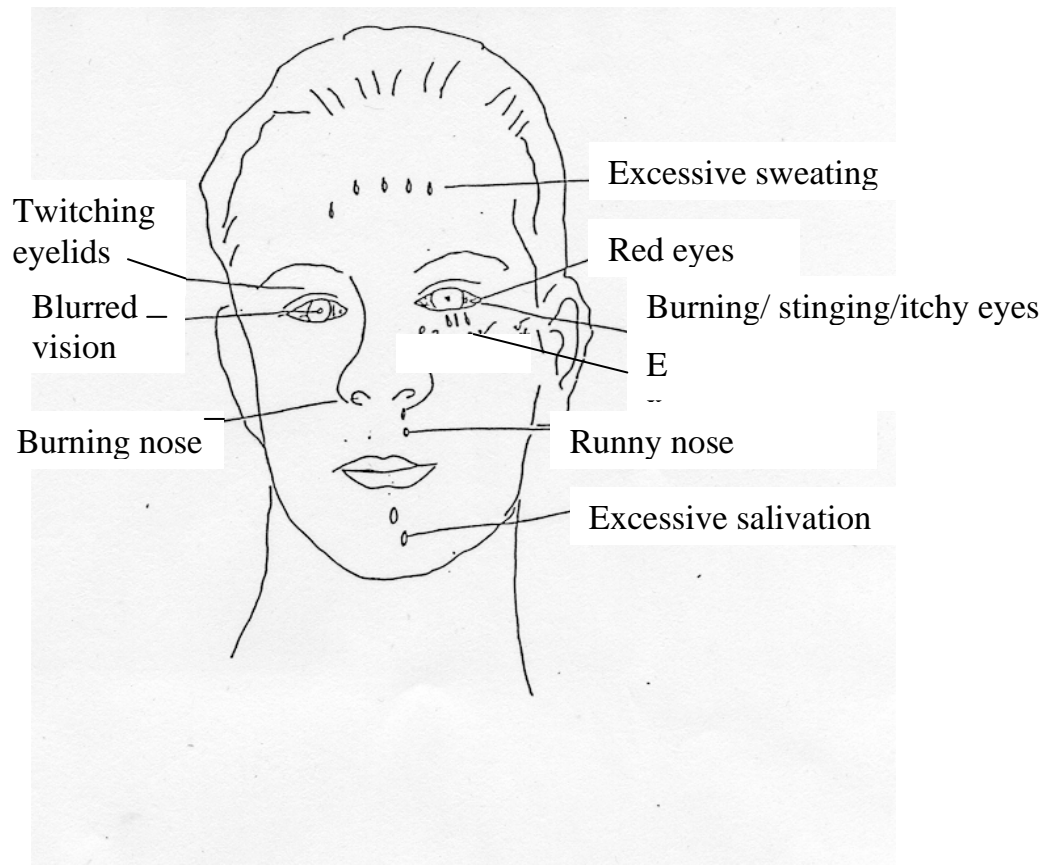
Method:

1. Distribute body maps to each farmer.
2. Explain DEFINITION of each sign and symptom.
 - Discuss *other* conditions or illnesses that could cause the sign and symptom.
 - Discuss how to identify the signs
3. Ask each farmer to circle or check each sign and symptom 'ever experienced' during or up to 24 hours AFTER spraying.
 - If any farmers are illiterate divide into groups with one reader per group.
 - Farmer may add a sign and symptom not on the list that they feel are associated.
4. Tabulate results on a master picture by a raise of hands poll.
5. Calculate percent ($\#/\text{total farmers} \times 100$) of each sign and symptom experienced.
6. Poll farmers level of pesticide poisoning:
 - Mild poisoning = only (1's) marked
 - Moderate poisoning = at least one (2) marked
 - Severe poisoning = at least one (3) marked
7. Discuss definition of mild, moderate and severe
 - Mild = sign or symptoms only from irritation or a vague symptom
 - Moderate = a nervous system sign or symptom
 - Serious = loss of consciousness or seizure

Discussions:

Discuss how through IPM experimentation with non-chemical alternatives to pest management, pesticide related illness can be eliminated.





Yearly pesticide liters of exposure

Adopted from Helen Murphy's Guide for Farmer-to-Farmer IPM Health Studies

Objectives:

- Calculate yearly pesticide liters of exposure
- Discuss how through IPM farmers can reduce spray frequency (or totally eliminate spraying depending on future non-chemical IPM alternatives discovered through IPM experimentation)

Materials:

Calculators

Method:

1. Ask farmers to calculate yearly pesticide liters of exposure using the table below:

| AMOUNT OF PESTICIDES USED BY FARMERS | | | | | | | |
|--------------------------------------|--------------------|---------------------------------|-----------------------------------|-------------------------|---|---------------------|----------------------------------|
| Crop | a. Tank size (li.) | b. # of tanks per spray session | c. ♦ # of spray sessions per week | d. ♦ # weeks per season | e. # of spray sessions per season (c*d) | f. seasons per year | Liters exposure per year a*b*e*f |
| | | | | | | | |
| | | | | | | | |
| | | | | | | | |
| TOTAL | | | | | | | |

♦fill in column 'c.' and 'd.' only if spraying on a weekly basis. Otherwise use column 'e.' showing how many spray sessions per season.

Discussions:

1. Discuss how through IPM farmers can reduce spray frequency (or totally eliminate spraying depending on future non-chemical IPM alternatives discovered through IPM experimentation).
2. Ask farmers to recalculate yearly pesticide liters of exposure using a lower number of spray sessions per season.
3. Compare the difference in liters of exposure between first and second calculation. Talk about the health and economic benefits if a farmer sprays less often.

Household storage and disposal of pesticides

Adapted from Helen Murphy's Guide for Farmer-to-Farmer IPM Health Studies

Objectives:

Discuss how farmers can reduce risks of pesticide poisoning through proper household storage and disposal of pesticides

Materials:

Paper and markers

Method and Discussions:

1. Ask each farmer to draw a picture of his house and farm showing the locations of the following:
 - Food storage
 - Food preparation
 - Food consumption
 - Drinking and cooking water source
 - Drinking and cooking water storage
 - Drinking and cooking water use
 - Location of all farm animals (chickens, ducks, cows, etc.)
 - Where pesticide containers are thrown out
 - Where tank is stored
 - Where pesticides are stored (and height from ground)
2. Display each drawing on the walls.
3. Each farmer walks around the room (like an art gallery) and answers the following questions (yes/no) for each household drawing:
 - Storage:
 - a. Is pesticide storage safe for children?
 - b. Does pesticide storage prevent drinking and cooking water contamination?
 - c. Does pesticide storage prevent food contamination?
 - d. Is pesticide storage safe for farm animals?
 - Disposal:
 - e. Is pesticide disposal safe for children?
 - f. Does pesticide disposal prevent drinking and cooking water contamination?
 - g. Does pesticide disposal prevent food contamination?
 - h. Is pesticide disposal safe for farm animals?
1. Tally the results on a master list (number and %)
2. Discuss why the answers are 'no' picture by picture. Define with group what constitutes unsafe storage and disposal.

Pesticides and pest resistance

Introduction

A pest is said to have developed resistance to a certain pesticide when it loses sensitivity to the material. Resistance may develop in relation to the actual dose, the concentration, or the exposure time to a certain pesticide. To measure the degree of resistance that a pest has developed, it is necessary to have a control treatment. It is also important to remember that when doing experiments especially with insect pests, care must be taken so that the insects are not damaged during handling resulting in deaths not directly related to pesticides.

Objective:

Try out different dosages of chemical insecticides on different larval stages to observe the development of pest resistance to chemical insecticides

Materials:

Chemical pesticides (five different kinds of chemical insecticides commonly used in the area for each group)

Larvae at different stages

Hand sprayers (one liter capacity)

Masks

Plastic or rubber gloves

Scissors

Forceps

Long disposable stirrers for mixing pesticides

Containers for mixing pesticides

Plastic cups with organdie sheets and rubber bands

Pens

Notebooks

Method:

In the session room:

1. All groups should collect fresh leaves from the upper part of the cotton plant. Cut leaves into 1" diameter sections.
2. Participants should prepare the hand sprayers before setting up the exercise. If the sprayers have been used before, wash them thoroughly with detergent. Use gloves when washing the sprayers. Check to see if the hand sprayers are working properly by pumping and spraying water. This will also clean the hose of the sprayer. If necessary, use water or a soft probe like a weed stem to clean the hose and clear the holes of the sprayer.
3. Participants should carefully read the instructions on how to use each pesticide product as printed on its label. Following the recommended dose (This differs from product to product!), participants will prepare the different pesticides. Trainers should call attention of participants to the correct dosage and appropriateness of mixing procedures – whether the pesticide product is in powder or suspension form!
4. Also pay attention to how participants calibrate the pesticide to use in one liter of water. (Note: If using a suspension, make sure to shake the bottle thoroughly to disperse the solution evenly. Choose a syringe with a needle hole big enough to take up the suspension. If using a pipette, take up more than the volume required and release the excess back into the suspension bottle to get the exact quantity. Never use the mouth to suck the suspension into the pipette!)
5. The members of the group handling the pesticides should put on masks (Homemade masks made of several layers of cloth or thick discarded pads are useful!) and plastic or rubber gloves. Use a long disposable stirrer for each kind of product that will be used to prepare the solution and properly dispose of the stirrer.
6. Each group should do five treatments, i.e., use five different kinds of chemical insecticides commonly used in the area. Each treatment should have three replications. Each group should then have a total of fifteen cups labeled accordingly. Members of the group who will

set up the treatments should also use masks and gloves as well as handle leaf sections with forceps.

7. Using hand sprayers, spray the chemical insecticides on the leaf sections and let dry. When dry, put a leaf section into each plastic cup lined with tissue paper and introduce 10 – 20 larvae (depending on the stage of the larvae). Each cup should be covered with organdie sheets held securely with rubber bands.
8. Observe after 12 hours, 24 hours, 48 hours, and 72 hours. Record observations on table suggested below taking note of the leaf damage, frass production, and the state of the larvae.
9. Remember: Dispose properly of empty pesticide containers to prevent pollution of the environment and any possible contamination. If you need to store unused pesticides keep them in a cool place that is safe for people (especially children) and animals. Wash hands thoroughly with soap and water after doing the exercise and each time you handle pesticides!
10. Trainers should call attention of participants to the need to handle, use, dispose, and store pesticide products properly and with caution. These are poisons!

In the field:

1. Each group should select fifteen individual plants in the field to carry out five treatments using five different kinds of chemical insecticides commonly used in the area. The same products used in the session room exercise may be used for the field exercise. Each treatment should have three replications.
2. Observe the pest population on each plant and classify the larval stages. Note down the pest population according to the larval stages.
3. Participants should prepare the hand sprayers before setting up the exercise. If the sprayers have been used before, wash them thoroughly with detergent. Use gloves when washing the sprayers. Check to see if the sprayers are working properly by pumping and spraying water. This will also clean the hose of the sprayer. If necessary, use water or a soft probe like a weed stem to clean the hose and clear the holes of the sprayer.
4. Participants should carefully read the instructions on how to use each pesticide product as printed on its label. Following the recommended dose (This differs from product to product!), participants will prepare the different pesticides. Trainers should call attention of participants to the correct dosage and appropriateness of mixing procedures – whether the pesticide product is in powder or suspension form!
5. Also pay attention to how participants calibrate the pesticide to use in one liter of water. (Note: If using a suspension, make sure to shake the bottle thoroughly to disperse the solution evenly. Choose a syringe with a needle hole big enough to take up the suspension. If using a pipette, take up more than the volume required and release the excess back into the suspension bottle to get the exact quantity. Never use the mouth to suck the suspension into the pipette!)
6. The members of the group handling the pesticides should put on masks (Homemade masks made of several layers of cloth or thick discarded pads are useful!) and plastic or rubber gloves. Use a long disposable stirrer for each kind of product that will be used to prepare the solution and properly dispose of the stirrer.
7. Members of the group who will set up the treatments should also use masks and gloves. Using hand sprayers spray the chemical insecticides on the individual plants and label treatments accordingly. Spray the chemical on the upper side of leaves moving from the top towards the bottom portion of the plant. Then spray the chemical on the underside of leaves moving from the bottom towards the top portion of the plant. Make sure that both sides of the leaves are drenched with the solution. Spray following the direction of the wind. Wash hands thoroughly with soap and water, and change clothes after spraying.
8. Observe after the first 24 hours and continue taking observations until the fifth day. Record observations on table suggested below taking note of the leaf damage, frass production, and the state of the larvae.
9. Remember: Dispose properly of empty pesticide containers to prevent pollution of the environment and any possible contamination. If you need to store unused pesticides keep them in a cool place that is safe for people (especially children) and animals. Wash hands thoroughly with soap and water after doing the exercise and each time you handle pesticides!
10. Trainers should call attention of participants to the need to handle, use, dispose, and store pesticide products properly and with caution. These are poisons!

Discussions:

1. Describe differences in the treatments in the cups and in the field.
2. What do these observations imply for crop production?

| TREATMENT | Items | Replication | Hours | Pesticide A | | | Pesticide B | | | Pesticide C | | | Pesticide D | | | Control (water) | | |
|------------------|-------|-------------|-------|-------------|----|---|-------------|---|---|-------------|---|---|-------------|---|---|-----------------|---|---|
| | | | | 1 | 2 | 3 | 1 | 2 | 3 | 1 | 2 | 3 | 1 | 2 | 3 | 1 | 2 | 3 |
| | | | | Leaf damage | 12 | | | | | | | | | | | | | |
| 24 | | | | | | | | | | | | | | | | | | |
| 48 | | | | | | | | | | | | | | | | | | |
| 72 | | | | | | | | | | | | | | | | | | |
| Frass production | 12 | | | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | | | |
| State of larvae | 12 | | | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | | | |
| Total | 12 | | | | | | | | | | | | | | | | | |
| | 24 | | | | | | | | | | | | | | | | | |
| | 48 | | | | | | | | | | | | | | | | | |
| | 72 | | | | | | | | | | | | | | | | | |

SCORING SYSTEM

LEAF DAMAGE

- 1: low
- 2: moderate
- 3: high

Frass production

- 1: none
- 2: little
- 3: much

State of larvae

- 1: dead
- 2: moribound
- 3: active

Effect of pesticides on predators

Method:

Recent studies on cotton suggest that ground predator fauna is rich in unsprayed fields in Asia, but is strongly eliminated by insecticides.

1. All groups choose four 10x10 m plots inside a homogenous cotton field (two sprayed as farmers' practice, two unsprayed). If possible, have a 3-5 m stretch as a buffer zone between the plots.
2. Participants weekly record predator densities (coccinellids, black predator ants, lycosid spiders, other spiders, carabid beetles, predator bugs, etc.) at dusk, or very early morning/evening, using 50x50x30 cm quadrants.
3. Group members take about ten samples from each treatment, sprayed and unsprayed (i.e. five samples per plot) and compare the seasonal densities.
4. [Additional pitfall traps (e.g. 250 ml plastic or glass jars half filled with water, buried in the soil up to the rim of the jar) may be put in a regular pattern within the same plots to record the activity of ground predators. Pitfall traps containing water should remain in the field for only about two days to avoid decomposition of trapped arthropods (If poisonous formaline is used, the pitfall trap can remain in the field for 1 week periods).]
5. In addition to sampling predators, this trial can be used to study the effect of spraying on *Spodoptera* and on the parasitism level.

Effects of pesticides on natural enemies

Objective:

Evaluate the effect of sprayed leaves on the survival of natural enemies

Materials:

Four jars with lids
Four pieces of Muslin cloth with rubber bands, to close jars (Method 2)
Labels
Scissors
Forceps
Long disposable stirrers for mixing pesticides
Masks
Plastic or rubber gloves
Paper, pen
Four small hand sprayers (0.5 liter)
Small amounts of insecticides

Method 1:

1. Participants prepare four hand sprayers before setting up the exercise. If the sprayers have been used before, wash them thoroughly with detergent. Use gloves when washing the sprayers. Check to see if the sprayers are working properly by pumping and spraying water. This will also clean the hose of the sprayer. If necessary, use water or a soft probe like a weed stem to clean the hose and clear the holes of the sprayer.
2. Participants should carefully read the instructions on how to use the product printed on the label. Following the recommended dose at field rate concentrations (This differs from product to product!), participants should prepare the different pesticides. Trainers should call attention of participants to the correct dosage and appropriateness of mixing procedures – whether the pesticide product is in powder or suspension form!
3. Also pay attention to how participants calibrate the chemical to use in 0.5 liter of water. (Note: If using a suspension, make sure to shake the bottle thoroughly to disperse the solution evenly. Choose a syringe with a needle hole big enough to take up the suspension. If using a pipette, take up more than the volume required and release the excess back into the suspension bottle to get the exact quantity. Never use the mouth to suck the suspension into the pipette!)
4. The members of the group handling the pesticides should put on masks (Homemade masks made of several layers of cloth or thick discarded pads are useful!) and plastic or rubber gloves. Use a long disposable stirrer for each kind of product that will be used to prepare the solution and properly dispose of the stirrer.
5. Each group should prepare three hand sprayers with commonly used insecticides for example: pyrethroid, carbamate (chemical insecticides), NPV or Bt (biological insecticide) and one hand sprayer with water (control). That means that each group will carry out four treatments (three with chemical insecticides and one control).
6. Members of the group who will set up the treatments should also use masks and gloves. Select four plants in the field: one plant per spray treatment. Using hand sprayers spray the chemical insecticides on the individual plants and label treatments (plants) accordingly. Spray the chemical on the upperside of leaves moving from the top towards the bottom portion of the plant. Then spray the chemical on the underside of leaves moving from the bottom towards the top portion of the plant. Make sure that both sides of the leaves are drenched with the solution. Spray following the direction of the wind. Wash hands thoroughly with soap and water, and change clothes after spraying.
7. Let the leaves dry on the plant.
8. Pick one or several leaves from each treatment and transfer to glass jars. (Use gloves!) Label the jars. Each group should have one jar of each spray treatment (four jars in total). Try to get the leaf to lie flat on the inside surface of the jar.
9. Collect several predators from the field. Transfer them to the jars. Use the same predator species in all treatments. Close the jar with the lid, and place a piece of tissue paper between the jar and the lid to avoid condensation inside the jar.

10. Check and record the condition of the predators after eight hours and after 24 hours. Count the number of dead insects. It may be necessary to touch the insect with a pen or pencil to determine if it is dead. If it does not walk off in a normal manner, then record it as dead.
11. Remember: Dispose properly of empty pesticide containers to prevent pollution of the environment and any possible contamination. If you need to store unused pesticides keep them in a cool place that is safe for people (especially children) and animals. Wash hands thoroughly with soap and water after doing the exercise and each time you handle pesticides!
12. Trainers should call attention of participants to the need to handle, use, dispose, and store pesticide products properly and with caution. These are poisons!

Method 2:

1. Participants prepare four hand sprayers as in Method 1.
2. Members of the group who will set up the treatments should use masks and gloves. Spray each piece of muslin cloth with a sprayer and let the cloth dry. Label the cloth.
3. All groups collect several predators from the field and transfer them to four jars per group. Use the same predator species for all treatments. Close the jar with the sprayed muslin cloth.
4. Group members check and record the condition of the predators after eight hours and 24 hours. Count the number of dead insects. It may be necessary to touch the insect with a pen or pencil to determine if it is dead. If it does not walk off in a normal manner, then record it as dead.
5. Remember: Dispose properly of empty pesticide containers to prevent pollution of the environment and any possible contamination. If you need to store unused pesticides keep them in a cool place that is safe for people (especially children) and animals. Wash hands thoroughly with soap and water after doing the exercise and each time you handle pesticides!
6. Trainers should call attention of participants to the need to handle, use, dispose, and store pesticide products properly and with caution. These are poisons!

Poison sprayer maintenance

Introduction

Pesticides are not medicines! They are poisons to be used and handled with great care. Some granular chemicals can be broadcast (using gloves and boots). But many compounds are liquid and need to be sprayed on the crop.

Proper maintenance of the sprayer is necessary to avoid direct exposure from leaking valves, leaking hoses, bad nozzles, and bad rubber rings on the tank openings. Many sprayers spill chemicals on to the back of farmers as they are spraying.

Proper maintenance is also required to have complete coverage of the plant. A nozzle that is old or clogged will not give good coverage of the plant. Many poorly maintained sprayers put out a stream of poison like a person urinating. This wastes money, and exposes the farmer to heavy dripping of the poisons. The spray should be small droplets and spread over the entire spray path.

In this activity, we will look at how to maintain a sprayer.

Objectives:

- Identify parts and function of a sprayer
- Take apart and put a sprayer back together

Materials:

Sprayer
Bucket
Hand tools for disassembling the sprayer
Large piece of cloth or paper

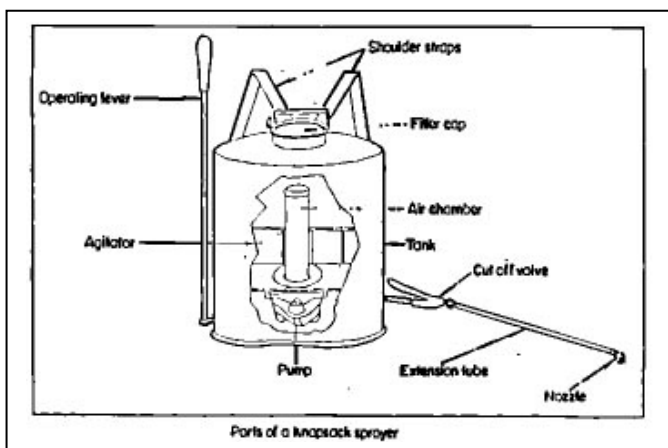
Time: 90 minutes

Method:

1. In a shady place, sit down in a group with one sprayer. Have one bucket of water and tools ready to be used before beginning.
2. The sprayer is made of many parts. Look at the sprayer and identify all the parts and their functions.
3. Fill the tank with clean water and operate the sprayer. Test the pump and valves. Note any leaks when the sprayer is being operated and when it is on its side.
4. Now empty the sprayer back into the bucket. Begin to disassemble the sprayer. Someone should keep track of how the sprayer is built so that it can be put back together. Make sure the pieces are placed on the cloth or paper. Any dirt on the pieces will cause problems when the sprayer is put back together.
5. Locate all the potential places for leaks.
6. Practice changing the rubber rings. Practice explaining what the rubber rings are used for.
7. Now look at the nozzle.
8. After you have examined all the parts, put the sprayer back together. Fill the tank with water. Check for leaks and check to see if the sprayer is working.

Discussions:

1. How do the rubber rings appear? Are they new or old? Were all the parts put together tightly so that the rubber ring is compressed? If the rubber ring is old, where do you buy a new one? Can you make one from an inner tube of tire? Are the seals on the top of the tank still good? If the farmer bends over with the sprayer on his back does the pesticide go on his back or head? Is there any corrosion on or in the tank?
2. Can the nozzle be adjusted? If so, how? What size of wire is needed to clean the nozzle (the wire should be smaller than nozzle hole)?
3. How often should a sprayer be checked for problems? When should the rings be changed? When should the nozzle be changed?
4. What is the cost of a new rubber ring? What is the cost of a new nozzle? What is the cost of pesticide poisoning from a leaking tank?



Weather

General introduction and effect on diseases:

Climate and weather are important for the growth and development of diseases, insects, natural enemies and plants. Climate is the long term general pattern of the daily weather patterns. For example, the climate of the South of Vietnam is hotter than the climate of the North of Vietnam. On a particular day however, the weather in the North may be hotter than in the South. The weather is difficult to predict, while climate is easier to forecast. In fact, weather scientists cannot reliably predict the weather for more than about 24 hours, even with super computers and vast information networks. Yesterday's and today's weather are still the best predictors of tomorrow's weather.

Weather is very important for determining the development and growth of disease and insects. It is not surprising then that diseases and insects are also very difficult to predict. Prediction is even more difficult because the short-term pattern of weather is also important (ex. 4 rainy days versus 1 day rain - 2 days cold - 1 day rain) but impossible to predict. Besides our lack of prediction ability, scientists do not know the actual effect of certain weather patterns on the development of disease.

Weather can be measured. Temperature ($^{\circ}\text{C}$ and degree days), rainfall (mm/hour or mm/day), solar radiation (joules/cm²/day), hours of cloud cover, relative humidity (%), atmospheric pressure (mbar), wind speed (m/minute), wind direction, and daylength (hours) are some of the parameters that are used to define and measure the weather. "It is a hot and windy day" can be described as "The maximum temperature is 35 $^{\circ}\text{C}$, wind direction south-east blowing at 5 meters per minute, with cloud cover after 3 p.m."

So what are the important concepts to have when considering the weather effects on living organisms? First is the rate of chemical reactions inside organisms. For most chemical reactions the hotter the chemical, the faster the reaction. Thus rice cooks faster on a hot fire, than on a warm fire. Plants, insects, spiders, bacteria, fungi and viruses also "cook" faster, meaning they develop faster. Rice matures a couple of days faster in hot areas. Insects develop from egg to adults in shorter times in hot areas. Fungi grow more quickly on food left on the table than food placed in the refrigerator. However, every organism has an optimal temperature for best growth and development. Too hot burns rice, and kills plants, insects, etc.. Some plants grow better at 25 $^{\circ}\text{C}$ than at 30 $^{\circ}\text{C}$, because the plants have an optimum growth temperature (actually the plant's enzymes have an optimal reaction temperature). This is true for insects and disease organisms. This explains why different plants, insects and diseases are found on mountain tops than at low elevations. Mammals are able to regulate body temperature so that we are less affected by outside temperature changes.

Temperature can be accumulated. This is called degree-day. Thus one day at 20 degrees may be the same as two days at 10 degrees, depending on the growth rate of the organism at different temperature. Degree-days are used in forecasting models to predict the development rate of insects and diseases in different environments.

Another key concept is water. Water is important for all life. Water can be in the form of water on a surface that is important for roots, for insects, and for germination of disease organisms. Water is also in the air as moisture. Low moisture (low humidity) means the air is dry. High humidity means there is a lot of water in the air. However, the amount of water that can be held in the air depends on how hot the air is. The hotter the air, the more moisture can be held. A cold glass collects water on the outside because the air around the cold glass becomes cold, and moisture in the air becomes water on the glass. Humidity is important for the development of microorganisms, especially bacteria and fungi.

Effect on disease organisms

Disease organisms for plants include bacteria, fungi, virus, and sometimes nematodes (some plant pathologists don't consider nematodes as 'disease' - in the field it doesn't really matter). Weather can affect the processes of the disease cycle in the following ways:

Transport/movement: Disease organisms are moved by the wind, by splashing rain water, and by flowing or flooding rain water moving soil, plants and disease organisms. The level of humidity, temperature and solar isolation determines the survival of the disease organisms during movement and before a host is available for infection.

Germination: Is mostly determined by the availability of an appropriate host. However, the ability to germinate is sometimes determined by temperature, humidity, water on the surface after rain or night dew, and solar radiation. The germination of fungi and bacteria is the first step in infection and usually means that a part of the disease organism is developing for entering an opening in the plant tissue or making an opening.

Infection: Success and failure of infection may depend on the growth rate of the disease organism in relation to the defense rate of the host plant. Success of infection is determined more by the plant condition than by the disease organism.

Incubation: Incubation is the time required for an infection to cause symptoms. The development of symptoms is also a function of the plant type and condition, but also a function of the relative development rates of the plant and the disease. Disease is like a race between the plant and the disease organism. If the weather is better for the plant than for the disease (in terms of optimum temperature, water, sunlight, etc.) the plant may never show symptoms or only show minor symptoms. However, if weather conditions are best for the disease organism, the disease may quickly have symptoms, quickly develop inoculum that is ready to be moved to other leaves or plants.

Inoculum development/reproduction: Production of fruiting bodies can be a function of temperature, sunlight and relative humidity. Movement of inoculum returns us to beginning of disease cycle.

In this activity we will note the specific effect of weather on the disease cycle process for the major diseases of cotton.

Objective:

Describe the effect of weather on the disease cycle processes for at least one major cotton disease

Materials:

Big paper, markers

Method:

1. Choose a major disease on cotton that you are studying (caused by bacteria, fungi, virus or nematodes).
2. On a large piece of paper, make two columns. On the top left column write "disease process" and list the disease cycle process in the left column.
3. On the top of the right column, write "Effect of disease" and write the name of the disease selected. In the column, write the effect of weather for each of the disease cycle processes. For example: Transport/Movement - Wind moves rust fungi from leaf to leaf, plant to plant, and field to field...
4. Present your opinions to the group.

Effect on Insects

Weather is important for determining the growth and development of diseases, insects and plants. In the introduction we explored the effect of weather on diseases. Our main ideas were that diseases are bags of chemicals. Their reactions are determined by temperature and water. The effect on the plant and the health and variety of the plant determine the development of disease symptoms.

Insects can also be considered bags of chemicals. The growth and development is determined by temperature and humidity. Temperature of course must be measured at the place where the insect is living. This means an insect on the leaf surface in the hot sun is growing faster than the same species hiding in the cool soil. The development of an insect species in a cool environment will be slower than the same insect species in a hot environment. Of course, there is also an optimal temperature for growth.

What other factors determine insect growth and development, especially as populations? One is the wind. Wind currents move insects from one area to another. Most insects can only stay in the air and let the wind push them about. The wind is also important for carrying the scent of host plants, or females of the same species. Plant scents allow insects to find their host plant. Some natural enemies also use the plant scent to find their prey when the prey is usually on a particular type of host plant (for example the parasite of Diamondback Moth that only occurs on cabbage). The scent of female insects is also carried in the wind. This scent is followed by males until he finds his mate.

Rain is another important part of the weather that strongly influences insects. Direct kill of insects by rain is important for small insects (why do most insects feed on the bottom of leaves?). Moisture is important for ending aestivation ("sleeping") stages of some larvae/pupae that survive in soil or plants during the dry season. Rain after a dry period causes some kinds of nitrogen to be more available to the plants so that the plants suddenly become more green and active in growth. This is an indirect positive influence on insect population growth.

So the weather influences the growth of insects in many ways. In this activity, we will discuss and develop a list of ways not listed above.

Objective:

Describe the effect of weather on the growth and development of insect populations

Materials:

Big paper, markers, ruler

Method:

1. On a large piece of paper, make two columns. On the top of the left column write "Insect development process" and list the following processes in the column:
 - * Migration (long distance)
 - * Movement (short distance)
 - * Birth rate
 - * Death rate
 - * Development rate (egg to adult stages)
 - * Aestivation
2. On the top of the right column, write "Effect of Weather". Make a list of weather influences on each parameter of population development in the left column. Try to give an example for each item.
3. Try to make a "Big Picture" of population development in relation to the development of the crop. For example, what would be the difference if the weather were hotter, or wetter, or dryer, or colder? Discuss the difference. What have been your experiences?
4. Present your results to the group.

Weather, Insects and Pathogens

Measuring the Micro-habitat

We discussed the influence of weather on organisms in the first sections on weather, insects and diseases. We said insects (and diseases) were like "bags of chemicals" with optimal reaction temperatures, humidity and moisture. What about in the field? Are there major differences in temperature, and moisture on a small scale (several ha)?

Differences in weather are usually seen over large areas. We will explore different sites and measure the micro-habitat. We will look at both temperature and moisture over several hectares on fields around the Training Center. Micro-climates often are different because of exposure to sunshine or rain. The micro-climate will change when moving from place to place and moving through time in the same place. Remember that an insect or pathogen moves its place when going from the top of a leaf to the bottom of the leaf. Small changes for us, are tremendous changes for insects or pathogens moving with the wind, rain, or by themselves.

Objective:

Demonstrate how weather parameters (temperature and moisture) are different over small areas of several meters.

Materials:

Big paper, pencil

Thermometers (2 per group), plastic bags, scales

Method:

1. Choosing sites: choose two locations. The locations should be in the following places:
 - a. place with no shade and high on a slope
 - b. place with lots of shade and high on a slope
 - c. place with no shade and low on a slope
 - d. place with lots of shade and low on a slope
2. Soil moisture: at each site, place some soil in a plastic bag. One handful is enough. Close the bag. Weigh the content of the bag. Dry the soil in the sun. Weigh the soil with the bag again after drying. The difference in weight is the amount of water in the soil. The original soil weight was the weight of water plus the non-water portion. {Original % moisture = [(weight of water)/(original weight - water weight)] x 100}.
3. Temperature: use the thermometer to measure three positions for each site:
 - a. soil temperature
 - b. air temperature above a plant
 - c. air temperature at the bottom of the plant
4. Make a table of temperatures and soil moisture for the sites.
5. Can you explain why there are differences in soil moisture at each site? Remember recent rains, irrigation, etc.. Are these differences important for the growth and development of insects and diseases? Especially in terms of the soil organisms!
6. What are the differences in temperatures? How do you think these values will change over a one-day period? Make a hypothetical graph of these changes. Are these temperature differences important for insect and disease development?
7. Is there an interaction between temperature and moisture for any site? What is the interaction?
8. Can you give examples from your own experience where disease and insects seem to be more dense or less dense because of differences in the micro-climate or micro-habitat? Why do some insects stay at the bottom and some stay at the top of the plant?

Weather and plants

We have discussed the effect of weather on the important processes of insect development (individuals and populations) and the effect of weather on the important processes on disease development. We began by making a general model of development for insects (egg - larvae/pupae - adult) and diseases (germination - infection - incubation - reproduction and movement). Next we considered the effect of weather (temperature, rain, humidity, solar radiation, etc.) on each of the general processes.

Why did we work this way? First is that we must learn to take big problems and break them into smaller problems. This is a problem solving method and very important for considering complex problems and interactions. Second is the importance of generalizing. You have all learned much specific information but you must be able to put this information into a general framework that relates to other problems.

Now to the main topic. These exercises are getting longer, but hopefully you are getting the skills and concepts.

In this activity we will look at the important stages of plant growth and development and discuss the influence of weather.

Plant stages (in general)

1. *Transport of seeds or vegetative parts.* This is especially important for weeds and other wild plants.
2. *Germination*
3. *Vegetative phase.* This is the important stage for building a foundation for the reproductive stage. More flowers and fruits will be produced when more starch is stored in the plant (how does the plant make starch?). Think about the vegetative phase as the population of stems, leaves and roots in the field. This is something like the larvae/pupae of insects.
4. *Flowers.* These are important organs on a plant. For determinant plants the flowers are produced only for a short period and are therefore very sensitive to harsh changes in weather. Think about these as the population of reproductive parts. This is something like female adults in insect populations.
5. *Maturation of seed.* Seeds must be produced and matured to be viable for the next growth. This is similar to populations of eggs produced by adult insects (fecundity).

In this activity consider the influence of weather on each stage. Use a systematic approach. Give examples. Break the big problem to smaller problems that can be discussed.

Objective:

Systematically explain the effect of weather on plant development

Materials:

Big paper, markers

Method:

1. On the piece of paper, make three columns. Label one column "Plant development". Label the second column "Weather parameter" and the third column "Effect of weather".
2. Discuss and fill in the chart. Give examples from different crops and weeds.
3. Each group should then give a systematic explanation of the effects of weather on plants to the other groups.

Diseases

Identification

Identification of disease symptoms

Introduction

This exercise shows that one can group types of diseases and learn about the developmental stages of a disease in the field without knowing the names of diseases.

Objectives:

- Distinguish between different groups of disease symptoms
- Compare developmental stages of each disease group

Materials:

Cotton field with different diseases in different progressive stages
Hand lens (at least one per group)
Drawing paper and crayons

Method:

Visit the field and ask each group to collect as many different disease or disease-like symptoms in different progressive stages as can be found (so not only leaf spots but also other disease symptoms such as deformed roots, discoloured leaves, etc.) Also collect plants with symptoms that might be caused by nutritional deficiencies.

Observations:

In the classroom, group the disease symptoms based on symptom groups like leaf spot diseases (including molds/mottling), stem disorders, wilts, boll rots and root disorders. Assign each disease group to a group of trainees. Ask each group to rank the symptoms in order of severity. Use the hand lens to check for spores of fungi. (Spores can sometimes be seen as moldy dusty appearance on a diseased area.)

Ask groups to draw the details of the different symptoms and disease development in color. Avoid the use of scientific terms such as Latin names of diseases.

Discussions:

1. Which diseases or disease groups are present?
2. What are the local names of the diseases?
3. Were there also symptoms caused by nutritional deficiencies or mechanical damages? Can you always distinguish these from diseases?
4. How do the symptoms look like? How do they start? Which plant parts are affected by the different diseases?
5. How do the diseases reproduce and spread? How can one find out?
6. Which are the most problematic diseases? Why?
7. How does the weather influence the development of diseases? When are the diseases most severe?
8. How can cultural practices influence the development of a disease? Which non-chemical disease management practices are known to control the disease?
9. Which method can be used for a short-term control, which ones for a long-term management?

Disease groups

Introduction

In order to be able to discover about disease management, one should appreciate information that is already available on life cycles of diseases. This exercise taps the information known by members of the group and links it up to practical field school situations. The exercise should not/does not “test” participants’ knowledge on diseases but summarizes the knowledge available and triggers creative thinking about how to find out about and manage diseases.

Objective:

List down available information on disease ecology and management of diseases

Materials:

Drawing paper and markers

Method:

List down diseases of the crop using the following guide questions. Remind trainees that “I don’t know.” is a truly valid answer and a better answer than “I guess. . .” at all times.

- Which diseases of this crop do you know? (Use local names.)
- What are the symptoms of the disease/s?
- When does it occur?

When the list is completed, classify them according to disease group using the question:

- Is the disease caused by a fungus, bacterium, virus or nematode? (Some participants may know if the disease is caused by a fungus, bacterium, virus, or nematode. Some may not. Ask participants to recall earlier discussions on symptoms of the disease/s. Remember that the exercise should not/does not “test” participants’ knowledge on diseases but summarizes the knowledge available and triggers creative thinking about how to find out about and manage diseases.)

After completing the task of grouping the diseases according to disease groups, focus on the method of spread. Ask participants to recall their observations of diseases in the field and how they spread. Use the following questions:

- Does the disease spread through water?
- Does the disease spread through infected seeds?
- Can it survive and multiply on weeds?
- Can it survive on plant residues?
- Can insects spread the disease?
- Can humans spread the disease?

If they are not certain, follow up each question with:

- What experiments can be designed and conducted to find out about this?

List information about disease groups on poster paper and put them up on the walls. These posters may be used as reference during future sessions.

Disease groups game

(This activity can be used as an icebreaker to start a session on disease groups.)

Objective:

Illustrate a simplified distinction of disease groups

Method:

Demonstrate how to make movements to represent different disease groups. For example:

- a bacterium places her hand behind the back, wiggles it like a tail, while circling in a spot
- a fungus outstretches arms and fingers like a tree
- a virus stands rigid and tall like a rod-structured virus particle
- a nematode moves one arm like a snake

Everyone stands in a circle. An *'it'* stands in the center of the circle and gives instructions. First, everyone in the group makes the gestures that signify the disease group that is called out by the instructor. When everyone is familiar with the gestures, the game can start.

The *'it'* points randomly at a participant and calls out a disease group, e.g., nematode. The participant must show the gesture that signifies the group mentioned, in this case, move one arm like a snake. In case the participant fails to show the correct gesture, she is eliminated from the circle. The game continues until only one player is left.

Study of symptom development of leaf spots: session room exercise

(This exercise is best done at the same time as the field exercise.)

Objective:

Observe the symptoms of leaf spot diseases

Materials:

Cotton field with symptoms of leaf spot diseases

Petri dishes, jars, clear plastic boxes

Tissue paper

Labels/tags

Poster paper, crayons, ruler, hand lens

Method:

Visit the field and collect leaves with small leaf spots (early stage of disease).

In the session room:

Use whole leaves or cut leaf portions with small leaf spots onto discs of e.g. 10cm diameter. Using a marker, draw a big circle around the leaf spot that you want to study on each leaf or leaf portion. Place each leaf or leaf portion in a petri dish lined with moist tissue paper. Close the lid. In case petri dishes are not available, one can use clear plastic boxes with lids or clear plastic bags that can be closed tightly. Leave some air inside!

Observations:

Draw each leaf spot and the area around the spot in detail, using crayons. Measure the diameter of the leaf spot. Use a hand lens to see whether you find any granular structures in the leaf spot (sporulation). Observe the leaf spot each or every other day and regularly draw and measure the size of the leaf spot. After one week, groups can be asked to present their findings.

Discussions:

1. What happens with a leaf spot over time (color, structure, area around spot)?
2. What is the difference between a fungal leaf spot and a bacterial leaf spot?
3. What is the difference between a disease spot and insect injury? If the spot is caused by an insect injury, do you think it would increase over time? Why or why not?
4. Is the leaf spot disease harmful to the crop?

Study of symptom development of leaf spots: field exercise

(This exercise is best done at the same time as the session room exercise.)

Objective:

Observe the symptoms of leaf spot diseases

Materials:

Cotton field with symptoms of leaf spot diseases

Labels/tags

Poster paper, crayons, ruler, hand lens

Method:

In the field, select a plant with a few small leaf spots on preferably young leaves. Label the plant. Tag the leaf with a small leaf spot. Using a marker, draw a wide circle to mark the spot.

Observations:

Draw each leaf spot and the area around the spot in detail, using crayons. Measure the diameter of the leaf spot. Use a hand lens to see whether you find any granular structures in the leaf spot (sporulation). Observe the leaf spot each or every other day and regularly draw and measure the size of the leaf spot. After one week, groups can be asked to present their findings.

Discussion:

1. What happens with a leaf spot over time (color, structure, area around spot)?
2. How can one recognize the first beginnings of a leaf spot?
3. What is the difference between a fungal leaf spot and a bacterial leaf spot?
4. What is the difference between a disease spot and insect injury?
5. What was the effect of the weather during the experiment?
6. If the exercise was done simultaneously with the session room exercise: Was there a difference in the development of leaf spots in the field and in the session room? If yes, why?

Study of symptom development of bacterial wilt disease

In the past, wilt disease had serious influence on cotton plants. The disease may be caused by other factors like fungus but the main agent was bacteria. Nowadays, however, varieties are available that are resistant to bacterial wilt. In this activity we learn more about wilt caused by bacteria by doing some pot experiments.

Objectives:

- Diagnose symptoms of bacterial wilt disease on cotton plants
- Observe spread of bacterial wilt disease
- Discuss management practices for bacterial wilt disease

Materials:

Field of cotton plants with different diseases

Two transparent plastic glasses per group

Toothpicks (at least six pieces per group)

Clean water

Potted healthy plants (4 per group)

Drawing paper, crayons

Knife

Method: Part I - Extracting the Inoculant

At the beginning of the season, each group should establish seedlings in four pots.

When disease starts to be observed in the field, visit the field and collect 3-5 infected plants per group. Carefully observe the appearance of the infected plants exhibiting symptoms of wilt disease. In the session room, set up the experiment following the steps:

1. Cut off roots and leaves of the plant. Cut the stem into pieces of 10 cm each. (*Note: Do not wash or clean the knife. This will be used to infect healthy plants in the second part of the experiment.*) Stick in three pieces of toothpicks into the stem to form a tri-pod so that the stem can be set up in the glass vertically.
2. Half fill the glass with clean water and put the stem in. About 3-5 cm of the stem should be in the water. Put the glass in a well-lighted area of the room for easier observation.
3. Observe after thirty minutes. Participants should not move the experiment so that the water is not disturbed.
4. Based on results of the experiment, i.e., when participants have established that the plant is infected by bacterial wilt disease, use water to establish pot wilt disease study. (*Note: Plants infected by bacterial wilt disease will have ooze coming out of the stem and moving into the water as a white substance.*)

Part II - Spreading Bacterial Wilt Disease

To set up pot studies on spread of wilt disease, use the following steps:

1. Each group should set up four pot studies to observe spread of bacterial wilt disease using the seedlings established in pots at the beginning of the season.
2. Infect using the following methods:
 - 2.1. Pot 1: Using inoculant (water) from Part I of the experiment inject ooze solution in collar of plant.
 - 2.2. Pot 2: Using inoculant (water) from Part I of the experiment inject ooze solution into leaves.
 - 2.3. Pot 3: Damage roots under the soil by a syringe. Put cotton with ooze solution to wound.
3. Keep another pot for control/comparison.
4. Observe results.

Observations:

Weekly draw appearance of the plant from the time that the inoculant was introduced. As the infected plants start to wilt, do the diagnostic exercise to check the cause of infection (look for bacterial ooze).

Discussions: Display results to whole group.

1. What were the symptoms of bacterial wilt disease in the field? How is this distinguished from symptoms caused by nutritional deficiencies or mechanical damages?
 - * How do you identify the disease (what are the symptoms, where are the symptoms located)?
 - * Where does the disease come from?
 - * How does the disease spread?
 - * How does the disease enter the plant?
2. Why did we use potted plants and not the field for the infection study?

Spread

Factors that influence disease development - exercise 1

Introduction

As you are aware, many factors affect disease development. This includes the environment (soil, weather, wind). In this activity, we will consider various factors and see how they affect disease.

Method:

1. Divide into five groups. Discuss and list down all factors that affect disease development.

DISEASE: Leaf disease like blight

(Use the same table for stem or root diseases such as wilt, rot, etc.)

| Factors | Favorable | Unfavorable |
|--|------------------|--------------------|
| Fertilizer | | |
| Weather (e.g., temperature, humidity, dew period, rain, wind) | | |
| Microenvironment (the conditions right around the plant that could be affected by number of leaves, plant density, etc.) | | |
| Soil conditions | | |
| Water conditions | | |
| Other factors | | |

2. For each factor and disease discuss and write down which aspects are favourable and unfavourable to disease development.
3. Present results to the whole group and discuss together. As groups present, summarize results into one table.
4. Supply additional information and correct misconceptions, if necessary.

If many diseases are important, spread the exercise over 2 weeks.

Factors that influence disease development - exercise 2

(If the exercise is being done in a regular field school, the trainer should prepare the cards before the session to give farmers more time to discuss about factors that influence disease development instead of preparing the cards.)

Last week you made a list of all factors that influence disease development. Now, we will play a card game to allow us to consider what would probably happen to disease under various conditions. Some are listed below:

- a) Weather
 - i) Temperature
 - ii) Humidity
 - iii) Dew period
 - iv) Rain
 - v) Wind
- b) Soil conditions
 - i) Intrinsic soil properties
 - ii) Fertilizer applied
- c) Microenvironment - the conditions right around the plant that could be affected by -
 - i) Number of leaves
 - ii) Plant density
 - iii) Water conditions
- d) Water conditions
 - i) Drought stress
 - ii) Wet soil

Objectives:

- List different factors that could affect disease
- Discuss how each affects disease such as blight as well as other pests of interest
- Discuss the risks of disease associated with various situations

Materials:

Big piece of paper and pens
Paper cut into small pieces

Method:

1. List different factors that have an effect on disease, using the results of last week's exercise.
2. Organize the list into groups of related factors (classify by weather, microenvironment, etc.).
3. For each of the aspects, discuss how the factor affects disease. (See table below if ideas are needed.)
4. For each factor, set various conditions.

- a) Fertilizer
 - i) high amount of fertilizer
 - ii) medium amount of fertilizer
 - iii) low amount of fertilizer
- b) Temperature
 - i) high
 - ii) medium
 - iii) low
- c) Humidity
 - i) dry air
 - ii) wet air
- d) Rain
 - i) heavy rainfall
 - ii) moderate rainfall
 - iii) drought
- e) Variety
 - i) resistant variety
 - ii) moderately resistant variety
 - iii) moderately susceptible variety

- iv) susceptible variety
- f) Crop stage
 - i) seedling stage (square formation)
 - ii) 20-55 DAS (flowering stage)
 - iii.) 55-75 DAS (boll formation stage)
 - iv.) 75-95 DAS (boll opening stage)

5. For each factor, each group should make a pile of cards with different conditions written on different cards.
6. Within each small group, each person should pick a card from each pile. The combination of cards will specify the situation. Each person should describe to the group how much disease they expect to have and how they will handle the situation.

Discussions:

Example: If your situation is as follows, how much risk is there for a lot of disease?

- a) medium amount of fertilizer
- b) high temperature
- c) wet air
- d) moderate rainfall
- e) moderately resistant variety
- f) square formation stage

After finishing the exercise, assign certain diseases to small groups. Ask groups to make tables summarizing how different factors influence the different diseases assigned to them. For example, how high-medium-low level of fertilizer influences blight. You can use the following table:

Leaf Blight

| Factors | How they influence disease development |
|---------------|--|
| Fertilizer | |
| Plant density | |
| Weather | |
| Etc. | |

Each group should present their tables to the big group. Discuss and summarize results.

Demonstration of spread of diseases

Introduction

An important aspect of disease management is sanitation. In order to prevent spread of disease, roguing is practiced or farm tools are cleaned after cultivating a field with a history of disease. Sanitation, however, is often neglected and one of the reasons may be that farmers do not understand the mechanism of spreading of diseases. This exercise demonstrates the spread of splash-dispersed (such as leaf spot disease caused by a fungus), soil-borne (such as nematodes) and insect-vectored diseases (such as a virus).

Objective:

Demonstrate spreading of diseases by splashing water, soil cultivation and by insects

Materials:

Field with preferably young crop (weeded)
Watering can
Clean poster paper
Hoe or other soil cultivating tools
Wheat flour or fine seeds of a fast germinating crop (e.g., watercress)
Syringe or straw
Five glass or plastic vials, one with strong red dye, four others with clear water

Method:

I. Demonstration of spread of disease by splashing water

Make sure that the soil is dry. Fill the watering can with water. Place a sheet of poster paper in between plants within a row and water the crop to simulate rain. Observe soil splash from between plant rows to the poster paper within the plant rows and explain that soil-borne diseases spread in this way.

Also, try using two plots of dry, bare soil (each about 1 X 2 m².) Leave one plot bare and cover the other plot with mulch, e.g., rice straw, sugarcane bagasse, leaves from trees. Place sheets of poster paper along the 2m border of each plot. Water each plot and compare the soil splash on both pieces of poster paper.

II. Demonstration of spread of disease by soil cultivation

Make sure that the soil is dry. Sprinkle 1 kg of flour on the soil between several plant rows and explain that this represents spores of a fungal disease or nematodes. In one row, ask a participant to use the hoe or other farming tool/s (wet the tool first) and simulate weeding of the field. In another row, ask the participants to wet the soles of their shoes/boots/feet and walk through the flour on their way to inspect nearby plants. Observe spread of flour. In case the field is wet, replace the flour by fine seeds and observe after germination of weeds.

III. Demonstration of spread of disease by insects

Use the syringe or straw and the vials, one with strong red dye and the others with clean water. Demonstrate spread of insect-borne viruses with the syringe that represents the mouth parts of a sucking insect. The vial with red dye represents a virus diseased plant, the vials with clean water represent healthy plants. Draw red dye with the syringe and move to the first vial with water. Draw in some water, ejecting ('spitting') a little red dye into the vial. Observe the coloring of the water. (Healthy plant becomes infected with 'virus'.) Move to the other vials with clear water and infect them one by one. If you want to show dilution, you also have to draw in some water from each vial. Observe that the coloring of the water in the vials and the reduced inoculum in the syringe is diluted every time it is used with a 'healthy plant'.

Discussions:

1. What did you observe?
2. Which diseases do you know spread in this way, i.e., splash; soil; insect?
3. How might these methods of spreading disease affect crops in the field?
4. How could spread of disease be prevented?

Virus and vectors

Introduction:

One of the most difficult aspects of cotton growing is the presence of viruses. These viruses are moved from season to season and plant to plant either by the seed itself or by insect vectors.

In Vietnam, aphids and possibly jassids are insects that are able to transmit viruses from one plant to another. In other countries, whiteflies and flea beetles may be present. What do they have in common? Of course, they all have sucking mouth parts and enjoy feeding on several plants. Some viruses can be transmitted after just a few seconds of feeding (non-persistent viruses). This is more like the mouth parts of the insect sucking on a infected plant move the virus to another healthy plant when feeding a little later on this plant. When insects transmitting this type of virus are sprayed, the insects move from plant to plant and the result can be (but not always!) that there is actually more virus than if not sprayed! The virus is acquired quickly by the insect, and the ability to transmit the virus is quickly lost.

The other type of virus requires a longer period of feeding, usually minutes to hours for the vector to get enough virus to move to another plant. Probably, the virus must build up in the mouth parts of the insect so that when the insect moves to another plant there is enough inoculum to get an infection. Insects usually force saliva into the plant when beginning a hole for feeding. This outflow of saliva is sufficient to carry virus into the plant. This type of virus is called "persistent" because the insect can persistently transmit the virus to many plants after obtaining the virus from an infected plant. Virus between "non persistent" and "persistent" types are called "semi-persistent". Persistent viruses can be carried long distance by the vector because the virus is not easily lost from the vector. The vector must feed for a long time the first time to acquire the virus, and the vector must feed for a long time to be able to transmit the virus. Prevention of viruses is very difficult. The farmer has few choices but spray if he expects a lot of virus. As field observers, no good recommendation can be given due to the lack of information.

In the following activity, we will collect different types of sucking insects that can be vectors of viruses in order to observe their mouth parts. We will also demonstrate the transmittal process by using our own mouths.

Objectives:

Describe the shape and function of typical vector insects
Use straws to demonstrate the transmittal process by vectors

Materials:

Paraffin wax, heat source, magnifying glass or 10x dissecting microscope, straws, red dye

Method:

I. Observation of mouth parts

1. Collect aphids and jassids from the field and from other plants.
2. Bring the insects to the laboratory and kill them with alcohol.
3. Mount the insects on their back in the wax trays. This is done by partially melting the wax with a wire. Make sure the mouth parts are above the wax.
4. Observe the insects with the magnifying glass or microscope and draw their mouth parts.

II. Simulation of virus being transmitted by vector

1. Set up three groups of four clear cups. Put water in each cup. In the first cup, put a drop of food colouring ("virus").
2. Dip the straw ("mouth parts") in the first cup just for a moment. Then dip into the next three cups. What happens in each of the cups? What kind of transmission does this simulate?
3. Now place some cotton in the end of the straw ("mouth parts"). First dip the straw in the "virus" and then into the next cups. Is there any difference in the results from this and the previous treatment?
4. Leave the mouth parts in the virus for a minute. Now dip the mouth parts in the other plants and leave them for a minute in each cup. What is the result? What kind of transmission does this simulate?

Discussions:

1. In the field, just a few insects are able to transmit virus, but often there are many virus-infected plants. Why?
2. Why control virus vectors when a large number of plants are infected with the virus? Is there a reason to control the insects? What about the economic analysis of such a situation?
3. What are the important viruses transmitted by insects in cotton?

Study of spread of fungal leaf spot

Objective:

Observe the spread of a fungal disease from an infected to a healthy plant

Materials:

Four **healthy** potted plants
Leaf spot infected leaves
Clean water
Small hand sprayer
Clear plastic bags
Tissue
Labels

Method:

First day preparations:

Germinate spores by inserting the leaf spot infected leaves in a plastic bag with moist (not soaking wet) tissue paper. Close the bag tightly but leave some air inside to avoid rotting. Leave the bag overnight.

Second day:

Bring the potted plants into the classroom. Put clean tap water into the container of the small hand sprayer. Spray two potted plants with clean water. Label one plant '*healthy control, uncovered*'. Cover the other plant with a plastic bag and label the plant '*healthy control, covered*'.

Prepare the disease inoculum by grinding and squeezing out extract from leaves and adding this to water OR by stirring the leaf portions with leaf spots in a glass with clean water. Transfer the inoculum to the small hand sprayer. Spray the inoculum on the other two potted plants. (Note: Add sticky substance to the solution, e.g., Tween 80 Liquid Detergent, so that the spores will stick to the plant.) Label one plant '*leaf spot infected, uncovered*'. Cover the other plant with a plastic bag so that high humidity is maintained and label that plant '*leaf spot infected, covered*'. Clean the hand sprayer carefully after use. The plastic should not be removed, except for observations or watering of the plants.

On all four pots, spray water 6 - 8 times per day to create the environment for disease development.

Observations:

Observe the development of symptoms in both pots over time. Once the symptoms have been observed sufficiently, destroy the infected plants to avoid infection of other plants.

Discussions:

1. Why did we inoculate the plants inside the session room and not in the field?
2. Why did we set up covered and uncovered treatments?
3. How many days did it take before symptoms were visible?
4. How does a fungal leaf spot spread in a field?

Pot experiment to test whether root diseases are soil borne

Objective:

Demonstrate disease development of healthy plant material in contaminated soil

Materials:

Cotton seeds
Four or more large pots
Clean soil
Labels

Method:

Fill four pots with clean soil. Collect diseased plant and cut up the roots into many small parts and mix with the soil in two pots. Label the pots '*infected soil*'. Label the two other pots '*healthy soil*'. Sow seeds in each pot. Water the plants regularly (if necessary under a screen cage to keep insects out) until symptoms appear. Apply a little fertilizer, if needed. Observe plant development in the two treatments over time.

Discussions:

1. Why did we use potted plants and not a field for the infection study?
2. How many weeks did it take before symptoms became visible?
3. Can you estimate the yield loss in the field?
4. How does the disease spread in a field?

Test of cotton seed quality

Introduction

The use of clean seed is essential to control diseases. To separate light-weight, possibly disease infected seeds from healthy seeds, a separation test with water is used. The following exercise will show whether light-weight seeds have a lower germination capacity and whether discoloration or fungal growth upon germination can be seen.

Objective:

Test seed lots for germination capacity and infection with seed-borne diseases

Materials:

Cotton seeds
Tissue paper
Clear or black plastic bags
Clean water
Hand lens

Method:

Take a sample from the seed lot. Prepare a container with water. Put the seeds in the water container. Stir the seeds. Allow one or two minutes for the seeds to settle. After settling, carefully remove the floating seeds. Put these in a container labelled as “floating seeds”. Also collect the remaining seeds and label them as “sinking seeds”. Count 100 seeds of each seed lot.

For each seed lot:

Prepare two layers of tissue paper and carefully sprinkle clean water on the tissue. The tissue should be moist but not soaking wet. Position the 100 seeds on the tissue paper in 10 rows of 10 seeds with a distance between seeds of about 3 cm. The seeds will stick on the moist tissue. Cover the seeds with another layer of tissue paper and also slightly moisten the top tissue layer with clean water. Loosely roll up the tissue with the seeds inside into a ‘sausage’. Put the roll into the plastic bag to keep it moist. Close the bag but leave some air inside. Label the bag either “floating seeds” or “sinking seeds”, according to the seed lot inside. Keep the bags in a dark place.

Observations:

Daily observations on germination and growth of mold can start after one or two days. After each observation, note the number of germinated seeds and the number of seeds with mold growth (hairy fungal structures) or rot. Use the hand lens to check each seed carefully. After each observation, check the moisture of the tissue. If dry, sprinkle some more water on the tissue. Do not remove the seeds. Again, roll up the tissue and put back into the plastic bag for further (daily) observations. After seven days, if possible, measure root length on the germinated seeds. After one week, or longer if desired, results can be summarized in a bar graph (horizontal: days after ‘sowing’; vertical: cumulative % germination and cumulative % diseased seeds) on poster paper and presented per group.

Discussions:

1. Was there a difference in germination between the treatments?
2. Was there a difference in the number of seeds with mold growth or rot between treatments?
3. What would happen with the seeds of each lot in a seed bed?
4. Why should the use of diseased seeds be avoided?

Effects of inundation of fields on incidence of wilt diseases

Objective:

Demonstrate the reduction of soil-borne wilting diseases by rotation with paddy rice

Materials:

Dryland soil from a field continuously planted with cotton with a history of wilting disease

Wetland soil (of a similar soil type as the dryland soil) from a paddy rice field (preferably one that has been flooded with water for two to three months)

Cotton seeds

Method:

Prepare one seedbed in the field with soil that has been continuously planted with cotton. Label this as 'dryland soil'. Take soil from the paddy rice field and prepare another seedbed using the paddy soil. Label this as 'wetland soil'. Sow (drill) 100 seeds in rows in each seedbed using a space of at least 5 cm between each seed.

Discussions:

1. What are the differences between the two treatments?
2. Did wilting occur? If yes, was there a difference in wilting incidence between both treatments? Why?
3. Would it be possible for farmers to prepare seedbeds with wetland soil? What would be the advantages and what would be the disadvantages?
4. What is the importance of crop rotation on incidence of wilt diseases?

Management

Disease management

Introduction:

Diseases are an important part of crop protection, but are usually very difficult to understand in the field. This is partly because the causal organisms (bacteria, fungi, viruses) are very small and cannot be seen moving around like insects or rats. We must learn new ways of thinking about these organisms in order to better manage diseases.

Management includes prevention and slowing down epidemics. Diseases will never be completely eradicated - only populations reduced to very low levels. Management usually needs the cooperation of several farmers working together to reduce overall disease in an area.

What are management activities? Below are some activities.

1. Allowing only disease-free seed and planting materials into an area. This can be done at any level of organization: farmer group, village, district, province, national.
2. Careful purchase of materials in the market and plant sellers.
3. Selecting good varieties.
4. Sanitation is important for keeping inoculum from one crop to get into the next crop.
5. Destroy sources of inoculum such as material in nurseries and fields with diseases.
6. It is also important to keep nematode infested soil from moving from field to field on the shoes of farmers, on buffalos, and plows.
7. Deep burial of diseased plant materials by plowing, removal of diseased plants, burning harvested crop residues, and repeated plowing to expose the soil to sunlight
8. Proper fertilizer management. There are numerous examples in which addition of nitrogen, potassium or calcium actually reduces the effects of certain fungi.
9. Small areas planted to a particular crop before the main growing season for the crop should be avoided. These small areas build up inoculum that is then carried over to the main season.
10. Crop rotation using crops which are not infected by the same diseases.
11. Crop planting times should take into consideration dominant diseases in the area and the effect of the micro-climate.
12. Using appropriate planting densities.

Some of these activities are related to the management of disease by effecting some changes in the environment. Some have to do with the plant and others have to do with effecting changes in the disease organism or pathogen. In this activity, we will use a method called brainstorming to develop area management methods and activities. The process is as important as the content since management implies participation of many persons.

Objectives:

- Outline management activities that could be organized for an area to reduce disease incidence
- Use brain-storming techniques to develop inputs from all participants

Materials:

Big paper and markers

Method:

(Brain-storming is a method of getting lots of creative ideas. Many ideas will not be useful, but the ideas will act as seeds to other ideas. Discussion of ideas is allowed only after all ideas have been written down)

1. Assign one person as the secretary who will write on the large piece of paper. Do not use a small piece because the whole group should be able to read the paper. Assign another person to be the facilitator.
2. The secretary should write "Area-Wide Disease Management" on the top of a large piece of paper.
3. The facilitator should ask the group what methods farmers practice in their localities to manage diseases.
4. The group members should tell the secretary their ideas. The secretary will write down the ideas. No comments are allowed from other members at this point. If any member makes comments, the Facilitator must ask the person to be quiet.
5. Continue writing down ideas with no discussion until the first page is full.

6. After the page is full, discuss each idea beginning at the top of the list. The Facilitator should be sure each person can make some comments. The Secretary should summarize the discussion on each point. Write the summaries on another large piece of paper. The summary should be along the angles of the disease triangle, i.e., some activities are related to the management of disease by effecting some changes in the environment; some have to do with the plant and others have to do with effecting changes in the disease organism or pathogen.
7. If there is time, use the same process using the following question: "What can IPM trainers do to help manage diseases in our village?"

Disease triangle to explain disease management

Introduction

Results of earlier exercises may form the basis for a discussion on disease management. It shows that diseases only become problematic when the interaction between pathogen, crop and environment is optimal for the pathogen. The exercise calls attention to the fact that disease management basically consists of orchestrating the pathogen, crop and/or environment.

Objectives:

- Reinforce discussions on disease management
- List down management practices for each component of the disease triangle to 'inactivate' disease spread

Materials:

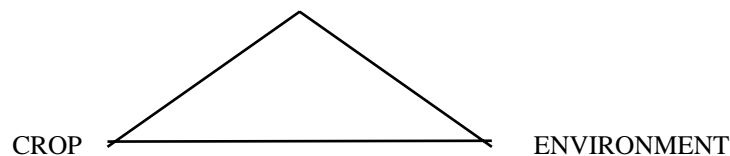
Big paper, pens, markers

Method:

Ask participants to recall the earlier discussion on disease management, i.e., that changes may be effected on the environment, plant or the pathogen to prevent disease. Also that for the development of disease, these three factors must be present or favourable. Ask for volunteers to give examples. For instance, a fungal disease that survives on crop residues in soil (Is the disease present? -> Yes) will definitely show when a susceptible crop (Is a susceptible crop present? -> Yes) is planted in a rainy season (Is a suitable environment present? -> Yes).

Draw the disease triangle:

DISEASE



Discussions:

Discussions may focus on the fact that the disease triangle helps us understand management practices that may be tried out or avoided to 'inactivate' at least one of the angles in the triangle. The following examples may be used to start a discussion on practical implementation of disease management strategies.

Disease angle (Is the disease present?):

1. To avoid a soil-borne disease, one could test the use of sub-soil in the nursery (Is the disease present? -> No? -> How would you apply this method in the field?).
2. To avoid an insect-transmitted virus disease, one could try to cover a nursery with screen-netting (Is the disease present? -> No? -> How would you apply this method in the field?).
3. A season with paddy rice can be considered as a season of inundation of soil with water. Certain soil-borne diseases are killed when soil is flooded for a period of time (Is the disease present? -> No? -> How would you apply this method in the field?).
4. By implementing sanitation measures such as removal of infected crop residues or diseased plant material in the field, one can test whether removal of sources of infection reduces disease (Is the disease present? -> No? -> How would you apply this method in the field?).

Crop angle (Is a susceptible crop present?):

1. Search for resistant hybrids by planting a portion of the field with other hybrids from neighboring areas and/or imported hybrids (Is the crop present? -> No? -> How would you apply this method in the field?).
2. Crop rotation by avoiding planting susceptible crops for several cropping seasons (Is the crop present? -> No? -> How would you apply this method in the field?).
3. Weeding of susceptible weeds (Is the crop present? -> No? -> How would you apply this method in the field?).

Environment angle (Is a suitable environment present?):

1. Choose a season that is not favorable for disease, e.g., the dry season (Is the environment favorable? -> No? -> How would you apply this method in the field?).
2. Change from overhead irrigation to flooding to reduce leaf wetness (Is the environment favorable? -> No? -> How would you apply this method in the field?).

After the discussion, divide the group into four. Refer to the session on disease/symptom groups. Assign one disease group to each group of participants. Ask each group to select one disease for a crop and to design a management measure that can be tested in the study field. Ask groups to present after they have completed the task. Discuss which angle of the disease triangle is avoided or inactivated. Try to implement the management measures that the groups present.

Management of different disease groups

Objectives:

- Identify different methods to manage diseases in the field
- Discuss how the methods can be applied for each of the disease groups in the field

Materials:

Big paper, pens, markers

Method:

With the whole group, first make a list of all possible methods that can be used to manage and control diseases in the field. Then each group selects one disease. For each disease discuss how you can use each of the possible methods that you listed. Whether certain methods can be used or not, depends very much on how a disease develops in the field. For the disease that your group selected discuss the questions in the following section.

Discussions:

1. How do you identify diseases (what are the symptoms, where are the symptoms located)?
 2. Where does the disease come from?
 3. How does the disease spread?
 4. How does the disease enter the plant?
 5. What development stages of the disease can you identify?
 6. What factors stimulate or inhibit development of the disease?
 7. What damage does it do to yield or quality of the crop (why, how)?
 8. What other information do you need to make a decision on management/control of the disease?
 9. How can you obtain/discover this additional information in your own field?
 10. What can you do with this information? How does it help you to make a better management decision?
- Summarize the results of your discussion on a big piece of paper. Each group will present the findings of their group.

Gender

Gender awareness

Background

The gender awareness exercises highlight the roles, responsibilities and relations between women and men in Vietnamese society in general and in agriculture in particular. Carrying out the exercises gives the opportunity to discuss ideas about these roles, responsibilities and relations and about the capabilities of women and men to participate in IPM training.

Objectives

1. to get insight into ideas about female and male characteristics, roles, responsibilities and relations;
2. to understand how these ideas influence opinions about the potentials of women and men to participate in IPM; and
3. to discuss women's and men's capabilities to participate in IPM training looking from a gender perspective.

The gender awareness exercises may be used in TOTs, Refresher courses, Farmer Field Schools or Planning Activities with farmer groups. For the TOT or Refresher training, it is recommended to start the gender awareness session with the first exercise about female and male characteristics/stereotypes.

Female and male characteristics and stereotypes

Objectives:

- Describe and analyze female and male characteristics
- Discuss ideas about female and male characteristics that can lead to stereotyping of women and men in our society
- Explain why we value certain female characteristics more than certain male characteristics and certain male characteristics more than female characteristics
- Discuss that women and men have different biological characteristics but can have similar social characteristics. Female and male biological characteristics cannot be (completely) changed, but female and male social characteristics have changed over time and are inter-changeable
- Discuss whether or not women can participate in IPM based on the results of the exercise

Materials:

Board, chalk or newsprint, markers

Time:

20 minutes

Method:

When you introduce this exercise

- Use your field experience and information given in the background section and notes for trainers.
- Questions such as: 'how many days in the year is women's day, how many days are men's days' or 'can you name a male leader, can you name a female leader' or 'who talks a lot in this field school, women or men', help to get participants into the exercise.

1. Make a list on the board or big paper of male and female characteristics, as follows:

| WOMEN | MEN |
|---------------|-----------------|
| - patient | - talk loudly |
| - sensitive | - strong |
| - gives birth | - grow mustache |
| - etc. | - etc. |

2. Ask the participants to add more characteristics for women and men, list them in the table. Note: among the characteristics listed there should be at least one relating to the sex characteristics of women and men (for example, getting pregnant, giving birth, breast feeding, growing a mustache or beard, etc.)
3. After finishing the list, go through each item under the heading women. Ask if men can also be patient, sensitive...? If so, mark the "yes" answer with "+". Mark the characteristics that are unchangeable (getting pregnant, growing a mustache etc.) with "-". Do it likewise with the men's column. Ask if women could talk loudly, be strong, etc. Continue to mark "+" for the "yes" answer until finished.

Discussions:

1. What female and male characteristics can not be changed? Which characteristics can both males and females have?
2. How would you react if a woman talks loudly and is stronger than a man? How would you react if a man does not talk loudly and is not as strong, but patient and sensitive?
3. Are women always patient, sensitive etc.? Do men always talk loudly, are strong etc? Why or why not?
4. Based on all the female and male characteristics identified, which ones are important for field school participation? Why? Can both women and men have these characteristics?
5. Do you think women are capable of participating in FFSs and follow-up activities? Are women still under-represented in FFS and follow-up activities? If so, why? If women are participating in FFS, do they feel shy to give their opinions? If so, why?

Optional

You can proceed with the following addition to this exercise. This will take 20 minutes more. This addition will focus on the criteria for the selection of field school candidates. It may be useful to carry it out in a village when the participation of women is still low, even after more than two seasons of farmer field schools. It may also be useful to carry it out or when the women continue to be very shy in the field school and the men continue to be dominant or make jokes that alienates women more.

1. Ask the participants what they think should be criteria for field school participation. Make a list of their ideas, not in detail, but use keywords. Make a table with four columns. List the criteria in the first column. The following table can be used to organize the opinions from the participants and the discussion based on their opinions.

| Criteria for FFS participation (stereotypes) | Opportunity for women to participate | Rationale for FFS participation | Opportunity for women to participate |
|---|--------------------------------------|---------------------------------------|--------------------------------------|
| Example: educated and able to understand and take notes..... | low | be a farmer and have field experience | high |

2. After you finish making the first column, ask the participants if the criteria they identified will make it easy or difficult for women to be selected or to participate in the field school. Why or why not? List in the second column whether based on the criteria listed the opportunity for women to be selected is high, medium or low.
3. After you finish the second column, ask the participants what should be the rationale for field school participation, that is, what should be a real requirement for field school participation as opposed to a stereotype assumption. List them in the third column. Note: request the participants to take into consideration the exercise they just carried out.
4. After finishing the third column, ask the participants if the rationale, the real requirement for field school participation would make it easier or more difficult for women to be selected or to participate in a field school. List in the fourth column the opportunity for women to participate.
5. Do you think that stereotype selection criteria make it more difficult for women to be selected for IPM training? If you reformulate the criteria, would it be easier for women to be selected? Do you think that reformulating the criteria, and using those for selection will result in getting capable participants in the Field School? What kind of criteria would you choose to use when you discuss with village leaders on the selection of participants for a FFS? Why?

Women's and men's roles in cotton cultivation: two generations

Objectives:

- Discuss and analyze the roles and responsibilities of women and men in cultivation, fieldwork and decision-making, over two generations: parents and our generation
- Compare the differences in roles and responsibilities of women and men over time and analyze what has changed
- Discuss who should participate in IPM, based on her/his role in growing cotton, not only in the field but also in decision making based on her/his knowledge about cotton cultivation

Materials:

Board, chalk, newsprint, markers, tape

Time: 30 minutes

Method:

When introducing this exercise:

- Use the information of the background section of this guide.
- Ask some questions such as: 'how long has cotton been cultivated in your village' or 'who worked in the cotton field some twenty years ago and some five years ago, women or men.'

1. Divide the participants in small groups of 5-7 persons. Ask the groups to discuss the roles and responsibilities in cotton growing of their mothers and themselves, and their fathers and themselves. Both the work in the field and decision making in cotton cultivation, e.g. who decided/decides on the variety to grow, on when to transplant and harvest, on the use of fertilizer or pesticide etc., are to be discussed. Use the following questions for the discussion:

- What work did your mother and father in growing cotton? What did they do in the field and who decided about cultivation?
- What work do you do as women and men in growing cotton? What do you do in the field and who decides about cultivation?

2. The groups will summarize their ideas on a big piece of paper. They can use the following table:

| Generations Women | Work in the field | Decision making | Generations Men | Work in the field | Decision making |
|----------------------|----------------------|--------------------|--------------------|----------------------|--------------------|
| Me-Mother | | | Bo-Father | | |
| Con- Farmer | | | Con-Farmer | | |

3. Each group will present their ideas to the whole group.

Discussions:

1. What are the major changes in cotton cultivation from the time of our parents to the present? What has changed for women, what has changed for men in their work in the field and in what they decide about cotton cultivation?
2. What have been the results of these changes for the relations between women and men?
3. When we look at the roles of women and men in growing cotton from our parents time to the present, is it important for women to participate in IPM field schools? Is it important for IPM to consider the field experience and knowledge of women?

Women's and men's roles and responsibilities in educating children in IPM

Objectives:

- Discuss and analyze the roles and responsibilities of mothers (and other female household or family members) and fathers (and other male household or family members) in educating their children in IPM
- Discuss how IPM knowledge is carried over to the future generation

Materials:

Board, newsprint, markers, tape

Time: 30 minutes

Method:

When introducing this exercise:

- Use the information of the background section of this guide.
 - Ask questions such as: "Who tells the children about growing cotton, women or men?", 'How do you tell your children about IPM...singing songs, lullabies...?'
1. Divide the participants into small groups. Each group will discuss the following questions:
 - What do you do as a mother (sister, cousin, mother) to teach the children to become cotton farmers?
 - What do you do as a father (brother, cousin, father) to teach the children to become cotton farmers?
 - How do you as a mother (sister, cousin, mother) teach the children to become cotton farmers?
 - How do you as a father (brother, cousin, father) teach the children to become cotton farmers?

2. Each group will summarize their ideas on a big piece of paper. They can use the following table:

| Mother and/or other females in the family | | Father and/or other males in the family | |
|---|---|---|--|
| What do I teach the children | How do I teach the children | What do I teach the children | How do I teach the children |
| Example: | | Example: | |
| Seed selection | They sit by me and see what I am doing and do what I do, etc. | Preparing the soil | They walk with me while I walk with the buffalo in the field, etc. |

3. Each group will present their ideas to the whole group.

Discussions:

1. How do women educate children in cotton growing? How do men educate children in cotton growing? Is it important for women to participate in IPM training? Why?
2. Will it be important for children to learn about IPM? Who will teach them?

Background notes for the trainers

Sex and gender

The unchangeable female and male characteristics are our biological characteristics with which we were born. It is what you are: a woman or a man. It is what we call sex. Female and male sex characteristics refer to our biological characteristics: women can get pregnant, men can grow a beard or mustache, etc.

The interchangeable characteristics, for example patience and eloquence, are those which we are not born with but raised with. They are socially constructed. It is what you can be as a woman or a man. It is what we call gender. Female and male gender characteristics refer to our social characteristics and our social roles as women and men. Female and male gender roles and relations vary within the same society or culture and between societies and cultures. Also, they change over time.

In most societies women and men tended and still tend to equate their female and male biological characteristics with their social roles and relations (and vice versa). For example, women cannot do the so called 'hard work', such as preparing the soil or carrying heavy loads because they have less muscular power than men. However, we observe that women farmers everywhere and in all times carried and carry out hard work normally done by men when male labour was/is not available.

This way of judging women and men normally leads to what is called stereotyping. That is to judge a woman or a man not for what s/he can be, but for what s/he is supposed to be based on her/his social roles which are derived almost solely from her/his biological characteristics. For example: a woman should not talk loudly, because she is a woman and she should behave properly; Or a man should talk loudly, because he is a man and should behave accordingly. Instead of looking at female and male characteristics from stereotype assumptions such as men are stronger than women and therefore women are weaker than men, it is more constructive to consider female and male characteristics, roles and relations by the way these can change and are inter-changeable and by the way we value these.

**Gender Division of Labour
or
The Roles and Responsibilities
of Women and Men
in Household – Agriculture – Commune**

Background

In this part, two exercises are presented which can be used as simple tools to gather data on the gender division of labour, or the work of women and men in agriculture, household and commune in general and in cotton (or other crop) growing in particular. The results of these activity profile tables allow for an analysis of gender roles and relations, and for a discussion on whether women should participate in IPM and the potentials of and the constraints on their participation.

Carrying out this exercise in field schools will give the trainer concrete data on the division of labour in various villages in the district where she works. These data can be used for discussions with village leaders when preparing for future farmer field schools. They will give information on the importance for participation of women in the field school.

Farmer groups that plan follow-up activities can carry out the exercise in combination with other planning tools in order to better decide who should participate in the activities, why and how this participation can be realized. Also, the exercise offers an opportunity to discuss the different needs women and men can have for follow-up activities and the content of those activities.

Gender division of labour in agriculture, household and commune

Objectives:

- Get information on the roles and responsibilities of women and men in agricultural, household and commune activities
- Discuss the potentials of women to participate in a farmer field school or follow-up activity
- Discuss possible constraints on the participation of women in a field school or follow-up activity and solutions for those constraints

Materials:

Board, newsprint, markers in two different colours, tape

Time:

5 - 10 minutes for the introduction

15 minutes for the table

Method:

There are two ways of presenting the exercise to the participants. The first one starts with a poem and can be used in a farmer field school as a more playful introduction to the exercise. The second one is a more straightforward introduction to the exercise.

First option to present exercise:

1. Write the poem on newsprint before the session so it can be readily presented to the participants. Ask one of the participants to read the poem.

((

Wake up!, Wake up!, 'Bo cu', it is already light
It's already Saturday. Don't you remember?
It's Saturday, isn't it?, You, 'Me no', are going to the field school, aren't you?
IPM!, IPM!, Having told by you, I feel a desire to know;
There are spider lads who spin webs to snare white butterfly ladies
The ants with the three segmented body swallow BPH still alive
However, up to now
I have thought they are from the same gang

Come on, let's move on time
I will take care of your housework
You go there and try to learn
If you are a good student, I will reward you

((

The poem was written by a farmer in Hai Hung province to "Motivate women to participate in the IPM programme".

2. Discuss what the poem is about. Allow some time for the comments of the participants. The following questions can help the participants to voice their opinions about the poem:
 - Are there female participants who have the same experience as the 'Me no' in the poem? Can they tell something about it?
 - Are there male participants who have the same experience as the 'Bo cu' in the poem? Can they tell something about it?
 - What does the last line of the poem mean?
 - What do you think about the poem?

Link the opinion of the participants about the poem with the exercise they are going to carry out. Explain that the exercise gives them an opportunity to look at the roles and responsibilities of

- women (Me no's) and men (Bo cu's) in the village: their work in agriculture, household and commune. During the exercise they will have an opportunity to discuss whether it is important for women to participate in IPM and what can be done for women to be able to participate.

Second option to present exercise:

- Prepare your own introduction:
 - Agricultural data about the role of women and men in agricultural activities in general and cotton growing in particular and IPM data on the participation of women and men in the programme.
 - Questions such as: Who works more in cotton growing, women or men? Who works more in the household, women or men? Who takes decisions about cotton growing and the household, women or men? Allow participants some time to give their opinions.
- Link your introduction and the opinions of the participants with the tables which will give the group an opportunity to analyze their work as women and men in agriculture, household and commune and to discuss who should participate in the field school or follow-up activity based on the work done.
- Divide the participants into small groups. Present the outline of the first table.

Table 1:
Gender division of labour in agriculture-household-commune

| ACTIVITIES | Women | | Men | |
|-------------|----------|--------|-------|-----|
| | 100----- | 0----- | ----- | 100 |
| AGRICULTURE | | | | |
| HOUSEHOLD | | | | |
| COMMUNE | | | | |

- Ask the participants to discuss about the labour division of men and women in agriculture, household and commune. They can still indicate some more detail for each of the categories, but not too much. For each of the category they should fill out the table. The 100-----0-----100 percentage scale indicates the work of women and men in agriculture-household-commune. Use the following rules:
 - 100=do all the work
 - 0=do no work
 - The work input of the women and men together adds up to 100%.
 - Use a red marker to shade the activities of the women and a blue marker to shade those of the men.
- Each group presents their results.

Discussions:

- Compare the tables of the groups; discuss the differences and similarities in their findings:
 - Who does more work in agriculture? in the household? in the commune?
 - What about the frequency of the work of women and of men? Is it done every day? every month? does it vary according to the season?
 - Who has to do more work, women or men? Comment on the statement men make that “women do more work but light work and that they do the hard work”. Does the woman's work stop when she is at home or does she continue until late in the evening? And can men sit and relax? Do women have time to go to the Bia Hoi or play cards or gamble with their friends? Do men have time to go to the Bia Hoi or play cards or gamble with their friends?
 - In the commune, are there differences between households in the work done by women and men? What kind of differences? (see suggestion below to elaborate on this question)
 - What should we conclude from this exercise with respect to the participation of women in farmer field schools or IPM follow-up activities?
 - Is it important for women to participate or not important for them to participate in IPM activities? Why?

Suggestion:

The table generates general data on the gender division of labour in the village. However, there are differences between families in the villages with regard to their possibilities to work in agriculture-household-commune. For instance, there are families where men are employed outside the farm for long periods of time and where women farm without adult men to support them. There are also families with a more diversified farming system and where men, for instance work in forestry activities and only marginally support the cotton cultivation which is done almost entirely by the women and their children. By adding extra space in the table for 'comments', participants can discuss the differences between families in the village and how these differences determine whether women or men should participate in IPM field school or follow-up activity.

Table 1: adjusted version to integrate information on differences in gender division of labour between families (the text shown is an example of how this table can be filled out)

| Activities | Women 100-----0-----100 | Men 0-----100 | Comments |
|---------------------------------|----------------------------|------------------|---|
| Agriculture Growing cotton | 80%////////\20% | | In this village many men are doing off-farm activities, they return to help with the harvest; Women, together with children carry out all cotton growing activities |
| Household | | | |
| Commune Participation in VWU | 100%///// | | All women in the village are members of the VWU, the Union is very strong here, the local leaders very active and supportive of female farmers to attend training and extension activities; the village administrative and political leaders value the work and opinions of the VWU members |

Gender division of labour in cotton growing

In the previous exercise participants have worked on a general picture of the division of labour between women and men in their village. In the following exercise they will elaborate in more detail the work which has to be done to grow cotton (or other crop or IPM related activity)

Objectives:

- Get information on the roles and responsibilities of women and men in cotton (or other crop) growing or other IPM related follow-up activity
- Discuss the potentials of women to participate in a farmer field school or follow-up activity
- Discuss possible constraints on the participation of women in a field school or follow-up activity and solutions for those constraints

Materials:

Board, newsprint, markers in two different colours, tape

Time:

25 minutes for the table

Method:

1. Follow the same procedure as in the previous exercise, but use the following table to summarize discussions in small groups:

Gender division of labour in growing ____ (fill out the crop or the follow-up activity)

| ACTIVITIES | Women | Men |
|------------------|----------------|----------------|
| | 100-----0----- | 0-----100----- |
| example | | |
| soil preparation | | |
| sowing seeds | | |
| etc. | | |

2. Ask the participants to discuss about labour division of men and women in cotton cultivation (or another crop), listing all the activities in cotton cultivation in detail. For each of the activities they should fill out the table. The 100-----0-----100 percentage scale indicates the work of women and men for each activity. Use the following rules:
 - 100=do all the work
 - 0=do no work
 - The work input of the women and men together adds up to 100%.
 - Use a red marker to shade the activities of the women and a blue marker to shade those of the men.
3. Each group presents their results.

Discussions:

1. Compare the results of the groups, discuss the similarities and differences in their findings.
 - Who works more in growing (fill out crop) or (fill out activity), women or men?
 - What can we conclude from this exercise with respect to the participation of women in a cotton or other crop farmer field school or in a follow-up activity?
 - Is it important for women to participate in the field school or the follow-up activity? Why?
 - Are there constraints to their participation? What are these? How can farmers and trainers help to solve those constraints? If you presented the poem in the previous exercise, what did the 'Bo cu' do to enable the 'Me no' to participate in the field school?

Suggestion:

A similar suggestion as for table 1 is given for this table.

The table generates general data on the gender division of labour in the village. However, there are differences between families in the villages with regard to their work in growing a certain crop. For instance, there are families where men are employed outside the farm for long periods of time and where women farm without adult men to support them or families where all the farm activities are entirely by the women and their children because they are widowed, or divorced or are abandoned by their husbands. By adding extra space in the table for 'comments', participants can discuss the differences between families in the village and how these differences equally determine whether women or men should participate in the field school or follow-up activity.

Table 2: adjusted version to integrate information on differences in gender division of labour between families (the text shown is an example of how this table can be filled out):

| Activity | Women | Men | Comments |
|------------------------------------|----------------|-----------|---|
| Cotton growing | 100-----0----- | 0-----100 | |
| seed selection etc. etc. | 100%//// | | |
| pesticide spraying etc. etc. | 70%////\30% | | Many women here spray pesticides because husbands work off-farm; some of the women work as pesticides sprayers for other families to earn money |

Notes:

- The tables without the extra column for comments work well with farmer groups. Immediately, they see the division of labour between women and men which guarantees a lively discussion about the roles and responsibilities of women and men and the relations between them in all life spheres: agriculture - household - commune. The more elaborate tables probably are more useful for awareness raising of IPM trainers and for giving them an opportunity to exchange their experiences and ideas about the districts where they work. These tables also can be presented in the field school or when planning a follow-up activity depending on whether or not the trainer thinks it useful for the group.
- It is important that the gender division of labour exercise is carried out for agricultural, household and commune activities and for the specific IPM activity. The results of the two tables have to be compared to be able to observe the potentials of and constraints on the participation of women in IPM. It is possible to combine the data on the specific IPM activity with the data on agricultural, household and commune activities in one table. For example, cotton growing in all its detail is shown under agriculture. The risk of integrating the specific activity into the agriculture-household-commune table is that the participants will focus solely on this activity and less on the others, which are equally important for the discussion. Nevertheless, it can be done under the condition that the trainer gives enough time for the participants to discuss and to fill out the whole table.

Background notes for the trainer

The gender division of labour tables help in classifying the daily roles and responsibilities of women and men.

Women often work continuously. They are responsible for agricultural, household and commune activities. In many cases men have more free time as they have less responsibility for the household activities.

Women and men may perform the roles in different ways. For example, men often make decisions at the village or commune level while women often do the work. On the other hand, often women manage the household economy.

Women usually do the majority of the household work and the commune work that deals with maintaining and strengthening the social cohesion of the commune. Men usually attend village or commune meetings where village or commune politics are discussed.

How does this affect planning for a farmer field school or follow-up activity?

The roles performed by women are always interconnected. When women participate in an IPM FFS, it will also affect other responsibilities. Women who attend the FFS one morning every week have less time for other agricultural and domestic responsibilities. Often this time constraint is mentioned by trainers as an obstacle for women's participation in IPM: 'Women are already busy, they do not have time to learn about IPM, that's why it is better that her husband participates'. The following quotation from the CFWS report on 'Women and IPM in Vietnam' shows that time constraint of women can be overcome if she gets practical and moral support from her family members and her husband:

"Most of the farmers who consider their attendance in the IPM training course favourably emphasized the fact that they were given support by other members of their families. For the men this support is mainly moral support. The members of the family may "jolly" ask them about the course, showing their concern with the course, etc. For women, the most important support from their families is revealed in specific attitudes and behaviour, either moral or practical, of other family members. The attitudes and the support from the husband are especially very significant for the participation of the woman in the training course."

A discussion with female trainees in Thang Binh revealed the following:

"Whether females can attend this kind of training depends on the attitudes of the husband. I am lucky, because my husband understands me and my desire for "improvement".

"Going out in the evening is also difficult for a woman if the husband does not agree. I had to find some way to explain to my husband not to be worried, otherwise he would think I am going to smile at others".

"When I go to the course, my mother and sister help me in the household work, otherwise I can not go all the time".

In contrast to the attendance of men in training courses, which is mainly the result of a personal decision, the participation of women in these courses are somewhat dependent on a collective decision of the whole family. While attending a course a woman has to spend more time on convincing other members of her family of the usefulness of her participation, encouraging them to understand her and gaining their moral and practical support.

**Gender Communication,
Facilitation and Sensitivity
Skills**

Background

In the previous exercises attention was paid to gender awareness, and division of labour between men and women. However, understanding more about roles of men and women, and the importance of women in agriculture and other activities is not enough to increase participation in Field Schools. In the following exercises we will pay attention on how to improve skills to discuss with village leaders when selecting participants for a FFS. We will also pay attention on how to make sure that women are encouraged to be active participants in a FFS. Gender communication, facilitation and sensitivity skills support the trainer when preparing a farmer field school or follow-up activity in discussions with village authorities.

Gender communication, facilitation and sensitivity skills are part of general communication skills:

- A trainer should be able to communicate and negotiate with local authorities the requirements needed for a potentially successful field school. One of those requirements is a fair chance for women to participate in a field school. A trainer may still feel not too comfortable talking about women's participation with the village authorities. This may relate to an earlier experience when the response of the authorities was negative or to a feeling of uncertainty about how to bring up the topic of women's participation anticipating a possible negative response from those authorities.
- In the field school sessions, particularly in a field school with few female participants, a trainer should facilitate the active participation of women. The trainer should create an atmosphere where women feel at ease to share their experiences, ask questions and say what they think without fear of being ridiculed by the male participants.

This session will support trainers in improving their gender communication, facilitation and sensitivity skills through role plays in which trainers perform their own professional role while being observed and analyzed by their colleagues.

The results of this session will be used together with those of the gender awareness and gender division of labour tables sessions to develop a trainer's gender action plan. This plan links with the general activity plan for future field school or follow-up activities.

Exercises in this part can be carried out in TOTs and Refresher courses for trainers.

Preparing for a Farmer Field School focusing on communicating and negotiating participation of women with village leaders

Objective:

Find out what constraints exist when discussing with village leaders the participation of women in field schools and how to solve those constraints

Materials:

Board or newsprint, markers, tape, other materials to stage the role play such as chairs, table, tea cups etc.

Time:

10 minutes for the introduction and explanation of the exercise

10 minutes for preparing the role play

15 minutes for carrying out the role play

20 minutes for analyzing the role play and discussion

10 minutes for wrap up

Method:

1. Explain that the participants will carry out a role play on preparing a farmer field school and discussing with village leaders about the participation of women in the school.
2. Ask for ten volunteers. If the group has less than 20 participants ask for eight volunteers.
3. The volunteers will go with you to another room. The rest of the participants are given points to observe in the role play (see next page).
4. Explain to the volunteers the case they have to perform:

You arrive at the village where you have a village meeting with the authorities to prepare a farmer field school. You know the area well. Many women are active in cotton growing. They sow the seeds, weed the fields, spray pesticides, fertilize the fields, harvest the field, and sell the produce. Some of the women even prepare the soil and irrigate the fields because they are the head of their household and have no male labour available to carry that out for them. You arrive at the Cooperative.

5. Request them to finish the case using the following data:
 - Among other things that need to be discussed before the start of the field school, you want to discuss the participation of women in the field school.
 - The village authorities are convinced that 'suitable field school' candidates should have 'communication skills' and at least seven years of formal education.

The group decides how many trainers and how many village leaders will be presented in the play.

6. While giving the group preparation time to prepare the role play go back to the class room and request the remaining participants to observe the role play using following guidelines:
 - What roles are performed?
 - What arguments do the trainers use?
 - What arguments do the village leaders use?
 - How do the trainers support their arguments?
 - How do the village leaders support their arguments?

Discussions:

1. Analyze the role play by going through each of the questions with the participants and note their answers on the board (key points, not too detailed)
2. After the analysis ask the participants:
 - Whether they agree with the arguments of the leaders and with how the leaders support their arguments.
 - If they do not agree with the arguments of the leaders or with how the leaders justify their arguments, what they would need to convince them that their arguments are not justified and how they would try to convince them. They now use their own experiences for the discussion.
 - List together actions that can be undertaken by trainers to improve the participation of women in the FFS. Discuss what can be done by the trainers themselves.
3. See background notes as well.

Facilitating a Farmer Field School focusing on creating a constructive atmosphere for women to participate actively

Objective:

Discuss difficulties of female participants in field schools and how to solve those difficulties

Materials:

Board or newsprint, markers, tape, other materials to stage the role play such as chairs, table, tea cups etc.

Time:

10 minutes for the introduction and explanation of the exercise

10 minutes for preparing the role play

15 minutes for carrying out the role play

20 minutes for analyzing the role play and discussion

10 minutes for wrap up

Method:

1. Explain that the participants will carry out a role play on facilitating a farmer field school where the IPM trainers want to create an atmosphere where women feel at ease to participate and do not feel inhibited to ask questions and voice opinions.
2. Ask for ten new volunteers. If the group has less than 20 participants ask for eight volunteers. The volunteers will go with you to another room. The rest of the participants are given points to observe during the role play (see next page).
3. Explain to the volunteers the case they have to perform:

You, IPM trainers, and the field school participants are now in the third week of the field school session. It is going well. The farmers are interested and enthusiastic. There are few women attending the school. They are: two married women who are supported by their husbands and families to attend the field school, one older woman who is a widow and heads a household of five children, one young single woman who lives with her parents and one woman whose husband is absent and heads a household of two children. The women are less dominant than the male participants. Only the widow speaks up quite often and tells the rest of the participants what she experiences and thinks. The two married women often talk to each other and stay together. The young woman is very silent, writes down everything in her notebook, never asks questions and absorbs everything said and explained. You noticed that the woman with the absent husband knows a lot about the cotton field but seldom shares her opinion with others. The men are dominant, they talk and comment a lot. Many of them think that they know everything and do not listen to the women's ideas. One of the men participants always makes jokes - sometimes these jokes are already annoying.

4. Request them to finish the case using the following data:
 - You want to improve the situation by getting the women to be more active in the field school.
 - You would like everyone in the group to respect and listen to each other.The group decides what field school session will be role played. Note: it should be one of the regular field school sessions.
5. While giving the group time to prepare the role play go back to the class room and request the participants to observe the role play using following guidelines:
 - What types of female and male participants attend this field school?
 - How did the trainers/facilitators deal with each type of participant?

Discussions:

1. Analyze the role play by going through each of the questions with the participants and note their answers on the board. You can use following table to organize the discussions:

| Types of female and male participants | How did the trainer deal with each type |
|---------------------------------------|---|
| | |

2. After the analysis of the role play ask the participants:
 - Are these the female and male types of participants in your field school? Do you want to add another type of female or male participant?
 - How would you have dealt with the types of participants based on your own experience?
 - What can you do to improve the participation of male and female participants.
3. You can add to the above table by giving some extra tools, if necessary, for the trainer to improve the facilitation of the field school session:

| Types of female and male participants | How did trainer deal with each type of participant | How to deal with each type of participant; extra tools |
|---------------------------------------|---|--|
| making jokes all the time (male) | ignore him others will not accept and will tell him to stop joking (trainer does not have to do much to prevent him from joking) | talk to him after the FFS explaining that sometimes jokes are nice, but that participants do not feel at ease to speak if he jokes all the time talk with the monitor of the group and ask advice on what to do |
| shy (woman) | say nice things to the person | encourage her to give her opinion; go to her and address her directly |

Notes:

- The above tools help the trainer in facilitating the regular field school sessions and in creating a favourable atmosphere for women to participate. However, it may be necessary to give some extra attention to the role of women in cotton cultivation and IPM so that they feel more confident about their participation in the field school.
- To support the female participants in the field school by showing them their value for the success of IPM, the trainer is strongly advised to carry out one of the gender awareness exercises as shown in the first session of the field guide. Women in field schools where these exercises were carried out were enthusiastic and felt recognized in their capabilities as field school participants. Also, the discussions between female and male participants in the school triggered off by the exercises were lively. Both enjoyed talking about and discussing their capabilities as IPM trainees and the importance of the participation of women for the success of the programme.

Background notes for the trainer

Tools to communicate with village leaders. For a trainer it can be important to:

- Have information to support your arguments. For example, if you want to convince leaders to involve more women in the field school, present data on the division of labour between women and men in cotton growing. If you conducted previous field schools you may have data collected by the farmers themselves through the gender division of labour tables which could support your arguments.
- Be able to negotiate your arguments. Negotiating is the process of convincing the village leaders of your arguments. Information at hand to support your arguments helps you to negotiate. Prepare the meeting together as an IPM trainers team. This will also support you in your discussion with the leaders. Anticipate what the village leaders will use as arguments and how they will justify their arguments, so you, or your team, will be better prepared to handle that. For example, if the leaders say that the men should participate in the field school because they are better educated and can therefore spread IPM information faster, explain that IPM is about training those farmers who are responsible for cotton growing and that formal education is less important than field experience. Therefore, women can be equally good participants for the FFS as men, depending on their responsibilities and experiences in cotton growing.
- Request before the meeting that a representative of the Women's Union (VWU) be present at the meeting. She will be able to support your arguments to ensure a fairer representation of women in the field school.
- Bring a female farmer who already participated in a field school to the meeting with the village leaders. She can relate her positive experiences as a field school participant to the leaders and thereby support arguments to ensure a fairer representation of women in the field school.
- Build up informal contacts with local VWU representatives besides building up informal contacts with other leaders and farmers, male and female. This can be done when you have carried out field schools in the village for more than one season and start to know the village better. Or when you already worked in the village before the start of the IPM programme.

Gender activity plan

Background

The trainers have gone through the gender awareness exercises, presented selected gender awareness exercises and the gender division of labour tables in the field school and performed the role plays on gender communication, facilitation and sensitivity. They now have (some) experience and ideas that can serve as inputs to their gender activity plan.

The gender activity plan is an integral element of the trainer's overall activity plan that is developed for the coming crop season.

Following are ideas that might support the trainer in elaborating the plan.

A gender activity plan can be made in TOTs, Refresher courses. It should be part of the provincial or district plan for IPM activities.

Ideas for elaborating the gender activity plan

Objective:

Formulate a plan and concrete actions that need to be taken to ensure participation of women in IPM activities

Materials:

Paper and markers

Time: 60 minutes

Method:

Whether or not the trainer has to do an effort to create opportunities for women to participate in IPM depends on the roles and responsibilities of women in cotton (or other crop) cultivation or other IPM related activity in the area where she works and on the actual participation of women in IPM. The following ideas can help the trainer decide whether specific actions have to be taken, that can create conditions for women to participate in IPM, and how to go about it.

1. Divide into small groups, by working area. Ask each group to discuss the following questions. Each group will summarize their ideas on a piece of big paper.

To make a gender activity plan, the following information will be needed:

- Information on the area where IPM activities will be organized:
 - * What is the situation in agriculture in your area? What is the participation of women and men in cotton (or other crop) growing, other agricultural activities and off-farm activities?
 - What has been the representation of women in IPM activities in the village or district before? Are women underrepresented, based on the amount of work they do, and decisions they make in agriculture?
 - If this information is not available, is there a possibility to collect data on participation of women and men in cotton (or other crop) growing and the participation of women in IPM activities. How?
 - How do you plan to improve the participation of women at:
 - * Village level:
 - Selection process of field school participants
 - In the farmer field school and follow-up planning
 - * District/Provincial level
 - Meetings with other trainers and sub PPD staff to discuss gender activity plans and actions
 - Discuss budget requirements for organizing activities to integrate women into IPM (FFS and follow-up)
 - What information are you going to use in the at village level, and district level? Is it available? If not how and where do you plan to get that information?
 - Who will you involve in the discussions with village leaders?
 - What other kind of information do you need?
2. After discussion each group will present their ideas, and will indicate how the gender activity plan will fit into their general work plan.

Background note for the trainer

To decide whether an effort should be made to create opportunities for women to participate in IPM activities is one element of the trainer's overall planning for IPM activities for the coming season(s). The trainer has to make an effort to obtain information and build up knowledge on the agricultural situation in the area of work. This information and knowledge help her to communicate and negotiate the participation of various categories of farmers in the field school with local authorities.

Having information and building up knowledge on the area of work not only helps the trainer decide whether (more) women should participate, but also what other categories of farmers should participate in FFS and IPM activities. These categories are determined by the socio-economic situation of farmers households or the potentials and interests of farmers to become IPM experts (richer and poorer farmers), experience in crop cultivation and being open to learn new practices (older and younger farmers) and gender or who does what and when in the farmers household (women and men). Gender is a category which runs through all other categories of farmers households. In almost all households: poorer or richer, with older or younger members and from different ethnic groups, there are women and men.

Soils

Water infiltration rates

Adapted from Richard Sikora's Soil Nutrients and Soil Health in Lowland Rice Production

Introduction

The water content of the soil is as much an important biological indicator of the soil condition as its soil respiration. Soil respiration allows microorganisms to perform their function of breaking down organic material that the plant can use for its development. Knowing more about the type of soil a farmer has in his field will help him make decisions about how he can improve it for his benefit.

Objectives:

- Observe the water holding capacity of different types of soil
- Discuss how to improve water holding capacity and its importance to overall plant health

Materials:

Soil samples from two areas mentioned in possible treatments under *Method*

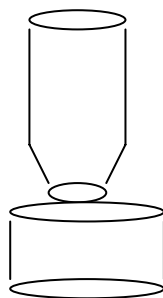
Plastic bottles approximately 1 liter size (two per group), pieces of cloth, beakers

Method:

1. Each group should set up two treatments each as follows:
 - Control (soil)
 - Soil + decomposed organic matter.
2. Take a used plastic bottle of approximately 1 liter size and cut the bottom off. Prepare two bottles per group for the treatments.
3. Put the bottle upside down and cover the bottle opening that is now at the bottom with a sheet of cloth.
4. Following the treatments, fill bottles with 1 kg air-dried soil to serve as a column. The column should now be brought into right position (see drawing) and a beaker is placed under the column to collect excessive water released by the column.
5. Finally pour slowly a predetermined volume of water (i.e. 500 ml) on the surface of the column and let the soil soak up the water.
6. Add more water until the predetermined amount of water is used up.
7. By subtracting the amount of water released by the column from the total amount of water you will get the water holding capacity.

Discussions:

1. Calculate the water holding capacity of each treatment.
2. Explain the importance of the soil's water holding capacity to the overall plant health.
3. How can the soil's water holding capacity be improved?



Existence of microorganisms

Adapted from Richard Sikora's Soil Nutrients and Soil Health in Lowland Rice Production

Introduction

Dead organic material of plant and animal origin will primarily be broken down by soil organisms, especially microorganisms. The end product of these biological processes is called humus and is an important factor for soil fertility. However, this is difficult for farmers to understand because farmers do not see the microorganisms.

Objectives:

- Visualize microbial growth
- Discuss the importance of microorganisms to soil fertility and to overall plant health

Materials:

50 g boiled rice

100 ml polyethylene bag

Steamer

Inoculum (bacterial wilt of tomato or potato)

Soil microbial solution (see process of preparation under *Method*)

Method:

1. Prepare the nutrient medium for the microbial. Each group should prepare three bags for three treatments. To do this, put 50 g boiled rice into a 100 ml polyethylene bag and seal it. Within 24 hours, for 20 minutes each sterilize the bag (nutrient medium) twice by steam-heating. After cooling down the white sterile medium is ready to use.
2. Set up three treatments by applying microorganisms from the following sources:
 - a) a drop of *Pseudomonas solanacearum* (bacterial wilt of tomato or potato). This is done by extracting inoculum from diseased plants
 - b) soil microbial solution. This is done by first mixing 50 g of field soil with 50 ml tap water. Stir the suspension and then let the soil settle for 10 minutes. Take 10 ml of the supernatant and pour on the sterile rice medium.
 - c) control. This is done by putting some soil supernatant in a bottle and heating it by steam for 20 minutes as described before. No microbial growth is expected in the control treatment.
3. To demonstrate microbial growth the plastic bags containing sterile rice are carefully opened and the inoculum is spread on the rice medium surface. The bags are closed again and let to sit in room temperature for approximately 7 days.
4. Microbial growth will become visible on the rice medium surface within 24 hours. A slimy lawn of various colors indicates bacterial growth whereas fungi appear to produce dry mycelia growing in the air best describable as a layer of fine cotton fibers.

Discussions:

1. Describe observations from the three treatments.
2. Where did the growth come from?
3. What effect would these microorganisms have on the soil? On crop development? Why?

Organic content of local soils

Adapted from Richard Sikora's Soil Nutrients and Soil Health in Lowland Rice Production

Introduction

Intensive farming practices which includes excessive chemical fertilizer and pesticide application, among others has brought with it a rapid decline in soil organic matter. The situation makes the crop susceptible to biotic and abiotic stress factors that leads farmers to continue increasing chemical inputs. Recent information on how to address this issue has called attention to the benefits that may be gained from restoring the organic matter in the soil especially when practiced with integrated pest and nutrient management. This exercise will allow farmers and trainers to gain ideas about why healthy soil means healthy crop.

Objectives:

- Observe the amount of organic matter present in the soil
- Discuss the importance of soil organic content to overall plant health

Materials:

Five areas to take samples from; identify areas where different soil amendments were used as mentioned in possible treatments under *Method*
Buckets, plastic foil, shovels, weighing scales

Method:

1. Identify areas different soil amendments were used. Possible treatments include:
 - Control (non amended soil)
 - Soil amended with straw
 - Soil amended with straw and urea
 - Soil amended with straw and compost
 - Soil amended with compost
2. Assign each group to a different area.
3. Each group should sample from the field of interest half a bucket of soil (approximately 5 kg).
4. Spread soil sample on a plastic foil and air-dry the soil in the sun.
5. Break clumps to get a homogeneous fine material.
6. Weigh 1 kg dried soil for the experiment.
7. Fill a bucket three-quarters with tap water. Stir the soil thoroughly.
8. Collect the organic matter from the water surface and air-dry it. (Organic matter is lighter than water and will accumulate on the water surface.)
9. Let the remaining soil settle to the bottom of the bucket for approximately ten minutes and carefully decant the water.
10. Air-dry the soil and take the dry weight of soil and organic matter separately.
11. Calculate percentage of organic matter based on total soil dry weight.

Discussions:

4. Which of the treatments had more organic matter content? Why do you think it was such?
5. Explain why healthy soil means healthy crop.

Nutrient uptake

Introduction

Plant nutrients are essential for plant growth. As the plant grows it uses up the nutrients in the soil. In the past this loss in nutrients was replaced by new nutrients delivered from the soil. However, in many cases intensive farming has depleted nutrients in the soil and these have had to be replaced. This can be done by inorganic fertilizers sold on the market or by organic fertilizer such as crop residues, manure, compost, etc. Whereas inorganic fertilizers are very specific and only provide one or two major nutrients, organic fertilizers cover a broad range of different nutrients. Inorganic fertilizers are mainly used when one certain nutrient needs to be applied at a high dosage, such as for nitrogen. In contrast, organic fertilizers have a more balanced nutrient content and besides providing essential macro- and micronutrients they also affect soil physical, chemical and microbial properties. Therefore, the best way to keep the soil fertile, achieve high yields and reduce fertilizer costs is a combination of adding organic matter and commercial inorganic fertilizer. Plants take up both organic and inorganic fertilizer in much the same way through a system of hose-like vessels between the roots and the top of the plant.

Objectives:

- Describe how nutrients from the soil move through the plant
- Explain the role of water in nutrient uptake from the soil

Materials:

Water, red ink or dye, two cups per group, plants and two straws

Method:

1. Go outside by group and find many kinds of plants including rice seedling, rau muong, celery, grasses and other plants.
2. Add water to the two cups and place several drops of the red food coloring. The water should be dark red.
3. Place the plants in the cups with the stems in the cups. Also place the straws in the cups. One straw should be flattened first. Place the plants in a bright place.
4. Wait 90 minutes and observe the plants. What happened to the color of the leaves? How has the red coloring moved in the plants? What does this imply about nutrient uptake from the soil?
5. How do plants take nutrients from the soil?