Discovery Learning Manual For Cabbage Pest Management

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CAB International

A compilation based on three publications of CAB International:
Crop Protection Compendium
Vegetable IPM Exercises
Understanding Natural Enemies

About this manual

Integrated Pest Management (IPM) and Integrated Crop Management (ICM) strategies are now widely recognized as key components in the move towards more sustainable and environmentally friendly approaches to crop production. In many developing countries however, these knowledge intensive strategies have not been widely adopted by farmers. To address this concern, Farmer participatory (FP) approaches to training and research are fast gaining acceptance.

The FP approach aims to build farmers' capacity to make their own crop management decisions, based on a better understanding of the agro-ecology of their own fields, and according to their own unique set of circumstances and priorities. This is in direct contrast to more conventional extension approaches where research recommendations are passed on to farmers in a top down manner. With FP approaches, the role of the extension service becomes more of a facilitator of a learning process by the farming community, and less of a messenger from the research community.

FP approaches rely heavily on non-formal education methods, and learning through doing, generally known as discovery learning. This manual provides technical information on major cabbage pests and beneficials and a range of discovery learning exercises and field experiments. It is aimed principally at National IPM programmes, IPM trainers, farmer trainers, and others interested in IPM training and farmer participatory approaches.

This manual draws heavily on three existing resources:

1. **Crop Protection Compendium, CABI**
   An interactive, multimedia knowledge base available on CD-ROM and on the Internet, containing a wide range of science-based information on all aspects of crop protection.

2. **Vegetable IPM Exercises: Protocols, Implementation and Background Information. (1998) JGM Vos, CABI Bioscience/FAO. pp 674**
   A manual of IPM exercises developed for the FAO Regional project for IPM of vegetables in Asia.

3. **Understanding Natural Enemies. Working with Natural Enemies Series, Bulletin No 1. (2001) Technical Support Group CABI Bioscience. pp 74**
   A training bulletin explaining the basic principles of biological control in a non-specialist way.

This Discovery Learning Manual aims to complement the Cabbage Integrated Pest Management: An Ecological Guide, developed by F.M. Praasterink, independent consultant to FAO.

The manual also aims to test whether a science-based electronic knowledge resource such as the Crop Protection Compendium can provide a satisfactory source for a learning manual in a farmer participatory context.

The manual consists of two major parts: Part I (sections 2, 3 and 4) provide the technical background on the biology and management of some major key pests (including diseases) and beneficials, linked to a set of farmer participatory exercises in Part II (sections 5, 6 and 7). Most exercises have been field-tested in one or several countries in Asia and Africa.

The information in either Part I or II is not intended to be comprehensive. Rather, Part I should be viewed as supporting the exercises in Part II, which are designed to facilitate an understanding of the ecological principles underpinning pest ecology and management through experimentation. By the same token, the exercises should be viewed as guidelines and sources of inspiration rather than as rigid instructions. We hope that the manual will have global relevance, as the exercises can and should be adapted to local conditions, such as available materials, prevalent pest problems, local knowledge and experience within the farming community.

A manual such as this is never truly finished. It needs to be constantly updated as practitioners around the world modify existing exercises and develop new ones. As compilers of this manual, we would like to ask for your help in keeping us abreast of new developments and informed of modifications or additional materials.

Future development

Modifications and additional materials are requested to be sent to the editors at CABI International.

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The following people/organizations are acknowledged for their photographs and illustrations:

D Agassiz (*Agrotis segetum, Hellula undalis*); ARS/USDA (*Helicoverpa zea*); Aventis CropScience (*Campoletis* sp., *Heliothis virescens, Mamestra brassicae, Pieris brassicae, Plasmodiophora brassicae, Pythium*); ID Bedford (*Bemisia tabaci, Eretmocerus* sp. on *B. tabaci*); Biologische Bundesanstalt Dossenheim (*Spodoptera littoralis*); Nigel Cattlin/Holt Studios (*Aphidius* sp., aphid parasitized by *Aphidius* sp., *Cotesia plutellae, Diadegma semiclausum, Encarsia formosana, E. formosa, Myzus persicae, Pieris rapae, Plutella xylostella, Spodoptera exigua*); A Cherry (*Diadegma insulare*); Ching-Chin Chien (*Pachyneuron muscarum*); CIMMYT (*Helicoverpa zea*); M Cock (*Aleurodicus dispersus*); Cornell University (*Crocidolomia pavonana*); CSL (*Agrotis segetum*, *Brevicoryne brassicae, Episyrphus balteatus, Liriomyza trifolii, Pieris brassicae*); AMI de Farias (*Aphelinus asychis*); Denver Natural History Museum (*Exorista bombycis*); SV Fowler (*Pardosa*); G Goergen/IITA Insect Museum (*Spodoptera littoralis*); KM Harris (*Aphidoletes aphidimyza*); S Hassan (eggs parasitized by *Trichogramma*); Y Hirose (*Thrips palmi*); M Holderness (*Peronospora parasitica*); WF Hsiao (*Crocidolomia pavonana*); ICRISAT (*Helicoverpa armigera, Spodoptera litura*); Iowa State University (*Xanthomonas campestris* pv. *campestris*); DG James (*Pieris rapae*); ABS King (*Helicoverpa armigera*); Clive Siu Ki Lau (*Phyllotreta striolata*); H Legutowska (*Thrips tabaci*); J Litsinger (*Micraspis crocea, Solenopsis geminata, Tetrastichus schoenobii*); M Mazzotti (*Trichogramma brassicae*); CM Mguni/C Nieves (*Xanthomonas campestris* pv. *campestris*); Natural History Museum, London (*Brachymeria* sp.); E Neering (*Spodoptera litura*); J Ogrodnick (*Orius* sp.); A Polaszek (*Telenomus* sp.); J Porter (*Agrotis segetum*); A Shelton (*Trichoplusia ni*); AL Snowdon (*Alternaria brassicicola*); KA Spencer (*Liriomyza sativae, L. trifolii*); JP Tewari (*Alternaria brassicicola*); University of Nebraska-Lincoln (*Agrotis ispilon*); USDA (*Coccinella septempunctata*); J Waller (*Alternaria* leafspot); Yu Yan-fen (*Thrips tabaci, T. palmi*).

Contents

Part II
Exercises for Cabbage Training of Trainers Courses and Farmer Field Schools

5. Agro-Ecosystem Analysis (AESA)

Objective: To learn more about the situation in the field and get a clear understanding of the occurrence of insect pests, diseases and natural enemies.

Materials needed:

- Cabbage field, divided into Farmers' Practice (FP) plot, IPM plot and others
- Notepaper, pen
- Colour crayons
- Large piece of paper (about 60 x 80 cm)
- Magnifying glass or hand lens

Procedure:

1. Each group samples 10 plants from the field plot. To select a plant, e.g. count the 5th plant from the edge, then to select the next plant, move 1-2 rows and then count the 5th plant from that position, etc., until 10 plants are sampled. By selecting plants according to plant and row numbers, we avoid selecting preferred plants that might not give a realistic picture of the crop as a whole. If plot sizes are very small or very long, select the plants at appropriate distances along a diagonal.

Participants preparing for the AESA of a cabbage field in India

2. Carefully observe and record any pests, defenders and diseases found on the selected plant. Start with the outer leaves and work towards the centre. Winged and wingless aphids should be counted separately (if there are too many aphids to count, use scale 0:4, where 0 = no aphids; 1 = 1-10 aphids; 2 = 10-100 aphids; 3 = 100 – 200 aphids; 4 > 200 aphids). In case of leaf spots, the number of infected leaves should be counted. Sampling should not be destructive, so do not bend the young leaves as this would cause damage.

Participants take notes for the AESA in Vietnam

3. Also observe the soil surface for any ground-dwelling pests or defenders.

4. Make records of the crop stage, weather condition, soil condition, water availability, cultural practices, and presence of weeds.

Participants collect samples for the AESA in the Philippines

5. After observing the 10 sample plants, uproot one normal (average) plant and one plant with obvious disease symptoms (wilting, growth disorder) (*only if the farmer allows uprooting!!!*). To uproot, use a shovel to make sure that the root system is not damaged. Carefully remove most of the soil from the roots to allow for drawing.

6. Cut the main root of the two uprooted plants open over the total length, so that the inside of the root is visible. Observe differences in colour.

Drawing:

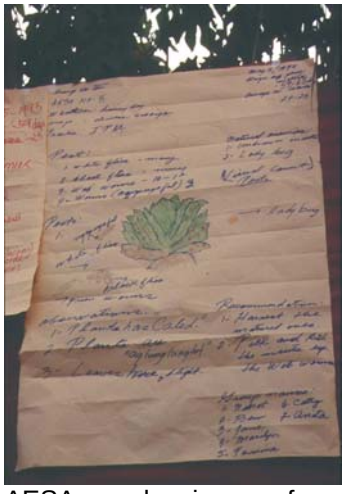
Find a place to sit as a group and make colour drawings on the large piece of paper. Draw the plants with injury symptoms (disease symptoms, holes due to insect feeding). On the left-hand side of the plant, draw the pest insects (which might be collected in small plastic bags or vials) and disease symptoms found (use hand lens!):

Small sucking pests, mites and leaf spots:
- indicate the total number found on all leaves, and the total number of leaves checked (for example, 15 jassids per 30 leaves)
- calculate the average per leaf (for example, 0.5 jassids per leaf).

Other pests and diseases:
- indicate the total or average number of pest insects found on all sample plants
- indicate the total or average number of leaves infected with disease on all plants and the percentage of plants with wilting symptoms
- if a wilted plant, that was uprooted, shows root damage, draw the damaged root. Carefully indicate root deformation, colour differences, etc.
- draw the disease symptoms

On the right-hand side of the plant, draw the beneficials (defenders) found. Again indicate the total numbers found (and calculate the average per plant). If weeds are present, draw some different weed species next to the plant.


Participants discussing the AESA of tomato in Trinidad


AESA drawing from cabbage during training in the Philippines

Discussion:

? How would you describe the general condition of the plant?
? What do you think are the most important factors affecting your crop at this stage?
? What, if any, measure should be taken?

Presentation:

When all groups have finalized their drawings and answered the questions, the groups present their work in front of the other groups. They explain the sampling, explain the drawings and discuss the answers to the three questions.

One group for each treatment (IPM vs. FP) presents its results. Each week, a different person from each group should do the presentation. In Farmer Field Schools, the Ecosystem Analysis drawings of the previous weeks should be available for comparison and to discuss development of the crop and insect populations. It is easily forgotten what the field looked like earlier in the season, what insect populations were found, and when control measures were taken. After the group presentations, a facilitator should summarize the answers to the three questions and decide with the groups which measures are to be taken in the different treatments.

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6. Insect Management Exercises

General background

Insects belong to the arthropods, which vary in size from tiny thrips that can hardly be seen with the naked eye, to quite large, such as the preying mantid. Studying their movements, feeding habits and reproduction patterns gives us information regarding the biology and ecology. An advantage of working with arthropods is that they usually can be studied using a hand lens only, unlike pathogens. Using arthropods, the concept of biological control can easily be demonstrated in a so-called insect zoo. The beneficial arthropods are the so-called predators (= insects or spiders that feed on other insects, particularly pests) or the so-called parasitoids (= insects that lay eggs in or on its host so that the host provides food for the young stages of the parasitoid). Predators are usually more easy to study, because they are usually larger than parasitoids. Another concept that can be studied using arthropods is the effect of pesticides (biological as well as chemical) on pests and beneficials. The biological pesticide B.t. (Bacillus thuringiensis) is rather easily available in various countries and therefore used in quite a number of exercises.

The following exercises are included:

Exercise	Page	Name	Time required
6.18	155	Effect of parasitisation on feeding behaviour of diamondback moth	½ day + ≤ 1 wk mon.
6.19	157	Rainfall as mortality factor	1 day + 4–8 wks mon.
6.20	159	Hand picking of eggs and caterpillars	Season-long, weekly mon.
6.21	161	Integrated management of web worm on cabbage	Season-long, weekly mon.
6.22	163	Use of screen in the nursery	1 day + 4–8 wks mon.
6.23	165	Mixed cropping example: cabbage with mustard	Season-long, weekly mon.
6.24	167	Comparison of biological and chemical pesticides used in caterpillar control	½ day + ≤ 1 wk mon.
6.25	169	Assessment of viability of B.t.	½ day + ≤ 1 wk mon.
6.26	171	Inhibition of larval feeding by B.t.	½ day + ≤ 1 wk mon.
6.27	173	Sensitivity of B.t. to sunlight	½ day + ≤ 1 wk mon.
6.28	175	Life cycle and biology of the parasitoid *Diadegma semiclausum*	½ - 1 day + 2-3 wks mon.
6.29	177	Life cycle and biology of the parasitoid *Cotesia plutellae*	½ - 1 day + 2-3 wks mon.
6.30	179	Life cycle and biology of the parasitoid *Diadromus collaris*	½ - 1 day + 2-3 wks mon.
6.31	181	Life cycle and biology of the parasitoid *Cotesia glomerata*	½ - 1 day + 2-3 wks mon.
6.32	183	Preference of host stages by *Diadegma semiclausum* (or *Cotesia plutellae*) (1)	½ - 1 day + 2-3 wks mon.
6.33	185	Preference of host stages by *Diadegma semiclausum* (or *Cotesia plutellae*) (2)	½ - 1 day + 2-3 wks mon.
6.34	187	Competition between *Diadegma semiclausum* and *Cotesia plutellae*	½ - 1 day + 2-3 wks mon.
6.35	189	Predation on sucking insects in insect zoo	½ day + ≤ 1 wk mon.
6.36	191	Cage exclusion of natural enemies in the field	½ day + ≤ 1 wk mon.
6.37	193	Screen caging in nursery	1 day + 4–8 wks mon.
6.38	195	Thrips feeding symptom development	1 day + 4–8 wks mon.
6.39	197	Rainfall as thrips' mortality factor	1 day + 4–8 wks mon.
6.40	199	Use of light reflective mulch to manage thrips and/or aphids	Season-long, weekly mon.
6.41	201	Parasitism of white fly	1-2 days
6.42	203	Parasitism of leaf miners	½ - 1 day + 2-3 wks mon.
6.43	205	Effect of pesticides on spiders and other natural enemies	½ - 1 day + 2-3 wks mon.
6.44	207	Role play on insecticide resistance	½ day or less
6.45	211	How to collect and recognise insect pathogen groups	½ day or less
6.46	215	How to isolate and culture viruses and bacteria	½ - 1 day + 2-3 wks mon.
6.47	217	How to test the infectivity of nematodes to target insects	½ day + ≤ 1 wk mon.

mon. = monitoring

6.1 Insect zoo

Some arthropods are pests, feeding on plant parts, others feed on insect prey, others live inside other arthropods and again others come from weeds or neighbouring crops, and are simply resting in the cabbage crop. To learn about the biology of arthropods, the insect zoo and variations on the insect zoo can be conducted. This exercise can be used for most pests, and especially for caterpillars.

Objective: To study arthropods, their feeding and life cycles

Materials needed:

- Small plastic vials and bags
- Tissue paper
- Camel- or fine hair brush
- Labels
- Hand lens
- Insect collection box
- Pins

Procedure:

To find out whether an arthropod is a pest, collect it in a vial, give it some food (leaves, stems and/or fruits of the studied crop). Close the tube and 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. Observe whether the insect feeds and on what it feeds; check again after some time.

To find out whether an arthropod is a predator, collect it in a vial, give it some prey (aphids, eggs or small larvae). Close the tube and 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. Observe whether the insect feeds and on what it feeds; check again after some time.

To find out about the developmental stages of arthropods, collect eggs, larvae/nymphs or pupae encountered in the field and rear them in vials through the next stages until the adult stage. Feed the larval stage on appropriate food (leaves, fruits, insect prey in case of predators) every day, and observe the arthropods during development. It is important to always place a piece of tissue paper between the vial and the lid to avoid condensation inside the tube.


Insect zoo made from plastic lemonade bottles

Another way to build ‘insect zoos’ is to transfer plants to pots, remove all arthropods on the plants and cover the plants with large transparent plastic covers (make a few windows with fine screen to avoid condensation). Insert the arthropods

that you want to study and daily observe the zoo.

It is a good idea to build up a reference collection of pests and natural enemies during a field school season. To make a reference collection, pierce the dead insects on insect pins or fine tailor pins (pierce the pin through the thorax, the middle part of the body) and add a small paper label to the pin with details of the collection date, place and crop.

Observations:

Record the local name of the arthropod that was collected and the location where it was collected and describe your observations on poster paper. Explain in presentation sessions which arthropod(s) you collected, where you collected them, what they were feeding on, whether they changed development stages and how long they remained in certain development stages. Illustrate the observations with drawings of each developmental stage of the studied arthropod.

Discussion:

- Did you learn more about the arthropod you studied in the insect zoo?
- Was the studied arthropod a ‘friend of the farmer’ or an ‘enemy of the farmer’?
- How could the information about duration of development stages help you in the management of arthropod pests?

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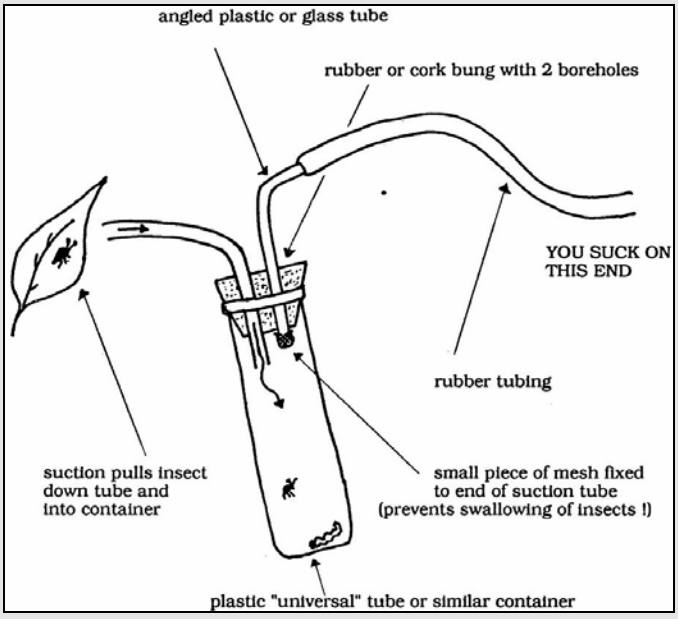
6.2 Collecting, preserving and culturing insect natural enemies

These are basic tips for people who have not worked with insects before. The information here is simply to allow you to keep insects alive for a few days after capture. The collecting method you choose will depend on the lifestyle of the natural enemy you are interested in, and whether you want to collect live specimens or not. Observing crop plants and pests will reveal many natural enemies, especially if you sample at different times of day and throughout a growing season. The most useful piece of equipment is a simple hand-held aspirator, designed specially for sucking up insects from leaves or the ground. It is small enough to carry in your bag or pocket. Most entomologists use home-made aspirators because they are not commercially available. Notes on how to make an aspirator are given below.

Materials needed:

- sweep nets for flying insects or foliage-dwelling species
- beating tray and stick
- plastic cups for pitfall traps
- aspirator for sucking up insects from leaves/ground (see diagram below)
- small specimen tubes
- filter paper
- cotton wool
- honey solution
- alcohol
- hand lens
- entomological or other fine pins

How to make a simple aspirator



Procedure:

Step 1. Collecting insects

Sweep nets

These are useful for sturdy predators, e.g. bugs and beetles active on foliage, but not so good for delicate flying insects because it is difficult not to damage them.

- Walk through the vegetation swinging the net from side to side with a regular motion.
- Use a small tube or jar to capture the insects once they are inside the net.

Wash or replace the net frequently if you collect in vegetation that has been heavily sprayed with pesticide.

Beating trays

These are useful for collecting many plant-dwelling species hiding amongst leaves etc. It is often easier to work in pairs with a beating tray, especially when collecting fast-moving species.

- Hold the tray under the plants or tree to be sampled, or simply spread an old white sheet on the ground underneath.
- Shake the plant or use a stick to beat the vegetation vigorously so that the insects fall onto the white sheet where you can spot them easily—*make sure your first attack is strong because some insects will cling tightly to foliage once they are slightly disturbed!*
- Collect the insects with your aspirator—*take care with aspirators not to inhale dust and do not use them with insects that secrete unpleasant chemicals.*

Pitfall traps

These are good for collecting predators that spend a lot of time on the ground, e.g. beetles and spiders. You will need to inspect your traps every day if you want to collect live specimens. If you leave a trap for several days you are more likely to find a rotting mess of half-eaten insects!

- Dig a hole and sink a smooth-sided plastic cup into the ground. Make sure that the rim of the cup is smoothly flush, or level, with the soil surface—*if the cup is sticking up above the surface the insects won't fall in!*
- Leave overnight.
- Inspect the next day and remove all captive insects.

- *If you do not need live specimens you can put some preservative (e.g. dilute formalin or ethylene glycol) into the water to prevent captured predators from eating each other.*

Rain is a problem with pitfall traps. You can experiment with simple roof designs, using plastic or a piece of flexible tin.

Hand collecting

You can collect many predators active on plants by carefully examining plants. This is often effective for nocturnal species such as syrphid and chrysopid larvae, which may be hiding in leaf folds or buds.

It may be easier to remove and bag samples of foliage to examine later back at base. This will also help you to quantify sampling.

Aerial traps

These can be used to sample some kinds of predatory flies, wasps and beetles. Aerial traps may be non-attractive (e.g. standing nets like the Malaise trap) or can be made attractive by a visual stimulus, such as a bright yellow or white board, or a light source.

You cannot use aerial traps to measure the density of a natural enemy, unless you know the range over which it attracts them.

Step 2. Preserving insects and other arthropods

You can preserve any arthropod in 80% alcohol for identification, but some groups are better preserved dry. The best way to kill arthropods for preservation is to drop them into alcohol (you can pull them out, once dead, if you want them to dry out).

Caution!

Do not use the old-fashioned 'killing jars' often mentioned in insect books.

The substances used in these jars (cyanide, carbon tetrachloride, ethyl acetate or chloroform) are highly dangerous to you too.

Large predatory bugs, flies, lacewings, wasps, earwigs

Use this method if the insect is 5 mm long, or more.

- Pin specimen through the thorax and dry.
- Store in sturdy insect boxes with a piece of mothball (napthalene) to prevent attack by stored product pests.

Large beetles

- Pin the specimen through one of its wing cases and dry—*do not pin through the thorax because this may be used for identification.*
- Store as above.

Small bugs, ants, spiders and mites

- Preserve directly in 80% alcohol.

Small beetles and larger parasitoids

- Put a very small spot of glue on the tip of a triangular point of card or stiff paper and attach the insect to this.
- Store the mounted specimens in collection boxes by pinning through the other end of the card point.

Very small parasitoids

- Preserve *half* the specimens in 80% alcohol.
- Preserve the remainder as dry specimens in tubes with sufficient crushed tissue paper to prevent them from moving.

Step 3. Keeping arthropod natural enemies alive

Food

It is best to provide natural enemies with their usual food source but this is not always possible or convenient. Adult parasitoids can usually be kept alive for several days with a honey solution, which is similar to the nectar they would feed on in the wild. Predators will often survive on a wide range of prey (dead or alive), processed meaty foods or honey solution.

- For a rapid daily food supply for parasitoids, make a 20% (by volume) solution of honey in water. Soak small pieces of cotton wool in the solution, and cover the cotton wool with gauze to prevent the adults from sticking to the honey.
- For a longer-lasting food source, make a mixture of honey-sugar-agar-water at a ratio of approximately 10:20:1:100. Heat these ingredients and then pipette drops onto cardboard strips—in the high relative humidity of the rearing cages the food absorbs sufficient water to become palatable to the insects.
- Increase the proportion of agar to make solid honey-agar, which you can cut into cubes. These are very acceptable to many predators.
- For maintaining predators, also experiment with the following: fish paste, chopped earthworms or maggots, moistened commercial pet food biscuits and housefly pupae.

Cages

- Temporary cages are easy to improvise using jars, plastic containers, etc.
- Provide ventilation by either piercing small holes in a plastic container or using a lid with a hole tightly covered with fine gauze net. Do not cover jars with gauze alone and an elastic band, as they are not insect proof.
- You may need to keep the humidity high to prevent the insects drying out. Place a piece of moist filter paper or cotton wool in the container, but do not allow the bottom to get too wet or you may damage or drown the occupants. If the atmosphere inside becomes too humid, you risk fungal contamination.

Handy Hints

☛ When collecting any specimens, make sure they have been fully labelled with date and location of collection, name of collector, crop or plant where collected and any associated hosts or prey; for example, "feeding on *Spodoptera* larva". Include all these details when sending specimens for identification.

☛ Always pack specimens for posting very well, using tissue paper to stop them from shifting position during transport. Tissue paper is better than cotton wool because legs, etc. can get tangled in cotton wool. It is a good idea to include a mothball against insect attack.

☛ When mounting specimens, try to use water-soluble glue so that a taxonomist can re-mount them later, if necessary.

☛ Take care when using honey solution in hot climates as it soon ferments or goes mouldy. You should supply fresh honey each day in the tropics.

☛ Never smear undiluted honey on cage surfaces as a food source because parasitoids and other delicate insects tend to get stuck to it.

☛ Never leave temporary cages or specimen tubes containing live insects in the sun or in a hot, unventilated vehicle. Some form of cool box is useful for transporting field-collected specimens back to base in tropical climates.

☛ Avoid glass containers for fieldwork—they frequently get broken.

If staff regularly have to spend many hours collecting insects with an aspirator, particularly in lab cultures, it may be better to use a small electric pump-operated version. Some organizations insist on this for safety reasons.

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6.3 Sampling for arthropods with light trap

As much information as possible on the abundance of pests and their natural enemies in the field is considered desirable for making a well-informed and good decision in pest management. Different trapping methods have different specific advantages, but each gives only a partial picture. For example, light trap will generally catch only flying adults (night-active moths, aphids, whiteflies). Together with other methods, the catches can provide a fuller picture of the agro-ecosystem.

Objective: To learn how to use a light trap and discover which arthropods are trapped using a light trap

Materials needed:

- Light trap (to be constructed as described below)
- Plastic bags (to collect the catches)
- Light-coloured (preferably white) plastic trays (for separating and sorting the catches)
- Camel- or fine hair brushes
- Forceps
- Pin mounted on a pencil-like wood (as handle) for separating the specimen
- Vials, containers (for keeping specimen, if needed for future reference)
- 1 pencil and paper for labels
- Detergent

Procedure:

The light trap can be cheaply constructed using locally available materials: using a simple kerosene lamp above a water basin supported by a tripod made with bamboo sticks or branches collected in the field. An illustration of a more sophisticated set-up is given on the following page. Prepare and set up the light trap during the day in the selected site (or a site in the vicinity). The lamp should be at a height above the crop level. Ensure that some detergent is added to the water in the basin.

Light the kerosene lamp only in the evening (at dusk) when the sun sets. On the next morning, pour the catches from the basin into plastic bag(s) together with some water. All excess water should be discarded. Back in the classroom, pour the catches into the trays. Separate and sort the catches into the various arthropod/insect groups (e.g., wasps, moths, leafhoppers, flies, etc.). Count the numbers caught for each group and tabulate the results for discussion.

Note: *For purposes of comparison or to complement the catch information by other means (sampling methods), the light trap may also be set up at about the same time as other trapping devices, such as sticky board, water pan trap, pitfall trap*.

Discussion:

- What does the catch consist of mostly (larva/nymph or adult)?
- Which are the main groups of insects/arthropod caught?
- Which group is most and which is least prevalent? And what is the ranking (in abundance) of the others?
- Since all these are caught using the light trap, what can you conclude?
- Can you relate any of these with the crops in the area where the trap is set up?
- What particular groups (stage and types) are not caught? And what can you conclude from this?
- In what way is the light trap useful and what are its limitations?
- If other traps are also set up (or other assessment methods done), how do the catches of the light trap compare with them? What can you conclude?

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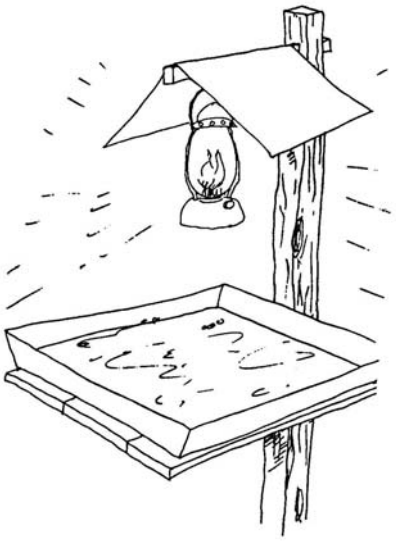

Example of a locally made insect light trap
in Indonesia

6.4 Sampling for arthropods with sticky board

As much information as possible on the abundance of pests and their natural enemies in the field is considered desirable for making a well-informed and good decision in pest management. Different trapping methods have different specific advantages, but each gives only a partial picture. For example, yellow sticky board will generally catch only flying adults (wasps, flies, night-active moths, aphids, whiteflies, flea beetles, etc.). Together with other methods, the catches can provide a fuller picture of the agro-ecosystem.

Objective: To learn how to use sticky boards and discover which arthropods are trapped using sticky boards

Materials needed:

- Sticky board (preferably yellow). Ready made commercial ones may be used or others which can be prepared using appropriate glue and board/tin plate of yellow or white colour.
- Plastic bags (to collect the catches)
- Camel- or fine hair brushes
- Forceps
- Pin mounted on a pencil-like wooden handle (for separating the specimens)
- Vials, containers (to keep specimen if needed for future reference)
- 1 pencil and paper for labels
- 1 marker pen
- Pieces of bamboo or wooden sticks (to hold up traps in the field)

Procedure:

The sticky board can be hung from or tied onto a wooden stick(s) or bamboo which is firmly poked into the ground where the trap is being set up (either in the field or in the vicinity). Keep the board in the vertical position and a little above the crop. It is best to set up the sticky trap in the morning and to collect it later in the day before dark. During collection, each board can be slipped into a clear plastic bag and labelled before taking it to the lab/classroom for checking/counting of the catch.

To facilitate counting, grid lines may be drawn with marker pen over the plastic bag (without removing the sticky board). Counts from each square are taken and subsequently pooled together for each group of arthropod (leafhoppers, flies, wasps, etc.) caught. They are then tabulated and the results analysed and discussed.

Note: *For purposes of comparison or to complement the catch information by other means (sampling methods), the sticky board may also be set up at about the same time as the others, such as water pan trap, pitfall trap, light trap*.

Discussion:

- What does the catch consist of mostly (larva/nymph or adult)?
- What are the main groups of insects/arthropod caught?
- Which group is most and which is least prevalent? And what is the ranking (in abundance) of the others?
- Since all these are caught using the sticky board, what can you conclude?
- Can you relate any of these with the crops in the area where the trap is set up?
- What particular groups (stage and types) are not caught? And what can you conclude from this?
- In what way is the sticky board useful and what are its limitations?
- Why should we regularly inspect the traps?
- If other traps are also set up (or other assessment methods done), how do the catches of the sticky board compare with them? What can you conclude?
- Can you design other sticky traps based on what you have learnt?

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6.5 Sampling for arthropods with water pan trap

As much information as possible on the abundance of pests and their natural enemies in the field is considered desirable for making a well-informed and good decision in pest management. Different trapping methods have different specific advantages, but each gives only a partial picture. For example, the water pan trap will generally catch only flying adults. A yellow trap will be suitable for catching aphids. Together with other methods, the catches can provide a fuller picture of the agro-ecosystem.

Objective: To learn how to use a water pan trap and discover which arthropods are trapped using the water pan trap

Materials needed:

- Light colour (yellow or white) shallow dish (eg. those used for serving food)
- Detergent
- Light-coloured (preferably white) plastic trays (for separating and sorting the catch)
- Plastic bags (to collect the catches)
- Camel- or fine hair brushes
- Forceps
- Pin mounted on a pencil-like wooden handle (for separating the specimen)
- Vials, containers (to keep specimen if needed for future reference)
- 1 pencil and paper for labels
- Short bamboo or wooden sticks (for making a simple tripod to support the dish)

Procedure:

Cross 3 pieces of short sticks or bamboo in such a way that they form a tripod on the ground to support the water pan trap. Then place a dish on the tripod. Fill the dish halfway with water and add some detergent.

The water pan trap should be set up in the open area with no vegetation hanging over. It may be located between crop rows or in the vicinity just outside the field, depending on the study objective.

The trap may be set up in the morning and collected later in the day before dark. During collection, pour the catch (including the water) into a plastic bag. Label the plastic bag before taking it to the lab/classroom for checking/counting of the catch. For sorting the catch, pour the catch into the tray. Remove excess water. Separate the catch into the various arthropod/insect groups (e.g., wasps, moths, leafhoppers, flies, etc.). Count the numbers caught for each group and tabulate the results for discussion.

Note: *For purposes of comparison or to complement the catch information by other means (sampling methods), the water pan trap may also be set up at about the same time as the others, such as sticky board, pitfall trap, light trap*.

Discussion:

- What does the catch consist of mostly (larva/nymph or adult)?
- What are the main groups of insects/arthropod caught?
- Which group is most and which is least prevalent? And what is the ranking (in abundance) of the others?
- Since all these are caught using the water pan trap, what can you conclude?
- Can you relate any of these with the crops in the area where the trap is set up?
- What particular groups (stage and types) are not caught? And what can you conclude from this?
- In what way is the water pan trap useful and what are its limitations?
- If other traps are also set up (or other assessment methods done), how do the catches of the water pan trap compare with them? What can you conclude?

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6.6 Studying predators in the field

Predators ('friends of the farmer' who are feeding on pests) are not so easy to observe in cabbages. However, they are present and often play a very important role in keeping pest species under control. In this exercise, two methods of collecting predators are used (hand collecting and pitfall traps).

Objective: To learn methods to collect and observe predators

Materials needed:

- Clear plastic containers (e.g. empty film containers) and bags
- 1 10x magnifying glass or hand lens
- Camel- or fine hair brush
- Labels
- Tissue paper

Procedure:

Go to the field and search for predators. Collect them in the clear plastic containers without touching the insects (use brush). Any unfamiliar arthropod on the cabbage plants should be collected and brought back to the classroom for study. For each insect to be studied, place it inside a plastic bottle together with parts of the plant and some known insect pests. Observe for 3 days and record whether the test insect feeds on plant or other insects. Experience will enable trainers and farmers to help other farmers understand which insect is a pest and which is a predator.

Besides using plastic bottles and hair brush to collect predators, trainers and farmers may use the pitfall traps to determine what predators are active at the soil level and to provide an indication of the number of ground dwelling predators present. A pitfall trap is a cup with straight sides, of about 12 cm high and 6 cm wide (diameter). It is buried up to the brim in the soil, usually between plants in a plant row. Live predators can be collected if no water is placed inside the cup. However, if numbers of predators are to be assessed, place water mixed with some liquid detergent to collect all insects that fall into the cup. Check the cups in the morning after leaving them overnight. Predators caught in pitfall traps will help complement the visual counts of predators during field sampling.

Discussion:

? What kind of predators are present in cabbage fields?
? Which predator is most common? Which insects are eaten by the predator?
? What is caught in pitfall traps? Are there more predators in pitfall traps than in visual counts?
? Are all insects found in pitfall traps predators? If not, what are their functions?
? Did you find new predators? Which stages of insect pests did it feed on?

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6.7 Soil-dwelling predators

Soil-dwelling predators may occur in unsprayed cabbage fields, but not in sprayed fields. The following exercise may illustrate this.

Objective: To discover the effect of pesticide spraying on soil-dwelling predators

Materials needed:

- Unsprayed or IPM cabbage field
- Sprayed cabbage field
- Quadrants of e.g. 50 x 50 cm
- Pitfall traps

Procedure:

Choose a 10x10 m plot inside an unsprayed (IPM) cabbage field and one plot inside a sprayed (Farmers Practice) field. Weekly count numbers of predators (crickets, red fire ants, black predator ants, spiders, carabid beetles, staphylinid rove beetles). Observations are best conducted very early morning or at sunset, using zinc quadrants (e.g. 50 x 50 cm), 10 samples per plot.

Pitfall trap made from plastic bottle and protected from rain by cover

Pitfall trap made from plastic container

In addition, pitfall traps could be installed in each plot. A pitfall trap is a cup with straight sides, of about 12 cm high and 6 cm wide (diameter). It is buried up to the brim in the soil, usually between plants in a plant row. Live predators can be collected if no water is placed inside the cup. However, if numbers of predators are to be assessed, place water mixed with some liquid detergent to collect all insects that fall into the cup. Check the cups in the morning after leaving them overnight. Predators caught in pitfall traps will help complement the visual counts of predators during field sampling.

Discussion:

- Which predators were found?
- What are the local names of the predators?
- What would they feed on and how many prey could they consume in 24 hours (you could check this in an insect zoo!)?
- Were all the different predators present in both fields? If no, why not?
- Were there as many of the different predators in the sprayed as in the unsprayed field?
- What does this mean for the pests in the unsprayed and in the sprayed fields?

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6.8 Direct observations of consumption rates of predators in the field

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: Study predators in the field and learn about their feeding habits

Materials needed:

- Hand lens
- Watch
- Whistle

Procedure:

Early morning at 7 a.m., the trainees are briefed, and are divided into two groups that will each observe a particular predator species (see section on Recognising Important Predator Groups p.89) in an unsprayed field:

A Syrphid larva (large instar)
B Chrysopid larvae (large instar)
C Coccinellid larva (large instar)

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 minute observation, and everyone follows his predator and observes the prey species, number and sizes of the prey, it eats within a 10 minute interval. The predators should not be disturbed and should not be given prey, because we want to observe the natural feeding behaviour.

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.

The same activity is repeated at 9.30 a.m. and (if possible) at 6.30 p.m., in order to compare the activity of predators at different times of the day.

After each observation, results are compiled and discussed in the "field class".

Discussion:

- How many can each predator eat?
- What is the preferred prey of each predator species?
- Are there differences in feeding rates at different times of the day?
- Which predator is the most active searcher?
- When the pests are less common, would the predators eat the same numbers of prey or less?

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6.9 Micro habitat distribution of pests and natural enemies within the plant

So far we have recorded pests and natural enemies, or defenders, by sampling the whole plant (or by sampling leaves only for small sucking pests). We made no distinction between different parts of the plant; whether insects occur on the fruits, leaves, in the top or at the bottom of the plant. To increase our understanding of the ecology of pests and interactions with the plant and with the defenders, we will look at more detail into their position within the plant. If defenders are found on the same plant parts as pests, there is more chance they will meet and consume the pests.

Objectives:

1. To understand on what parts of the plant pest stages are mostly found;
2. To understand whether predator species are found on the same plant parts as pests.

Materials needed:

- One blank data sheet for each subgroup
- Charts with drawing pens

Procedure:

Divide each group into 2 or 3 subgroups that can separately make their observations. Assign one species (pest or defender) from the following list to each subgroup (only this species has to be recorded, all other insects can be ignored). Select those species that are available in the field.

Number of plants to be sampled: 10 (larger species) or 5 (small species). Some pests or defenders are small but quite common, sampling 5 plants will be sufficient in these cases. Examples of the number of plants sampled for each species is indicated below.

Walk through the field and select plants that are relatively erected. Measure the plant and divide the plant into three equal parts, top, middle and bottom. The best way to sample may be to start at the growing top, then observe the stem, then all the fruits and finally the 5 leaves from that section of the plant (top, middle or bottom). For small species, leaves should be picked so that you can observe more closely. Because it is very tedious to sample all leaves of the plant, it suffices to sample 5 leaves from the top, 5 leaves from the middle and 5 leaves from the bottom of the plant.

To analyse the results, calculate the total and the average number on each plant part. Each group should prepare a chart for presentation.

Species	*Number of plants to be sampled*
Aphids	5
Thrips	5
Whitefly adults	5
Whitefly nymphs	5
Flea beetle adults	5
Diamondback moth eggs	10
Diamondback moth larvae	10
Webworm eggs	10
Webworm larvae	10
Cutworm eggs	10
Cutworm larvae	10
Armyworm eggs	10
Armyworm larvae	10
Cabbage looper eggs	10
Cabbage looper larvae	10
Coccinellid larvae and adults	10
Chrysopa eggs	10
Chrysopa larvae	10
Ants	10
Spiders	10
Predatory bugs (anthocorids, *Geocoris*)	10

Discussion:

- What is the distribution of each pest within the plant?
- Why are pests found on specific plant parts?
- Are predators found on the same or on different parts of the plant as the pests? What could this mean?

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6.10 Importance of flowers as food source to adult parasitoids (1)

Many adult parasitoids require food in the form of sugary solutions for their survival. Given such foods, parasitoids may greatly increase their life span. In this exercise, parasitoids of the important cabbage pest diamondback moth are used.

Objective: To demonstrate the effect of provision of honey solution on the life span of parasitoids

Materials needed:

- Clear plastic cages and potted cabbage plants
- 1 Magnifying glass (10x)
- Camel- or fine hair brush
- 1 pencil and paper for labels
- Honey solution
- Pieces of clear plastic sheet

Procedure:

Set up 4 or 6 cages, each enclosing a potted young cabbage plant. Provide diluted honey in fine droplets on plastic sheet in half of the cages, but not in the others. Label the cages accordingly. Introduce 10 pairs of newly emerged adult parasitoids (*Cotesia plutellae*, *Diadegma semiclausum* or *Diadromus collaris*) into each cage. Carefully observe and record the number of living and dead parasitoids everyday until all have died.

Discussion:

? Do you see parasitoids feeding on the honey droplets?
? How many days after do the parasitoids begin to die? And in which of the cages?
? Do more parasitoids die over time?
? Are the numbers of dead parasitoids the same in all the cages?
? Which type of cages have more dead parasitoids at any one time?
? What can you conclude from the results?

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6.11 Importance of flowers as food source to adult parasitoids (2)

Many adult parasitoids require food in the form of sugary solutions for their survival. Given such foods, parasitoids may increase greatly their life span. In nature, the parasitoids usually visit flowers to feed on the honey or pollen. In this exercise, parasitoids of the important cabbage pest diamondback moth are used.

Objective: To demonstrate the benefits of flowers as a food source to adult parasitoids

Materials needed:

- ✔ Clear plastic cages with screened windows to provide for good ventilation
- ✔ Potted flowering plants (species which provide a good source of honey)
- ✔ Plastic/glass vials
- ✔ Camel- or fine hair brush
- ✔ 1 pencil and paper for labels

Procedure:

Set up 4 or 6 cages, half of which enclose a potted flowering plant each. In the other half, there are no flowering plants enclosed. Label the cages accordingly. Introduce into each cage 10 pairs of newly emerged adult parasitoids (*Cotesia plutellae*, *Diadegma semiclausum* or *Diadromus collaris*). Carefully observe and record the number of living and dead parasitoids every day until all have died.

Note: This study should be repeated using different flowering plants (including weeds and cultivated crops) each time.

Discussion:

? Do you see parasitoids visiting the flowers?
? After how many days do the parasitoids begin to die? And in which of the cages?
? Do more parasitoids die over time?
? Are the numbers of dead parasitoids the same in all the cages?
? Which type of cages have more dead parasitoids at any one time?
? From the results, what can you conclude with regard to the role of flowering plants?
? What would you advise with regard to flowering plants often found in the border areas of cultivated crops?

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6.12 Life cycle of caterpillar pests

Objective: To understand the life cycle and development stages of an insect with complete metamorphosis.

Materials needed:

- Clear plastic bottles, vials or bags
- Cages (covered with screen) and potted cabbages
- Unsprayed cabbage field
- Sweep net to collect moths
- 1 Magnifying glass (10x)
- Camel- or fine hair brush
- Tissue paper

Procedure:

To study egg laying, collect moths from the field using the sweep net and place them inside a potted, screen-caged cabbage plant. Leave the moths inside the cage for 24 hours. After 24 hours, remove the moths and observe for eggs laid on the plant. Observe the eggs and make drawings. Don't touch the eggs! Continue to monitor.

Cabbage leaf with *Plutella* larva (left) and pupa (right)

Plutella adult on cabbage leaf in field

Also take a few leaves or leaf parts with eggs from the potted plant and place them into clear plastic bottles that are lined with slightly moist tissue paper. Label the bottles (name, date, crop). Keep them in the classroom or in the shade. Monitor the eggs and observe time of hatching.

After hatching observe the larvae in the bottles as well as on the caged plants. In the bottles, regularly provide new cabbage leaves for food. Regularly make drawings. Note dates of pupation. Monitor the progress closely, as the experiment can fail due to diseases, predation or escape.

Discussion:

Egg stage:

? Where are the eggs laid - on the plant or on the soil?
? If on the plant - which part of the plant? If on the leaf - which part of the leaf? If on the stem, which part of the stem?
? How many eggs were laid?

? What is the shape and colour of the eggs? Are there differences in colour?
? How many days does it take for the eggs to hatch?

Larval stage:

? What is the size of the caterpillar at hatching?
? Where do the larvae feed? Which part of the plant?
? Do the larvae change skin? Why?
? What are the sizes of the larval stages?
? What happens when the caterpillar is fully grown?
? How many days does the larval stage last?

Pupal stage:

? Where does the caterpillar pupate?
? Does it make a cocoon? What is the colour, shape and size of pupa and cocoon? What is the function of the cocoon?
? How many days does the pupal stage last?

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6.13 Diamondback moth injury symptoms on cabbage

This exercise is applicable only to cabbage fields with diamondback moth, but could be adapted to other caterpillar pests as well. If the risk of losing the crop due to the introduction of caterpillar pests is perceived to be too high, use screen cages to cover the treated plants.

Objective: To study the symptoms caused by diamondback moth (DBM) on cabbage

Materials needed:

- Field with 4 weeks old cabbage
- 32 to 62 3rd-4th instar DBM larvae
- 8 stakes and labels

Procedure:

Cabbage damaged by *Plutella* larvae

Select 8 uninjured cabbage plants in the field. Mark the plants with stakes and labels (write treatment and date on label). Transfer the DBM larvae to the selected plants (2 plants do not receive DBM larvae, 2 plants receive 1 larva each, 2 plants receive 5 to 10 larvae each, 2 plants receive 10 to 20 larvae each). Alternatively, the selected plants with DBM larvae could also be caged so that the larvae cannot migrate to other plants.

If necessary, spray the field with a B.t. product after a few weeks.

Observations:

Weekly observe the extent of injury done by the DBM larvae and compare the different treatments. Make drawings of the injury symptoms. At harvest, weigh the cabbages per treatment and compare the yields.

Discussion:

? On which part of the plant did the DBM feed?
? What was the damage observed on each plant?
? Was there a difference between the treatments (yield)?
? What might increase or decrease the damage caused by DBM (rain/sunshine)?

Possible modifications of/additions to the exercise:

1. Prepare insect zoo (see exercise 6.1) with DBM

2. Do field walk and count DBM larvae on slightly and heavily injured plants
3. Test effect of plant age on injury by DBM by raising cabbages in pots at intervals and use these plants in insect zoos

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6.14 Plant compensation study

A healthy crop usually can compensate for caterpillar feeding injury that occurs. However, cabbages with holes often give lower market prices. It is important to understand what kind of injury can be compensated for, at what stage, and what kind of injury causes loss of yield and/or quality. First assess where the insects usually occur on the plant, on shoots, on older or younger leaves. Also assess the average percentage of leaves per plant that are affected.

Objective: To study compensation by a cabbage crop after defoliation due to leaf feeding insects

Materials needed:

- ✔ Vegetable field (circa 1 month after planting)

Procedure:

Choose an appropriate layout for the experiment, e.g.:


Manual leaf removal (50%) to mimic plant defoliation by insects

A. Defoliation:
Select 50 plants in one plot. Remove half of each leaf along the main vein (= 50% defoliation) or remove every other leaf entirely (= 50% defoliation) of 5 plants at 1 week after planting, of another 5 plants at 2 weeks after planting, of another 5 plants at 3 weeks after planting, of another 5 plants at 4 weeks after planting, of another 5 plants at 5 weeks after planting. Leave the remaining 25 plants unharmed. Label or tag all treated plants. In addition, another batch of 50 plants could be selected in another plot to do the same exercise cutting only 25% leaves at the same intervals as above.

B. Removal of shoot / growing point:
Select 50 plants in one plot. Remove the shoot by cutting the growing point of 5 plants at 1 week after planting, of another 5 plants at 2 weeks after planting, of another 5 plants at 3 weeks after planting, of another 5 plants at 4 weeks after planting, of another 5 plants at 5 weeks after planting. Leave the remaining 25 plants unharmed. Label or tag all treated plants.

Observations:

At weekly intervals after removal of plant parts, observe the average plant height and the average number of branches and / or leaves per plant in each treatment. At harvest, observe the yield in the different treatments. Present the results and compare the different treatments.

Discussion:

- ? Was there a difference in yield between the different treatments? How much?
- ? What would have happened if we had continued the defoliation until harvest time?
- ? Does this trial simulate loss of foliage due to insect feeding?
- ? Does this trial simulate loss of energy production by the plant due to defoliation?
- ? Up till what stage was the crop able to compensate for shoot or leaf loss?

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6.15 Assessment of impact of ground-dwelling predators

Objective: Study the importance of ground-dwelling predators in the management of leaf feeders

Materials needed:

- ✔ Unsprayed cabbage field with caterpillar infestation
- ✔ Exclusion fences (zinc sheets, about 20 cm high, with Tanglefoot = sticky glue at the higher edge)

Procedure:

A few weeks after planting, select 20 plants (same growth stage) with similar caterpillar densities. Label 10 plants as "control" and 10 plants as "predator excluded". Position exclusion fences around each "predator excluded" plant. Remove any predators inside the barrier or on the plant. Do not disturb the caterpillars!

Observations:

Weekly observe the number of caterpillars and, if appropriate, pupae on each plant. At harvest, assess the yield and marketing grade of each treatment.

Discussion:

? What are the differences between caterpillar populations in both treatments?
? What are the differences in yield between the treatments? Why?
? How important are ground-dwelling predators in the management of caterpillar pests?

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6.16 Measuring the parasitism level of caterpillars

This exercise can be conducted any time when caterpillars (either boll worms, borers or leaf worms, such as semi-loopers or army worms) are common.

Objectives:

1. To find out which parasitoids attack caterpillars (leaf worms or borers)
2. To assess the importance of parasitism
3. To find out about some aspects of the biology of the parasitoid species, for example, do they attack young or older stages of the host

Materials needed:

- Plastic tubes, with labels
- Fine hair brush
- Tissue paper
- Fresh plant material for larval feeding

Procedure:

Each group should select two species of larva that are common in the field. Collect from an unsprayed field:

- 5 small (< 0.8 cm) larvae of each species
- 5 medium (0.8 - 1.5 cm) larvae of each species
- 5 large (> 1.5 cm) larvae of each species

If plenty of tubes are available, each group could collect more larvae. Use a brush to transfer the larvae into the tubes, don't touch or injure them!

Put the larvae one-by-one 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 or other plant parts for food, and secure a piece of tissue paper between the lid and the tube to prevent condensation.

Observe each tube daily and replace food regularly. Observe carefully whether parasitoids emerge from the caterpillar, whether the caterpillar has pupated, or whether the adult has emerged. If parasitoids emerge, count them and keep them for identification.

Continue these observations until parasitoids 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:

$$\%\,\text{parasitism} = \frac{\text{parasitised larvae}}{\text{total larvae}} \times 100\%$$

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.

Discussion:

- What parasitoids were found?
- What were the parasitism levels of each pest species?
- Did you find different parasitoid species in small compared to large stages of the host?
- Describe how each parasitoid species developed in the tubes (e.g., was development mostly inside or outside the host; how many parasitoids emerged per host; from which host stage did the parasitoid emerge).

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6.17 Parasitisation on diamondback moth of cabbage

This exercise has been developed for diamondback moth on cabbage, but could be conducted for other caterpillars on other crops as well.

Objective: To learn about the level of parasitisation of diamondback moth (DBM) by natural enemies

Materials needed:

- Bottles or vials
- Screen gauze
- Honey solution and cotton bolls
- Unsprayed cabbage field

Procedure and observations:

In the unsprayed field, sample 10 DBM larvae (5th instar), observe them carefully (colour differences that show parasitisation). Pull the guts and assess the percentage parasitisation (destructive, direct method) by counting the number of larvae with parasitoid larvae inside their guts. Alternatively, rear the larvae and assess the percentage of adult parasitoids emerging (not destructive, indirect method; possibility to release the parasitoids).

Sample 5 parasitised DBM larvae and store them in a bottle with cabbage leaf material, closed with gauze. Wait until adults emerge, provide honey solution on cotton bolls and, when possible, release the adult parasitoids in the field.

Discussion:

? What is the parasitisation rate in the unsprayed field?
? What is the effect of natural enemies on the DBM population?
? What would be the effect of spraying pesticides on the DBM parasitoids?

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6.18 Effect of parasitisation on feeding behaviour of diamondback moth

This exercise has been developed for diamondback moth on cabbage, but could be conducted for other caterpillars on other crops as well.

Objective: To study the effect of parasitisation on larval feeding behaviour of diamondback moth (DBM) on cabbage

Materials needed:

- Bottles or vials
- Screen gauze
- Unsprayed cabbage field

Procedure and observations:

Sample 5 un-parasitised (no discolouring of the larva) 4th instar DBM larvae, and store them in a bottle or vial with cabbage leaf material, closed with gauze. Sample 5 parasitised (some discolouring in the gut of the larva) 4th instar DBM larvae, and store them in a separate bottle with cabbage leaf material, closed with gauze. Observe the feeding and movements of the larvae in the different bottles. Observe the moment of pupation of the larvae and the emerging adults.

Discussion:

? How much leaf material is eaten in the different bottles or vials?
? Could the parasitised larvae still injure cabbage plants?
? Would the crop be able to compensate for the injury due to parasitised or un-parasitised larvae?

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6.19 Rainfall as mortality factor

To evaluate what portion of the eggs and larvae of a small-size caterpillar pest, such as the DBM, are washed off the plant during rains, plastic roofs can be used to exclude rainfall during a rainy season (N.B.: roofs do alter the light intensity and thus the growth of the plant).

Objective: Study the impact of rain on caterpillar populations

Materials needed:

- ✔ Cabbage field with caterpillar infestation
- ✔ Plastic roofs (rain-shelters) that can protect one or two rows of cabbages from rain

Procedure:

The roofing experiment is best conducted when the caterpillar population is high. Plastic roofs are used to cover rows of cabbage plants. Select 4 rows of 5 plants each to cover and another 4 rows of 5 plants to be used as control. Label the selected rows. The roofed rows need to be watered (by flooding) regularly. When there is not much rain, simulate rain in the un-roofed treatment by overhead irrigation. Do not apply any insecticides.

Observations:

Weekly record numbers of caterpillar larvae and, if appropriate, pupae in each treatment. Also record the weather conditions (sunny, cloudy, rainy).

Discussion:

- ? Was there a difference in incidence of the studied caterpillar pest between the roofed and un-roofed treatments?
- ? What was the effect of rain? Why?
- ? How does this relate to the incidence of the caterpillar pest during dry and rainy seasons?

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6.20 Hand picking of eggs and caterpillars

Objective: To study the impact of mechanical removal of eggs and caterpillars from a field

Materials needed:

- ✔ Unsprayed cabbage field with caterpillar infestation

Procedure:

Allocate two plots of about 25 m^2 in the cabbage field. Label one plot "No hand picking" and the other plot "Hand picking". Do not apply pesticides in both plots.

In the "Hand picking" plot, weekly monitor the plot and remove all eggs and caterpillars by hand (=sanitation). In the "No hand picking" plot, weekly monitor but do not undertake any action against eggs and caterpillars.

Discussion:

? Was there control of the caterpillar injury in the sanitation plot?
? What was the effect of the hand picking on crop development and yield?
? Is hand picking an economical method of pest management?

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6.21 Integrated management of web worm or heart caterpillar on cabbage

This exercise is only suitable for cabbage with web worm.

Objective: To study the mechanical and biological control of web worm or heart caterpillar on cabbage

Materials needed:

- Young cabbage field
- B.t.
- Small sprayer

Procedure:

Divide the field into two plots.

Spray one plot regularly with insecticides ("farmers' practise").

Observe the other plot ("IPM") weekly and remove by hand the egg clusters of web worm or leaf parts with just emerged and gregariously feeding web worm larvae (look for "windows in leaves"). When the crop is 40 days old, start to spot treat (= spray the infected plants only) with B.t. when later instar larvae are present.

Observations:

Count the number of pesticide applications in the farmers' practise plot and the number of B.t. applications in the IPM plot. Measure yield in both plots.

Discussion:

? What are the differences between the two treatments?
? What is the effect on natural enemies in both plots?
? What is the difference in cost and labour intensity between the two treatments?

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6.22 Use of screen in the nursery

In order to protect seedlings from excessive sunlight or rainfall, roofs are often constructed above seed beds. When roofs are used, care should be taken not to shade the seedlings too much as elongated and etiolated seedlings may not survive the transplanting shock in the field. An alternative is the use of screen cages, not only to provide shade (not too much shade) for the seedlings, but also to protect seedlings from caterpillars and other pests. The exercise below tests both roofs and screen cages. The exercise is applicable only to transplanted crops.

Objective: To manage caterpillars on cabbage seedlings

Materials needed:

- Cabbage seeds
- Soil medium (preferably subsoil, mixed with compost/manure (mature) and sand
- Screen netting (fine mesh – holes should be smaller than the average size of an aphid)
- Roof of local material (banana leaves, straw, clear plastic)

Procedure:

Choose a location for the nursery with good access to irrigation. Prepare the soil medium (according to local practices but preferably choose subsoil instead of topsoil). Prepare two identical seedbeds for broadcasting seeds according to the local practices of about 1 x 1 m^2 each. Label both plots.

Count 400 seeds, using only undamaged seeds. Prepare a nursery cage with screen covering the seed bed (dimensions of nursery: about 2 x 2 m^2 soil surface, height 0.5-1 m). Irrigate the nurseries. Broadcast 200 seeds in each seedbed. Cover seeds with fine soil. Cover the seed beds with banana leaves, straw or other material. Place nursery cages over one seed bed.

After germination, the seed bed cover can be removed (about 7 days after sowing). Irrigate regularly and apply nutrients according to local farmers' practices. Do not apply pesticides. By the end of the nursery phase, about 4 days before planting, the screen cage should be removed in order to harden the young plants.

Nursery with screened versus open plot in India

Cabbage nursery with and without screen in Thailand

Observations:

Weekly observe: total number of seedlings growing. Count number of seedlings with symptoms of caterpillar feeding. Are there any other insects or diseases present?

After 4 to 6 weeks (transplanting time), count the leaves per seedling on 25 randomly chosen seedlings and measure seedling height.

Calculate ratio of successfully raised seedlings out of 200 seeds sown per treatment, average number of leaves per seedling, average seedling height, ratio of seedlings with caterpillar feeding injury. Present the results.

Discussion:

- What are the differences between the treatments (emergence, seedling vigour)?
- What will happen with the plants after planting in the field (think of survival rates)?
- What are other possible modifications of the nursery practices?
- What is the difference in cost and labour intensity between the two treatments?

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6.23 Mixed cropping example: cabbage with mustard

Diversity in an agro-ecosystem has a stabilising effect, especially on crop diseases. Multiple cropping however is often more labour intensive than monoculture. In the multiple cropping exercises, economical advantages or disadvantages should be included in the evaluation. Legume crops usually make good intercrops as the nitrogen that they fix will benefit the other crop. In cabbage-tomato intercropped fields, incidence of diamondback moth was found to be less than in monocropped cabbage fields. In this interactive session, ideas are exchanged regarding possible benefits of intercrops.

Objective: To manage diamondback moth, web worm and aphids on cabbage

Materials needed:

- Field (2 plots of each about 10 x 10 m^2)
- Cabbage seedlings
- Mustard seeds

Procedure:

Prepare the field following farmers' practices. Label one plot as "cabbage monoculture" and the other plot as "cabbage & mustard polyculture". Two weeks before planting cabbage, in the "cabbage polyculture" plot, sow one row of mustard after every nine rows of (not yet planted) cabbage. Plant cabbage in both plots according to farmers' practices, leaving one row after every nine rows of cabbage for the mustard in the polyculture plot. Again in the polyculture plot, two weeks after planting cabbage, sow a second row of mustard next to the previous one. Tag 10 sample cabbage plants at random in each plot. Apply weeding, fertilising and irrigation following farmers' practices. Do not apply pesticides, except for the bio-pesticide B.t. if needed. Harvest the plots separately.

Observations:

Weekly observe the 10 sample plants per plot and diamondback moth and web worm larvae as well as aphids. At harvesting, assess the yield and the market grade of the cabbage in the monoculture as well as in the polyculture plot.

Discussion:

? What were the differences in the ecosystem between the cropping systems?
? Was there an effect on crop health?
? Which system would give the highest benefit to farmers?
? Was there control of pests? How about other crop health problems?
? Was there a difference in yield and/or market grade? Why?

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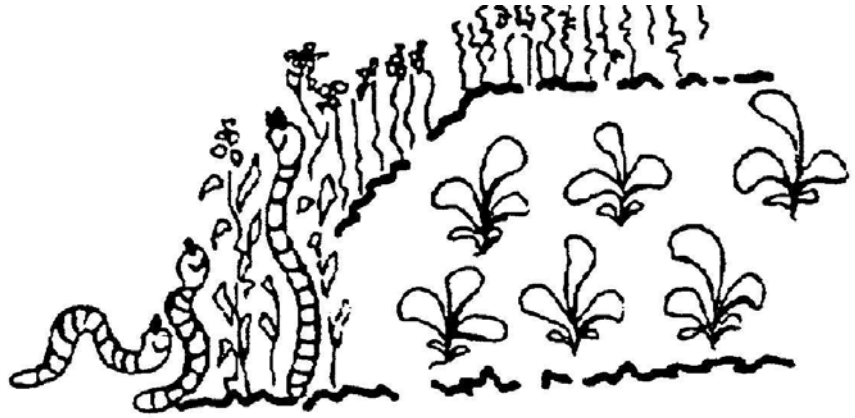

Trap crops such as mustard will attract pests so that the cabbage crop will remain protected for a while and will allow for timely intervention

6.24 Comparison of biological and chemical pesticides used in caterpillar control

Both biological and chemical agents are used to control insect pests. This study will compare the effectiveness of the biological control agent, B.t. and the chemical agent monocrotophos in controlling a cabbage pest such as web worm on cabbage. Biological control agents have been found to be safe to humans and the environment, yet effective in controlling larval pests, caterpillars in particular. They also preserve natural enemies. The gut of larval pests is attacked indicating that the B.t. must be eaten. Chemical agents work by attacking the nervous system of all animals, including humans. This study will demonstrate the effectiveness of a safe insecticide, B.t., which is a biological agent. In this exercise, web worms are used, but it can be adapted to other caterpillar pests as well.

Materials:

- Cabbage crop (15-30 days after planting)
- 2 Hand sprayers (1 litre)
- 1 Package B.t. and 1 package monocrotophos
- 1 Bucket water
- 3 Aqua cups with plastic sheet and rubber bands
- 1 Fine hair brush and scissors
- 9 Web worm larvae (similar size)
- Tissue paper
- Protective clothing: masks, gloves, aprons
- Soap and plenty of water for washing afterwards

Procedure:

This study should be executed in the late afternoon (B.t. breaks down under bright sunlight). Use protective clothing since the experiment will include the spraying of a chemical. Mix B.t. at recommended rate and spray cabbage plants (2-3 rows) at one side of the field. Mix monocrotophos at recommended rate (use gloves while mixing!) in the other hand sprayer and spray cabbage plants (2-3 rows) at the opposite side of the field. Wash with soap after spraying the poison. Allow to dry for 1 hour.

Collect 9 web worm larvae in a cup (from the unsprayed portion of the field). Cut 5 x 5 cm square sections from the upper part of a B.t. sprayed leaf and transfer the leaf portion to an Aqua cup with humid tissue paper inside. Put 3 web worm larvae inside the Aqua cup and close the cup with plastic sheet, secured with a rubber band. Label the cup "B.t.". Cut 5 x 5 cm square sections from the upper part of a monocrotophos sprayed leaf and follow the same steps as above. Label the cup "monocrotophos". Cut 5 x 5 cm square sections from the upper part of an unsprayed leaf and follow the same steps as above. Label the cup "control". Store the cups in a cool, shaded place and observe the behaviour of the larvae.

Wash with soap after handling the poisoned leaves.

Discussion:

- Did the larvae feed on the leaves?
- Why did we insert a humid tissue paper inside the Aqua cups?
- Why did we store the cups in a cool, shaded place?
- Did the larvae in the B.t. treatment behave different from the ones in the other treatments?
- Did all the larvae die? At what time? Why (not)?

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6.25 Assessment of viability of B.t.

This exercise will use living organisms to determine if B.t. has maintained its toxicity in storage and / or whether the B.t. purchased from a store is still useful for application in the field. Cabbage pests such as DBM have become resistant to a wide range of chemical insecticides. B.t. has been shown to effectively control DBM and other caterpillar pests. However, since it is a sensitive biological agent, it is subject to breakdown and loses its killing power. Use of B.t. is part of an IPM programme and works together with other natural enemies to control cabbage pests. Chemical insecticides do not do this. Therefore, this exercise is to determine/demonstrate the toxicity of B.t. After this exercise, you should be able to determine whether the B.t. you bought from the store is effective against your caterpillar pest. The exercise can be adapted to other caterpillar pests as well.

Materials needed:

- 1 Unsprayed cabbage plant
- 1 Fine hair brush
- Scissors
- 10 Aqua cups with a plastic sheet and rubber bands
- 1 Package of B.t.
- 6 Web worm larvae (or larvae of another pest); preferably small ones
- Tissue paper

Procedure:

Fill 2 Aqua cups with water. Mix 1/4 teaspoon B.t. with the water in one cup. Label the cup "B.t." and the other cup "water".

Collect inner leaves from the unsprayed cabbage plant. Cut the leaves into 8 sections of 5 x 5 cm. Dip 4 leaf sections into the "B.t." cup and 4 other leaf sections into the "water" cup. Label all leaf sections. Allow the leaf sections to dry in a cool, shaded place.

Put moist tissue paper in the 8 left over Aqua cups. Place each "B.t." leaf section into one cup (label!) and each "water" leaf section into other cups (label cups!). Place 3 small web worm larvae (or larvae of another pest) in each cup using the brush. Avoid damaging the caterpillars. Cover the cups with the plastic sheet and secure with a rubber band. Position the cups in a cool shaded place.

Observations:

Check the cups every 10 - 12 hours for 3 days and observe feeding and larval death. Usually, differences can be seen within 1.5 days.

Discussion:

? What happened to the larvae in the two treatments?
? Is your B.t effective in controlling the pest?
? Why did we put moist tissue paper in the Aqua cups?

? Why did we place the cups in the shade?
? Why did we include a comparison with "water" in the experiment?

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6.26 Inhibition of larval feeding by B.t.

This study will show how B.t. inhibits larval feeding. Many farmers spray B.t. without seeing immediate kill of the target pest. This is because B.t. acts slower than conventional chemical insecticides. Before actual larval death occurs, feeding by larvae stopped. This often causes farmers to think that B.t. is not effective. However, the benefits of B.t. use (conservation of parasitoids and predators, overall safety to farmers and consumers, safe for the environment) far outweigh those of chemical insecticides. Moreover, resistance in the target pests to chemical insecticides have rendered them less effective than B.t.. After this exercise you should be able to understand how B.t. kills the target pest and realise that B.t. makes the pest stop feeding before it kills the pest, hence there is less damage caused. The exercise can be adapted to other caterpillar pests as well.

Materials:

- 1 Unsprayed cabbage plant
- 2 Fine hair brushes
- Scissors
- 10 cups with plastic sheets and rubber bands
- 20 Large web worms or other caterpillars (of similar size)
- Tissue paper
- 1 Package of B.t.
- 2 Litre water
- 1 Plastic pail
- 1 Long wooden stirrer

Procedure:

Collect leaves from the upper part of the cabbage plant. Cut leaves into 5x5 cm sections. Using the pail, pour a litre of water and mix the recommended dose of B.t. on the label. Mix well, using the long wooden stirrer. Dip 4 leaf sections in the B.t. solution. Dip other leaf sections in clean water. Allow the leaf sections to dry in a cool, shaded place. Place each leaf section into a plastic cup lined with tissue paper. Label "B.t." or "water" according to the treatment. Collect 20 large web worm or other caterpillars (of similar size) using the brush. Place 2 larvae in each cup and cover with plastic, secured with a rubber band.

The next morning, check for feeding and/or larval death. Replace the leaf sections (B.t. treated ones in the B.t. cup and untreated ones in the untreated cup). At noon, check for feeding and/or larval death. Again replace the leaf sections. Using paper and pencil, trace the area of the leaf section from the B.t. cup and the untreated cup to indicate feeding quantity. Compare the leaf tracings from both B.t. treated and untreated leaf sections. Observe the amount of frass in the B.t. cup and untreated cup. Repeat the last steps in the late afternoon and continue for 3 days.

Discussion:

- Were there any differences in feeding between B.t. treated and untreated leaf sections?
- When did these differences occur?
- Were there any differences in amount of frass?
- What do these differences indicate? Suggest why the larvae stopped feeding?

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6.27 Sensitivity of B.t. to sunlight

This study will show how sunlight breaks B.t. down. Since B.t. is a biological agent, it is sensitive to sunlight. In bright sunlight, it loses its effectiveness and strength to kill insects. This study shows the effect of sunlight on the effectiveness of B.t. as well as the most appropriate time of the day to apply B.t. The exercise can be adapted to other caterpillar pests as well.

Materials:

- 2 Distant rows of cabbage plants (about 15-30 days after planting) in a not recently sprayed cabbage field
- 9 Small web worm or other caterpillar larvae of similar size
- 1 Hand sprayer (1 litre)
- 1 Package of B.t.
- 3 Fine hair brushes
- Scissors
- 1 Bucket of water
- 12 Cups with plastic sheets and rubber bands

Procedure:

Mix B.t. at recommended rate in the bucket and spray cabbage plants on one row (3-5 m) early in the morning. Do the same in the late afternoon, using another row (3-5 m).

One hour after the last spray, select a plant in each of the treated rows and one plant in an untreated row. Collect a leaf from the upper part of each selected plant. Label the leaves.

Cut a section of 5x5 cm from each leaf and place each in a separate cup (label the cups "Bt - sun", "Bt - no sun", "No Bt"). Add 2 - 3 web worm larvae to each cup and cover with plastic, secured with a rubber band. Store the cups in a cool and shaded place.

Daily observe signs of feeding in each cup and death of the larvae for 2 to 3 days.

N.B. 1. This study should be repeated if it rains on the day of the study.
2. This study may be repeated at two-day intervals to determine the effectiveness of B.t. on cabbage in the field.

Discussion:

? Did the larvae feed on the leaves?
? Did any of the larvae die? In which treatment?
? What was the effect of sunlight on the killing effect of B.t.?
? Why should we repeat the study if it rains on the day of the B.t. application?
? When is the best time of the day to apply B.t.?

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6.28 Life cycle and biology of the parasitoid *Diadegma semiclausum*

Objective: To understand the life cycle and development stages of *Diadegma semiclausum*, a larval-pupal parasitoid of diamondback moth (DBM). At the end of the study, participants will know how to rear the parasitoid using an insect zoo technique.

Materials needed:

- Clear plastic cages and potted cabbage plants with newly hatched larvae of DBM
- Additional clear plastic containers to study collected specimen
- 1 Magnifying glass (10x)
- Camel or fine hair brush
- 1 pencil and paper for labels
- 1 note book
- 1 roll tissue paper
- Honey solution
- Pieces of clear plastic sheet

Procedure:

Collect adult *D. semiclausum* (preferably mated ones and not too old) from the field or a laboratory culture. Keep them (preferably 2 pairs per cage) in a well ventilated cage. Provide diluted honey in fine droplets on plastic sheet. Place in the cage a potted cabbage plant with up to 50 DBM larvae of 2-3 days old. During this period, observe carefully what the parasitoids do and make notes.

Diadegma release house in young cabbage field

Interior of *Diadegma* release house with parasitised *Plutella* pupae

After 24 hours, remove the parasitoids and rear the DBM larvae until they form cocoons.

Discussion:

? How frequently do the female parasitoids visit the cabbage leaves?
? What happens when the parasitoids come near DBM larvae?

- How do the parasitoids attack larvae?
- How do DBM larvae try to escape from being attacked? By staying still or falling over?
- How many days (from exposure) before cocoons are formed?
- How many cocoons are formed from each DBM larva?
- What colour are the cocoons and where are they usually found?
- How long (no. of days) before the adult insect emerges from the cocoon?
- What is the total life cycle (no. of days) of the parasitoid?
- From the adult emergence, determine the level of parasitism (%)
- From your observation, what are the DBM stages in which the parasitoid develops? So, what kind of parasitoid is it?

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6.29 Life cycle and biology of the parasitoid *Cotesia plutellae*

Objective: To understand the life cycle and development stages of *Cotesia plutellae*, an endo-larval parasitoid of diamondback moth (DBM). At the end of the study, participants will know how to rear the parasitoid using an insect zoo technique.

Materials needed:

- Clear plastic cages and potted cabbage plants with newly hatched larvae of DBM
- Additional clear plastic containers to study collected specimen
- 1 Magnifying glass (10x)
- Camel- or fine hair brush
- 1 pencil and paper for labels
- 1 note book
- 1 roll tissue paper
- Honey solution
- Pieces of clear plastic sheet

Procedure:

Collect adult *C. plutellae* (preferably mated ones and not too old) from the field or a laboratory culture. Keep them (preferably 2 pairs per cage) in a well ventilated cage. Provide diluted honey in fine droplets on plastic sheet. Place in the cage a potted cabbage plant with up to 50 DBM larvae of 2-3 days old. During this period, observe carefully what the parasitoids do and make notes.

After 24 hours, remove the parasitoids and rear the DBM larvae until they form cocoons.

Cotesia parasitoid pupae on larvae of cabbage pest in field

Cotesia adults reared from pupae

Discussion:

? How frequently do the female parasitoids visit the cabbage leaves?
? What happens when the parasitoids come near DBM larvae?
? How do the parasitoids attack larvae?

- How do DBM larvae try to escape from being attacked? By staying still or falling over?
- How many days (from exposure) before cocoons are formed?
- How many cocoons are formed from each DBM larva?
- What colour are the cocoons and where are they usually found?
- How long (no. of days) before the adult insect emerges from the cocoon?
- What is the total life cycle (no. of days) of the parasitoid?
- From the adult emergence, determine the level of parasitism (%)
- From your observation, what are the DBM stages in which the parasitoid develops? So, what kind of parasitoid is it?

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6.30 Life cycle and biology of the parasitoid *Diadromus collaris*

Objective: To understand the life cycle and development stages of *Diadromus collaris*, a pupal parasitoid of diamondback moth (DBM). At the end of the study, participants will know how to rear the parasitoid using an insect zoo technique.

Materials needed:

- Clear plastic cages and potted cabbage plants with newly hatched larvae of DBM
- Additional clear plastic containers to study collected specimen
- 1 Magnifying glass (10x)
- Camel- or fine hair brush
- 1 pencil and paper for labels
- 1 note book
- 1 roll tissue paper
- Honey solution
- Pieces of clear plastic sheet
- Pre-pupae of DBM
- 2-3 Petri dishes

Procedure:

Collect adult *D. collaris* (preferably mated ones and not too old) from the field or a laboratory culture. Keep them (preferably 2 pairs per cage) in a well ventilated cage. Provide diluted honey in fine droplets on plastic sheet. Place 2-3 Petri dishes with up to 50 DBM pre-pupae spread out in the dishes. The Petri dishes should be placed over yellow papers (to attract the adult parasitoids). During this period, observe carefully what the parasitoids do and make notes.

After 48 hours, remove the parasitoids and rear the DBM larvae until they form cocoons.

Discussion:

? How frequently do the female parasitoids visit the cabbage leaves?
? What happens when the parasitoids come near DBM larvae?
? How do the parasitoids attack larvae?
? How do DBM larvae try to escape from being attacked? By staying still or falling over?
? How many days (from exposure) before cocoons are formed?
? How many cocoons are formed from each DBM larva?
? What colour are the cocoons and where are they usually found?
? How long (no. of days) before the adult insect emerges from the cocoon?
? What is the total life cycle (no. of days) of the parasitoid?
? From the adult emergence, determine the level of parasitism (%)
? From your observation, what are the DBM stages in which the parasitoid develops? So, what kind of parasitoid is it?
? What happens when the parasitoids come near DBM pre-pupae?

? How do the parasitoids attack pre-pupae and how long (minutes) do they stay with it?
? How many cocoons are formed from each pre-pupa?

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6.31 Life cycle and biology of the parasitoid *Cotesia glomerata*

Objective: To understand the life cycle and development stages of *Cotesia glomerata*, an endo-larval parasitoid of *Pieris* (cabbage white butterfly). At the end of the study, participants will know how to rear the parasitoid using an insect zoo technique.

Materials needed:

- Clear plastic cages and potted cabbage plants with newly hatched larvae of DBM
- Additional clear plastic containers to study collected specimen
- 1 Magnifying glass (10x)
- Camel- or fine hair brush
- 1 pencil and paper for labels
- 1 note book
- 1 roll tissue paper
- Honey solution
- Pieces of clear plastic sheet
- Young larvae (preferably 2nd instar) of *Pieris*

Procedure:

Collect adult *C. glomerata* (preferably mated ones and not too old) from the field or a laboratory culture. Keep them (preferably 2 pairs per cage) in a well ventilated cage. Provide diluted honey in fine droplets on plastic sheet. Place in the cage a potted cabbage plant with up to 50 *Pieris* of 2-3 days old. During this period, observe carefully what the parasitoids do and make notes.

After 24 hours, remove the parasitoids and rear the DBM larvae until they form cocoons.

Discussion:

? How frequently do the female parasitoids visit the cabbage leaves?
? What happens when the parasitoids come near DBM larvae?
? How do the parasitoids attack larvae?
? How do DBM larvae try to escape from being attacked? By staying still or falling over?
? How many days (from exposure) before cocoons are formed?
? How many cocoons are formed from each DBM larva?
? What colour are the cocoons and where are they usually found?
? How long (no. of days) before the adult insect emerges from the cocoon?
? What is the total life cycle (no. of days) of the parasitoid?
? From the adult emergence, determine the level of parasitism (%)
? From your observation, what are the DBM stages in which the parasitoid develops? So, what kind of parasitoid is it?
? Do the *Pieris* larva try to escape from the attack? How?

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6.32 Preference of host stages by *Diadegma semiclausum* [or *Cotesia plutellae*] (1)

Objective: It is frequently asked which larval stage(s) of the diamondback moth are attacked or preferred by the parasitoid *Diadegma semiclausum* [or *Cotesia plutellae*]. This exercise will help participants find out what stages are attacked and preferred when the parasitoid is not given a choice of the host larval stages.

Materials needed:

- Clear plastic cages with screened windows for ventilation
- Potted cabbage plants
- Camel- or fine hair brush
- 1 pencil and paper for labels
- Honey solution
- Clear plastic sheets

Procedure:

Collect adult *D. semiclausum* [or *C. plutellae*] (mated ones and not too old) from the field or a laboratory culture. Keep one pair in a well ventilated cage. Provide diluted honey in fine droplets on plastic sheet. Place a potted cabbage plant with 20 first-instar larvae of diamondback moth in the cage. During this period, observe carefully what the parasitoids do and make notes. After 24 hours, remove the parasitoids and rear the diamondback moth larvae until they form cocoons. Note the number of parasitoid cocoons formed.

Repeat the study using other larval instars (separately) of the diamondback moth.

Discussion:

- Describe how the parasitoids attack the host larvae?
- How many parasitoid cocoons did you find for each larval instar exposed to the attack?
- Are they the same? Which exposed instar gives rise to more parasitoid cocoons? And how different are the numbers (= what is the level of parasitism)?
- Is there any larval stage that the parasitoid did not attack?
- From the results, what can you conclude on the host stages which the parasitoid will attack or prefer?

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6.33 Preference of host stages by *Diadegma semiclausum* [or *Cotesia plutellae*] (2)

This exercise is a follow-up of the previous exercise (1).

Objective: It is frequently asked which larval stage(s) of the diamondback moth are attacked or preferred by the parasitoid *Diadegma semiclausum* [or *Cotesia plutellae*]. This exercise will help participants find out which stages are attacked and preferred when the parasitoid is given a choice of the different host larval stages together.

Materials needed:

- Clear plastic cages with screened windows for ventilation
- Potted cabbage plants
- Camel- or fine hair brush
- 1 pencil and paper for labels
- Honey solution
- Clear plastic sheets

Procedure:

Collect adult *D. semiclausum* [or *C. plutellae*] (mated ones and not too old) from the field or a laboratory culture. Keep 3-4 pairs in a well ventilated cage. Provide diluted honey in fine droplets on plastic sheet. Place in the cage a potted cabbage plant with 20 each of the different instar larvae of diamondback moth. During this period, observe carefully what the parasitoids do and make notes. After 24 hours, remove the parasitoids. Remove the larvae to different cages, each with a potted cabbage plant. Keep all the larvae of the same instar together in the same cage. Rear the larvae until they form cocoons. Note the number of parasitoid cocoons formed in each cage.

Discussion:

? Describe how the parasitoids attack the host larvae?
? How many parasitoid cocoons did you find for each larval instar exposed to the attack?
? Are they the same? Which exposed instar gives rise to more parasitoid cocoons? And how different are the numbers (= what is the level of parasitism)?
? Is there any larval stage that the parasitoid did not attack?
? From the results, what can you conclude on the host stages which the parasitoid will attack or prefer?

Notes:

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6.34 Competition between *Diadegma semiclausum* and *Cotesia plutellae*

Objective: It is frequently asked: What happens when a larva of diamondback moth is attacked by both *Diadegma semiclausum* and *Cotesia plutellae*? Will the two parasitoids emerge or only one, and which one? This exercise will help participants find out what happens and understand how the two parasitoids compete with each other.

Materials needed:

- Clear plastic cages with screened windows for ventilation
- Potted cabbage plants
- Camel- or fine hair brush
- 1 Pencil and paper for labels
- Honey solution
- Clear plastic sheets

Procedure:

Collect adult *D. semiclausum* [or *C. plutellae*] (mated ones and not too old) from the field or a laboratory culture. Keep 3-4 pairs in a well ventilated cage. Provide diluted honey in fine droplets on plastic sheet.

Place a potted cabbage plant (with 1-2 leaves) in a clear plastic cage (not too large to enable observation). Introduce onto the plant a second-instar larva of diamondback moth. Then place in the cage a female *D. semiclausum* (mated one and not too old). Observe carefully to see when the parasitoid attacks the larva. After the attack, remove the parasitoid and introduce a female *Cotesia plutellae* (mated one and not too old). Observe carefully to check that it attacks the larva. After the attack, remove the parasitoid and rear the larva until it forms the parasitoid cocoon. Record what parasitoid cocoon is formed (*D. semiclausum* or *C. plutellae*). Replicate this 10 times.

Repeat the above, but reversing the sequence of the parasitoid attacks, i.e., introduce *C. plutellae* first and followed by *D. semiclausum*.

Discussion:

- ? Describe how the parasitoids attack the host larvae?
- ? How many parasitoid cocoons does each larva produce?
- ? What parasitoid cocoons were obtained when *D. semiclausum* attacked first?
- ? What parasitoid cocoons were obtained when *C. plutellae* attacked first?
- ? Which of the two parasitoids has a higher level of success in parasitising the larvae of diamondback moth?
- ? What can you conclude from the results?

Notes:

6.35 Predation on sucking insects in insect zoo

Predators may feed on sucking insects in the field. To learn about their consumption rate we can conduct a simple study with predators that feed on small sucking pests such as aphids, leafhoppers or white flies.

Materials needed:

- Clear plastic or glass vials with lids (5 per group)
- Some tissue paper
- Fine brush
- Hand lens
- Labels

Procedure:

Collect leaves in the unsprayed field plot with plenty of small sucking pests.

a. In case of aphids, remove the winged adults with a fine brush, and remove any other insects so that exactly 10 aphids are left per leaf.

b. In case of white flies, remove any other insects with a fine brush, so that 10 white flies per leaf are left.

Be careful not to damage the prey. Insert each leaf in a vial so that per group there are 4 vials with leaves each with 10 of the same prey species. Insert a single leaf without prey in the 5th vial (control).

Also collect different species of predators in the unsprayed field plot, with 5 individuals of each species. Each group could choose their own predator species, for example, chrysopid larvae, syrphid larvae, *Paederus* adults, coccinellid larvae or adults, or *Orius*.

Add one predator to each vial. Label the vials with date, time, group name, predator species and prey species. Place a piece of tissue paper between the vial and the lid to avoid condensation. Keep the vials in a shaded place (away from direct sunlight).

Observe the predators for a while, to see if they feed on the prey.

After 24 h, carefully count and record the number of prey (that are alive) inside the vial. Check whether you can retrieve any remains of pests that have been killed.

Discussion:

? How many pests did each predator consume in 24 h?
? Which predator species ate most and which ate least?
? What happened in the control vial? What is the value of having a control without a predator?

? Do predators behave differently inside 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.

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6.36 Assessing natural enemies: cage exclusion of natural enemies in the field

Some arthropods are pests, feeding on plant parts, others feed on insect prey, others live inside other arthropods and again others come from weeds or neighbouring crops, and are simply resting in the cabbage crop. To learn about the effect of natural enemies on pest populations in the field, cage exclusion practices can be used.

Objective: To study arthropods, their feeding and life cycles

What You Need

- ✔ cabbage field with aphid infestation
- ✔ nylon mesh
- ✔ bamboo sticks

Procedure:

Day 1: Setting up exclusion cages

1. Prepare 10 nylon mesh cages (50 x 50 x 70 cm), supported by four bamboo sticks to cover individual plants.
2. Select 10 plants with high numbers of pests (e.g. aphids). Randomly label five of the plants as ‘without predator' and five as ‘with predator'.
3. Remove all predators from all the plants and from the soil underneath, and cage each plant. Bury the nylon mesh carefully into the soil to prevent access of any insects (ants may gain access through small crevices in the soil).
4. Collect 20 active predators (e.g. large ladybird or lacewing larvae) and introduce four predators into each ‘with predator' cage.

Day 4: Assessing the impact of predators

1. After 4 days, remove the cages. Carefully count the number of pests on each plant in each treatment, and record whether the introduced predators are still alive.
2. Calculate the average number of aphids in each treatment.

Discussion:

? How many pests were found on the different plants in the different treatments?
? What can we conclude about the role of predators in the field?

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6.37 Screen caging in nursery

Objective: To manage aphids and whiteflies on cabbage seedlings / transplants

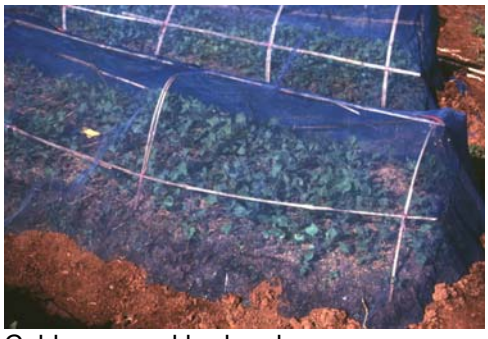
Materials needed:

- Wood or bamboo for nursery frame
- Aphid-proof (fine mesh) nylon screen
- Healthy seeds (virus-free!) of a virus susceptible cultivar
- Soil medium, preferably a mixture of subsoil, sand and compost or manure

Procedure:

Choose a location for the nursery with good access to irrigation. Prepare a nursery cage with screen covering the seed bed. Prepare the soil medium according to local practices, but preferably choose subsoil instead of topsoil to avoid damping-off or other soil-borne diseases.

Prepare two identical seedbeds for broadcasting seeds according to the local practices of about 1 x 1 m^2 each. Label one plot 'uncovered seed bed' and the other 'screen covered bed'. Irrigate the seed beds. Count 400 seeds, using only undamaged seeds.


Cabbage seed bed under screen

Broadcast 200 seeds in each seed bed. Cover seeds with a thin layer of fine (sieved) soil. If common practice, cover the seed beds with any mulch material to maintain humidity. Place the nursery cage over one seed bed. Make sure that there are no holes in the screen and bury the edges of the screen into the soil so that insects cannot enter the cage.

After germination, the seed bed mulch can be removed (about 7 days after sowing). Irrigate regularly (above the cage) and apply nutrients according to local practices. Do not apply any pesticides. Towards the end of the nursery phase, about 4 days before planting, the screen cage could be removed in order to harden the plants.

Observations:

Observations can be conducted after 4 to 5 weeks: count leaves ready for planting. Randomly choose 25 seedlings and count leaves per seedling and measure seedling height. Count seedlings with mosaic or other virus symptoms. Count seedlings with aphids. Are there any other pests or diseases present? Record symptoms. Calculate the percentage of successfully raised seedlings out of 200 seed sown, average number of leaves per seedling, average seedling height, percentage of seedlings with virus symptoms, aphids and other pests/diseases.

NB. Beware of different types of symptoms and confusion that may arise with symptoms caused by sucking insects!

Discussion:

- What were the differences between the 2 treatments?
- What is the difference in labour/cost requirements between the 2 treatments?
- What are advantages and disadvantages of the nurseries?
- What are other possible modifications of the nursery practices?
- What would be the effect of spraying on aphids and/or whiteflies in the farmers' nursery without screen?

Notes:

6.38 Thrips feeding symptom development

Objective: To learn the specific symptoms of thrips feeding injury

Materials needed:

- ✔ Fine screen cage of about 1 m high and 50 cm diameter (1 per group)
- ✔ Healthy, unsprayed cabbage seedlings (about 5 weeks old) (2 per group)
- ✔ Cabbage field with thrips

Procedure:

Collect cabbage leaves and/or flowers with thrips in the field. Do not touch the thrips as they are very sensitive to handling. Transfer the leaves / flowers with the thrips inside to a cabbage seedling (position the leaves / flowers somewhere in the top of the seedling). Cover the seedling with the screen cage. Cover another seedling with a screen cage as a control plant. Make sure there are no thrips on the control plant!

Observations:

Daily observe symptom development on the thrips-infected plant. Check the upper sides and under-sides of all leaves. Always compare with the control plant. Check whether you can find any thrips. Irrigate the plants daily after the observation.

Discussion:

? After how many days did you find symptoms?
? What are thrips feeding injury symptoms?
? Where are the symptoms found?
? Where are the thrips found?

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6.39 Rainfall as thrips' mortality factor

Objective: To show the influence of weather conditions on thrips outbreaks

Materials needed:

- ✔ Plastic roofs of about 1 m high, to cover circa 5 plants in a row
- ✔ Young cabbage field

Procedure:

Install the plastic roofs in the field. If there is lack of rain, simulate rain with daily overhead irrigation.

Observations:

Weekly observe the cabbage plants under the roof and compare them with other cabbage plants. Check the undersides of young leaves for thrips and feeding injury. Rate as follows: 0 = no symptoms; 1 = slight symptoms; 2 = moderate symptoms; 3 = severe symptoms.

Discussion:

- ? What are the differences between the roof-covered and non-covered plants? Why?
- ? Were there differences in numbers of thrips found on the leaves? Why?
- ? Were there differences in amount of thrips feeding injury found on the leaves? Why?
- ? What was the effect of rain/overhead irrigation?

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6.40 Use of light-reflective mulch to manage thrips and/or aphids

Objective: To demonstrate repellence of thrips and/or aphids by reflective mulch

Materials needed:

- ✔ Healthy seeds or seedlings (preferably raised in screen-covered nursery)
- ✔ Silvery or white plastic mulch

Procedure:

Prepare (harrowing plus fertiliser) a field roughly 50 m^2 before planting. Divide the field into two plots and assign one plot to the mulch and one to the control (and label them). Make plant holes in the mulch. Sow the seeds or plant the healthy seedlings in both plots. Apply exactly the same management practices in both plots.

Observations:

Weekly observe the plants in both treatments. Turn young leaves to check for thrips and feeding injury symptoms. Rate as follows: 0 = no symptoms; 1 = slight symptoms; 2 = moderate symptoms; 3 = severe symptoms. Weekly assess the number of plants with aphids. At harvest, record the yield and market grade of both treatments.

Discussion:

- ? What are the differences between the mulched and non-mulched plants? Why?
- ? Were there differences in numbers of thrips and aphids found on the leaves? Why?
- ? Were there differences in amount of thrips feeding injury found on the leaves? Why?
- ? What were other effects of the mulching?
- ? What is the difference in cost and labour intensity of the mulch (try to quantify)?
- ? Would the mulch be cost-effective?

Notes:

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6.41 Parasitism of white fly

Objective: To measure the parasitism level of white fly.

Materials needed:

- ✔ Cabbage field with white fly infestation
- ✔ If available, dissecting microscope

Procedure:

The simplest way to measure the parasitism level of white fly nymphs is by picking leaves with high numbers of white fly nymphs, and counting the number of healthy nymphs (whitish/yellowish in colour) and parasitised nymphs (brownish/black in colour). The advantage of this method is that it is simple, and results are directly obtained. The disadvantage is that it is not very accurate, because we underestimate parasitism since some nymphs may just have been parasitised but still appear as healthy nymphs. Another practical problem is that it is very difficult to distinguish in the field the white nymphs from empty nymphal remains.

Observations:

Leaves can also be picked and taken to the laboratory to be checked under the microscope. Empty skins can now be distinguished from healthy pupae.

Alternatively, leaves with nymphs can be taken to the laboratory to be kept in vials until healthy nymphs or parasitoids emerge.

Discussion:

? Were parasitised nymphs found? If yes, how did they look like exactly?
? What percentage of the nymphs was parasitised? Is this high or low?
? Which of the above three methods was most convenient? Which was most accurate?

Notes:

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6.42 Parasitism of leaf miners

This exercise is similar to exercise 6.41: Parasitism of white fly.

Objective: To study the parasitism levels of leaf miners of cabbages

Materials needed:

- IPM (or unsprayed) cabbage field with the studied crop
- FP cabbage field with the studied crop
- 20 vials
- Tissue paper
- Honey

Procedure:

Collect 10 leaves with a leaf miner (keep the leaf against the light to check whether the leaf miner is present) in the IPM cabbage field. Transfer the leaves to vials with a piece of tissue paper inside (1 leaf per vial). Label the vials with the date of collection, "IPM field" and your name. Follow the same procedure for the FP cabbage field and label the vials with "FP field". Store the vials in a cool, shaded place. When parasitoids emerge, honey solution can be fed to keep them alive for later release in the field.

Observations:

Daily check the vials for emergence of adult leaf miners or parasitoids. Record the date of emergence and the number of leaf miners and parasitoids from the "IPM field" and the "FP field".

Discussion:

? Were there any parasitoids? How many in the different fields?
? What is the effect of the parasitoids on the leaf miner population?
? What was the effect of pesticides on the level of parasitism?
? How can the level of parasitism be increased?

Notes:

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6.43 Effect of pesticides on spiders and other natural enemies

When pesticides are applied in the field, they also spread into the environment. Generally, pesticides reach the soil either through application on the soil or through run-off. Gaseous chemicals may escape into the air. In the soil, pesticides can bind to soil particles and/or move into groundwater. When a pesticide is highly persistent in the environment, undesirable biological effects may be caused, such as negative effects on soil flora and fauna, on aquatic life, on ecological diversity and air quality (pollution). From the crop management viewpoint, there are some additional, serious disadvantages of the use of chemical pesticides. In addition to the target pest, pesticides kill beneficials such as natural enemies and antagonistic fungi.

Objective: To evaluate the effect of sprayed leaves on the survival of natural enemies

Materials needed:

- Vegetable field, preferably unsprayed
- 4 Jars with lids
- 4 Pieces of Muslin cloth with rubber bands, to close the jars
- Labels
- Note paper, pen
- 4 Small hand-sprayers ((0.5 l), shared between groups)
- Small amounts of different insecticides
- Bio-pesticide (e.g. B.t.)

Procedure:

METHOD I

1. Prepare 4 hand sprayers before the practical. If a sprayer has been used before, wash it thoroughly with detergent. Prepare and fill 3 hand sprayers with commonly used insecticides, at field rate concentrations, for example: monocrotophos (organophosphate), cypermethrin (pyrethroid), *Bacillus thuringiensis* (biological insecticide). Fill 1 hand sprayer with pure water (control). Label the hand sprayers to avoid confusion!

2. Select 4 plants, at a reasonable distance from each other, in the field: one plant per spray treatment. Label each plant with the name of the treatment. Spray the labelled plants with the corresponding spray solution and let the leaves dry on the plant.

3. Pick one or several leaves from each treatment and transfer these per treatment to glass jars (use gloves). Label the jars. Each group should have one jar of each spray treatment (4 jars in total). Try to get the leaf to lie flat on the inside surface of the jar.

4. Collect predators, e.g. spiders or lady beetles, from the field. Try not to touch the predators but use a brush to collect them in jars. Carefully transfer them to the treatments (one of each species per jar). Use the same predator species in all treatments and make sure they are of similar size. Close the jar with the lid, and

place a piece of tissue paper between the tube and the lid to avoid condensation inside the jar.

METHOD II

1. Prepare 4 hand sprayers as in 1. of Method I.

2. Spray each piece of muslin cloth with a sprayer and let the cloth dry. Label the cloth (use gloves!).

3. Collect several predators from the field as in 3. of Method I (handle them carefully!) and transfer them to 4 jars per group. Use the same predator species for all treatments and make sure that they are of similar sizes. Close the jar with the sprayed muslin cloth.

Observations:

Check and record the condition of the predators after 8 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.

Discussion:

- What happened to the predators in the different jars? Why?
- What happens in the field when a farmer sprays against a certain pest?
- What will happen in a field 1, 2, 3 weeks after spraying?

Notes:

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6.44 Role play on insecticide resistance

When pesticides are used on a frequent basis, there is a risk of build-up of pest resistance against pesticides. Serious outbreaks of pests, e.g. diamond back moth on cabbage and brown plant hopper on rice, have been documented in several SE Asian countries after intensive use of chemicals resulted in the reduction of natural enemies, and meanwhile building up of pest resistance to pesticides. Last but not least, farmers tend to increase the frequency and dosage of pesticide applications when crop health problems persist. As farmers get caught in the 'pesticide treadmill', costs of production escalate. In this role play, experiences are shared about the reduction of effectiveness of insecticides due to build-up of pest resistance.

Objective: To understand how insect populations become resistant to insecticides.

Materials needed:

- 1 'Poison-sprayer' = Hand sprayer filled with water
- 14 Caps

Procedure:

Organise the group for the role play. You will need the following volunteers:

1 Participant acting as the story teller
1 Participant acting as the farmer (he will keep the 'poison sprayer' with him)
7 Participants, to be 'ordinary worms', who do not wear caps
14 Participants, to be 'super worms', who do wear caps
A group of 'observers' (all remaining participants), who will take notes of what happens

Ask the 'ordinary worms' to stay at one side of the room and the 'super worms' on the opposite side. The middle of the room is the farmers field (you could draw a boundary on the floor, using chalk, being the edge of the field, and put some chairs or stools as plants in the field).

The story teller starts reading the play and gives instructions to the group (instructions in italic):

> In the first week of the season, the farmer went to the field and he found 5 worms. He did not know it, but 1 of these, a 'super worm', was resistant to the pesticide that he usually used. All the others were 'ordinary worms'.
>
> *(1 'super worm' and 4 'ordinary worms' go into the field. After that, the 'farmer' comes in and acts as if he is observing his crop)*
>
> The farmer became very worried that his crop would be eaten by the worms, and he decided to spray poison immediately. One lucky 'ordinary worm' managed to escape the poison by hiding under a plant.
>
> *(The 'farmer' brings the 'poison sprayer' into the field and sprays all the worms except one 'ordinary worm')*

All but one of the ‘ordinary worms’ died of poisoning and the ‘super worm’ happily survived because of the resistance he/she has against the poison.

(All ‘ordinary worms’, except one, die, while the ‘super worm’ shows his cap to the public as his protection and smiles)

Now the farmer was happy, so he went away for a week. In that week, the remaining worms pupated, became adults and then started mating in order to make babies. Each adult could make 3 babies, so in the next generation of worms, there were 3 ‘ordinary worms’ and 3 ‘super worms’. After the mating and making babies, the adult insects died.

(Surviving worms rest, as if they are pupating, then arise, then get babies by inviting 3 more ‘ordinary worms’ and 3 more ‘super worms’ into the field, then fly away and die)

The next week the farmer came to the field and found 6 worms. Of course he did not know that among the 6, there were 3 ‘super worms’ who were resistant against poison. Again he was worried and he decided to spray. This time he mixed the poison a bit stronger and took care to cover all areas of plants where the worms could be hiding.

(Farmer looks around carefully and sprays all the worms, not excluding anyone)

All ‘ordinary worms’ died of the poison spray, but the ‘super worms’ survived.

(‘Ordinary worms’ die, while the ‘super worms’ again show their caps to the public and smile)

Again the remaining worms (3 ‘super worms’) pupated and emerged as adults, mated and made babies. As before, each adult made 3 babies, flew away and died. Because all the parents were ‘super worms’, the 9 new worms were all ‘super worms’.

(Surviving ‘super worms’ rest, as if they are pupating, then arise, then get babies by inviting 9 more ‘super worms’ into the field, then fly away and die)

The next week, the farmer visited the field again. Now he found 9 worms. He sprayed again with an even stronger poison, but now, none of the worms died!

(Farmer takes his poison sprayer, looks around carefully and sprays all the worms, not excluding anyone. The 'super worms' again show their caps to the public and smile)

What should the farmer do now?

(End of the role play: all players stand up and all observers applaud)

Observations:

Get the observers to report their observations. Use the following guide questions for the following discussion.

Discussion:

- ? How many worms died out of how many in each generation?
- ? How and why did this change between the generations?
- ? What would happen if the farmer continued spraying pesticides?
- ? What else could the farmer try to do?

Some individuals in a pest population have genetic traits that allow them to survive a pesticide application.

A proportion of the survivors' offspring inherit the resistance traits. At the next spraying these resistant individuals will survive.

If pesticides are applied frequently, the pest population will soon consist mostly of resistant individuals.

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6.45 How to collect and recognise insect pathogen groups

To study the biocontrol potential of different groups of pathogen or parasite, you will want to investigate dead or dying insects that you think are diseased or infected. You may have found these during the course of fieldwork or surveys. It is always worth asking colleagues to look out for insect corpses or sickly individuals when they are in the field.
This practical explains what to collect and how to identify which pathogen or parasite group you are dealing with, using macroscopic features (what you can observe without the aid of a microscope). It also outlines how to store freshly collected material before sending it for specialist identification.

Materials needed:

- Plastic or glass collecting tubes (take plenty, to keep individual specimens separate)
- Tissue paper (under tube lids to prevent condensation)
- Paper bags, envelopes or small packets
- Hand lens
- Healthy host insects and their plant food
- Cool bag or similar for keeping material fresh if you have no access to a fridge

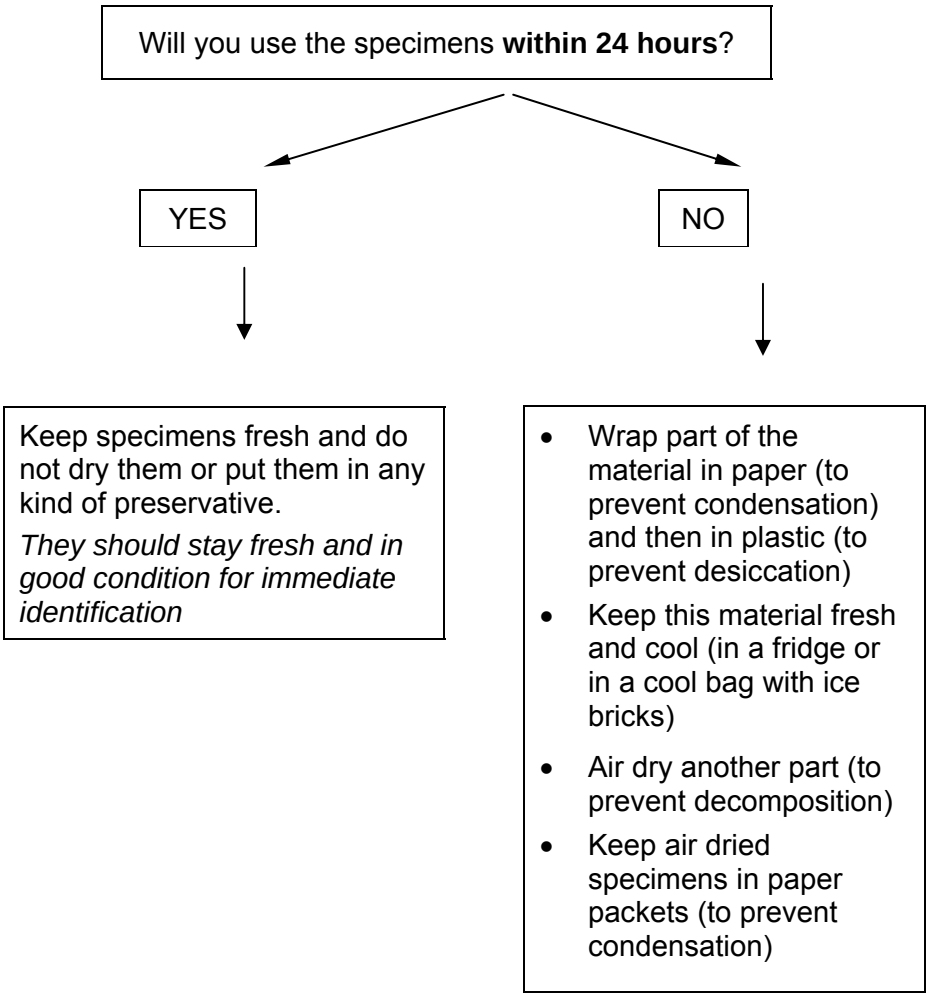
Procedure:

Step 1. Collecting material in the field

Collect as many diseased insects as possible because you may need to do several different things with them—by collecting many specimens you may find insects showing different life stages of the pathogen, which will help in identification.
Collect living and apparently healthy insects in a separate container—you can use the healthy ones for re-isolating the pathogens later. Some may also be infected but not yet showing symptoms, and these may develop later in the lab. This can be a useful extra source of material, especially if you cannot return to the site.

Don't forget to collect some food plant material for your living insects!

What you do with your specimens depends on how long you are going to be in the field. Decide how to keep your diseased specimens by following the chart on the next page.



Step 2. Identifying the likely pathogen group

Viruses

- Disease occurs mainly in the larval stage.
- Larvae often become pale and flaccid just before they die, turning dark after death.
- With baculovirus infections, the body contents of the host liquefy and the larva may hang by its prolegs, forming an upside-down V, often oozing a white fluid.

Bacteria

- Disease occurs mainly in the larval stage.
- Larvae remain normal colour until death and then darken to the brownish-black common with bacterial decomposition.
- Dead larvae often become flaccid, but do not liquefy.

Fungi

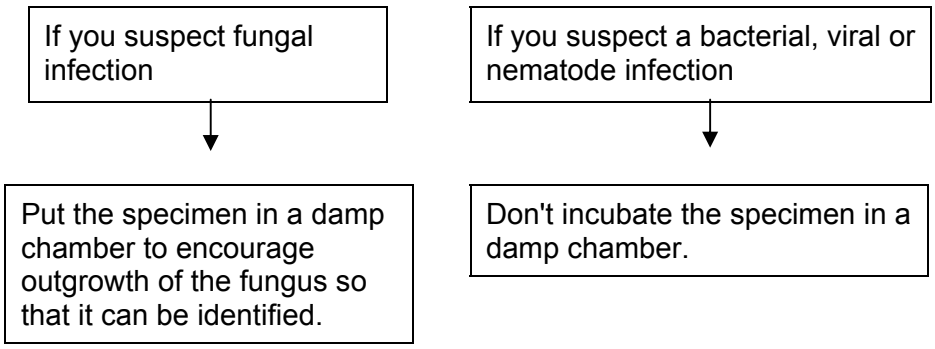
- Disease can be at any stage of the host.
- Dead insects become desiccated and rigid, never soft.
- Fungal mycelium or spores may be visible growing over the corpse.

Nematodes

- Infection can be at any stage of the host.
- Nematodes can often be seen through the insect cuticle.
- Some nematodes cause the dead host to change colour (to grey for *Steinernema*, to red for *Heterorhabditis*).

Step 3. Studying specimens

When you are not sure what you have…



When you know what kind of pathogen you have…

- ✔ Try to re-infect some healthy insects to produce the disease—you can do this using the field-collected insects.
- ✔ Preserve some material in your collection for future reference.
- ✔ Send some material away to specialists for full identification.

Handy hints:

> ☛ **Caution!**
> Never put pathogen-infected specimens in alcohol or expose them to heat or sunlight. Alcohol or other preservatives may destroy the important features needed in identification and will kill the living organism. Heat and UV light rapidly deactivate many pathogens.

- ☛ Viruses with inclusion bodies keep well in the decomposed bodies of their hosts. If you have some soft-bodied insects with suspected virus, or any host which might contain protozoa, you can keep samples in sealed glass tubes, kept cool if possible but this is not essential. The inclusion bodies or spores may remain intact (for microscopic examination) and infective (for re-infecting hosts) for several weeks, but make sure they do not dry out or are exposed to sunlight.
- ☛ The best way to air-dry material is to let it dry naturally (not in the sun, or in an oven—or abandoned in a hot vehicle!) for 3-4 days in a separate paper bag or envelope, which will prevent condensation forming. Air-drying should prevent material decomposing so that you can examine it microscopically back at base. It is possible to retrieve living pathogens, especially protozoa, from dried material.
- ☛ It is easy to re-infect insects with fungal pathogens without culturing the fungus first. You can try putting some healthy hosts into a small pot along with an infected corpse to see if they become infected. A more reliable way is to scrape some spore material off the specimen and suspend this in water, and then dip healthy hosts briefly into the fungal suspension using a paintbrush. You can also dip food plant leaves into the suspension, and then allow healthy hosts to feed on these and pick up spores by contact. The leaf-dip method can also be tried for re-infecting hosts with bacterial or viral material from a diseased specimen.
- ☛ For re-infecting other pathogen groups, see the culturing techniques described in the following exercise.
- ☛ Remember to label your tubes and packets of pathogen material, as you would for insect specimens, with date, location, habitat and host information.

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6.46 How to isolate and culture viruses and bacteria

It is easy to contaminate the insect's food supply, whether artificial diet or plant parts, with pathogen material. This bioassay technique will show whether your virus or bacteria isolate can infect a particular insect species.

Materials needed:

- Pathogen-killed larvae
- Water
- Blender
- Insect zoo
- Plant sprayer
- Healthy host insects and their plant food

Procedure:

Prepare two different sets of insect zoos. One will serve as a control in which the food is sprayed with water without entomopathogens.
For the other treatment, blend pathogen-killed larvae (or other stages) in some water. Spray the mixture onto leaves of the particular host's food plant using a domestic plant sprayer and feed the leaves to healthy hosts.

Observations:

Observe the insects for 1 to 3 weeks. Get the observers to report their observations. Use the following guide questions for the following discussion.

Discussion:

? What was the difference between the two treatments?
? What was the cause of death?
? How many insects died?
? How long did it take for the insects to die?
? What does it teach us when we apply entomopathogens in the field?

Healthy insects feeding on these contaminated leaves should become infected and die. In this way, you can set up continuous transfer from diseased to healthy insects.

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6.47 How to test the infectivity of nematodes to target insects

This bioassay technique will show whether your nematode isolate can infect a particular insect species. You will need some specialist materials and equipment (see below). You can also use it to find out about dose rate (the concentration of nematodes applied) or to compare isolates or different targets. The principle is to provide an 'infection arena' containing a water film, which allows the nematodes to swim to the test larvae. The best way to do this is to 'inoculate' damp sand (or filter/tissue paper) with the nematodes and then add the larvae. If the nematodes are pathogenic they will invade the larvae and kill them within 48 hours.

Materials needed:

- Moist sterilized sand (water content approximately 4% by volume)
- Plastic screw top 'universal' tubes
- Test larvae
- Nematode suspensions of known concentration
- Dissecting needles
- Beakers of approximately 250 ml volume with graduations

Procedure:

Step 1. Infecting the larvae

1. Fill each universal tube with moist sand.
2. Make a hole in the centre of the sand and add a given dose of nematodes in 1 ml tap water.
3. Tap the tube to fill in the hole and leave the nematodes to disperse (usually overnight).
4. Add a single test larva to each tube and screw the lid on.
5. Leave for 4-5 days at room temperature.

Step 2. Assessing infectivity

1. Remove the larvae from the tubes and wash in tap water.
2. Record mortality and any change in colour or shape.
3. Using the microscope, dissect each larva in a small quantity of water in a dish and count the number of nematodes infecting it.

Step 3. Assessing nematode survival

1. Empty the sand from each tube into a separate beaker, rinsing the tube with water to remove any remaining nematodes.

2. Make up the water volume to 100 ml and swirl the suspension vigorously.
3. Allow the sand to settle for a few seconds, then pour off the water containing the nematodes.
4. Count the number of nematodes in the water (see below).
5. Wash the sand again, and again count the nematodes in the water you pour off.
6. Calculate mean and standard error for the number of nematodes per insect host in each dose.
7. Calculate mean and standard error for the number of nematodes remaining in the sand at each dose.

Handy hints:

- ☛ This bioassay design allows you to study the relationship between the dose of nematodes applied to each larva and the number of nematodes that manage to infect the host. You can use it in a training context with different doses, e.g. 10, 100, 500, 1,000, 5,000 or 10,000 nematodes. Whether you want to compare different doses, different target hosts or different nematode isolates, you should set up at least 3 repeats of each treatment.
- ☛ You can sometimes see the colonizing nematodes through the cuticle of the dead larva, and eventually the nematodes will stream out into the water film when the larval body contents are consumed.
- ☛ If you do not have a lens grid in your microscope, you can count nematodes using a simple grid drawn on a Petri dish lid.
- ☛ If a large numbers of nematodes are present, you can count only a subsample of the nematode water suspension eg. 5 ml from a total of 100 ml and multiply by 20 to get the final figure.

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7. Disease Management Exercises

General background

The symptoms caused by diseases are variable but can easily be classified as leaf spots, damping-off, fruit spots and rots, root disorders, shoot disorders, mottling and mosaics, regardless of the causal agent (see exercise 'Identification of disease symptoms', p.221. This classification helps us to group exercises that can be done in order to find out about the life cycle of diseases and ways to manage a disease. In the following section, general exercises are described for identification of disease symptoms, leaf spot diseases, damping-off, and root and shoot disorders.

The exercises are listed below.

Exercise	Page	Name	Time required
7.1	221	Identification of disease symptoms	½ day
7.2	223	Pathogen groups	½ day
7.3	229	Disease triangle to explain disease management	½ day
7.4	231	Demonstration of spread of pathogens	½ day
7.5	233	Study of symptom development of leaf spots: classroom exercise	½ day + ≤ 1 wk mon.
7.6	235	Study of symptom development of leaf spots: field exercise	½ day + ≤ 1 wk mon.
7.7	237	Effect of infection of the seed bed	1 day + 4–8 wks mon.
7.8	239	Effect of the use of infected planting material	1 day + 4–8 wks mon.
7.9	241	Test effect of hot water seed treatment	1 day + 4–8 wks mon.
7.10	243	Use of subsoil to manage leaf spot diseases in the nursery	1 day + 4–8 wks mon.
7.11	245	Soil solarisation to manage leaf spot diseases in the nursery	1 day + 4–8 wks mon.
7.12	247	Steam sterilisation to manage leaf spot diseases in the nursery	1 day + 4–8 wks mon.
7.13	249	Test effect of infected crop debris in the field	Season-long weekly mon.
7.14	251	Effect of rain on the spread of leaf spot	Season-long weekly mon.
7.15	253	Spread of black rot disease by farming tools	1 day + 4–8 wks mon.
7.16	255	Test different cultivars for resistance to leaf spots	Season-long weekly mon.
7.17	257	Restricted fungicide use to manage leaf spots	Season-long weekly mon.
7.18	259	Study of spread of a fungal leaf spot	1 day + 4–8 wks mon.
7.19	261	Seed drenching/coating to manage damping-off	½ day + 4–8 wks mon.
7.20	263	Use of subsoil to manage damping-off in the nursery	1 day + 4–8 wks mon.
7.21	265	Soil solarisation to manage damping-off in the nursery	1 day + 4–8 wks mon.
7.22	267	Steam sterilisation of soil to manage damping-off in the nursery	1 day + 4–8 wks mon.
7.23	269	Raised plant beds to reduce damping-off incidence	1 day + 4–8 wks mon.
7.24	271	Effects of inundation of fields on incidence of damping-off diseases	1 day + 4–8 wks mon.
7.25	273	Pot experiment to test whether root diseases are soil-borne	1 day + 4–8 wks mon.
7.26	275	Use of subsoil to manage clubroot in the nursery	1 day + 4–8 wks mon.
7.27	277	Steam sterilisation of soil for the nursery	1 day + 4–8 wks mon.
7.28	279	Soil solarisation to manage root diseases in the nursery	1 day + 4–8 wks mon.
7.29	281	Test effect of soil solarisation in the field	Season-long weekly mon.
7.30	283	Effect of liming on clubroot of cabbage	Season-long weekly mon.
7.31	285	Spread of cabbage soft rot by farming tools	1 day + 4–8 wks mon.
7.32	287	Effect of rain on the spread of cabbage soft rot	Season-long weekly mon.
7.33	289	Sanitation measures to manage cabbage soft rot	Season-long weekly mon.

mon. = monitoring

7.1 Identification of disease symptoms

Once the importance of field inspections in relation to disease identification and management has become evident, the following exercise could be conducted to learn about different types of disease symptoms and about stages of severity of diseases in the field. The exercise shows that, without knowing names of diseases, one can group types of diseases and learn about the developmental stages of a disease in the field.

Objective: To distinguish between different groups of disease symptoms and learn about the developmental stages.

Materials needed:

- Vegetable field with different diseases in different progressive stages
- Hand lens (at least 1 per group)
- Poster paper
- Colour crayons
- Plastic bags

Procedure:

Participants discussing the symptoms of clubroot in the Philippines

Visit the field and ask each group to collect as many different disease 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.).

In the 'classroom', the disease symptoms should be grouped into leaf spot diseases, wilts, rots, root and shoot disorders. Assign each disease group to a group of trainees. Request the trainees per group to rank the disease symptoms in order of severity. Use the hand lens to check whether any spore structures can be found.

Ask each group to make drawings of progressive stages of the disease. Have a presentation on the disease symptoms and disease development per group. Avoid the use of scientific terms such as Latin names of diseases.

Discussion:

? Which diseases were present? What are the local names of the diseases?
? How do the symptoms look like? How do they start?
? Which plant parts are affected by the different diseases?
? How do the diseases reproduce and spread? How can one find out?
? Are the described diseases problematic? If yes, why?
? During which season are the diseases most severe?

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7.2 Pathogen groups

After the previous exercises, trainees are usually eager to learn more about disease management. In order to be able to discover about disease management, one should appreciate information that is now already available on life cycles of pathogens (the disease-causing organisms). Usually trainees know more than they realise about pathogen ecology, e.g. life cycles. This exercise taps the information present in the class and links it up to practical field school situations. The exercise should not / does not "test" the participants' knowledge on pathogens, but summarises the knowledge available and triggers creative thinking about 'how to find out' and 'what is the importance of knowing scientific names of diseases'. In the annex to this exercise, background information is given for each pathogen group. Usually, participants greatly appreciate a copy of this annex.

Objective: To become aware of available knowledge on disease ecology and to concentrate on management rather than control of diseases

Materials needed:

- Poster paper
- Markers

Procedure:

In an interactive classroom session, pathogen groups can be discussed as an introduction to disease management. First, a list of diseases of the crop concerned can be made together with the trainees. If the training session covers more than one crop, choose and focus on only one crop to avoid confusion. Use 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?
? What are the local names?
? What are the symptoms?
? When does it occur?

When the list is complete, assign the diseases according to the four disease groups, using for each disease the question:

? Is the disease caused by a fungus, bacterium, virus or nematode?

Again, "I don't know" is a valid answer. It triggers the creation of exercises or tests on "how to find out about it"! You could ask participants to come forward and make a schematic drawing of pathogens belonging to the four pathogen groups on a poster (see examples in the Annex to this exercise).

The following discussion should be focused on the method of spreading of each pathogen group. Use questions, such as:

? Does it spread through water?
? Does it spread through infected seeds?

? Can it survive and multiply on weeds?
? Can it survive on plant residues?
? Can insects spread the pathogen?
? Can humans spread the pathogen?

List all this information on poster paper and paste the posters on the wall of the classroom so that one can refer to them during future disease sessions.

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Annex: General background information to pathogen groups

Fungi

Fungi are living organisms. Many of the fungi are useful organisms, e.g. some enrich the soil, and some produce medicinal substances. Only one in ten fungi causes plant disease. A few examples of fungal diseases are: late blight of tomato and potato, downy mildew of cabbage, clubroot of cabbage, cercospora leaf spot of hot pepper. Fungi usually grow in threadlike structures, called mycelium, and produce spores (like seeds) with which they reproduce. It is possible to see fungal structures using a hand lens or, even better, with a microscope. Individual spores can only be seen when a microscope is used.

Symptoms that may be caused by fungal plant diseases:

> Fruits show dark, dry spots

> Roots become dry and sometimes have a white collar

> Stems become dry and have dark spots

> Leaves have dark spots with sometimes dots inside

> Undersides of leaves are covered with a mould

Survival and multiplication of fungi that cause plant diseases:

> Fungi produce spores (‘seeds’) when the humidity is high enough

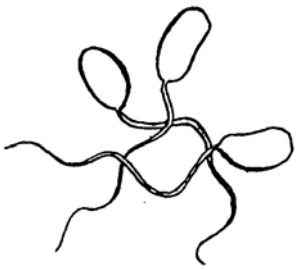
> The spores spread from one plant to the other by wind or rain splash

> The fungi consume the plant juices to produce spores on the diseased plant tissue

> The spores stay alive in crop residues in the soil and attack new host plants when they are grown on that same field

Bacteria

Bacteria are living organisms. Many of the bacteria are useful organisms, e.g. some enrich the soil, and some produce medicinal substances. Only one in ten bacteria causes plant disease. A few examples of bacterial diseases are: bacterial wilt of tomato and hot pepper, black rot of cabbage, soft rot of cabbage. Bacteria do not form spores but they can survive adverse conditions by coating themselves. Bacteria can only be seen using a powerful microscope.



Symptoms that may be caused by bacterial plant diseases:

> Fruit rots with a bad smell

> Leaves have water-soaked spots

> The roots rot and smell bad

Survival and multiplication of bacteria that cause plant diseases:

> Bacteria live and multiply under humid conditions

> The bacteria spread from plant to plant in water (irrigation, rain)

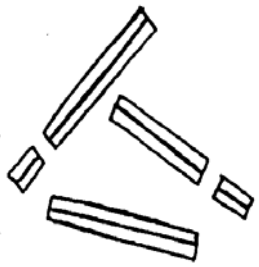
> Bacteria often enter the plant through injuries and suck plant juices

> As the plant dies, the bacteria stay in the soil ready to attack new host plants when they are grown on that same field

Viruses

Viruses are living organisms, but cannot live without other organisms (such as plants or insect-vectors). A few examples of viral diseases are: mosaic of cucumber, mosaic of potato. Viruses do not form spores, they duplicate themselves inside their hosts. Viruses are so minute, that they can only be seen with a very powerful electron-microscope.

Symptoms that may be caused by viral plant diseases:



> Curling of leaves or production of small leaves

> Mosaic or mottling of leaves

> Small fruits or colourless fruits

> Stunting of plant and reduced numbers of fruits produced

Survival and multiplication of viruses that cause plant diseases:

> Viruses live in infected plants and weeds

> Viruses are carried by insects from one plant to another or by humans

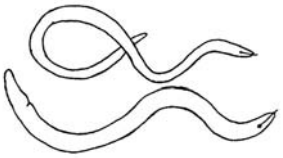
> Within the plant, viruses use plant components to multiply, and plant growth is reduced

> As the plant matures, some viruses can move to the seeds and be spread by seeds

Nematodes

Nematodes are living organisms that look like little worms and usually live in the soil. Many of the nematodes are useful organisms, e.g. some enrich the soil, and some are biological control agents. Only one in ten nematodes causes plant disease. An example of a nematode disease is: tomato root knot. Nematodes have life cycles like insects: they usually mate, females lay eggs, after hatching there are larval stages that mature into the adult stage. Nematodes that cause disease usually suck the roots of plants and cause gall-like structures on the roots. It is possible to see individual nematodes with a microscope.

Symptoms that may be caused by nematode plant diseases:



> Roots have small tumours

> Leaves stay small

> Fruits stay small and do not taste good

> The plant doesn't grow well and stays small

Survival and multiplication of nematodes that cause plant diseases:

> Nematodes live in roots of plants and suck the root juices

> Nematodes spread through soil that is carried from one field to another by humans (on shoes or farming implements)

> Nematodes can stay in soil in resting structures that germinate as new crops grow in the same field

7.3 Disease triangle to explain disease management

The information gathered in the previous exercises forms the basis for a discussion on disease management. The discussion can continue on a different day in a follow-up disease session as described here on the disease triangle and disease management. It shows that diseases only become problematic when the interaction between pathogen, crop and environment is optimal for the pathogen. In addition, it is stressed that disease management basically consists of manipulation of the pathogen, crop and / or environment. This is again an interactive classroom session. It is important to have the posters that were produced in previous disease sessions exhibited on the walls of the classroom and use them during this session as reference material.

Objective: To simplify the disease management background and promote creative thinking

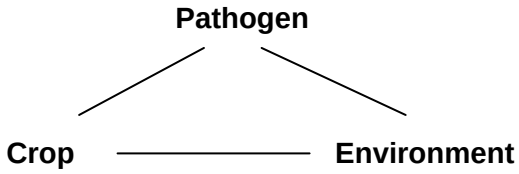
Materials needed:

- Poster paper
- Markers

Procedure:

Explain that for the development of a disease, there are three factors (pathogen, crop, environment) that must be present or favourable. Give examples, such as a fungal disease that survives on crop residues in soil (Is the pathogen present? -> Yes), which 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:



The disease triangle helps us in understanding management practices as we should try to avoid or 'inactivate' at least one of the angles in the disease triangle. The following examples may be useful to start a discussion on practical implementation of examples given:

Pathogen angle (Is the pathogen present?):

To avoid a soil-borne disease, one could test the use of subsoil in the nursery (Is the pathogen present? -> No? -> How would you apply this method in the field?).
To avoid an insect-transmitted virus disease, one could try to cover a nursery with screen-netting (Is the pathogen present? -> No? -> How would you apply this method in the field?).
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 pathogen present? -> No? -> How would you apply this method in the field?).
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 pathogen present? -> No? -> How would you apply this method in the field?).

Crop angle (Is a susceptible crop present?):

Search for resistant cultivars, by planting a portion of the field with other land races from neighbouring areas and/or imported cultivars (Is the crop present? -> No? -> How would you apply this method in the field?).
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?).
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?):

Choose a season that is not favourable for disease, e.g. the dry season (Is the environment favourable? -> No? -> How would you apply this method in the field?).
Change from overhead irrigation to flooding to reduce leaf wetness (Is the environment favourable? -> No? -> How would you apply this method in the field?).
Test mixed cropping, so that the disease cannot spread easily (Is the environment favourable? -> No? -> How would you apply this method in the field?).

After this discussion, divide the group into four. Refer back to the session on disease symptom groups and assign one symptom group to each training group. Ask each group to select one disease of the crop concerned and to design a disease triangle plus a disease management measure that can be tested in the trial field. Ask the groups to present the information after 1 or 2 hours and facilitate the discussion afterwards. Discuss which angle of the disease triangle is avoided or inactivated. Try to implement the management measures that the groups present in the trial field.

Note: It is very important to finish this theoretical session with some practical experiments that can be done in the field, such as an infection experiment and field + classroom. In that way, the theory is linked to practice as well as tested under local field conditions.

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7.4 Demonstration of spread of pathogens

An important aspect of disease management is the sanitation: in order to prevent spread of disease, roguing should be practised or farm tools should be 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 pathogens. This exercise session symbolises the spread of splash-dispersed (such as a leaf spot disease caused by a fungus), soil-borne (such as a nematode disease) and insect-vectored diseases (such as a virus).

Objective: To demonstrate spreading of pathogens by splashing water, soil cultivation and by insects in three different exercises.

Materials needed:

- 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
- 5 Glass or plastic vials, 1 filled with strong coffee or tea, the others with clear water

Procedure:

1. 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.

If the crop is not suitable for the exercise as described under 1., try this: use two plots of dry, bare soil of each about 1 x 2 m^2. 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 2 m border of each plot. Water each plot and compare the soil splash on both poster papers.

2. 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 (wet the tool first with water) and simulate weeding of the field. In another row, ask the participants to wet the soles of their shoes/boots/feet with water and walk through the flour on their way to inspect nearby plants. Observe spread of flour and also look at the farming tool and/or the soles of the shoes after the exercise. In case the field is wet, replace flour with fine seeds (such as watercress) and observe after germination of the seeds.

3. Use the syringe or straw and the vials, one of which is filled with coffee or tea and the others with clean water, to demonstrate spread of insect-borne viruses. The syringe represents the mouth parts of a sucking insect. The vial with coffee

represents a virus-diseased plant, the vials with clean water healthy plants. Suck coffee with the syringe and move to the first vial with water. Suck water, ejecting (‘spitting’) a bit of coffee, from the first vial. Observe the colouring of the water -> ‘healthy plant’ becomes infected with ‘virus’. Move to the other vials with clear water and ‘infect’ them one by one. Observe the colouring of the water in the vials and the reduced inoculum in the syringe as it is diluted every time it is used with a ‘healthy plant’.

Discussion:

- What did you observe?
- Which diseases do you know that spread in this way (splash - soil - insect)?
- How might these methods of spreading of pathogen affect crops in the field?
- How could spread of pathogens be prevented?

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7.5 Study of symptom development of leaf spots: classroom exercise

To learn more about the symptom development of leaf spots, a few basic exercises can be conducted. Similarly to the insect zoo, a disease zoo can be set up using infected leaves or plants to study the symptom development. These experiments help to recognise diseases in their early phases before they become severe. It would be best to execute this classroom exercise at the same time as the field exercise described hereafter.

Objective: To learn more about the symptomatology of leaf spot diseases

Materials needed:

- Cabbage field with leaf spots
- Petri-dishes, jars, clear plastic boxes, plastic bags
- Tissue paper
- Labels / tags
- Poster paper, crayons, ruler, hand lens

Procedure:

'Disease zoo' vase to monitor black rot development

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

In the classroom:

Use whole leaves or cut leaf portions with small leaf spots into discs of e.g. 10 cm diameter. Mark the leaf spot that you want to study on each leaf or leaf portion with a marker by drawing a big circle around the leaf spot. Place each leaf or leaf portion in a Petri-dish lined with moist tissue paper. Close with the lid. In case Petri-dishes are not available, one could also use clear plastic boxes with lids or clear plastic bags (that can be closed tightly - BUT leave some air inside).

Drawing exercise in classroom in Philippines

Drawing disease symptoms

Disease symptoms drawn from life samples (bean)

Observations:

Draw each leaf spot and the area around the spot in detail, using colour crayons. Measure the diameter of the leaf spot. Use a hand lens to see whether you find any mouldy and / or 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:

- What happens with a spot over time (colour, structure, area around spot)?
- How can one recognise the first symptom of the spot?
- What is the difference between a fungal spot and a bacterial spot?
- What is the difference between a disease spot and insect injury?
- Is the spot disease harmful to the crop production? Why?

Notes:

7.6 Study of symptom development of leaf spots: field exercise

It would be best to execute this field exercise at the same time as the classroom exercise.

Objective: To learn more about the symptomatology of leaf spot diseases

Materials needed:

- Cabbage field with leaf spot disease
- Labels / tags
- Drawing paper, crayons, ruler, hand lens

Procedure:

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 and mark the leaf spot with a marker (draw a wide circle around the spot).

Observations:


Trainees in the Philippines draw leaf spots

Draw each leaf spot and the area around the spot in detail, using colour crayons. Measure the diameter of the leaf spot. Use a hand lens to see whether any mouldy or granular (spores?) structures can be found in the leaf spot. Observe the leaf spot each or every other day and regularly draw and measure the size of the leaf spot. Also note the weather conditions (sunny, clouded, rainy). After one week, groups can be asked to present their findings.

Discussion:

? What happens with a spot over time (colour, structure, area around spot)?
? How can one recognise the first beginning of a spot?
? What is the difference between a fungal spot and a bacterial spot?
? What is the difference between a disease spot and insect injury?
? What was the effect of the weather during the experiment?

If the classroom exercise on symptom development was done simultaneously:

? Was there a difference in development of spots in the field and in the classroom? If yes, why?

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7.7 Effect of infection of the seed bed

Warning: This exercise is not always successful and hence it is advised that different groups try different leaf spot diseases.

Objective: To understand more about the life cycle of leaf spot diseases

Materials needed:

- Site in the field for a nursery
- Leaves with leaf spots (can be dead leaves) of the studied cabbage crop
- Seeds of a susceptible cabbage cultivar
- Subsoil, compost / manure, sand
- Labels / tags
- Hand lens

Procedure:

Keep the infected leaves overnight in a plastic bag with moist tissue paper to promote sporulation.

Prepare a site for a nursery. Prepare a soil mixture of subsoil mixed with compost / manure and sand (is supposed to be free of diseases).

Prepare one seed bed (possibly in a simple tray with holes for drainage) with the soil mixture and sow 200 seeds. Label the seed bed ‘Control seed bed’.

Prepare another seed bed at least 25 cm apart from the control seed bed. Shallowly incorporate the infected leaves into the soil mixture. Sow 200 seeds. Label the seed bed ‘Infected seed bed’.

Fertilise, irrigate and apply other nursery practices following farmers' practices. Do not apply any pesticides. If necessary, construct a cage with a screen net to keep insects out.

Observations:

Observe the seed beds weekly. Count the emerging seedlings and observe whether any leaf spots appear. Count the leaf spots. Observe whether any leaves drop off. After 4 to 5 weeks the experiment can be terminated. Sample 25 seedlings at random per treatment and assess the average number of leaf spots per seedling.

Discussion:

? Are there differences in numbers of seedlings between the treatments? Why?
? Was there any leaf drop in both seed beds?
? How many days did it take before leaf spots became visible?
? What would happen with the seedlings after transplanting in the field (disease development)?

? What is the importance of nursery practices in relation to disease management in the field?

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7.8 Effect of the use of infected planting material

Objective: Discover the importance of starting with healthy planting material

Materials needed:

- Site for the experiment in the field
- Plastic foil + bamboo sticks
- 10 Healthy seedlings
- 10 Seedlings with leaf spots

Procedure:


Pot experiment with black rot infected seedling in sterile soil

Choose a location for the trial with two plots (if needed with access to irrigation). Prepare the soil following farmers' practices. Plant the 10 healthy plants / plant parts and label that first plot "healthy planting material". Fence the first plot with plastic foil, supported by 4 bamboo sticks. Plant the 10 infected seedlings in the second plot and label it "infected plant material". Fence the second plot with plastic foil, supported by 4 bamboo sticks. Apply crop management practices following farmers' practices, but do not apply any pesticides.

Observations:

One week after planting, the observations can start. Weekly monitor the number of plants with lesions and estimate the percentage of diseased leaves per plant. When the plants mature, harvest the crop. Count and weigh the harvested product.

Discussion:

? What are the differences between the treatments in terms of crop health?
? What are the differences in terms of yield?
? Is it possible to grow a healthy crop from infected plant material?

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7.9 Test effect of hot water seed treatment

This exercise is applicable only to cabbage crops that are susceptible to seed-borne diseases, such as bacterial leaf spot on hot pepper or black rot on white cabbage. Do not use pre-treated (coated) seeds.

Objective: To demonstrate the effect of hot water treatment on cabbage seeds and seed-borne leaf spot diseases.

Materials needed:

- ✔ Seeds of a susceptible cultivar of the cabbage crop studied
- ✔ Thermos flask with warm / hot water (circa 50°C)
- ✔ Seed bed, prepared with subsoil/compost/sand mixture

Procedure:

Count 2 sets of 200 seeds.

Insert 200 seeds in the thermos flask with hot water and leave the seeds in the hot water for 25 to 30 minutes. Let them air-dry. Sow the 200 hot-water-treated seeds in a portion of the seed bed. Label the plot "hot water treated seed".

Sow 200 untreated seeds in another portion of the seed bed. Label "untreated seed".

Apply irrigation and other nursery practices following farmers' practices. Do not apply any pesticides. If needed, construct a cage with a screen net to keep insects out.

Observations:

After 4-6 weeks, observe the number of seedlings growing in each treatment. Calculate the percentage germination per treatment. Observe any leaf spots or other abnormalities in both treatments.

Discussion:

- ? Was there a difference in germination between the treatments?
- ? Were there any abnormalities during growth of seedlings in the different treatments?
- ? Were there any leaf spots in the nursery? If so, where did it come from?
- ? What other seed treatments do you know and could they be tested in a similar way?

Notes:

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7.10 Use of subsoil to manage leaf spot diseases in the nursery

The exercise is applicable only to transplanted crops and in this context only to soil-borne leaf-spot diseases.

Objective: To test the use of subsoil in the management of leaf spots in the nursery

Materials needed:

- Cabbage seeds
- Top soil (from a field cultivated regularly with cabbage and with a history of leaf spot disease infection)
- Sub soil (from the same field, but taken from the soil layer beneath the top 30 cm of soil)
- Compost/manure (mature)
- Sand

Procedure:

Choose a location for the nursery with access to irrigation. Prepare two types of soil medium:

a. using topsoil, sand and compost/manure
b. using subsoil, sand and compost/manure

Label the two heaps of soil mixture. Prepare 2 seed beds and label them as well.

Maintain a border between 2 seed bed treatments of at least 30 cm. Broadcast sow 200 seeds in each seed bed. Apply water and fertiliser when needed.

Observations:


Trainees in India filling pots with subsoil

After 3-5 weeks, the observations can be conducted. Count the total number of seedlings in the two seed beds and calculate the percentage of seedlings grown. Also observe the number of weeds in each seed bed. Randomly choose 25 seedlings from each treatment. Count the number of leaves and calculate the average number of leaves per seedling in each treatment. Also compare the length of the root system. Count the seedlings with leaf spots and calculate the percentage per treatment.

Discussion:

- What are the differences between the treatments (emergence, seedling vigor,…)?
- Can we improve the soil mixture with other additions?
- What will happen with the plants after planting in the field (think of survival rates)?
- What are other possibilities to modify nursery practices?
- Was there a difference in crop health between the treatments? Why?

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7.11 Soil solarisation to manage leaf spot diseases in the nursery

Objective: To test soil solarisation in the management of leaf spots in the nursery

Materials needed:

- Seed bed in a cabbage field with a history of leaf spot disease (untreated topsoil)
- Transparent polyethylene plastic (about 50 µm)
- Cabbage seeds of a leaf spot susceptible cultivar

Procedure:

Also see the illustrations on the following page.

Prepare a seed bed of about 2 x 5 m^2 (+ border) according to farmers' practices, including harrowing and fertilising, using the topsoil. Measure two plots of 2 x 2.5 m^2 each (make sure that the field is level, in order to avoid water flow from solarised to control portion). Irrigate one plot and apply the transparent plastic. The plastic must be secured along the border of the nursery by burying all four edges of the plastic well into the soil so that no heat can escape from underneath the plastic. Leave the other plot (control) fallow. Record the weather during the following weeks (sunny / cloudy / rain). After 4 to 6 weeks, the plastic can be removed. Sow 200 seeds in each plot. Apply normal irrigation and other nursery practices (not spraying!) during the seedling raising period. If needed, construct a cage with screen net to keep insects out.

Observations:

At removal of the plastic, check the plastic in the soil solarisation experiment for holes. Record in case there are holes (these will negatively influence the effect of the solarisation). Depending on the crop, after 4 to 6 weeks, record numbers of growing seedlings, of weeds and of diseased seedlings. Calculate the overall percentage of healthy seedlings. Randomly uproot 25 seedlings per treatment. Assess the average number of leaves per seedling and measure root length. Prepare presentations in the form of an agro-ecosystem analysis poster with, per treatment, a drawing of a seedling and all the data grouped around the seedling.

Discussion:

? What are the differences between the different treatments?
? Was there difference in seedling growth and health? Why?
? What is the cost and labour needed for the solarisation?
? What will happen to the beneficials (incl. natural enemies) in the topsoil?
? What are other advantages and disadvantages of the treatment? Was there control of disease? Why?

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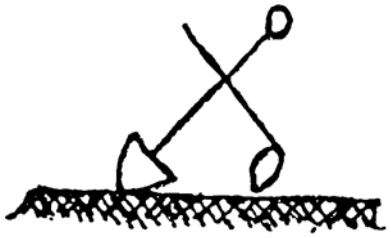
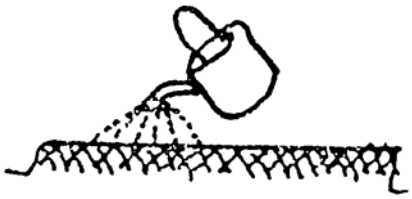
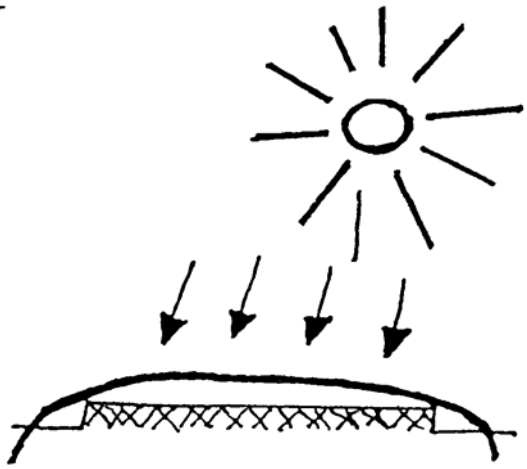


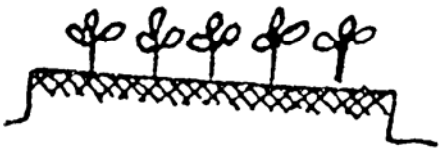
Soil preparation

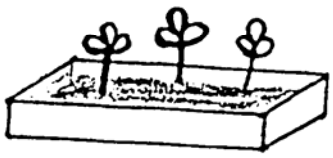


Irrigation



Solarisation





Sowing / Planting

7.12 Steam sterilisation to manage leaf spot diseases in the nursery

The exercise is applicable only to transplanted crops and in this context only to soil-borne leaf spot diseases.

Objective: To test steam sterilisation of soil in the management of leaf spot in the nursery

Materials needed:

- Seed bed in a cabbage field (untreated topsoil taken from a field with a history of leaf spot disease infection)
- Empty oil drum, bricks and plank
- Jute bags
- Fire place
- Cabbage seeds of a leaf spot susceptible cultivar

Procedure:

Mix the topsoil with compost/manure and sand as general practice when preparing soil medium for a nursery. Divide the soil mixture into two equal portions.

Clean the empty oil drum. Position the oil drum above a burner or wooden fire place. Insert water (a layer of about 10 cm in the drum). Put some bricks and cover the water layer with a wooden plank. Transfer one portion of the topsoil mixture into bags. Close the bags tightly. Transfer the bags to the drum (there must be enough air circulation between the bags). Cover the drum with a cover / lid and secure the lid with some bricks (see illustration on the following page). Light the fire and steam sterilise the soil in 3-4 hours. Let the soil cool down inside the bags. Open the bags and transfer the soil to another clean drum or container (leave open). The soil can be used 3 to 4 days later.

Prepare a seed bed of about 2 x 2.5 m^2 (+ border) according to farmers' practices, including harrowing and fertilising. Measure two plots of 2 x 2.5 m^2 each (make sure that the field is level, in order to avoid water flow from control to the steam sterilised portion). Prepare one seed bed with the untreated topsoil mixture and label ‘untreated soil’. Prepare the other seed bed (same size) with the steam sterilised soil mixture and label ‘sterilised soil’. Sow 100 seeds in each seed bed. Apply normal irrigation and other nursery practices (not spraying!) during the seedling raising period. If needed, construct a cage with screen netting to keep insects out.

Observations:

After sowing, monitor the different treatments weekly. Record numbers of growing seedlings, of weeds and of diseased seedlings. After 4 to 5 weeks, calculate the overall percentage of healthy seedlings in both plots. Randomly uproot 25 seedlings per treatment. Assess the average number of leaves per seedling and measure the shoot and root length.

Discussion:

? What are the differences between the different treatments?
? Was there difference in seedling growth and health? Why?
? What are the advantages and disadvantages of the treatments?
? What will happen to the beneficials (incl. natural enemies) in the topsoil?
? What is the difference in cost and labour needed for the soil treatments?
? Was there control of disease? Why?

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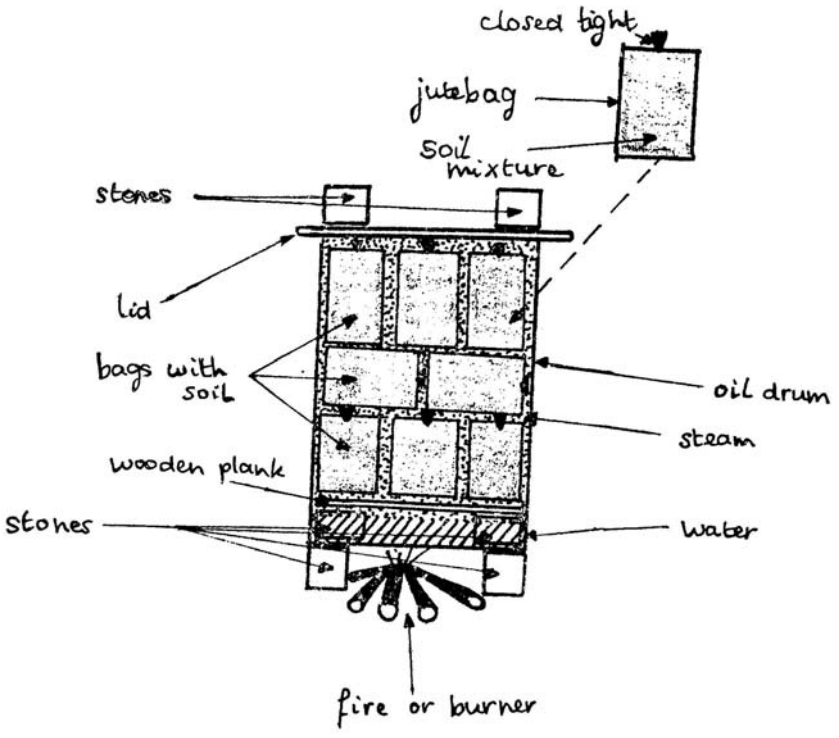

Locally devised steam sterilisation equipment

7.13 Test effect of infected crop debris in the field

This exercise is best done during a relatively dry period in an area where farmers usually leave the crop residues in the field after harvesting. When pathogens causing leaf spot diseases are present in those crop residues, they can often survive crop-free periods on those crop residues and cause disease outbreaks in a following cropping season.

Objective: Discover survival and spread of pathogens from infected crop debris

Materials needed:

- Cabbage residues with leaf spots, e.g. old leaf spot infected leaves
- Field of a susceptible cultivar, preferably just planted
- If possible, polyethylene plastic foil and 16 sticks

Procedure:

Make 4 field plots of each about 2 x 2 m^2 and label the plots as follows:

A. Control, no rain
B. Control, rain simulation
C. Crop debris, no rain
D. Crop debris, rain simulation

If possible, make a kind of fence around each plot with polyethylene plastic foil and 4 sticks. In plot B, overhead irrigation is performed daily to simulate the rainy season. In plot C and D, old infected leaves are deposited between the plants. In plot D, overhead irrigation is performed daily.

Observations:

Carefully monitor the plants weekly. Observe when leaf spots occur and how many leaf spots per plant or per leaf are developing. Try to distinguish the different diseases in case of multiple infection. At harvest, measure final yield and weight of the produce in each plot and note market grade (if applicable).

Discussion:

? What are the differences between the treatments?
? Which treatment first showed leaf spots? Which treatment showed leaf spots latest? Why?
? Was there an effect of the overhead irrigation?
? What was the difference in yield and market grade? Why?

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7.14 Effect of rain on the spread of leaf spot

Contrary to the previous experiment, this experiment can only be conducted during a rainy season.

Objective: To study the effect of rain on the spread of leaf spot disease

Materials needed:

- ✔ Plastic roof of about 1 m high, to cover 5 to 10 plants in a row
- ✔ Cabbage field (young crop, susceptible cultivar)

Procedure:

Each group of participants could install and monitor one roofed and one control treatment. Install the plastic roof in one row in the cabbage field and label the roofed plot "Without rain". Choose another plot of 5 to 10 plants, in another row at e.g. 1 m distance from the "Without rain" plot and label the plot "With rain". The roofed treatment should be irrigated, if needed, on the soil (avoid soil splash!). If there is lack of rain, simulate rain with daily overhead irrigation in the "With rain" plot. Do not spray with fungicides in either plot. All other crop management practices (such as fertilisation) should be the same in both plots.

Observations:

Weekly observe the plants in both plots. Check the number of plants with leaf spots, the number of leaf spots per plant and/or number of leaves with leaf spots per plant. Note the location of leaves with spots (lower, middle or top part of the plant). After 2 or 3 months, prepare to present the results by plotting the data over time in graphs. Compare the development of disease over time in the different plots and present the results per group of participants.

Discussion:

? Were there differences in number of leaves with leaf spots?
? Were there differences in numbers of leaf spots found on the leaves?
? Where were the leaf spots found? Would they cause yield reduction?
? Are leaf spot diseases more problematic during rainy seasons? Why?

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7.15 Spread of black rot disease by farming tools

Objective: To demonstrate the mechanical spread of cabbage black rot by farming tools

Materials needed:

- Cabbage field with black rot incidence
- Per group of participants, 4 potted young plants (healthy!) of a susceptible cultivar
- Scissors
- Alcohol
- Clean (tap) water
- 2 Big plastic bags
- Labels and tape

Procedure:

Bring the potted plants into the classroom. Water the plants. Label the potted plants as follows:

A. Healthy control (dry)
B. Healthy control (wet)
C. Black rot infected (dry)
D. Black rot infected (wet)

Water the potted plants. Clean the scissors with alcohol. After cleaning, dip them in clean (tap) water. Use the scissors to cut the leaves of both "healthy control" plants (A and B). Make incisions in different parts of several leaves so that in one leaf the main vein is cut, in another leaf only a side vein, in another leaf only intra-veinal cuts, in another leaf cuts into the edges of leaves. Before each new cut, clean the scissors with alcohol and dip them in water. Cover plant B with the plastic bag and secure the bag with tape.


Participant cuts with scissors in black rot infected leaf


Participant consequently cuts with same scissors in leaf of healthy plant


Black rot symptoms have developed on mechanically infected leaf

Visit the black rot infested field and collect leaves with clear black rot symptoms. Bring the leaves to the classroom. Clean the scissors with alcohol. After cleaning, dip them in clean (tap) water. Use the scissors to cut once across a lesion and immediately afterwards cut a leaf of plant C. Before each new cut, clean the scissors with alcohol, dip them in water and again cut across a lesion, and immediately afterwards a leaf of plant C. Make incisions in different parts of several leaves so that

in one leaf a main vein is cut, in another leaf only a side vein, in another leaf only intra-veinal cuts, in another leaf cuts into the edges of leaves. Repeat this for plant D. Cover plant D with the plastic bag and secure the bag with tape.

Keep the potted plants outside in the shade or in the classroom and water when necessary.

Observations:

Monitor the four plants and observe any lesions on the leaves.

Discussion:

- Was there transmission of disease? Why?
- How would the disease spread in the field?
- Was there a difference between the ‘wet’ and ‘dry’ treatments? If so, what does this mean?

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7.16 Test different cultivars for resistance to leaf spots

Host plant resistance can play a major role in disease management: resistant or less susceptible cultivars may have fewer leaf spots or only develop leaf spots at a later stage or develop leaf spots that are less infectious. Tolerant cultivars do show leaf spots, but do not give a lower production. Differences in resistance or tolerance to leaf spot between cultivars are investigated in this exercise.

Objective: Discover difference in host plant resistance or tolerance to leaf spot diseases under local conditions

Materials needed:

- ✔ Seed of locally used cabbage cultivars, and, if available, seed of other cultivars (if possible certified seed). If possible try to find several different cultivars with known resistance to leaf spot diseases.

Procedure:

Discuss the characteristics of the locally used cultivars. Identify which other cultivars may be acceptable by both farmers and consumers and which may have additional advantages.

Sow small quantities of seed of each cultivar (up to 4 or 5 maximum) in separated seed beds in the case of a transplanted vegetable crop or plant beds in the case of a direct seeded crop.

Label the different plots with the names of the cultivars. Apply the same crop management practices for all plots.

Observations:

Select and label 10 sample plants per cultivar. Weekly observe the sample plants and monitor the crop stages. At harvest, measure final yield and note the market grade. Prepare presentations with the crop growth and development over time as well as the yield and market grade.

While doing the observations on 10 sample plants, try to distinguish the different diseases in cases of multiple infection. Count the number of leaf spots and number of leaves with leaf spots per disease. Measure the diameter of 10 leaf spots. Prepare presentations with the development of disease over time.

Discussion:

? What are the differences between the cultivars?
? Was there a difference in earliness of the cultivars?
? Was there a difference in yield of the cultivars?
? Which cultivar would be preferred by farmers? Why?
? Which cultivar would be preferred by consumers? Why?
? Which cultivar first showed leaf spots? Which cultivar showed leaf spots latest?
? Was there a difference in leaf spot size between the different cultivars?

? Which cultivar was more resistant to leaf spot?
? Which cultivar will be grown again in another season?

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7.17 Restricted fungicide use to manage leaf spots

Objective: Minimise fungicide use in the management of leaf spot diseases

Materials needed:

- Susceptible cabbage crop in the field (young crop)
- Preventative and curative fungicides
- Plastic foil and 12 bamboo sticks

Procedure:

Discuss with the participants about threshold treatments and assess the most practical and, according to the participants, most appropriate threshold treatments. In this example, 10 and 5 % threshold treatments are used.

In the cabbage field, allocate three, distant plots of each about 5x5 m^2. Fence the plots with plastic sheet, supported with bamboo sticks. Label one plot as "farmers practice" or "conventional practice", another plot as "threshold treatment 10%" and the third plot as "threshold treatment 5%". In the "farmers practice" plot, apply the usual farmer's spray regime to control leaf spots (most probably frequent sprays using fungicide cocktails). In the other plots, monitor the disease development and apply a fungicide (one chemical only) at the recommended rate only when the threshold is reached. E.g. in the "threshold treatment 10%", apply a fungicide when 1 lesion is found per 10 plants, but not more often than every 5 days. In the "threshold treatment 5%" monitor the disease development. Apply a fungicide when 1 lesion is found per 20 plants, but not more often than every 5 days. In the threshold treatments, if possible, alternate the fungicides used.

Observations:

In addition to the disease development, record the fungicides used, the number of applications per plot and, at harvesting, the total yield per plot. If possible also include observations on other diseases, plant development, natural enemies, arthropod pests, etc. During the season, after 2 or 3 months, prepare presentations with the disease development over time and the total fungicide usage.

Discussion:

? What are the differences between the treatments? Why?
? What were the differences in input + labour costs?
? What were the differences in yield and market grade?
? Which treatment could be adopted again in another season?

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7.18 Study of spread of a fungal leaf spot

Objective: To observe spread of a fungal disease from an infected to a healthy plant

Materials needed:

- 4 Potted young cabbage plants (healthy!)
- Leaf spot infected leaves
- Clean water
- Small hand sprayer
- Clear plastic bags
- Tissue
- Labels

Procedure:

Day 1. Pick leaves with different lesions in the field. Place leaves or leaf portions with lesions in separate plastic bags. Also insert some moist (not soaking wet) tissue in each bag. Close the bags tightly but leave some air inside to avoid rotting. Keep the bags in a cool, shady place. Leave the bags overnight.

Day 2. Bring the potted plants into the classroom. Insert clean (tap) water into the container of the small hand sprayer. Spray two potted plants with the 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 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. 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.

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.

Discussion:

? Why did we inoculate the plants inside the classroom and not in the field?
? How many days did it take before symptoms were visible?
? How does a fungal leaf spot spread in a field?
? What can be recommended to a farmer with a leaf spot in his field?

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7.19 Seed drenching/coating to manage damping-off

Commercial seed companies often coat seeds with a fungicide layer to manage diseases during germination of seeds. The seed coating treatment should be mentioned on the package label. One can recognise whether or not seed is coated from the colour of the seed skin: when it is not the natural seed colour, then it is coated. This exercise is only suitable for un-coated seeds.

Objective: Use small quantities of fungicide to protect seedlings from damping-off

Materials needed:

- 3 Trays for seedling raising (size about 50 x 75 cm, with holes for drainage in the bottom)
- Cabbage seeds (un-coated, healthy looking seeds)
- Damping-off infected soil
- Fungicide (e.g. propamocarb-hydrochloride)
- 2 Jars or vials
- Clean water
- Tissue paper

Procedure:

Prepare the seedling raising trays with the damping-off infected soil. Label one tray "fungicide treated seeds", another tray "water treated seeds" and the third tray "untreated seeds".

Count 3 sets of 100 seeds. Prepare a fungicide solution according to the recommendations on the label in one jar or vial, labelled "fungicide treated seeds". Prepare another jar or vial with clean water and label "water treated seeds".

Soak 100 seeds in the "fungicide treated seeds" labelled jar for 10 minutes. Soak another batch of 100 seeds in the jar or vial labelled "water treated seeds" for 10 minutes. After 10 minutes, let the seeds air-dry on the tissue paper (label to avoid confusion).

Sow the seed lots in the corresponding trays. In the "untreated seeds" labelled tray, sow the last batch of 100 seeds. Place the seedling trays in a nursery area, if appropriate under shade and/or under a screen net to keep insects out. Irrigate and fertilise the seedling raising trays when needed.

Observations:

Weekly record the number of growing seedlings as well as the number of seedlings killed due to damping-off. After 4 to 5 weeks, calculate the overall percentage of seedlings killed due to damping-off.

Discussion:

? What are the differences between the different treatments?

- Was there reduction of disease? Why?
- Was there difference in seedling growth? Why?
- What are the disadvantages of the fungicide treatment (think of beneficial fungi in the soil)?
- What is the difference in cost and labour needed for the seed treatments?
- What are alternative, non-chemical methods to control damping-off?

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7.20 Use of subsoil to manage damping-off in the nursery

Objective: To manage damping-off diseases

Materials needed:

- Cabbage seeds
- Top soil (from a regular cultivated field and with a history of damping-off disease infection)
- Sub soil (from the same field, but taken from the soil layer beneath the top 30 cm of soil)
- Compost/manure (mature)
- Sand

Procedure:

Choose a location for the nursery with access to irrigation. Prepare two types of soil medium:

a. using topsoil, sand and compost/manure
b. using subsoil, sand and compost/manure

Label the two heaps of soil mixture. Prepare 2 seed beds and label them as well.

Maintain a border between 2 seed bed treatments of at least 30 cm. Broadcast sow 200 seeds in each seed bed. Apply water and fertiliser when needed.

Observations:

After 3-5 weeks, the observations can be conducted. Count the total number of seedlings in the two seed beds and calculate the percentage of seedlings grown. Also observe the number of weeds in each seed bed. Randomly choose 25 seedlings from each treatment. Count the number of leaves and calculate the average number of leaves per seedling in each treatment. Also compare the length of the root system. Count the seedlings with damping-off symptoms and calculate the percentage per treatment.

Discussion:

? What are the differences between the treatments (emergence, seedling vigor,…)?
? Can we improve the soil mixture with other additions?
? What will happen with the plants after planting in the field (think of survival rates)?
? What are other possibilities to modify nursery practices?
? Was there a difference in crop health between the treatments? Why?

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7.21 Soil solarisation to manage damping-off in the nursery

Objective: To test soil solarisation in the management of damping-off in the nursery

Materials needed:

- Seed bed in a cabbage field with a history of damping-off disease (untreated topsoil)
- Transparent polyethylene plastic (about 50 µm)
- Cabbage seeds of a damping-off susceptible cultivar

Procedure:

Also see the illustrations in the exercise on soil solarisation for leaf spots.

Prepare a seed bed of about 2 x 5 m^2 (+ border) according to farmers' practices, including harrowing and fertilising, using the topsoil. Measure two plots of 2 x 2.5 m^2 each (make sure that the field is level, in order to avoid water flow from solarised to control portion). Irrigate one plot and apply the transparent plastic. The plastic must be secured along the border of the nursery by burying all four edges of the plastic well into the soil so that no heat can escape from underneath the plastic. Leave the other plot (control) fallow. Record the weather during the following weeks (sunny / cloudy / rain). After 4 to 6 weeks, the plastic can be removed. Sow 200 seeds in each plot. Apply normal irrigation and other nursery practices (not spraying!) during the seedling raising period. If needed, construct a cage with screen net to keep insects out.

Observations:

At removal of the plastic, check the plastic in the soil solarisation experiment for holes. Record in case there are holes (these will negatively influence the effect of the solarisation). Depending on the crop, after 4 to 6 weeks, record numbers of growing seedlings, of weeds and of diseased seedlings. Calculate the overall percentage of healthy seedlings. Randomly uproot 25 seedlings per treatment. Assess the average number of leaves per seedling and measure root length. Prepare presentations in the form of an agro-ecosystem analysis poster with, per treatment, a drawing of a seedling and all the data grouped around the seedling.

Also carefully remove the soil from the roots of the 25 seedlings per treatment. Observe any root deformations. Make a bar graph per treatment of the number of growing seedlings and the number of wilted seedlings over time and present the findings.

Discussion:

? What are the differences between the different treatments?
? Was there difference in seedling growth and health? Why?
? What is the cost and labour needed for the solarisation?
? What will happen to the beneficials (incl. natural enemies) in the topsoil?
? What are other advantages and disadvantages of the treatment?
? Was there control of disease? Why?

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7.22 Steam sterilisation of soil to manage damping-off in the nursery

Objective: To test steam sterilisation in the management of damping-off in the nursery

Materials needed:

- Seed bed in a vegetable field (untreated topsoil taken from a field with a history of damping-off disease infection
- Empty oil drum, bricks, plank
- Jute bags
- Fire place
- Cabbage seeds of a damping-off susceptible cultivar

Procedure:

Mix the topsoil with compost/manure and sand as general practice when preparing soil medium for a nursery. Divide the soil mixture into two equal portions.

Clean the empty oil drum. Position the oil drum above a burner or wooden fire place. Insert water (a layer of about 10 cm in the drum). Put some bricks and cover the water layer with a wooden plank. Transfer one portion of the topsoil mixture into bags. Close the bags tightly. Transfer the bags to the drum (there must be enough air circulation between the bags). Cover the drum with a cover / lid and secure the lid with some bricks (see illustration on page 248). Light the fire and steam sterilise the soil in 3-4 hours. Let the soil cool down inside the bags. Open the bags and transfer the soil to another clean drum or container (leave open). The soil can be used 3 to 4 days later.

Prepare a seed bed of about 2 x 2.5 m^2 (+ border) according to farmers' practices, including harrowing and fertilising. Measure two plots of 2 x 2.5 m^2 each (make sure that the field is level, in order to avoid water flow from control to the steam sterilised portion). Prepare one seed bed with the untreated topsoil mixture and label ‘untreated soil’. Prepare the other seed bed (same size) with the steam sterilised soil mixture and label ‘sterilised soil’. Sow 100 seeds in each seed bed. Apply normal irrigation and other nursery practices (not spraying!) during the seedling raising period. If needed, construct a cage with screen netting to keep insects out.

Observations:

After sowing, monitor the different treatments weekly. Record numbers of growing seedlings, of weeds and of diseased seedlings. After 4 to 5 weeks, calculate the overall percentage of healthy seedlings in both plots. Randomly uproot 25 seedlings per treatment. Assess the average number of leaves per seedling and measure the shoot and root length.

In addition: Observe any discoloration of the root systems. Make a bar graph per treatment of the number of growing seedlings and the number of wilted seedlings over time and present the findings.

Discussion:

- What are the differences between the different treatments?
- Was there difference in seedling growth and health? Why?
- What are the advantages and disadvantages of the treatments?
- What will happen to the beneficials (incl. natural enemies) in the topsoil?
- What is the difference in cost and labour needed for the soil treatments?
- Was there control of the disease? Why?

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7.23 Raised plant beds to reduce damping-off incidence

Damping-off is often enhanced in fields with a bad soil structure. Improvement in drainage can reduce incidence of damping-off.

Objective: To test the effect of changes in soil structure on wilting diseases

Materials needed:

- Cabbage field with a history of damping-off
- Hoes or spades or other tools to work the soil
- Cabbage seeds or seedlings

Procedure:

In the area of the field where wilting plants had been observed previously, prepare two plots of 1 x 2 m^2 each as plant beds in the field: one plot level and the other plot raised (15-30 cm). Sow seeds or plant seedlings and raise the crops in each plot using common farmers' practices, including regular watering during dry periods.

Trainees in India prepare raised seed bed

Commercial seedling production on raised cabbage beds in Vietnam

Observations:

Weekly observe both plots and record number of wilted plants. Pull out the wilted plants and study the root system. Record any abnormalities on the roots. After 1 or 2 months, pull out all the remaining plants and study the roots. Measure and compare the root length in both plots. Also measure and compare the plant length in both plots.

Discussion:

? Was there a difference in disease incidence between both plots?
? Was there a difference in root length in both plots? Why?
? Was there a difference in plant length in both plots? Why?
? What other measures could improve drainage and/or reduce wilt incidence?

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7.24 Effects of inundation of fields on incidence of damping-off

Objective: To demonstrate the reduction of damping-off diseases by rotation with paddy rice

Materials needed:

- Dry land soil from a field grown continuously with cabbages with a history of damping-off
- Wetland soil (of a similar soil type as the dry land soil) from a paddy rice field (preferably one that has been flooded with water for 2-3 months)
- Cabbage seeds of the studied crop

Procedure:

Prepare one seed bed in a field that has been continuously planted with cabbages with the soil in that field. Label ‘dry land soil’. Take the soil from the paddy rice field and prepare the second seed bed using the paddy soil. Label 'wetland soil'. Sow (drill) 200 seeds in rows in each seed bed using a space of at least 5 cm between each seed.

Observations:

Weekly record total numbers of seedlings as well as numbers of wilted seedlings in both plots. After 4 - 6 weeks, uproot all seedlings in both plots and observe the roots. Do you see any discoloration of the roots? Measure root length as well as shoot length of 25 randomly chosen seedlings.

Discussion:

- What are the differences between the two treatments?
- Did wilting occur? If yes, was there a difference in wilting incidence between both treatments? Why?
- Would it be possible for farmers to prepare seed beds with wetland soil? What would be the advantages and what the disadvantages?

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7.25 Pot experiment to test whether root diseases are soil-borne

This exercise is useful when soil-borne diseases are found in the field that can be recognised on the above ground plant parts. Take some soil from such a "focus" of infection for comparison with clean soil.

Objective: To show disease development of healthy plant material in contaminated soil

Materials needed:

- Seeds of susceptible cabbage cultivar
- Four or more large pots
- Infected soil (e.g. from a dry land cabbage field with root disease symptoms)
- Clean soil (e.g. from a wetland paddy field)
- Bucket with water
- Labels

Procedure:

Fill 2 or more pots with infected soil and label them "Infected soil" and 2 or more other pots with clean soil (if necessary mixed with some clean sand) and label them "Healthy soil". Sow 1 to 3 seeds in each pot. Water the plants regularly and keep them in a shady place (if necessary under a screen cage to keep insects out) until symptoms appear. Apply fertilisers (a little) if needed.

Observations:

Observe the above ground plant parts regularly. Check differences in plant height between the treatments and observe wilting symptoms. Depending on the crop, after 1 or 2 months the root system should be observed for symptoms. Carefully remove the pots. Gently wash the soil from the roots in the bucket with water and study the roots. Make drawings of the symptoms and present those together with the results of the observations on the above ground plant parts.

Discussion:

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

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7.26 Use of subsoil to manage clubroot in the nursery

Objective: To manage clubroot in cabbage

Materials needed:

- Cabbage seeds
- Top soil (from a regularly cultivated field and with a history of clubroot infection)
- Sub soil (from the same field, but taken from the soil layer beneath the top 30 cm of soil)
- Compost/manure (mature)
- Sand

Procedure:

Choose a location for the nursery with access to irrigation. Prepare two types of soil medium:

a. using topsoil, sand and compost/manure
b. using subsoil, sand and compost/manure

Label the two heaps of soil mixture. Prepare 2 seed beds and label them as well.

Maintain a border between 2 seed bed treatments of at least 30 cm. Broadcast sow 200 seeds in each seed bed. Apply water and fertiliser when needed.

Observations:

After 3-5 weeks, the observations can be conducted. Count the total number of seedlings in the two seed beds and calculate the percentage of seedlings grown. Also observe the number of weeds in each seed bed. Randomly choose 25 seedlings from each treatment. Count the number of leaves and calculate the average number of leaves per seedling in each treatment. Also compare the length of the root system.

Count the seedlings with wilting symptoms and calculate the percentage per treatment. Observe the roots of all seedlings (carefully remove the soil from the roots) and calculate the percentage of clubroot disease infected seedlings per treatment.

Discussion:

? What are the differences between the treatments (emergence, seedling vigor,…)?
? Can we improve the soil mixture with other additions?
? What will happen with the plants after planting in the field (think of survival rates)?
? What are other possibilities to modify nursery practices?
? Was there a difference in crop health between the treatments? Why?

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7.27 Steam sterilisation of soil for the nursery

Objective: To test steam sterilisation in the management of clubroot in the nursery

Materials needed:

- Seed bed in a cabbage field (untreated topsoil taken from a field with a history of clubroot infection)
- Empty oil drum, bricks, plank
- Jute bags
- Fire place
- Cabbage seeds

Procedure:

Mix the topsoil with compost/manure and sand as general practice when preparing soil medium for a nursery. Divide the soil mixture into two equal portions.

Clean the empty oil drum. Position the oil drum above a burner or wooden fire place. Insert water (a layer of about 10 cm in the drum). Put some bricks and cover the water layer with a wooden plank. Transfer one portion of the topsoil mixture into bags. Close the bags tightly. Transfer the bags to the drum (there must be enough air circulation between the bags). Cover the drum with a cover / lid and secure the lid with some bricks (see illustration on page 248). Light the fire and steam sterilise the soil in 3-4 hours. Let the soil cool down inside the bags. Open the bags and transfer the soil to another clean drum or container (leave open). The soil can be used 3 to 4 days later.

Prepare a seed bed of about 2 x 2.5 m^2 (+ border) according to farmers' practices, including harrowing and fertilising. Measure two plots of 2 x 2.5 m^2 each (make sure that the field is level, in order to avoid water flow from control to the steam sterilised portion). Prepare one seed bed with the untreated topsoil mixture and label ‘untreated soil’. Prepare the other seed bed (same size) with the steam sterilised soil mixture and label ‘sterilised soil’. Sow 100 seeds in each seed bed. Apply normal irrigation and other nursery practices (not spraying!) during the seedling raising period. If needed, construct a cage with screen netting to keep insects out.

Observations:

After sowing, monitor the different treatments weekly. Record numbers of growing seedlings, of weeds and of diseased seedlings. After 4 to 5 weeks, calculate the overall percentage of healthy seedlings in both plots. Randomly uproot 25 seedlings per treatment. Assess the average number of leaves per seedling and measure the shoot and root length.

In addition: Observe any deformation and/or discoloration of the root systems. Make a bar graph per treatment of the number of growing seedlings, the number of wilted seedlings and the number of seedlings with clubroots over time and present the findings.

Discussion:

- What are the differences between the different treatments?
- Was there difference in seedling growth and health? Why?
- What are the advantages and disadvantages of the treatments?
- What will happen to the beneficials (incl. natural enemies) in the topsoil?
- What is the difference in cost and labour needed for the soil treatments?
- Was there control of disease? Why?

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7.28 Soil solarisation to manage clubroot in the nursery

Objective: To test soil solarisation in the management of root diseases in the nursery

Materials needed:

- Seed bed in a cabbage field with a history of clubroot disease (untreated topsoil)
- Transparent polyethylene plastic (about 50 µm)
- Cabbage seeds

Procedure:

Also see the illustration in the exercise on soil solarisation to manage leaf spot diseases.

Prepare a seed bed of about 2 x 5 m^2 (+ border) according to farmers' practices, including harrowing and fertilising, using the topsoil. Measure two plots of 2 x 2.5 m^2 each (make sure that the field is level, in order to avoid water flow from solarised to control portion). Irrigate one plot and apply the transparent plastic. The plastic must be secured along the border of the nursery by burying all four edges of the plastic well into the soil so that no heat can escape from underneath the plastic. Leave the other plot (control) fallow. Record the weather during the following weeks (sunny / cloudy / rain). After 4 to 6 weeks, the plastic can be removed. Sow 200 seeds in each plot. Apply normal irrigation and other nursery practices (not spraying!) during the seedling raising period. If needed, construct a cage with screen net to keep insects out.

Observations:

At removal of the plastic, check the plastic in the soil solarisation experiment for holes. Record whether there are holes (these will negatively influence the effect of the solarisation). Depending on the crop, after 4 to 6 weeks, record numbers of growing seedlings, of weeds and of diseased seedlings. Calculate the overall percentage of healthy seedlings. Randomly uproot 25 seedlings per treatment. Assess the average number of leaves per seedling and measure root length. Prepare presentations in the form of an agro-ecosystem analysis poster with, per treatment, a drawing of a seedling and all the data grouped around the seedling.

Also carefully remove the soil from the roots of the 25 seedlings per treatment. Observe any root deformations. Make a bar graph per treatment of the number of growing seedlings, the number of wilted seedlings and the number of seedlings with root disease over time and present the findings.

Discussion:

? What are the differences between the different treatments?
? Was there a difference in seedling growth and health? Why?
? What is the cost and labour needed for the solarisation?
? What will happen to the beneficials (incl. natural enemies) in the topsoil?

- ? What are other advantages and disadvantages of the treatment? Was there control of disease? Why?
- ? Was there control of the disease? Why?

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7.29 Test effect of soil solarisation in the field

Objective: To test soil solarisation in the management of clubroot in the field (see illustration in the exercise on soil solarisation to manage leaf spot diseases, page 246)

Materials needed:

- A clubroot infected field
- Transparent polyethylene plastic (about 50 µm)
- Healthy seeds or seedlings of a susceptible cultivar

Procedure:


Soil solarisation experiment

Prepare a field of about 100 m^2 (+ border) according to farmers' practices, including harrowing and fertilising. Measure two plots of 5 x 7.5 m^2 each (make sure that the field is level, in order to avoid water flow from the solarised to the control plot). Irrigate one plot and apply the transparent plastic. Make sure that the edges of the plastic are buried well into the soil so that the wind cannot lift the plastic. Label the plot "solarised". Leave the other plot fallow (label "control"). After 4 to 6 weeks (depending on the ambient temperatures), the plastic can be removed. Before removal of the plastic, check the plastic for holes. Sow or plant seeds or seedlings in each plot. Make sure that the seedlings are not infected with any root disease! Follow farmers' practices in crop management during the rest of the season.


Cabbage infected by clubroot in field which was not solarised

Observations:

Monitor both plots weekly and observe number of plants with yellowing leaves or wilting symptoms. At harvesting, randomly uproot 25 plants per treatment and count the number of plants with root deformations. Assess final yield.


Good crop performance in soil solarised field

Discussion:

? What are the differences between the two treatments?
? Was there reduction of disease? Why?

- Was there difference in yield? Why?
- What are the advantages and disadvantages of the solarisation treatment?
- What is the difference in cost and labour needed?

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7.30 Effect of liming on clubroot of cabbage

Objective: To test liming in the management of clubroot of cabbage

Materials needed:

- A clubroot infected field
- Cabbage seeds
- Lime
- If available: soil pH meter or a soil test kit

Procedure:

Prepare a field of about 75 m^2 (+ border) according to farmers' practices. If a soil pH meter is available: cross the field diagonally and take samples for measuring soil pH at regular intervals (e.g. every 5 metres). Take samples until 15 cm deep, mix samples (at least 10) and measure average pH using the pH meter or a soil test kit.

Measure two plots of 5 x 7.5 m^2 each (make sure that the field is level, in order to avoid water flow from limed to control plot). Calculate how much lime is needed for the surface of one plot, using the below table:

Hydrated lime rates at different soil pH levels (source: Sherf and Macnab)

Soil pH	Hydrated lime required (kg/ha)	Hydrated lime required (kg/37.5 m^2)
5.0	5600	21
5.5	4490	17
6.0	3370	13
6.5	2240	8
7.0	1680	6
7.2	1680	6
8.0	0	0

Apply lime in one plot (limed treatment) and work lime into the soil. Preferably leave the field for about one month before planting the cabbage crop. Raise 200 seedlings of cabbage in a disease-free nursery (use subsoil?) for about 30-40 days. Check for disease! Transplant 100 seedlings in the limed treatment and 100 seedlings in the un-limed, control treatment. Follow local farmers' practices in fertilising and irrigating the crop.

Limed treatment in the Philippines

Unlimed treatment in the Philippines

Observations:

Monitor the field weekly and observe number of plants with yellowing leaves or wilting symptoms. If possible, measure soil-pH every two weeks. At harvesting, randomly uproot 25 plants per treatment and count the number of plants with clubroot symptoms. Assess final yield and weight per cabbage head. Assess the soil pH again in each treatment after the final harvest.

Discussion:

- What are the differences between the two plots?
- Was there control of disease? Why?
- Was there a difference in yield? Why?
- Why did we monitor soil pH?
- What are the advantages and disadvantages of the liming treatment?
- What is the additional cost and labour needed for the liming treatment?
- Would the liming be cost-effective?

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7.31 Spread of cabbage soft rot by farming tools

Objective: To study the spread of cabbage soft rot through mechanical injury

Materials needed:

- Cabbage field with soft rot incidence
- 4 Potted young plants (healthy!) of a susceptible cultivar
- Scissors
- Alcohol
- Clean (tap) water
- 2 Big plastic bags
- Labels and tape

Procedure:

Bring the potted plants into the classroom. Water the plants. Label the potted plants as follows:

A. Healthy control (dry)
B. Healthy control (wet)
C. Soft rot infected (dry)
D. Soft rot infected (wet)

Clean the scissors with alcohol. After cleaning, dip them in clean (tap) water. Use the scissors to cut the leaves of both "healthy control" plants (A and B). Make incisions in different parts of several leaves so that in one leaf the main vein is cut, in another leaf only a side vein, in another leaf only intra-veinal cuts, in another leaf cuts into the edges of leaves. Before each new cut, clean the scissors with alcohol and dip them in water. Cover plant B with the plastic bag and secure the bag with tape.

Visit the soft rot infested field and collect plants or plant parts with clear leaf spot symptoms. Bring the plants to the classroom. Clean the scissors with alcohol. After cleaning, dip them in clean (tap) water. Use the scissors to cut once across a rotting lesion and immediately afterwards cut a leaf of plant C. Before each new cut, clean the scissors and again cut across a lesion and immediately afterwards a leaf of plant C. Make incisions in different parts of several leaves so that in one leaf a main vein is cut, in another leaf only a side vein, in another leaf only intra-veinal cuts, in another leaf cuts into the edges of leaves. Before each new cut, clean the scissors with alcohol and dip them in water. Repeat this for plant D. Cover plant D with the plastic bag and secure the bag with tape.

Keep the potted plants in the classroom and water when necessary.

Observations:

Monitor the four plants and observe any development of lesions on the leaves.

Discussion:

- Was there transmission of disease? Why?
- How would the disease spread in the field?
- Was there a difference between the ‘wet’ and ‘dry’ treatments? If so, what does this mean?

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7.32 Effect of rain on the spread of cabbage soft rot

This exercise should be conducted during the rainy season.

Objective: To study the effect of rain on the spread of cabbage soft rot

Materials needed:

- Plastic roof of about 1 m high, to cover 5 to 10 plants in a row
- Cabbage field (young crop, susceptible cultivar). Make sure that the cabbage field has only a few sources of infection (very few soft rot infected plants)

Procedure:

Each group of participants could install and monitor one roofed and one control treatment. Install the plastic roof in one row in the cabbage field and label the roofed plot "Without rain". Choose another plot of 5 to 10 plants, in another row at e.g. 1 m distance from the "Without rain" plot and label the plot "With rain". The roofed treatment should be irrigated, if needed, on the soil (avoid soil splash!). If there is lack of rain, simulate rain with daily overhead irrigation in the "With rain" plot. Do not spray with fungicides in either plot. All other crop management practices (such as fertilisation) should be the same in both plots.

Observations:

Weekly observe the plants in both plots. Check the number of plants with leaf spots, the number of leaf spots per plant and/or number of leaves with leaf spots per plant. Note the location of leaves with spots (lower, middle or top part of the plant). After 2 or 3 months, prepare to present the results by plotting the data over time in graphs. Compare the development of disease over time in the different plots and present the results per group of participants by plotting the data over time in graphs.

Discussion:

? What are the differences between the roofed and non-covered plants? Why?
? Were there differences in numbers of plants with rot symptoms? Why?
? Is the cabbage soft rot disease generally more problematic during rainy seasons? Why?
? What is the effect of rain?

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7.33 Sanitation measures to manage cabbage soft rot

Objective: To test the impact of sanitation on spread of bacterial soft rot diseases

Materials needed:

- Cabbage field with very early soft rot infection
- Large plastic bags

Procedure:

Start the experiment when the crop is still young. Allocate two plots of each about 100 plants and label one plot "Without sanitation (control)" and the other plot "With sanitation". Maintain a border between the two plots of about 1 m wide.

In the plot without sanitation, do not remove plants when they wilt and die. In the plot with sanitation, weekly monitor the plants and when plants wilt, pull them out. Insert the pulled plants into a plastic bag and remove them from the field in order to avoid spread of the infection. Check for mould at the soil surface and on the plant. The wilting plant must be removed with the soil attached to the roots.

Gathering crop residues in Vietnam after harvest to mix with manure for composting

Crop residues with among others black rot infection

Observations:

Record the weekly observed percentage infected plants in the "Without sanitation" plot and the accumulated percentage infected (=removed) plants of the "With sanitation" plot. At harvesting, assess the final yield and market grade of both plots.

Discussion:

? Was the disease present in both plots?
? Where did the disease come from?
? Were there differences in disease incidence between both plots? If yes, why?
? Were there differences in yield between both plots? If yes, why?
? What are different, appropriate methods to get rid of the infected plant material?
? What are the costs and benefits of the sanitation practice?

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