PART 1: BASIC TOOLS AND GENERAL TECHNIQUES

Chapter 1: Equipment and Collecting Methods

Collecting methods may be divided into two broad categories. In the first, a collector actively finds and collects the insects with the aid of nets, aspirators, beating sheets, or whatever apparatus suits particular needs. In the second, a collector participates passively and permits traps to do the work. Both approaches may be used simultaneously, and both are discussed in the following pages. Using as many different collecting methods as possible will permit a collector to obtain the greatest number of specimens in the shortest period of time.

Catching specimens by hand may be the simplest method of collecting; however, this method is not always productive because of the evasive behavior of many insects. Some insects are not active at times and places that the collector finds convenient. Some insects cause injury or discomfort through bites, stings, repulsive chemicals, or urticating setae. Often, particular kinds of equipment and special methods are needed. Equipment and methods described here have general application. Advanced studies of specific insect or mite groups may have developed unique procedures for collecting and surveying. For example, Agosti (2001) outlines procedures for surveying ground-nesting ant biodiversity. Clever collectors will make adaptations to fit their specific purposes and resources.

The equipment used to assemble an insect or mite collection is not necessarily elaborate or expensive. In many instances, a collecting net and several killing bottles will suffice. However, additional items will permit more effective sampling of a particular fauna. Many collectors carry a bag or wear a vest in which they store equipment. The following items usually are included in the general collector's bag.

1.1 Equipment

1.1.1 Forceps

Fine, lightweight forceps are strongly recommended for any collector. Specialized forceps may be selected depending upon individual needs (Fig. 1). Lightweight spring-steel forceps are designed to prevent crushing of fragile and small insects. Extra-fine precision may be obtained with sharp-pointed "watchmaker" forceps; however, care must be taken not to puncture specimens. When possible, grasp specimens with the part of the forceps slightly behind the points. Curved forceps often make this easier. When the forceps are not in use, their tips should be protected. This can be accomplished by thrusting the tips into a small piece of styrofoam or cork, or by using a small section of flexible tubing as a collar.

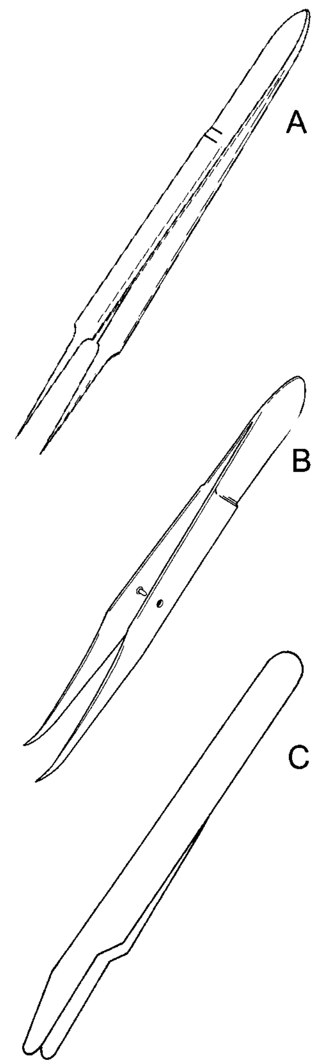


Figure 1.1
Forceps for insect collecting.
A. fine watchmaker forceps
B. curved metal collecting forceps
C. soft forceps

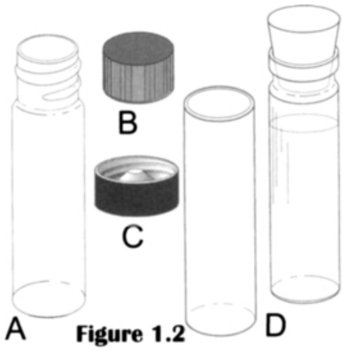


Figure 1.2
Sample Vials.

1.1.2 Sample Vials

Sample vials of various sizes containing alcohol or other preservatives are necessary for collecting many species and life stages of insects and mites. Leak-proof caps are recommended for both field and permanent storage (Fig. 1.2).

1.1.3 Killing Bottles

Killing bottles or killing jars in various sizes are important to preserve specimens quickly (Fig. 1.3).

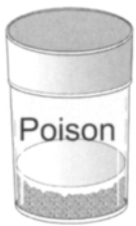


Figure 1.3
An example of a killing bottle appropriately affixed with a "Poison" label.

1.1.4 Small Containers

Small crush-proof containers are necessary for storing and protecting specimens after their removal from killing bottles (Fig. 1.4). These containers may be made of cardboard, plastic, or metal and should be partly filled with soft tissue paper to keep specimens from becoming damaged. Some collectors do not recommend the use of cotton in storage containers because specimens become entangled in the fibers and may become virtually impossible to extricate without damage. However, some collectors of minute or fragile insects find that specimens stored in a few wisps of cotton are better protected from damage.

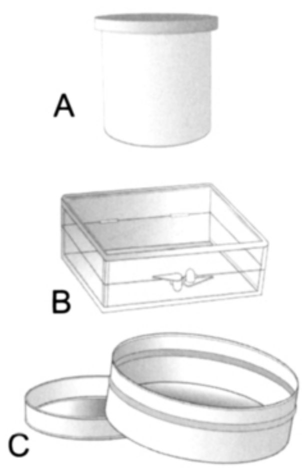


Figure 1.4
Examples of small crush-proof collecting containers.
A. empty film canister
B. plastic box
C. tin pill box

1.1.5 Small Envelopes

Small envelopes are useful for temporary storage of delicate specimens (Fig. 1.5). Specially designed glassine envelopes, which prevent undue dislodging of butterfly and moth scales, are available from biological supply houses.

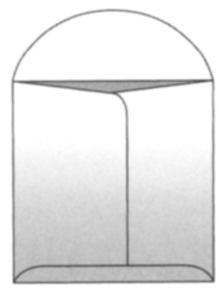


Figure 1.5
Glassine envelope designed for field storage of moths and butterflies.

1.1.6 Aspirators

Aspirators are necessary for collecting many kinds of small-bodied or agile insects and mites.

1.1.7 Absorbent Tissue

Absorbent tissue is highly recommended for use in killing bottles and aspirators.

1.1.8 Notebook

A notebook and writing equipment are essential for jotting down notes and label data (Fig. 1.6).

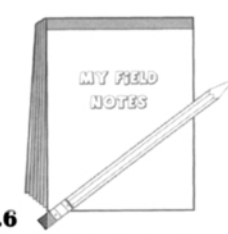
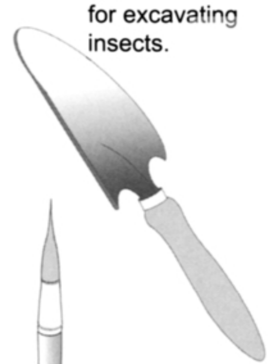


Figure 1.6
An example of a field journal for recording specimen label data and other notes.

1.1.9 Tools for Cutting or Digging

A knife or plant clippers or both are necessary for opening galls, seed pods, twigs, and other kinds of plant material. In addition, a small gardener's trowel (Fig. 1.7) for some kinds of excavation and a heavy knife or small hatchet may be helpful for searching under bark or in decaying logs.

Figure 1.7
A gardener's trowel for excavating insects.



1.1.10 Brush

A small, fine brush (camel's hair is best) is needed to aid in collecting minute specimens (Fig. 1.8). By moistening the tip, tiny specimens will adhere to it and may be quickly transferred to a killing bottle or vial.

Figure 1.8
A fine-tip camel's-hair brush.

1.1.11 Bags

Bags for retrieving plant material, rearing material, or Berlese samples are a good idea (Fig. 1.9). Remember that samples stored in plastic may decompose within a few short hours. Samples must be transferred to more permanent containers immediately upon returning from the field. For collecting much plant material, a botanist's vasculum (tin box) is advisable.

1.1.12 Hand Lens

A hand lens is helpful and will quickly become an indispensable aid to collectors (Fig. 1.10). A lens worn on a lanyard is convenient and prevents its loss while in the field.

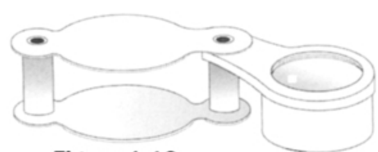


Figure 1.10
A collapsible hand lens.

1.1.13 Summary

This list may be modified according to the special kinds of insects or mites to be collected. For example, a plant press to prepare plant specimens for determination or as voucher specimens, especially when leaf-mining insects are studied, may be needed. When collecting at night, a flashlight or headlamp is essential; the latter is especially useful because it leaves the hands free.

Much of the basic collecting equipment may be obtained from ordinary sources, but equipment especially designed for collecting insects often must be bought from biological supply houses. In some cases, addresses may be found on the Internet or in the yellow pages of telephone directories under "Biological Laboratory Supplies" or "Laboratory Equipment and Supplies." Biological and entomological publications often carry advertisements of equipment suppliers. Because these firms are located in many parts of the country and change names and addresses fairly often, it is not practical to list them here. Biologists at a local university usually can recommend a supplier in their area.

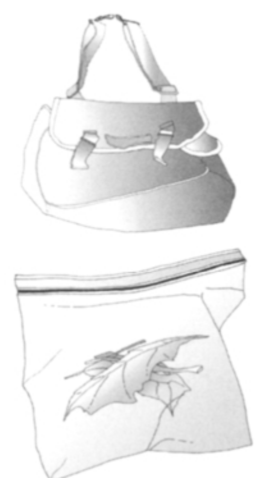


Figure 1.9
Bags for collecting miscellaneous plant or soil materials from the field.

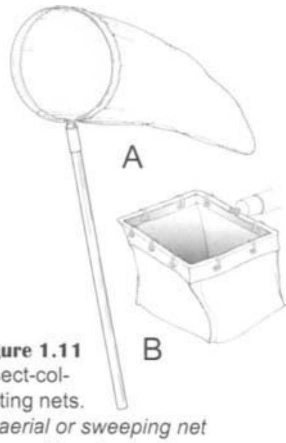


Figure 1.11 Insect-collecting nets.
A. aerial or sweeping net
B. aquatic net

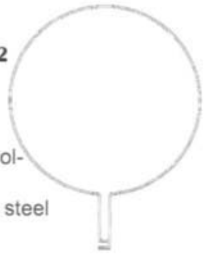


Figure 1.12 A common design for the rim of an insect-collecting net made from steel wire.

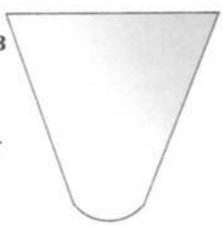


Figure 1.13 General shape of the bag on an aerial or sweep net.

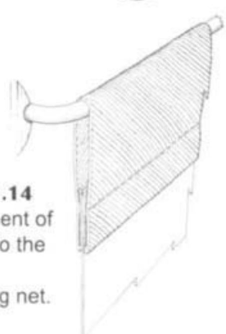


Figure 1.14 Attachment of the net to the rim of a collecting net.

1.2 Collecting Nets

Collecting nets come in three basic forms: aerial, sweeping, and aquatic (Fig. 1.11). The aerial net is designed especially for collecting butterflies and large-bodied flying insects. Both the bag and handle are relatively lightweight. The sweeping net is similar to the aerial net, but the handle is stronger and the bag is more durable to withstand being dragged through dense vegetation. The aquatic net is used for gathering insects from water and is usually made of metal screening or heavy scrim with a canvas band affixed to a metal rim. A metal handle is advisable because wooden ones may develop slivers after repeated wetting. The net chosen depends on the kind of insects or mites intended for collection.

Several kinds of nets, including collapsible models with interchangeable bags, are available from biological supply houses, but anyone with a little mechanical ability can make a useful net. The advantage of a homemade sweep net is that its size and shape can be adapted to the needs of the user, to the kind of collecting intended, and to the material available. Net-constructing materials include the following.

1. A length of heavy (8-gauge) steel wire for the rim, bent to form a ring 30–38 cm in diameter (Fig. 1.12). Small nets 15 cm or smaller in diameter sometimes are useful, but nets larger than 38 cm are too cumbersome for most collecting.

2. A strong, light fabric, such as synthetic polyester, through which air can flow freely. Brussels netting is best but may be difficult to obtain; otherwise, nylon netting, marquisette, or good-quality cheesecloth can be used. However, cheesecloth snags easily and is not durable. The material should be folded double and should be 1.5–1.75 times the rim diameter in length (Fig. 1.13). The edges should be double-stitched (French seams).

3. A strip of muslin, light canvas, or other tightly woven cloth long enough to encircle the rim. The open top of the net bag is sewn between the folded edges of this band to form a tube through which the wire rim is inserted (Fig. 1.14).

4. A straight, hardwood dowel about 19 mm in diameter and 105–140 cm long (to suit the collector). For attachment of the rim to the handle, a pair of holes of the same diameter as the wire are drilled opposite each other to receive the bent tips of the wire, and a pair of grooves as deep and as wide as the wire are cut from each hole to the end of the dowel to receive the straight part of the wire (Fig. 1.15).

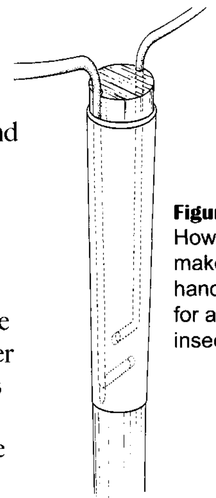


Figure 1.15 How to make a handle for an insect net.

5. A tape or wire to lash the ends of the rims tightly into the grooves in the end of the handle. This may be electrician's plastic tape or fiber strapping tape commonly used for packaging. If wire is chosen, the ends should be bound with tape to secure them and to keep them from snagging. A close-fitting metal sleeve (ferrule) may be slipped over the rim ends and held in place with a small roundheaded screw instead of tape or wire lashing.

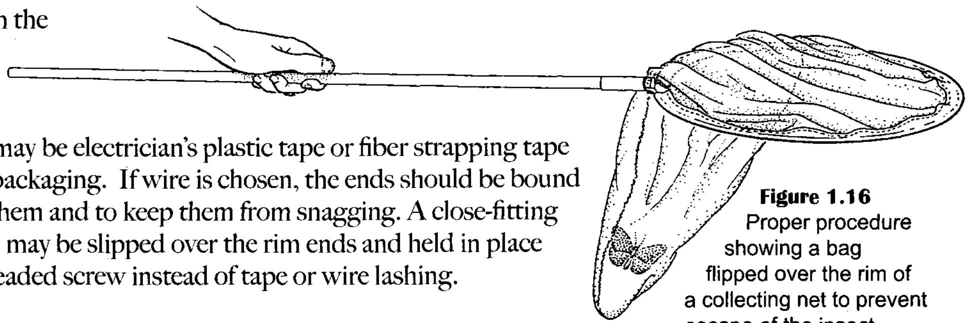


Figure 1.16
Proper procedure showing a bag flipped over the rim of a collecting net to prevent escape of the insect.

After the net has been placed on the rim, the ends of the band should be sewn together and the rim ends fastened to the handle. The other end of the handle should be filed to remove sharp edges. The net is then ready for use.

Efficient use of a net is gained only with experience. Collection of specimens in flight calls for the basic stroke: Swing the net rapidly to capture the specimen, then follow through to force the insect into the very bottom of the bag. Twist the wrist as you follow through so that the bottom of the bag hangs over the rim (Fig. 1.16); this will entrap the specimen. If the insect is on the ground or other surface, it may be easier to use a downward stroke, quickly swinging down on top of the specimen. With the rim of the net in contact with the ground to prevent the specimen from escaping, hold the tip of the bag up with one hand. Most insects will fly or walk upward into the tip of the bag, which can then be flipped over the rim to entrap the specimen.

Sweeping the net through vegetation, along the sand and seaweed on beaches, or up and down tree trunks will catch many kinds of insects and mites. An aerial net may be used in this way, but the more durable sweeping net is recommended for such rough usage. After sweeping with the net, a strong swing through the air will concentrate anything into the tip of the bag, and then, by immediately grasping the middle of the net with the free hand, the catch will be confined to a small part of the bag. Only the most rugged sweeping net may be used through thistles or brambles. Even some kinds of grasses, such as sawgrass, can quickly ruin a fragile net. Burrs and sticky seeds are also a serious problem.

The catch may be conveyed from the bag to a killing jar in a number of ways. Single specimens are transferred most easily by lightly holding them in a fold of the net with one hand while inserting the open killing jar into the net with the other. While the jar is still in the net, cover the opening until the specimen is overcome; otherwise, it may escape before the jar can be removed from the net and closed. To prevent a butterfly from damaging its wings by fluttering in the net, squeeze the thorax gently through the netting when the butterfly's wings are closed (Fig. 1.17). This will temporarily paralyze the insect while it is

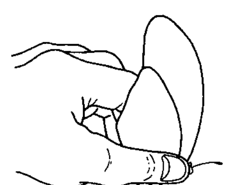


Figure 1.17
Technique for paralyzing a butterfly by squeezing its thorax between thumb and index finger.

being removed to the killing jar. Experience will teach you how much pressure to exert. Obviously, pinching small specimens of any kind is not recommended. When numerous specimens are in the net after prolonged sweeping, it may be desirable to put the entire tip of the bag into a large killing jar for a few minutes to stun the insects. They may then be sorted and desired specimens placed separately into a killing jar, or the entire mass may be dumped into a killing jar for later sorting. These methods of mass collecting are especially adapted to obtaining small insects not readily recognizable until the catch is sorted under a microscope.

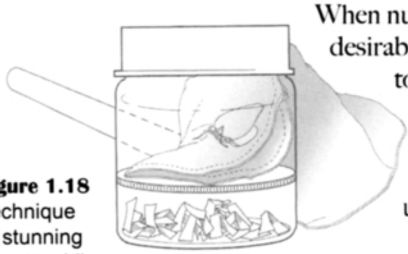


Figure 1.18 Technique of stunning insects while in collecting nets by using the killing jar.

Removal of stinging insects from a net can be a problem. Wasps and bees often walk toward the rim of the bag and may be made to enter a killing jar held at the point where they walk over the rim. However, many insects will fly as soon as they reach the rim, and a desired specimen may be lost. A useful technique involves trapping the insect in a fold of the net, carefully keeping a sufficient amount of netting between fingers and insect to avoid being stung. The fold of the net can then be inserted into the killing jar to stun the insect (Fig. 1.18). After a few moments, the stunned insect may be safely removed from the net and transferred to a killing jar. If the stunned insect clings to the net and does not fall readily into the jar, pry the insect loose with the jar lid or forceps. Do ***not*** attempt this maneuver with fingers because stunned wasps and bees can sting reflexively.

Several special modifications are necessary to adapt a net for aquatic collecting. Aerial nets made of polyester or nylon may be used to sweep insects from water if an aquatic net is not available. The bag will dry quickly if swept strongly through the air a few times. A wet net should not be employed for general collecting until thoroughly dry, or other specimens, especially butterflies, may be damaged.

For more specialized collecting, nets can be adapted in many ways. Nets can be attached to the ends of beams that are rotated about their midlength by a motor drive. Nets also can be adapted to be towed by or mounted on vehicles (Fig. 1.19).

Suggested Reading:

Collecting Nets

- Riley 1957
- McNutt 1976
- Martin 1977
- Rudd & Jensen 1977
- Dunn & Reeves 1980
- LeSage 1991

Vehicle Mounted Nets

- Traver 1940
- Gressitt et al. 1961
- Sommerman & Simmet 1965
- Sommerman 1967
- Hill 1971
- Almand et al. 1974
- Rudd & Jensen 1977
- Holzapfel et al. 1978
- Kronblad & Lundberg 1978
- Barnard 1979

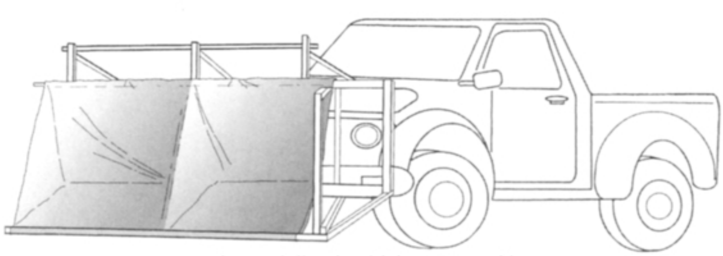


Figure 1.19 A specialized vehicle-mounted insect net.

1.3 Killing Containers and Agents

Killing an insect quickly is paramount to the preservation of a nice specimen. If an insect is allowed to beat its wings or crawl about for extended periods, it will inevitably harm its wings, break legs and antennae, or lose color.

1.3.1 Freezing Insects

When collecting around the home or school, insects can be killed effectively with minimum damage by placing them immediately into a freezer. This method has two distinct advantages: First, no messy and potentially dangerous chemicals are needed; second, insects may be left in the freezer for long periods of time and need only be thawed before pinning. This convenience alone makes the freezer method attractive to many collectors.

1.3.2 Injecting Insects with Alcohol

Most large insects and especially large moths are best killed with an injection of alcohol using a hypodermic needle and syringe. For most insects, inject into the ventral area of the thorax. For large beetles, inject alcohol into a coxal socket. Less than 1 cm^3 of alcohol is usually sufficient to kill large specimens.

1.3.3 Killing Jars for Field Collecting

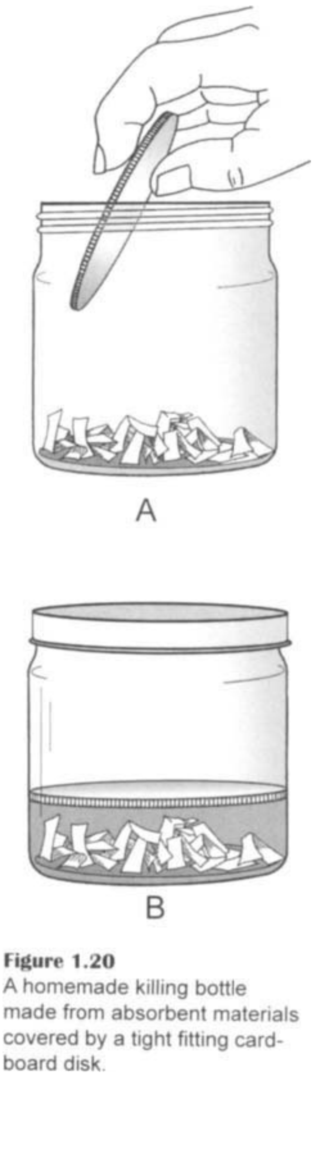


Figure 1.20
A homemade killing bottle made from absorbent materials covered by a tight fitting cardboard disk.

Any heavy, widemouthed glass jar or bottle with a tight-fitting stopper or metal screw top may be used as a killing container (Fig. 1.20). Glass is preferred over plastic, and jars with relatively thick glass are preferred over thin, fragile glass, for obvious reasons. Olives frequently are sold in bottles that make convenient killing containers. Tops that may be removed with only a quarter turn often are preferred but may not be obtained readily. This type of lid may be quickly removed and returned to the jar with a minimum of effort. Collectors interested in taking minute or small insects may prefer using small vials that can be carried in a shirt pocket. Parallel-sided vials can be closed with cork stoppers. When collecting small-bodied insects in vials, care must be exercised to ensure that the stopper seats firmly against the wall of the vial. Otherwise, specimens become wedged between the glass and stopper, resulting in damage to some specimens. A crumpled piece of tissue paper placed in the vial helps to maintain the specimens clean and disentangled.

Jars for use with liquid killing agents are prepared in one of two ways. One way is to pour about 2.5 cm of plaster of Paris mixed with water into the bottom of the jar and allow the plaster to dry without replacing the lid. Sufficient killing agent is then added to saturate the plaster; any excess should be removed. The lid is then replaced. This kind of jar can be recharged merely by adding more killing agent. A second method is to place a wad of cotton, paper, or other absorbent material in the bottom of a jar, pour enough liquid killing agent into the jar to nearly saturate the absorbent material, and then press a piece of stiff paper or cardboard, cut to fit the inside of the jar tightly, over it (Fig. 1.20). The paper or cardboard acts as a barrier between the insect and the killing agent, preventing the specimen from contacting the agent directly and keeping the agent from evaporating too rapidly.

1.3.4 Liquid Killing Agents

Popular liquid killing agents include ethyl acetate ($CH_3CO_2 \cdot C_2H_5Z$), carbon tetrachloride (CCl_4), ether (diethyl ether, $C_2H_5 \cdot O \cdot C_2H_5$), chloroform ($CHCl_3$), and ammonia water ($NH_4OH$ solution). Ethyl acetate is recommended by many entomologists as the most satisfactory liquid killing agent. The fumes of ethyl acetate are less toxic to humans than the fumes of the other substances. Ethyl acetate usually stuns insects quickly but kills them slowly. Specimens, even though they appear dead, may revive if removed from the killing jars too soon. However, a compensating advantage is that most specimens may be left in an ethyl acetate killing jar for several days and remain pliant. (If the ethyl acetate is allowed to evaporate from the killing jar, specimens will harden.) For these reasons, a killing jar with ethyl acetate is preferred by many entomologists over a cyanide jar, especially when the jar is used infrequently.

Hobbyists and other collectors have found that fingernail polish remover (acetone) also works as an alternative to ethyl acetate and is more commonly available, especially in an emergency situation.

Carbon tetrachloride was once very popular as a liquid killing agent because it is not flammable and was easily obtained as a spot remover for clothes. Carbon tetrachloride is no longer recommended, however, because it is a carcinogen and a cumulative liver toxin. Specimens killed with carbon tetrachloride often become brittle and difficult to pin.

Ether and chloroform are extremely volatile and flammable and should not be used near an open flame or lighted cigarette. Their high volatility makes them serviceable in a killing jar for only a short period of time. Perhaps the greatest hazard with chloroform is that even when stored in a dark-colored jar, it eventually forms an extremely toxic gas called phosgene (carbonyl chloride, $COCl_2$). Chloroform, however, is useful when other substances cannot be obtained. Chloroform stuns and kills quickly, but it has the disadvantage of stiffening specimens.

Ammonia is irritating to humans, does not kill insects very effectively, and spoils the colors of many specimens. However, ammonia is readily available and will serve in an emergency. Ammonium carbonate, a solid but volatile substance, also may be substituted.

Spray-dispensed insecticides may be used, if not to kill specimens, to at least "knock them down" into a container from which they may be picked up. If they are directed into a container topped with a funnel, they may be allowed to revive and treated further as desired (Clark & Blom 1979).

Killing agents may be any of various liquids. Never deliberately inhale the fumes, even momentarily. All killing agents are to some extent hazardous to human health. All killing jars or bottles should be clearly labeled "**POISON**" and kept away from persons unaware of their danger (Fig. 1.21).

Collectors who travel on airlines to collecting sites, especially on international flights, should be aware that the transportation industry now prohibits poisons on commercial passenger airliners in many parts of the world. Security at airports continues to increase, and the likelihood of being questioned about toxicants within carry-on baggage is high. Customs officials may also object to the importation of killing agents such as cyanides and ethyl acetate. When an international collecting trip is anticipated, travel agents and consulate officials should be consulted.

Never deliberately inhale the fumes, even momentarily.

All killing agents can be hazardous to human health.

All killing jars or bottles should be clearly labeled "POISON" and kept away from persons unaware of their danger!

1.3.5 Solid Killing Agents

The solid killing agents used in killing jars are cyanides: potassium cyanide (KCN), sodium cyanide ($NaCN$), and calcium cyanide ($Ca(CN)_2$). Potassium cyanide is the compound of choice among many collectors. Sodium cyanide is equally effective but it is hygroscopic (it absorbs water and makes the jar wet). Calcium cyanide is seldom available. Handle all cyanide compounds with extreme care. They are dangerous, rapid-acting poisons with no known antidote. If even a single grain touches the skin, wash the affected area immediately with water. To avoid handling the cyanide and storing or disposing of surplus crystals, you may be able to find a chemist, pharmacist, or professional entomologist who can make the killing jar for you. If this is not feasible, use utmost care in following the instructions given here.

To make a moderate-sized cyanide killing jar, place cyanide crystals about 15 mm deep in the bottom. A smaller amount of cyanide may be used for smaller jars. Cover the crystals with about 10 mm of sawdust and add about 7 mm of plaster of Paris mixed with water to form a thick paste, working quickly before the plaster solidifies. Then add crumpled absorbent paper to prevent water condensation on the inside surface of the glass. Instead of the plaster of Paris, a

Figure 1.21
An example of a properly labeled killing jar.

plug of paper or cardboard may be pressed on top of the sawdust. Be sure that it fits tightly. When the jar is ready for use, place several drops of water on the plaster or paper plug. In an hour or so, enough fumes of hydrocyanic acid will have been produced to make the jar operative. ***Do not test this by sniffing the open jar!***

Another substance that has been recommended as a killing agent is dichlorvos (2.2-dichloroethenyl dimethyl phosphate), also called DDVP, Vapona, Nogos, Herkol, and Nuvan. Polyvinyl chloride (PVC) resin impregnated with this chemical and sold commercially as bug strips or No-Pest Strips is long lasting and somewhat less dangerous than other killing agents, but its time-release aspect allows only small quantities of the active agent to be released over time, which may be too small to kill quickly. Therefore, PVC-impregnated dichlorvos is more effective as a killing agent in traps than in killing jars.

Every killing jar should be clearly and prominently labeled "**POISON**." The date the killing jar was fabricated can also be placed on the poison label. This will give the collector a rough idea of the life of a killing jar. The bottom outside of the jar must be reinforced with tape, preferably cloth, plastic, or clinical adhesive tape. The tape cushions the glass against breakage and keeps its dangerous contents from being scattered if the container breaks.

Laws involving toxic materials are changing, and local officials should be consulted as how best to proceed.

Killing jars or bottles will last longer and give better results if the following simple rules are observed.

1. Place a few narrow strips of absorbent paper in each killing jar to keep it dry and to prevent specimens from mutilating or soiling each other. Replace the strips when they become moist or dirty. This technique is useful for most insects, except Lepidoptera, which are difficult to disentangle without damage.

2. Do not leave killing jars in direct sunlight because they will "sweat" and rapidly lose their killing power.

3. If moisture condenses in a jar, wipe it dry with absorbent tissue.

4. Keep delicate specimens in separate jars so that larger specimens do not damage them.

5. Do not allow a large number of specimens to accumulate in a jar unless it is to be used specifically for temporary storage.

6. Do not leave insects in a cyanide killing jar for more than a few hours. The fumes will change the colors of some insects, especially yellows to red, and specimens will generally become brittle and difficult to handle.

7. If it is necessary to hold insects for more than several hours, place the specimens in another container and store them in a refrigerator.

8. Keep butterflies and moths in jars by themselves so that the setae and scales that they shed will not contaminate other specimens. Contaminated specimens often make viewing of taxonomic features more difficult. Further, removing scales and setae is time-consuming, often tedious, always annoying, and potentially destructive to the specimen.

9. Never test a killing jar by smelling its contents.

10. Old jars that no longer kill quickly should be recharged or disposed of in a legal and responsible manner. A cyanide jar that has become dry may be reactivated by adding a few drops of water.

Suggested Reading:
Killing Agents
- Lindroth 1957
- Frost 1964
- White 1964
- Pennington 1967
- Preiss et al. 1970
- Clark & Blom 1979
- Banks et al. 1981

1.4 Aspirators and Suction Devices

The aspirator (Fig. 1.22), known in England as a "pooter," is a convenient and effective device for collecting small or highly mobile insects and mites. (The device was named in honor of Frederick W. Poos, an American entomologist who employed the device to collect Cicadellidae.) The following materials are needed to construct an aspirator.

1. A vial 2.5–5 cm in diameter and about 12 cm long.

2. Two pieces of glass or copper tubing about 7 mm in diameter, one piece about 8 cm long and the other about 13 cm long. Copper tubing can be obtained from hobby shops and has the obvious advantage of being less fragile than glass.

3. A rubber stopper with two holes in which the tubing will fit snugly.

4. A length of flexible rubber or plastic tubing about 1 m long, with diameter just large enough to fit snugly over one end of the shorter piece of stiff tubing.

5. A small piece of cloth mesh, such as cheesecloth, and a rubber band.

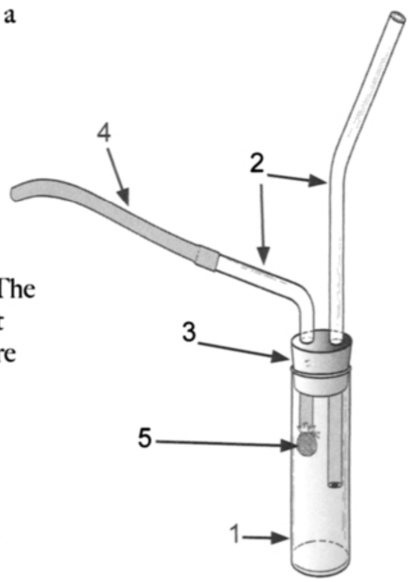


Figure 1.22
Components of a vial aspirator.
1. vial
2. glass or copper tubing
3. rubber stopper
4. rubber or plastic tubing
5. mesh cloth and rubber band

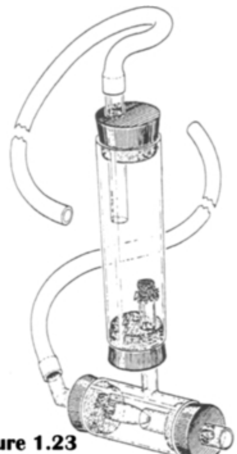


Figure 1.23
A blow-type aspirator.

To make an aspirator, bend the glass tubes as in Fig. 1.22. In bending or cutting glass tubes, always protect your fingers by holding the glass between several layers of cloth. Obtain the advice of a chemist or laboratory technician for cutting and bending glass. Moisten one end of the longer tube and insert it through one of the holes in the rubber stopper. Moisten one end of the shorter tube and insert it through the other hole in the stopper. Next, use a rubber band to fasten the cloth mesh over the end that was inserted through the stopper. (This will prevent specimens from being sucked into the collector's mouth when the aspirator is used.) Attach one end of the flexible tubing to the free end of this tube. The length, size and amount of bend in the tubing will vary according to the user's needs. To complete the assembly, insert the rubber stopper into the vial. To use the aspirator, place the free end of the flexible tubing in the mouth, move the end of the longer glass tube close to a small specimen, and suck sharply. The specimen will be pulled into the vial.

Instead of a vial, some workers prefer a tube. In either method, keep small pieces of absorbent tissue in the vial or tube at all times to prevent moisture from accumulating. Note that there is some danger of inhaling harmful substances or organisms when using a suction-type aspirator (Hurd 1954).

Either the vial- or tube-type aspirator (Fig. 1.22) may be converted into a blow-type aspirator by removing the 13-cm glass tube and substituting a T-shaped attachment (Fig 1.23). The flexible tubing is attached to one arm of the T, the opposite arm is left open, and the stem of the T is inserted into the vial and covered with mesh. Upon blowing through the flexible tubing, a current of air passes across the T and creates a partial vacuum in the vial, which produces the suction needed to draw specimens into the vial. This kind of aspirator eliminates the danger of inhaling small particles, fungus spores, or noxious fumes. Aspirators with a squeeze bulb sometimes may be purchased, or, if a valved bulb can be obtained, they may be used with either pressure or suction.

Collection traps also have been devised with the suction feature applied on a much larger scale than with the usual aspirator. These include the handheld converted dustbuster insect vacuums (Fig. 1.24), the D-vac, (which employs a backpack motor fan) (Dietrick et al. 1959, Dietrick 1961); the Insectavac (Ellington et al. 1984a, b); and another device developed for the collection of honey bees (Gary & Marston 1976). Suction produced by a fan has been employed in traps in conjunction with light or other attractants. Some of these traps are described in the following references and in Section 1.b on ***Traps***. Suction is created by a piston in a "slurp-gun" described for aquatic collecting (Gulliksen & Deras 1975). This principle could be adapted for use in air to gather insects and to deposit them in a vial attached to the side of the piston.

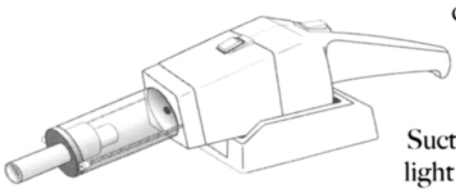


Figure 1.24
A motorized suction device (i.e. Dustbuster) modified to collect insects.

Suggested Reading:

Aspirators and Suction Devices

- Johnson 1950
- Hurd 1954
- Jonasson 1954
- Johnson & Taylor 1955
- Woke 1955
- Johnson et al. 1957
- Lumsden 1958
- Dietrick et al. 1959
- Dietrick 1961
- Minter 1961
- Taylor 1962a
- Singer 1964
- Wiens & Burgess 1972
- Coluzzi & Petrarca 1973
- Sholdt & Neri 1974
- Bradbury & Morrison 1975
- Evans 1975
- Azrang 1976
- Gary & Marston 1976
- Barnard & Mulla 1977
- Ellington et al. 1984a, b
- Summers et al. 1984
- Holtcamp & Thompson 1985
- Belding et al. 1991
- DeBarro 1991
- Governatori et al. 1993
- Moore et al. 1993
- Wilson et al. 1993
- Arnold 1994

1.5 Other Collection Devices

1.5.1 Beating Sheets

A beating sheet should be made of durable cloth, preferably white, attached to a frame about 1 m square, with two pieces of doweling or other light wood crossing each other and fitted into pockets at each corner of the cloth. Variations on the design can include a cloth stretched between two dowels or a triangular cloth that can be operated with one hand (Fig. 1.25). An ordinary light-colored umbrella also may be used as a beating sheet. Place the beating sheet or umbrella under a tree or shrub or limb and sharply beat the branches or foliage with a club or stick. Specimens that fall onto the sheet are easily seen against the light-colored material and may be removed by hand or with forceps, a moistened brush, or an aspirator. Locating specimens on the sheet is sometimes a problem because leaves or other unwanted material also drop onto the sheet. Watching for movement helps to locate specimens. Tilting the sheet displaces debris, leaving the insects and mites clinging to the cloth.

Beating sheets are especially useful for collecting beetles and are particularly effective early and late in the day and when the weather has turned cold. A "ground cloth" also is used in sampling crop fields (Rudd & Jensen 1977) in much the same manner as a beating sheet.

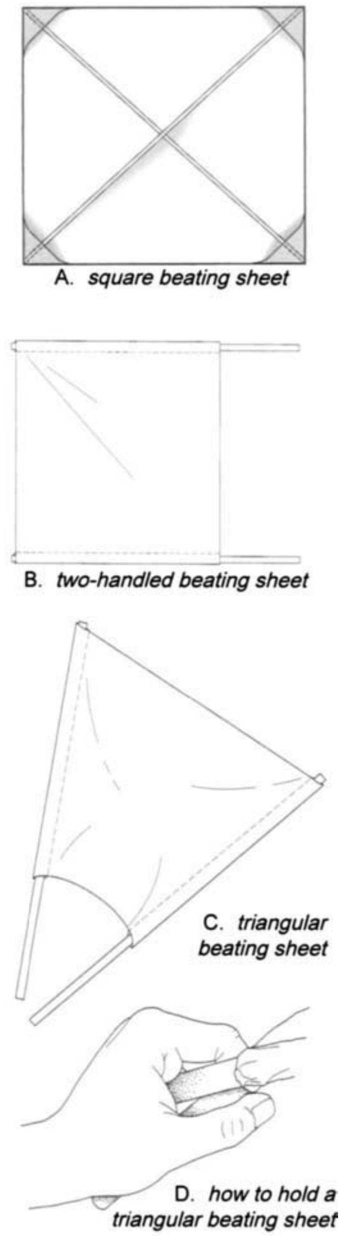


Figure 1.25
Beating sheets.

1.5.2 Drag Cloth

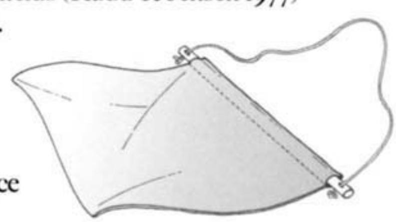


Figure 1.26
Drag cloth.

A drag cloth or flag (Fig. 1.26) made from durable light-colored cloth attached to a piece of doweling along one edge can be dragged through long grass or small shrubs to collect ticks. A variation on this method is to make a flag on a longer handle and to use it to brush against larger trees and shrubs. Questing ticks dislodge easily from the plants and collect on the cloth.

1.5.3 Sifters

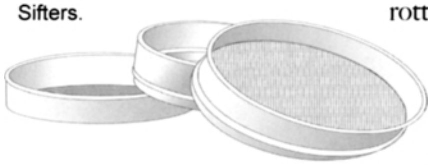


Figure 1.27
Sifters.

Sifters serve to collect insects and mites that live in ground litter, leaf mold, rotting wood, mammal and bird nests, fungi, shore detritus, lichens, mosses, and similar material (Fig. 1.27). Leaf litter sifters can be constructed very simply by collecting leaf litter on a container, shaking the contents through a wire screen such as a cooling rack to separate the large leaves and then retaining the reduced sample. Sifters also are especially useful for winter collecting to pick specimens in diapause.

Almost any container with a wire-mesh screen bottom will serve as a sifter. The size of the mesh depends on the size of the specimens sought. For general purposes, screening with 2.5–3 meshes per centimeter is satisfactory. To use the sifter, place the material to be sifted into the container and shake it gently over a white pan or piece of white cloth. As the insects and mites fall onto the cloth, they may be collected with forceps, a brush, or an aspirator.

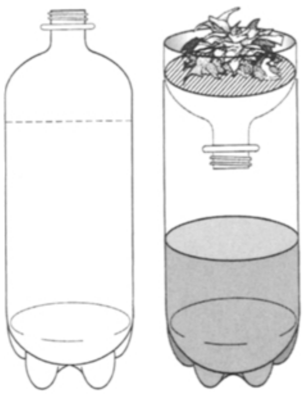


Figure 1.28
Pop bottle extractor.

A similar method is used chiefly to collect mites from foliage. Employing a sifter of 20-mesh screen (about 8 meshes per centimeter) with a funnel underneath that leads to a small vial, beat pieces of vegetation against the screen to dislodge the mites and cause them to fall through the screen and into the vial below.

1.5.4 Separators and Extractors

Somewhat similar to the sifter are various devices designed to separate or extract live specimens from substances in which they may be found, such as leaf mold and other kinds of vegetable matter, shore detritus, and dung. A simple extractor can be constructed from a 2-liter plastic pop bottle (Fig. 1.28). Separators are also used effectively in net sweepings that include so much foreign matter that it is difficult to pick out the insects. These devices usually depend on some physical aid, such as light, heat, or dryness, to impel the insects to leave the substrate. One of the simplest such devices is the sweeping separator (Fig. 1.29), which consists of a carton or wooden box with a tight-fitting lid. A glass jar is inserted near the top of the box on one side. If the jar is made with a screw top, a hole of proper diameter cut in the side of the carton will permit the jar to be screwed onto it. The cover ring, without the lid, from a home-canning jar may be nailed to the periphery of a hole in a wooden box and the jar then screwed onto the ring.

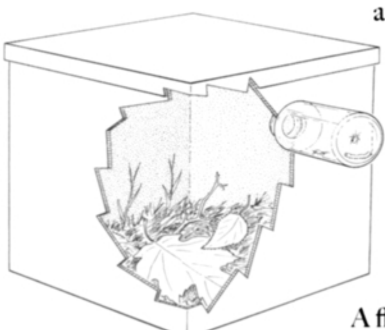


Figure 1.29
Sweeping separator box.

The sweepings are dumped into the box and the cover is quickly closed. A flashlight, table lamp, or other light source should be placed close to the jar. The insects in the darkened box soon will be attracted to the lighted glass jar. When all the insects appear to have entered the jar, it can be removed and its contents put into a killing jar. Alternatively, a jar cover containing a piece of blotting paper soaked with xylene may be placed over the jar for a short while to stun the insects, which may then be sorted.

The Berlese funnel (Fig. 1.30) and its modifications are cleaner and more efficient than sifting to separate insects and mites from leaf mold and similar materials. The sample is placed on a screen near the top of a funnel. A light bulb can be placed above the sample to produce heat and light, which drive the insects downward into the funnel. In other designs, heated coils or a jacket around the funnel can be used to dry the sample and make it inhospitable. The insects and mites are directed by the funnel into a container, sometimes containing alcohol, at the bottom of the funnel. Care should be taken not to dry the sample so rapidly that slow-moving specimens die before they can leave the sample. To prevent large amounts of debris from falling into the container, place the sample on the screen before the container is put in place.

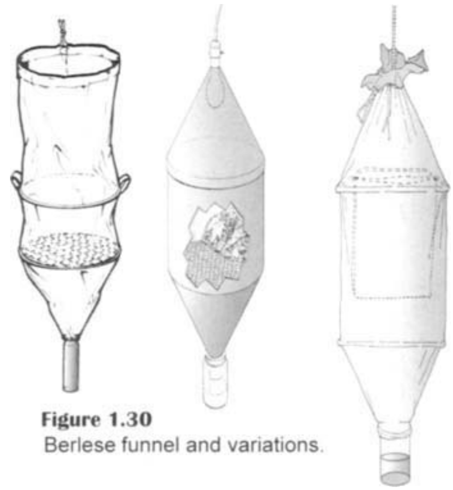


Figure 1.30
Berlese funnel and variations.

1.6 Traps

A trap is any device that impedes or stops the progress of an organism. Traps are used extensively in entomology and may include devices used with or without baits, lures, or other attractants. The performance of a trap depends on its construction, location, time of day or year, weather, temperature, and kind of attractant. A little ingenuity coupled with knowledge of the habits of the insects or mites sought will allow for modifications or improvements in nearly any trap or may even suggest new traps. Only a few of the most useful traps are discussed here; however, Peterson (1964) and Southwood (1979) include extensive bibliographies on insect trapping.

Suggested Reading:

Separators & Extractors

- Salmon 1946
- Kevan 1955, 1962
- Newell 1955
- Kempson et al. 1962
- Murphy 1962
- Woodring 1968
- Brown 1973
- Merritt & Poorbaugh 1975
- Gruber & Prieto 1976
- Lane & Anderson 1976
- Masner & Gibson 1979
- Arnett 1985
- Zolnerowich et al. 1990

Traps

- Barber 1931
- Williams & Milne 1935
- Chamberlin 1940
- Fleming et al. 1940
- Huffacker & Back 1943
- Broadbent et al. 1948
- Johnson 1950
- Williams 1951
- Whittaker 1952
- Dales 1953
- Dodge & Seago 1954
- Williams 1954
- Eastop 1955
- Woke 1955
- Heathcote 1957a
- Banks 1959
- Dodge 1960
- Race 1960
- Fredeen 1961
- Glasgow & Duffy 1961
- Gressitt et al. 1961
- Harwood 1961
- Morris 1961
- Taylor 1962b
- Hollingsworth et al. 1963
- Frost 1964
- Hanec & Bracken 1964
- Peterson 1964
- Thorsteinson et al. 1965
- Turnbull & Nicholls 1966
- Bidlingmayer 1967
- Kimerle & Anderson 1967
- Everett & Lancaster 1968
- Hartstack et al. 1968
- Nijholt & Chapman 1968
- Heathcote et al. 1969
- Herting 1969
- Thompson 1969
- Catts 1970
- Dresner 1970
- McDonald 1970
- Clinch 1971
- Gojmerac & Davenport 1971
- Hansens et al. 1971
- Nakagawa et al. 1971
- Emden 1972
- Pickens et al. 1972
- A'Brook 1973
- Ford 1973
- Klein et al. 1973
- Hienton 1974
- Weseloh 1974
- Yates 1974
- Acuff 1976
- Hargrove 1977
- Lammers 1977
- Martin 1977
- Rogoff 1978 *(includes extensive bibliography)*
- Stubbs & Chandler 1978 *(collecting and recording on pp.1-37)*
- Barnard 1979
- Meyerdirk et al. 1979
- Southwood 1979
- Hafraoui et al. 1980
- Howell 1980
- Sparks et al. 1980 *(includes pictures of several types of traps)*
- Banks et al. 1981

Suggested Reading:

Light Traps
- Glick 1939, 1957
- Frost 1957, 1958
- Blakeslee et al. 1959
- Meyers 1959
- Goma 1965
- Coon & Pepper 1968
- Fincher & Stewart 1968
- Stewart & Lam 1968
- Callahan et al. 1972
- Hocking & Hudson 1974
- Roling & Kearby 1975

Windowpane Traps
- Chapman & Klinghorn 1955
- Corbet 1965
- Lehker & Deay 1969
- Wilson 1969

Interception Nets and Barriers
- Parman 1931, 1932
- Gordon & Gerberg 1945
- Leech 1955
- Nielsen 1960
- Merrill & Skelly 1968
- Gillies 1969
- Nielsen 1974 *(describes catches of insects in funnel traps on trunks of beech trees)*
- Steyskal et al. 1981

The elevation or height above ground at which the trap is placed can be important in affecting the performance of traps, especially light traps. Optimum trap placement is a complex issue with many variables, including the behavior of the targeted insect, specific locality, and size and color of the trap, influencing its performance.

1.6.1 Windowpane Trap

This simple and inexpensive trap (Fig. 1.31) involves a barrier consisting of a windowpane held upright by stakes in the ground or suspended by a line from a tree or from a horizontal line. A trough filled with a liquid killing agent is placed so that insects flying into the pane drop into the trough and drown. They are removed from the liquid, washed with alcohol or other solvent, and then preserved or dried and pinned. This trap is not recommended for adult Lepidoptera or other insects that may be ruined if collected in fluid.

Figure 1.31
Windowpane trap.

1.6.2 Interception Nets and Barriers

A piece of netting about 1.8 m high can be stretched between three trees or poles to form a V-shaped trap, with the wide end of the V open. A triangular roof should be adjusted to slope gently downward to the broad open side of the V. A device of this type will intercept many kinds of flying insects, particularly if the trap is situated with the point of the V toward the side of maximum light and in the direction of air movement. A pair of such nets set in opposite directions or a single net in a zigzag shape will intercept specimens from two directions. Insects flying into such a net tend to gather at the pyramidal apex, so they are easy to collect. The so-called "funnel" or "ramp" traps are interception devices that direct insects to a central point, where a retaining device or killing jar may be placed.

1.6.3 Malaise Traps

A modification of the interception net led to the more complex trap developed by the Swedish entomologist René Malaise that now bears his name. Several modifications of his original design have been published, and at least one is available commercially (Townes 1962, 1972; Steyskal 1981). The trap, as originally designed, consists of a vertical net serving as a baffle, end nets, and a sloping canopy leading up to a collecting

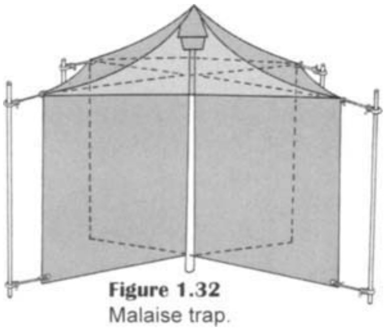
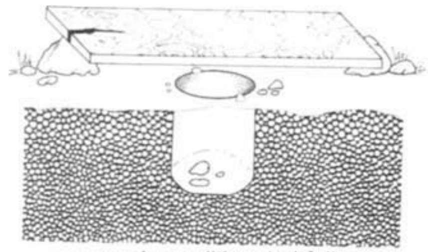


Figure 1.32
Malaise trap.

device (Fig. 1.32). The collecting device may be a jar with either a solid or evaporating killing agent or a liquid in which the insects drown. The original design is unidirectional or bidirectional with the baffle in the middle, but more recent types include a nondirectional type with cross-baffles and with the collecting device in the center. Malaise traps have been phenomenally successful, sometimes collecting large numbers of species that could not be obtained otherwise. Attractants increase the efficiency of the traps for special purposes.



A. *traditional pitfall trap*

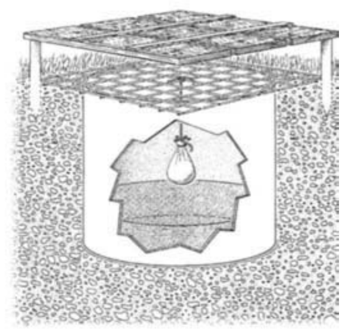
1.6.4 Pitfall and Dish Traps

Another simple but very effective and useful type of interception trap consists of a dish, can, or jar sunk in the ground and is called a pitfall trap (Fig. 1.33). Wandering arthropods fall into the trap and are unable to escape from the partly filled container of 70% ethyl alcohol or ethylene glycol (automobile antifreeze). The latter is preferred because it does not evaporate. A cover must be placed over the open top of the container to exclude rain and small vertebrates while allowing insects and mites to enter. Pitfall traps may be baited with various substances, depending on the kind of insects or mites the collector hopes to capture. Although most specimens that fall into the trap remain there, the trap should be inspected daily because specimens can decompose or become damaged. Specimens should be removed and placed in alcohol or in a killing bottle while they are in their best condition.

Other pitfall trap modifications include placing a bait within the collection container or hanging it directly above. The cereal dish trap, another modified pitfall trap, represents a simple but effective device for obtaining insects attracted to dung. This trap consists of a small dish, preferably with a rim, set in the ground (Fig. 1.33C) and partly filled with 70% ethyl alcohol or ethylene glycol. A piece of stout wire, such as a coat hanger, is bent to form a loop at one end that holds the bait receptacle. A few zigzag bends in the other end of the wire will keep the looped end from swinging after the wire is pushed into the earth. The bait receptacle may be a small plastic or metal cup (such as that used for medicine doses), a coffee creamer, or a cup formed from aluminum foil. When baited with animal or human feces, this trap attracts beetles (typically

Suggested Reading:
Pitfall and Dish Traps

- Broadbent et al. 1948
- Broadbent 1949
- Tretzel 1955
- Greenslade 1964
- Luff 1968, 1975
- Alderz 1971
- Briggs 1971
- Greenslade & Greenslade 1971
- Gist & Crossley 1973
- Greenslade 1973
- Schmid et al. 1973
- Loschiavo 1974
- Joossee 1975
- Morrill 1975 *(bibliography)*
- Muma 1975 *(includes bibliography of can and pitfall trapping)*
- Newton & Peck 1975
- Shubeck 1976
- Smith 1976
- Smith et al. 1977 *(describes low-cost carrying cages, pitfall traps, and rearing cages)*
- Thomas & Sleeper 1977
- Houseweart et al. 1979
- Reeves 1980



B. *baited pitfall trap*

C. *cereal dish trap*

D. *PVC pitfall trap*

Figure 1.33 Pitfall and dish trap

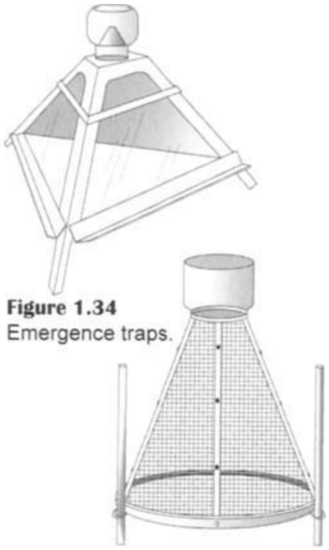


Figure 1.34
Emergence traps.

Scarabaeidae and Staphylinidae), springtails, ants, earwigs, some parasitic Hymenoptera, and several families of flies, including Phoridae, Sepsidae, and Muscidae. The larger, strong-flying calliphorid and sarcophagid flies seldom fall into the liquid, although they are attracted to the bait. The alcohol fumes may cause smaller flies to drop into it. The trap is made of easily obtained materials, is easily transported, and provides excellent results. It deserves wide use.

Modifications to the basic pitfall trap include a section (up to a meter) of PVC pipe, capped at both ends, and where a small channel is cut lengthwise along the pipe (Fig. 1.33D). Burying the pipe such that the open channel is at the soil surface allows wandering insects to fall into the pipe.

1.6.5 Emergence Traps and Rearing Cages

An emergence trap is any device that prevents adult insects from escaping when they emerge from their immature stages in any substrate, such as soil, plant tissue, or water (Yates 1974). A simple canopy over an area of soil, over a plant infested with larvae, or over a section of stream or other water area containing immature stages of flying insects will secure the emerging adults (Lammers 1977). If the trap is equipped with a retaining device (as in the Malaise trap), the adults can be killed and preserved shortly after emergence. Remember, however, that many insects should not be killed too soon after emergence because the adults are often teneral (soft bodied) and incompletely pigmented. For preservation, specimens must be kept alive until the body and wings harden completely and colors develop fully. Emergence traps and rearing cages (Fig. 1.34) enable the insects to develop naturally but ensure their capture when they mature or when larvae emerge to pupate.

Suggested Reading:

Lobster/Eel Traps

- Nicholls 1960
- Doane 1961
- Brockway et al. 1962
- Morrill & Whitcomb 1972
- Nielsen 1974
  (describes catches of insects in funnel traps on trunks of beech trees)
- Nakagawa et al. 1975
- Reierson & Wagner 1975
- Broce et al. 1977
- Steyskal 1977
- Becker et al. 1991

1.6.6 Traps Using the Lobster or Eel Trap Principle

This trap design can be made from any container that has its open end fitted with a truncated cone directed inward (Fig. 1.35). This trap design is commonly used to catch minnows, lobsters, and eels in water. An ordinary killing jar with a funnel fastened into its open end is an example. When the funnel is placed over an insect, the specimen will usually walk or fly toward the light and enter the jar through the funnel. Modified traps of this type include the Steiner and McPhail traps, which are used extensively in fruit fly surveys.

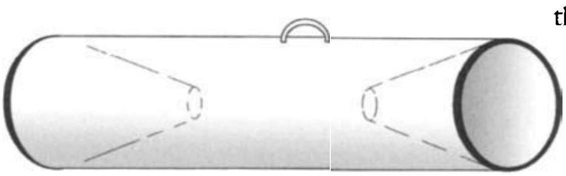


Figure 1.35
Eel trap.

McPhail was the first entomologist to use an invaginated clear glass trap for evaluating the attraction of fruit flies to various baits (Burditt 1982). The McPhail Trap (Fig. 1.36), as it is known today, is used extensively for monitoring fruit fly populations in the United States and abroad. A homemade version of the McPhail trap was devised by Burditt (1988), and a number of baits such as sucrose, yeast, or ethyl alcohol, can be used to attract and collect many kinds of insects. As an example, the McPhail trap was used in a survey to collect euglossine bees in tropical Amazonian forests (Becker et al. 1991). The inside of the Steiner trap usually has a sticky material containing a pheromone or other lure.

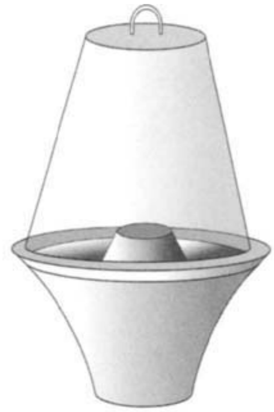


Figure 1.36
McPhail trap.

1.6.7 Light Traps

Light traps take advantage of the attraction of many insects to a light source and their attendant disorientation. Using various wavelengths of light as the attractant, a great variety of traps can be devised *using* a basic funnel and collection container design (Fig. 1.37).

Many traps can be constructed easily from materials generally available around the home. All wiring and electrical connections should be approved for outdoor use. Funnels can be made of metal, plastic, or heavy paper. Traps can be with or without a cover, but if they are operated for several nights, covers should be installed to exclude rain.

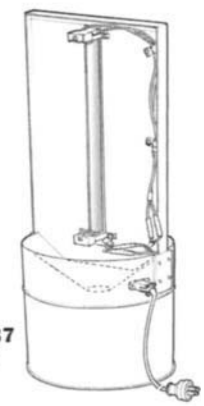


Figure 1.37
Blacklight trap.

The New Jersey trap (Fig. 1.38) includes a motorized fan to force insects attracted to the light into a killing jar. This trap has been especially useful for collecting small, nonscaly insects, such as midges and gnats. This type of light trap, in which the insects fall directly into a killing jar, is not recommended for use with moths because such delicate specimens may be badly rubbed or torn. If only small insects are desired, they may be protected from damage by larger insects by placing a screen with the proper-sized mesh over the entrance. The Minnesota trap is very similar to the New Jersey trap, but it does not include a fan or any motorized method of draft induction.

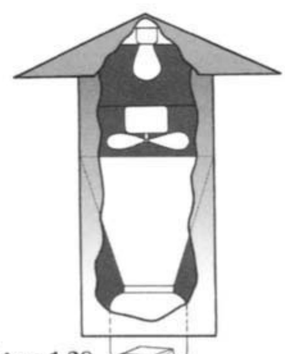


Figure 1.38
New Jersey trap.

The Wilkinson trap (Fig. 1.39) requires somewhat more effort to construct than the preceding traps, but it has the advantage of confining, but not killing, the trapped insects. Moths, therefore, can be collected in good condition if the trap is inspected frequently and desirable specimens are removed quickly through the hinged top and placed in a killing jar.

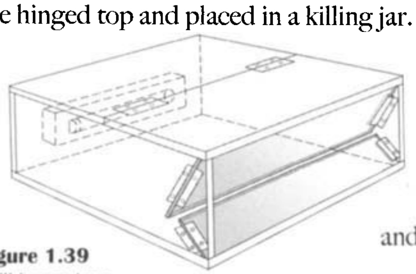


Figure 1.39
Wilkinson trap.

Several highly effective but more elaborate devices have been made for collecting moths and other fragile insects in good condition. Basically, they all use the principle of a funnel with a central light source above it and vanes or baffles to intercept the

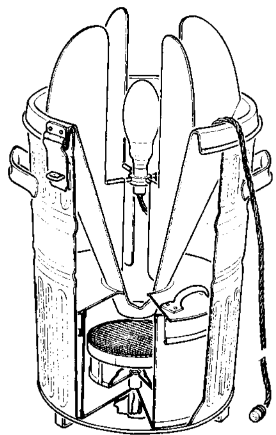


Figure 1.40
Mercury vapor light trap.

approaching insects, which are dropped through the funnel into the container beneath that may hold a killing agent. The nature of the container and the type of killing agent affect the quality of the specimens obtained. Some traps catch the insects alive in a large collection chamber (such as a garbage can) that is filled or nearly filled with loosely arranged egg cartons. Most moths will come to rest in the cavities between the egg cartons and will remain there until removed the next morning.

To prevent rainwater from accumulating in the trap, place a screen-covered funnel inside the collection chamber to drain the water out through a hole in the bottom of the trap. Sometimes a system of separators is added to guide beetles and other heavy, hard-bodied insects into a different part of the container than the moths and other delicate specimens.

The most effective light traps use lamps high in their output of ultraviolet light (Fig. 1.40). An example is the Robinson trap, which employs a 175-watt mercury vapor lamp. Other trap designs use ultraviolet fluorescent tubes (most commonly 15-watt), which also are effective and may be powered by a car battery or other portable source of electricity. High-wattage mercury vapor lamps emit more ultraviolet light than the more common fluorescent "blacklight" tubes, and many more insect specimens come to mercury vapor light setups. However, mercury vapor setups are not as portable and, in the field, require the use of a gasoline-powered generator. Self-ballasted mercury vapor lamps are not as effective on a watt-by-watt basis as those with a separate ballast.

Some collectors report that a few groups of insects respond best to incandescent lamps or even gas camping lanterns that do not emit ultraviolet light. However, for the majority of insects that may be taken with lights, high ultraviolet output is desirable.

Suggested Reading:

Light Traps

- Gui et al. 1942
- Mulhern 1942
- Pratt 1944
- Williams 1948
- Davis & Landis 1949
- Frost 1952, 1957, 1958, 1964, 1970
- Meyers 1959
- Fredeen 1961
- Graham et al. 1961
- Hollingsworth et al. 1961
- U.S. Department of Agriculture, Agriculture Research Service 1961
- Barr et al. 1963
- Belton & Kempster 1963
- Breyev 1963
- Kovrov & Monchadskii 1963
- Lowe & Putnam 1964
- White 1964
- Lewis & Taylor 1965
- Belton & Pucat 1967
- DeJong 1967
- Bartnett & Stephenson 1968
- Hardwick 1968
- Stewart & Lam 1968
- Powers 1969
- Wilkinson 1969
- Andreyev et al. 1970
- McDonald 1970
- Miller et al. 1970
- Stanley & Dominick 1970
- Barrett et al. 1971
- Carlson 1971
- Stewart & Payne 1971
- Hollingsworth & Hartstack 1972
- Tedders & Edwards 1972
- Clark & Curtis 1973
- Pérez Pérez & Hensley 1973
- Morgan & Uebel 1974
- Smith 1974
- Nantung Institute of Agriculture 1975
- Apperson & Yows 1976
- Onsager 1976
- Stubbs & Chandler 1978
- Burbutis & Stewart 1979
- Howell 1980
- Hathaway 1981

Except in special cases, light sources should be placed near the ground. This is counterintuitive to some collectors who try to place lights as high as possible. However, unpublished studies suggest that more specimens come to ground-level light sources, and species that tend to be "flighty" more reliably remain near these lights. Of course, specialized light collecting may require the placing of a light trap high in a tree or some other location.

A lightweight, spill-proof 12-volt battery, in which the acid electrolyte is a gel rather than a liquid, is far superior to the standard automotive battery for powering light traps. These new batteries are fairly expensive and require a special charger. Special lightweight, nickel-cadmium battery packs, used to power blacklights for collecting, are marketed by some dealers of entomological equipment.

1.6.8 Light Sheets

Another highly effective method of employing light to attract moths and other nocturnal insects involves a "light sheet" (Fig. 1.41). A particularly effective and efficient light sheet or "light net" design is one made of sturdy netting material with edges of ripstop nylon and having a rectangular cutout near the bottom in which a mercury vapor or other light source is placed. The netting and cutout greatly reduce flapping caused by wind, and the cutout allows one light source to shine in all directions. Ground sheets made of ripstop nylon also are useful, lightweight, easy to clean, and quick-drying.

An alternative light-sheet design employs light sources (ultraviolet fluorescent tubes, gasoline lanterns, or automobile headlights) placed in front of the light sheet. As insects are attracted and alight on the sheet, they are easily captured in cyanide bottles or jars by the collector, who stands in attendance or at least checks the sheet frequently. The sheet may be pinned to a rope tied between two trees or fastened to the side of a building, with the bottom edge spread out on the ground beneath the light. Some collectors use supports to hold the bottom edge of the sheet several centimeters above the ground so that specimens cannot walk into the vegetation under the sheet and be overlooked. Other collectors turn up the edge to form a trough into which insects may fall as they strike the sheet.

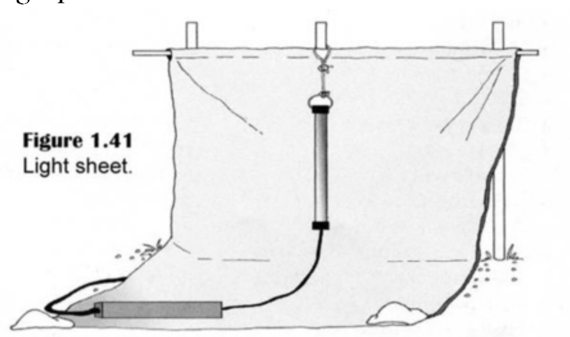


Figure 1.41
Light sheet.

The light sheet remains unsurpassed as a method of collecting moths in flawless condition or of obtaining live females for rearing purposes. Its main disadvantage is that species that fly very late or those that are active only in the early morning hours may be missed unless the collector is prepared

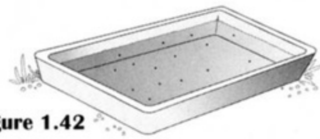


Figure 1.42
Pan trap.

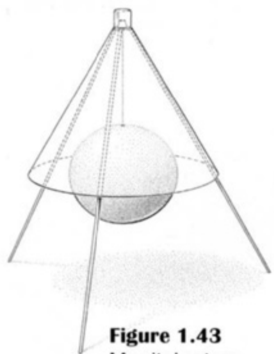


Figure 1.43
Manitoba trap.

to spend most of the night at the sheet. Many other insects besides moths are attracted to the sheet, and collectors of beetles, flies, and other kinds of insects would do well to collect with this method.

It should be emphasized that the phases of the moon profoundly influence the attraction of insects to artificial light. Attraction is inhibited by a bright moon. The best collecting period each month extends from the fifth night after the full moon until about a week before the next full moon.

1.6.9 Color Traps

Colored objects also serve as attractants for insects. A bright yellow pan (Fig. 1.42) containing water is used to collect winged aphids and parasitic Hymenoptera. The insects are attracted by the color and drown in the water. The trap is more effective when a little detergent has been put in the water to reduce surface tension and thereby cause insects to drown more quickly. Yellow seems to be the best color for traps, but various kinds of insects react differently to different colors. The Manitoba trap (Fig. 1.43) has a black sphere to attract horse flies (family Tabanidae), which are then captured in a canopy-type trap.

1.6.10 Sticky Traps

In its simplest design, this type of trap involves a board, a piece of tape, a pane of glass, a piece of wire net, and a sphere, a cylinder, or some other object (often painted yellow). The trap is coated with a sticky substance and suspended from a tree branch or other convenient object. Insects landing on the sticky surface cannot extricate themselves. The sticky material can be dissolved with a suitable solvent, usually toluene, xylene, ethyl acetate, or various combinations of these solvents. Later, the insects are washed in Cellosolve and then in xylene. Sticky traps should not be used to collect specimens such as Lepidoptera, which are ravaged by the sticky substance and cannot be removed without being damaged.

Various sticky-trap materials are available commercially, some with added attractants, including lights. However, exercise caution in selecting a sticky substance because some are difficult to dissolve. General techniques for cleaning specimens taken with sticky traps have been outlined (Murphy 1985), and a cleaning procedure involving an ultrasonic cleaner has been developed (Williams & O'Keeffe 1990).

SUGGESTED READING:

Color traps
- Gui et al. 1942
- Hottes 1951
- Brown 1954
- Granger 1970
- Kring 1970
- Beroza 1972
- Emden 1972
- Kieckhefer et al. 1976
- Hendrix & Showers 1990
- McClain et al. 1990

Sticky traps
- Golding 1941, 1946
- Johnson 1950
- Moreland 1955
- Heathcote 1957b
- Still 1960
- Murphy 1962, *pp. 226–227*
- Taylor 1962b
- Maxwell 1965
- Gillies & Snow 1967
- Lambert & Franklin 1967
- Prokopy 1968, 1973
- Dresner 1970
- Harris et al. 1971
- Mason & Sublette 1971
- Emden 1972
- Buriff 1973

1.6.11 Snap Traps

Two kinds of traps designed for quantitative sampling may be termed "snap traps." One (Menzies & Hagley 1977) consists of a pair of wooden or plastic disks, slotted in the center to fit on a tree branch and connected to each other by a pair of rods. A cloth cylinder is affixed at one end to one of the disks and at the other end to a ring sliding on the rods. After the cloth cylinder has been pulled to one end and secured in place, the ring is held by a pair of latches. When insects have settled on the branch, its leaves, or flowers, the latches are released by pulling on a string from a distance, and the trap is snapped shut by a pair of springs on the rods, capturing any insects present. One of the canopy traps (Turnbull & Nicholls 1966) operates in a similar fashion. When a remotely controlled latch is pulled, a spring-loaded canopy is snapped over an area of soil, and insects within the canopy are collected by suction or a vacuum device. This trap was designed for use in grasslands.

1.6.12 Artificial Refuge Traps

Many insects, especially beetles, are found under stones, planks, or rotten logs. Providing refuges (such as pieces of wood, cardboard, or complex traps) is also an effective form of trapping.

1.6.13 Electrical Grid Traps

In recent years, electrocuting insects has been used extensively in pest-control work. The insects are attracted to a device by a chemical (see Section 1.7), light, or other substance placed in a chamber protected by a strongly charged electrical grid (Fig. 1.44). This method is not effective in preserving insect specimens but may be useful for purposes such as surveying the arthropod fauna of an area.

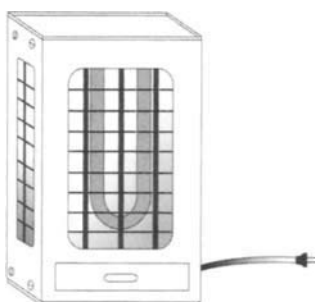


Figure 1.44
Electrical grid trap.

Sticky traps (continued)
- Chiang 1973
- Goodenough & Snow 1973
- Williams 1973
- Evans 1975
- Edmunds et al. 1976 *(methods of collecting and preservation on pp. 8-26)*
- Harris & McCafferty 1977
- Murphy 1985
- Bowles et al. 1990
- Knodel & Agnello 1990
- Vick et al. 1990
- Jenkins 1991
- Miller et al. 1993

Artificial Refuge Traps
- Glen 1976
- Shubeck 1976

Electrical Grid Traps
- Graham et al. 1961
- Hollingsworth et al. 1963
- Mitchell et al. 1972, 1973, 1974
- Goodenough & Snow 1973
- Hienton 1974
- Glen 1976
- Rogers & Smith 1977
- Stanley et al. 1977
- Kogan & Herzog 1980

1.7 Baits, Lures, and Other Attractants

Any substance that attracts insects may function as a bait. Natural products, chemicals derived therefrom or synthesized, and; secretions of insects may all be attractants (Jacobson & Beroza 1964, Jacobson 1972). Exposure of attractive substances is found in a variety of constructed traps.

Suggested Reading:
Baits and Lures
- Walsh 1933
- Golding 1941
- Dethier 1955
- Macleod & Donnelly 1956
- Atkins 1957
- Colless 1959
- Rennison & Robertson 1959
- Beroza & Green 1963
- Hocking 1963
- Mason 1963
- Wilton 1963
- Jacobson & Beroza 1964
- Coffey 1966
- Newhouse et al. 1966
- Sanders & Dobson 1966
- Strenzke 1966
- DeJong 1967
- Acree et al. 1968
- Everett & Lancaster 1968
- Fincher & Stewart 1968
- Morris & DeFoliart 1969
- Beroza 1970, 1972
- Nakagawa et al. 1971
- Wellso & Fischer 1971
- Beavers et al. 1972
- DeFoliart 1972
- Knox & Hays 1972
- Roberts 1972
- Beroza et al. 1974
- Debolt et al. 1975
- Howell et al. 1975
- Pinniger 1975
- Shorey & McKelvey 1977
- Bram 1978
- Zimmerman 1978
- Howell 1980
- Laird 1981

1.7.1 Sugaring for Insects

One of the oldest collecting methods involves the use of a specially prepared bait in which some form of sugar is an essential component. The bait may be refined or brown sugar, molasses, or syrup. Such substances often are mixed with stale beer or fermented peaches, bananas, or other fruit. Sugar-baited traps are most often used for moths and butterflies, but they are also effective for some flies (Dethier 1955) and caddisflies (Bowles et al. 1990).

One particularly effective recipe for Lepidoptera uses fresh, ripe peaches; culls or windfalls are suitable. Remove the seeds but not the skins, mash the fruit, then place it in a 4-liter (1-gal) container with a snugly fitting but not airtight cover. Avoid using metal containers that may rust or corrode. Fill each container only one-half to two-thirds full to allow space for expansion. Add about a cup of sugar and place in a moderately warm place for the mixture to ferment. The bubbling fermentation reaction should start in a day or so and may continue for two weeks or more, depending on the temperature. During that time, check the fermentation every day or every other day and add sugar until fermentation appears to have subsided completely. As the added sugar is converted to alcohol, the growth of yeast slows and eventually ceases.

If the mixture is allowed to run low in sugar during the fermentation process, vinegar will be produced instead of alcohol. Remember to smell the bait periodically and to add plenty of sugar to avoid vinegar formation. The amount of sugar consumed will be surprising, usually over 0.4 kg per liter (3.3 lb per gal). After fermentation ceases, the bait should remain stable and should be kept in tightly sealed containers to prevent contamination and evaporation. Canned fruit, such as applesauce, may also be used to make the bait, but such products are sterile and a small amount of yeast must be added to start fermentation. The bait should have a sweet, fruity, winelike fragrance. Although the bait may seem troublesome to prepare, it keeps for years and is available at any time, even when fruit is not in season.

Immediately before use, the bait may be mixed with 30% to 50% molasses, brown sugar, honey, or a mixture of these ingredients. This thickens the bait, retards drying, and makes the supply last longer. Set out the sugar bait during the early evening before dark. Apply the bait with a paintbrush in streaks on tree trunks, fence posts, or other surfaces. Choose a definite route, such as along a trail or along the edge of a field, so that later you can follow it in the dark with a lantern or flashlight. Experienced collectors approach the patches of bait cautiously, with a light in one hand and a killing jar in the other to catch moths before they are frightened off. Some collectors prefer to wear a headlamp, leaving both hands free to collect specimens. Although some moths will escape, a net usually is regarded as an unnecessary encumbrance because moths can be directed rather easily into the jar. Sugaring is an exceptionally useful way to collect noctuid moths, and the bait applied in the evening often will attract various diurnal insects on the following days. This bait has been used in butterfly traps with spectacular results. However, collecting with baits is notoriously unpredictable, being extremely productive on one occasion and disappointing on another, under apparently identical conditions.

Suggested Reading:
Feces
- Steyskal 1957
- Coffey 1966
- Fincher & Stewart 1968
- Merritt & Poorbaugh 1975

1.7.2 Feces

Animal, including human, feces attract many insects. A simple but effective method of collecting such insects is to place fresh feces on a piece of paper on the ground and wait a few minutes. When a sufficient number of insects has arrived, a net with its bag held upward can be brought carefully over the bait about 1 m above it. This will not disturb the insects, nor will they be greatly disturbed when the net is lowered gently about two-thirds of the distance to the bait. At this point, the net should be lowered quickly until its rim strikes the paper. The insects, mostly flies, will rise into the net, which may then be lifted a short distance above the bait and quickly swung sideways, capturing the insects in the bottom of the bag. Feces are most attractive to insects during the first hour after deposition, and many flies can be caught during that relatively short period of time. Because of this, the "baiting with feces" method may be used for quantitative studies.

Other insects may continue coming to the feces for a more extended period and may be captured by placing a canopy trap over the feces or by using the feces with the cereal dish trap (see Section 1.6.4). Emergence traps placed over old feces will capture adult insects emerging from immature forms feeding there. The same methods also may be used with other baits, such as decaying fruit, small dead animal carcasses, and a wide variety of other substances.

1.7.3 Oatmeal

Hubbell (1956) showed that dry oatmeal scattered along a path will attract such insects as crickets, camel crickets, cockroaches, and ants. Some of these insects feed only at night and may be best located by using a flashlight or headlamp. The specimens may be hand-collected, aspirated, or netted and placed into a killing jar.

1.8 Pheromones and Other Attractants

Insects rely heavily upon chemical communication. Over the last several years, the research into insect chemical communication has been phenomenal. New

Suggested Reading:

Pheromones

- Beaudry 1954
- Holbrook & Beroza 1960
- Jacobson & Beroza 1964
- Howland et al. 1969
- Beroza 1970
- Campion 1972
- Jacobson 1972
- Goonewardene et al. 1973
- Pérez Pérez & Hensley 1973
- Shorey 1973
- Beroza et al. 1974
- Birch 1974
- Campion et al. 1974
- Peacock & Cuthbert 1975
- Weatherston 1976
- Shorey & McKelvey 1977
- Steck & Bailey 1978
- Neal 1979
- Howell 1980
- Sparks et al. 1980
- Hathaway 1981
- Vite & Baader 1990
- Gray et al. 1991
- Mullen 1992
- Mullen et al. 1992
- Bartelt et al. 1994

Carbon Dioxide

- Reeves 1951, 1953
- Rennison & Robertson 1959
- Takeda et al. 1962
- Whitsel & Schoeppner 1965
- Newhouse et al. 1966
- Snoddy & Hays 1966
- Wilson et al. 1966
- Gillies & Snow 1967
- Morris & DeFoliart 1969
- Hoy 1970
- Stryker & Young 1970
- Davies 1971
- Batiste & Joos 1972
- Blume et al. 1972
- Roberts 1972;
- Wilson et al. 1972
- Debolt et al. 1975
- Gray 1985
- Carroll 1988

terms, new concepts, and the refinement of old ideas have been necessary. For instance, the term *allelochemical* has been proposed for chemicals secreted outside the body of an organism. *Semiochemicals* are compounds that influence insect behavior or that mediate the interactions between organisms. Currently, at least three categories of semiochemicals have been identified. A *synomone* is an interspecific chemical messenger that is beneficial to emitter and receiver. Examples include secondary plant chemicals that attract entomophagous insects to the plant and subsequently to prey or hosts. A flower fragrance that is attractive to a bee through nectar and pollen gained is also beneficial to the flower through pollination. In contrast, an *allomone* is an interspecific chemical that, in a two-species interaction, is beneficial only to the emitter. A *kairomone* is an interspecific chemical that is beneficial only to the receiver. Insect collectors are beginning to appreciate the role that interspecific attractants play in capturing insects, and in the future we will probably rely on them even more often. For instance, "Spanish fly" (cantharidin) is an irritant to vertebrates that is produced by meloid beetles but has recently come into use as an extremely effective attractant for other beetles (such as pedilids) and bugs (such as bryocerines).

Substances naturally produced and emitted by insects that cause a behavioral response in individuals of the same species are known as *pheromones*. Sex pheromones, in which the pheromone is emitted by one sex and attracts individuals of the opposite sex, are often used in traps to aid in controlling pest species. They also are emitted into the air from controlled-release dispersers throughout a particular crop to disrupt mating by misdirecting male insects to these synthetic sources as well as habituating males to the scent of their own females. Most pheromones are blends of several components; the blend is highly specific, attracting only one species. Pheromones are known to attract members of one sex, usually males, from great distances.

1.8.1 Carbon Dioxide

Host animals likewise may be used as bait for various bloodsucking insects, with or without constructed traps. Carbon dioxide (Fig. 1.44) in the form of dry ice, cylinder gas, or marble chips treated with an acid such as vinegar serves as an attractant for certain insects and has been very successful in attracting horse flies to Malaise and Manitoba traps. Dry ice is also an effective attractant for adult mosquitoes, horse flies, and parasites such as ticks.

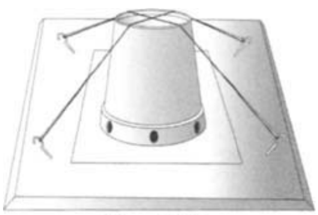


Figure 1.44
CO_2 trap.

1.8.2 Sounds

Sounds are produced by many insects to attract other members of the same species. These sounds are very specific in pitch, tempo, and duration. Sound sources once tape-recorded are now often synthesized and broadcast using high-tech electronic amplifiers. Photo cells often serve as timing devices to turn sounds on or off, rather than manual switches. Recordings of such sounds, played at the proper volume, have been effective in luring grasshoppers, mole crickets, and other kinds of insects.

1.9 Collecting Aquatic Insects

Insects and mites emerging from water may be collected by some of the same methods as terrestrial insects. Aquatic-insect-collecting dip nets and heavy-duty aquatic nets can be purchased commercially (Fig. 1.45 A&B). Kick screens (Fig. 1.45C) can be constructed easily from a fine-mesh window screen attached to two stout poles. Holding the screen on the bottom of the stream immediately downcurrent from where rocks and other debris are dislodged can yield a large number of aquatic organisms. Flume collecting is an old technique that has recently been revived. The extraction of insects and mites from flumes involves glass-topped sleeve cages, modified Malaise and tent traps, and Berlese photoattractive traps positioned directly above water. Other types of aquatic collecting techniques, such as tow traps and dredges (Fig. 1.46), require specialized equipment that we do not describe here. The collection of aquatic insects is of great importance in public health and general ecological studies.

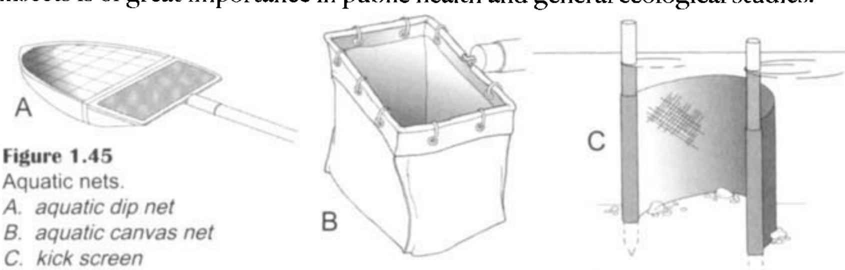


Figure 1.45
Aquatic nets.
A. aquatic dip net
B. aquatic canvas net
C. kick screen

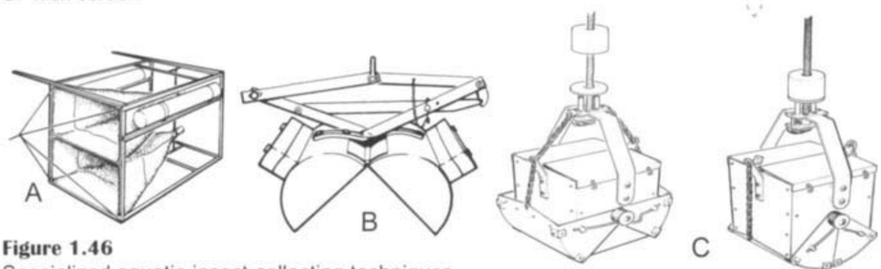


Figure 1.46
Specialized aquatic-insect-collecting techniques.
A. tow trap, B. dredge, C. modified Birge–Ekman dredge

Suggested Reading:

Sound Traps
- Belton 1962
- Cade 1975
- Walker 1982
- Bailey 1991
- Dethier 1992
- Parkman & Frank 1993
- Thompson & Brandenburg 2004

Aquatic Insects
- Hodgson 1940
- Welch 1948 (general)
- Jonasson 1954
- Mundie 1956, 1964, 1966, 1971
- Gerking 1957
- Essig 1958
- Lindeberg 1958
- Grigarick 1959
- Pieczynski 1961
- Sladeckova 1962
- Morgan et al. 1963
- Cushing 1964
- Macan 1964
- Corbet 1965
- Wood & Davies 1966
- Kimerle & Anderson 1967
- Tarshis 1968a, b
- Waters 1969
- Coulson et al. 1970
- Elliott 1970
- Carlson 1971
- Edmondson & Winberg 1971
- Mason & Sublette 1971
- Fahy 1972
- Finch & Skinner 1974
- Langford & Daffern 1975
- Murray & Charles 1975
- Apperson & Yows 1976
- Edmunds et al. 1976
- Landlin 1976
- McCauley 1976
- Masteller 1977
- Edmunds & McCafferty 1978
- LaGasa & Smith 1978
- Pennak 1978 (general)
- Ettinger 1979
- Lawson & Merritt 1979
- LeSage & Harrison 1979
- Wood et al. 1979 *(methods of collecting, preserving, and rearing on pp. 45-53)*
- McCafferty 1981
- Merritt et al. 1984 *(general)*
- Halstead & Haines 1987
- Weber 1987

Suggested Reading:

Collecting Soil Insects
- Davidson & Swan 1933
- Barnes 1941
- Salt & Hollick 1944
- Kevan 1955, 1962
- Newell 1955
- Turnock 1957
- MacFadyen 1962
- Murphy 1962
- Teskey 1962
- Brindle 1963 *(includes collecting methods)*
- Evans et al. 1964 *(techniques on pp. 61–88)*
- Kühnelt 1976 *(observation and collecting techniques on pp. 35–65, bibliography on pp. 385–466)*
- Lane & Anderson 1976
- Akar & Osgood 1987

Collecting Ectoparasites
- Banks 1909 *(mostly of historical interest, but describes the old methods and contains much general information about insects)*
- Klots 1932
- Cantrall 1939–1940, 1941
- Comstock 1940
- Chu 1949
- Grandjean 1949
- Lipovsky 1951 (mites)
- Cook 1954
- Williamson 1954
- Wagstaffe & Fidler 1955
- Balogh 1958
- Lumsden 1958
- Morgan & Anderson 1958
- Oldroyd 1958
- Fallis & Smith 1964
- Peterson 1964
- Urquhart 1965 *(elementary directions for making insect collections, pp. 1–19)*
- Knudsen 1966, 1972, pp. 128–176
- Norris 1966
- U.S. Department of Agriculture, Plant Pest Control Division 1966–1970
- Watson & Amerson 1967
- Lehker & Deay 1969
- Nicholls 1970

(continued next page)

1.10 Collecting Soil Insects

As with aquatic specimens, insects and mites that live on or under the soil surface require special techniques and equipment for their collection and study. Often separators, extractors and pitfall and Berlese traps can be utilized to collect soil-inhabiting insects. Various floatation techniques have been used to collect small insects and mites from soils and other substrates as well. Many soil-inhabiting species are of great economic importance because they devour the roots of crops. Many of these insects spend their immature stages in soil but emerge and leave the soil as adults. A considerable amount of literature on collecting soil insects has been published, the most useful of which is cited here.

1.11 Collecting Ectoparasites

Ectoparasites are organisms that live on the body of their host. Examples include lice and fleas. Some ectoparasites, particularly those that fly, may be collected in some of the traps discussed previously, using their hosts as bait; others may be collected either by hand or with special devices described in the listed readings.

1.12 Collecting Regulated Insects

Collecting insects or other arthropods at points of entry into the state or country or by way of specific and intensive surveys is an important part of regulatory officials' duties. Extreme care must be exercised when collecting, preserving, and indentifying the invasive organisms. Care must be taken to preserve the sample together with its collection information, in a manner that will allow others to also verify species identity. Both shipping and rearing insects (sections found in this book) are critical for regulatory entomologists to understand and utilize.

1.13 Collecting Insects for Pest Management Audits

Collecting tools and techniques are an important part of a pest management audit, regardless of the type of building or the commodities stored or manufactured in the facility. Intensive inspections require a solid understanding of how and why pests may enter a building. Understanding the importance of food, water, and harborage to pests' establishment is crucial in conducting pest audits. Understanding building construction and maintenance is also invaluable. Insect pest managers can take pest information observed during an audit and subsequently describe a correct course of action to eliminate existing and exclude other potential pests from facilities.

Using insect glue traps as monitors is very important to pest auditors. Because inspections occur only over the course of one or two days, they represent a snapshot in time of what is occurring in the building. Monitors, on the other hand, work 24 hours a day, seven days a week, and provide a record of events over a much longer period of time. Flashlights, screwdrivers, probes, flushing agents, scrapers, mirrors, collecting vials, clipboards, blueprints of the building, knee pads, and bump caps are all part of an auditor's routine inspection tool bag. On occasion, more sophisticated equipment is required to listen for or observe pests in areas where they hide. Auditors must be ever alert to signs of pest infestations, even when the pest cannot be found. Often alerting the owners to "conducive conditions" (those conditions that might enable a pest to enter or to flourish if introduced) can be of as much value as finding the pest.

1.14 Collecting Insects for Forensic or Medico-Criminal Investigations

Use of arthropods in forensic or legal investigations has become standard practice in recent years. Insects and mites are often the subject of lawsuits especially where contamination of products, misapplication of pesticides, transmission of disease, or human myiasis is concerned. Collection and documentation of specimens as evidence requires great attention to procedure and protocol.

Many different collecting techniques can be employed in death scene investigations. Direct observation or sweep net sampling can often provide important information of the adult stages of insects above the body if taken before the body is disturbed. These data are often lost evidence if not taken immediately. Direct collecting of insects from on, in, and around the body is one of the most effective means of obtaining reliable data. Often, collecting various stages of insects and recording the exact location of the collections are vital. Rearing a portion of the immature stages to adulthood can provide important times and identifications of insects. For detailed protocols about collecting and preserving insects and related organisms in forensic cases, please consult the cited references.

1.15 Rearing

Collectors should take every opportunity to rear insects and mites. Reared specimens generally are in the best condition. Further, rearing provides life stages that otherwise might be collected only rarely or with great difficulty. With respect to parasitic insects, such as some Hymenoptera and Diptera, rearing provides unambiguous proof of host associations and other scientifically important information.

Ectoparasites (continued)
- Brown 1973
- Ford 1973
- Cogan & Smith 1974, p. 152
- Thompson & Gregg 1974
- Cheng 1975
- McNutt 1976
- Service 1976 *(includes extensive bibliographies)*
- Stein 1976
- Martin 1977
- Bland & Jacques 1978
- Edmunds & McCafferty 1978
- Lincoln & Sheals 1979
- Southwood 1979 *(includes extensive bibliographies)*
- Banks et al. 1981
- Eads et al. 1982
- Pritchard & Kruse 1982
- Arnett 1985
- Stehr 1987, 1991
- Borror et al. 1989
- Dindal 1990
- Stehr 1991
- Dryden & Broce 1993
- Kasprzak 1993

Forensic Entomology
- Erzinclioglu 1983
- Anderson 1999
- Castner & Byrd 2001
- Greenberg & Kunish 2002
- Haglund & Sorg 2002
- Saferstein 2004
- James & Nordby 2005

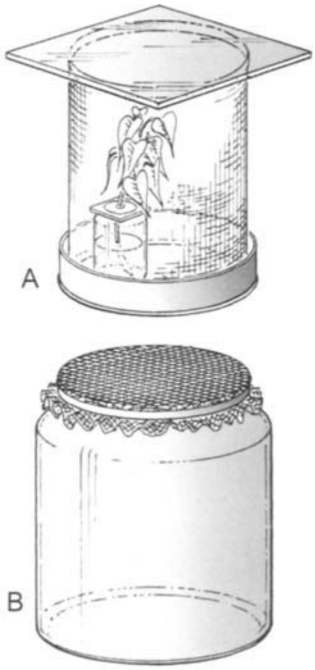


Figure 1.47
Rearing chambers.
A. screen cylinder capped on both ends
B. glass jar with ventilated lid

By preserving one or more specimens from each of the stages as they are reared, the collector can obtain a series of immature stages along with associated adults (Clifford et al. 1977). Such series are desirable, especially for species in which the adult is known but the immature stages are unknown or difficult to identify. Often, the converse is also true. Some species of insects, such as stem-mining flies, are fairly abundant in the larval stage. Sometimes we do not know whether the specimens represent a species that has been described and named from an adult whose life history is unknown. Because adults of these flies are seldom found, the easiest way to obtain the stage necessary for specific determination is to rear the larvae or pupae.

If only a few specimens are reared, then the shed "skins" (exuviae) and pupal cases of puparia should be preserved because they are valuable when properly associated with the reared adult. Do not preserve a pupa or puparium with an adult unless you are positive that the association is correct. Put pupae in separate containers so that adults or parasites that emerge are associated with certainty. When feasible, the parasite's host should be preserved for identification. Keep careful notes throughout the rearing so that all data relative to the biology of the species are properly correlated.

1.15.1 Containers for Rearing

To rear specimens successfully, rearing cages must simulate closely the natural conditions in which the immatures were found. A screen cylinder capped on both the top and bottom can be an effective rearing container (Fig. 1.47A). Almost any container will serve as a temporary cage for living insects or mites. A paper bag is a simple temporary cage that is very handy on field trips. Plant material or a soil sample containing insects or mites is placed in the paper bag, which is then sealed. A paper bag also can be placed over the top of a plant on which insects or mites are found. The bottom edge of the bag is tied tightly around the exposed stems. Stems then are cut and placed in a jar of water. Paper bags are not transparent and must be removed to observe the specimens or to determine when the foliage needs to be changed. Clear plastic bags are better suited to such viewing. However, they are not recommended for more than short-term use because they are airtight and specimens may be damaged by drowning in condensed water inside the bag.

A glass jar whose lid is replaced by a piece of organdy cloth or gauze held in place by a rubber band forms a simple temporary cage (Fig. 1.47B). A few such jars in a collecting kit are useful for holding live insects. For aquatic species, a watertight lid on the jars is advisable. Aquatic insects often die when transported over a considerable distance in water that sloshes in the container. Fewer specimens will die if the jar is packed with wet moss or leaves. After arrival at your destination, release the insects into a more permanent rearing container.

Certain aquatic insects may be reared readily indoors in an aquarium or a glass jar. The main goal is to duplicate their natural habitat. If the specimen was collected from a rapidly flowing stream, it probably will die indoors unless the water is aerated. Other insects do well in stagnant water. Aquatic vegetation usually should be provided in the aquarium, even for predaceous specimens, such as dragonfly nymphs, which often cling to underwater stems. Keep sufficient space between the surface of the water and the aquarium cover to allow the adult insect to emerge. The amount of space will vary according to the insect being reared. For example, a dragonfly needs considerable space and a stick, rock, or other object above the water on which to perch after emerging so that the wings will expand fully.

Aquatic insects can be reared in their natural habitat by confining them in a wire-screen or gauze cage. Part of the cage must be submerged in water and anchored securely. The screen used in aquatic cages should be coarse enough to allow food through yet fine enough to retain the insects being reared.

Most adult insects, both terrestrial and aquatic, are teneral when they first emerge and should not be killed until the body and wings harden and the colors develop fully. Hardening may require a few minutes, hours, or days. Keep even small flies alive for one full day after they emerge. Specimens killed while teneral will shrivel when mounted. Some insects, especially butterflies and moths, will beat their wings against the cage and lose many scales or tear their wings if kept in cages too long after emerging. Providing adequate space in which emerging insects may expand their wings fully and move about slightly is therefore critical in the design of rearing cages.

Some beetles and other boring insects often are abundant in bark and wood. Excellent adult specimens may be obtained if pieces of infested wood are placed in glass or metal containers. However, remember that rearing these insects sometimes requires considerable time. Cages made of wood or cardboard are not suitable for such insects because those found in wood or bark usually are well equipped, both in immature and adult stages, to chew their way through a cage made of such material and thus escape.

Figure 1.48
Flowerpot cage for long-term rearing.

A flowerpot cage is one of the best containers for rearing plant-feeding species over an extended period. The host plant, if its size and habitat permit, is placed in a flowerpot and a cylinder of glass, plastic, or wire screen placed around the plant (Fig. 1.48). Another type of flowerpot cage is made by inserting a cane or stick, taller than the plant, into the soil in the pot. One end of a net or muslin tube is fitted over the edge of the pot and is held in place by a string. The other end of the tube is tied around the top of the stick. An advantage of the flowerpot cage is that the plant is living, and so it is not necessary to add fresh plant material daily.

Plant-feeding mites will not wander far as long as suitable host material is available for them. Because mites are wingless even as adults, they can be confined in an open rearing container by making a barrier around the top edge or upper inner sides of the container with petroleum jelly or talcum powder.

Emergence cages are essentially rearing cages that are used when it is impractical or impossible to bring specimens indoors. Emergence cages may also be considered as traps and are discussed under that heading (see Section 1.6.5). With plant-feeding insects, a sleeve consisting of a muslin tube with open ends is slipped over a branch or plant and tied at one end (Fig. 1.49). The insects are then placed in the tube, and the loose end of the tube is tied. This cloth tube can be modified to allow observation of the insects by replacing the midsection with a "window" of clear plastic or wire screen. If the insects in the tube require duff or debris in which to pupate, the tube should be placed perpendicular to the ground and duff or debris placed in the lower end.

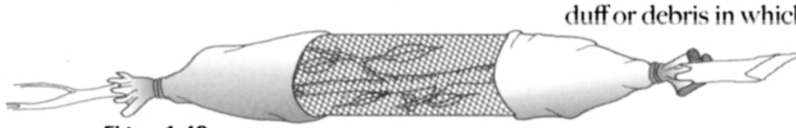


Figure1.49
Muslin sleeve cage.

1.15.2 Rearing Conditions and Problems

1.15.2.1 Moisture

The moisture requirements of insects and mites are varied. Examination of the habitat from which specimens were collected should provide clues about their moisture requirements in captivity. Many insects in the pupal stage resist desiccation. Species that normally infest stored foods also require very little moisture. In fact, many produce their own metabolic water. Most species found outdoors require higher levels of humidity than generally exist indoors. Additional moisture can be added to indoor rearing cages in several ways. To increase the humidity in a cage, keep a moist pad of cotton on top of the screen cover of the cage or place a moist sponge or a small glass vial filled with water in the cage. The mouth of the vial is plugged with cotton and the vial laid on its side so that the cotton remains moist. Pupae may be held for long periods in moist sawdust, vermiculite, sphagnum, or peat moss. In a flowerpot cage, the water used to keep the plant alive and moisture released through transpiration should provide sufficient moisture for the plant-feeding insects and mites. Spraying the leaves daily also may supplement moisture requirements in rearing cages. Too much moisture may result in water condensation on the sides of the cage, which may trap the specimens and damage or kill them. Excess moisture also enhances the growth of mold and fungus, which is detrimental to the development of most insects and mites. A 2–3% solution of table salt sprayed regularly in the cage will help prevent mold and fungus growth.

1.15.2.2 Temperature

Of all the environmental factors affecting the development and behavior of insects and mites, temperature may be the most critical. Because arthropods are cold-blooded, their body temperature is usually close to the temperature of the surrounding environment, and their metabolism and development are directly affected by increases and decreases in temperature. Each stage of an insect or mite species has a low and a high point at which development ceases. These are called *threshold* temperature levels.

Most species that are collected and brought indoors for rearing can be held at normal room temperature; the optimum temperature for rearing varies from species to species and with different stages of the same species. As with all rearing techniques, every attempt should be made to duplicate optimum natural conditions. Specimens that normally would overwinter outdoors should be kept during the winter in rearing cages placed in an unheated room, porch, or garage. Never place an enclosed rearing cage in direct sunlight; the heat becomes too intense and may kill the specimens.

1.15.2.3 Dormancy and Diapause

Insects and mites cannot control the temperature of their environment. Instead, they make physiological adjustments that allow them to survive temperature extremes. In regions with freezing winters, insects and mites have at least one stage that is resistant to low temperatures. The resistant stage may be egg, larva, nymph, pupa, or adult. When winter arrives, only the resistant stage survives. *Dormancy* is the physiological state of an insect or mite during a period of arrested development. *Diapause* is the prolonged period of arrested development brought about by such adverse conditions as heat, drought, and cold. This condition can be used to advantage in rearing. For example, if rearing cages must be unattended for several days, then many specimens can be refrigerated temporarily to slow their activity and perhaps force diapause. This measure should be used with caution because the degree and duration of cold tolerated by different species vary.

Dormancy: *the physiological state of an insect or mite during a period of arrested development.*

Diapause: *the prolonged period of arrested development brought about by such adverse environmental conditions as heat, drought, and cold.*

The reverse situation, that of causing diapause to end, is equally useful. Overwintering pupae that normally would not develop into adults until spring can be forced to terminate diapause early by chilling them for several weeks or months, and then bringing them to room temperature so that normal activity will resume. Often, mantid egg cases are brought indoors accidentally with Christmas greenery. The eggs, already chilled for several months, hatch when kept at room temperature, often to the complete surprise and consternation of the unsuspecting homeowner.

Artificial manipulation of the light period will control diapause in many species.

1.15.2.4 Light

Most species of insects and mites can be reared under ordinary lighting conditions. However, artificial manipulation of the light period will control diapause in many species. If the light requirements of the species being reared are known, then the period of light can be adjusted so that the specimens will continue to develop and remain active instead of entering diapause. Light and dark periods can be regulated with a 24-hour timing switch or clock timer. The timer is set to regulate light and dark periods to correspond with the desired lengths of light and darkness. For example, providing 8–12 hours of continuous light during a 24-hour cycle creates short-day conditions that resemble winter; providing 16 hours of continuous light during a 24-hour cycle creates long-day conditions that resemble summer. Remember that many insects and mites are very sensitive to light; sometimes even a slight disturbance of the photoperiod can disrupt their development.

1.15.2.5 Food

The choice of food depends upon the species being reared. Some species are detritovores and will accept a wide assortment of dead or decaying organic matter. Examples include most ants, crickets, and cockroaches. Other insects display food preferences so restricted that only a single species of plant or animal is acceptable. At the time of collection, carefully note the food being consumed by the specimen and provide the same food in the rearing cages.

Carnivorous insects should be given prey similar to that which they normally would consume. This diet can be supplemented when necessary with such insects as mosquito larvae, wax moth larvae, mealworms, maggots, and vinegar flies or other insects that are easily reared in large numbers in captivity. If no live food is available, a carnivorous insect sometimes may be tempted to accept a piece of raw meat dangled from a thread. Once the insect has grasped the meat, the thread can be gently withdrawn. The size of the food offered depends on the size of the insect being fed. If the offering is too large, the feeder may be frightened away. Bloodsucking species can be kept in captivity by allowing them to take blood from a rat, mouse, rabbit, or guinea pig. ***A human should be used as a blood source only if it is definitely known that the insect or mite being fed is free of diseases that may be transmitted to the person.***

Stored-product insects and mites are easily maintained and bred in containers with flour, grains, tobacco, oatmeal or other cereal foods, and similar products. Unless leaf-feeding insects are kept in flowerpot cages where the host plant is growing, fresh leaves from the host plant usually should be placed in the rearing cage daily and old leaves removed.

1.15.2.6 Artificial Diets

Some species can be maintained on an artificial diet. The development of suitable artificial diets is complex, involving several factors besides the mere nutritional value of the dietary ingredients. Because most species of insects and mites have very specific dietary requirements, information regarding artificial diets is found mainly in reports of studies on specific insects or mites.

1.15.2.7 Special Problems and Precautions in Rearing

Problems may arise in any rearing program. Cannibalism, for instance, is a serious problem in rearing predaceous insects and necessitates rearing specimens in individual containers. Some species resort to cannibalism only if their cages become badly overcrowded. Disease is also a problem and can be caused by introducing an unhealthy specimen into a colony, poor sanitary conditions, lack of food, or overcrowding.

Cages should be cleaned frequently and all dead or clearly unhealthy specimens removed. Exercise care not to injure specimens when transferring them to fresh food or when cleaning the cages. Mites and small insects can be transferred with a camel's-hair brush.

Attacks by parasites and predators also can be devastating to a rearing program. Carefully examine the host material when it is brought indoors and before it is placed in the rearing containers. This reduces the possibility of predators and parasites being introduced accidentally into the cages. Also, place rearing cages where they will be safe from ants, mice, the family cat, and other predators.

Suggested Readings:

Rearing

- Needham 1937
- Hodgson 1940
- Gerberich 1945
- Chu 1949
- Hopkins 1949
- Harwood & Areekul 1957
- Levin 1957
- Fischer & Jursic 1958
- Lumsden 1958
- Peterson 1964
- Krombein 1967
- U.S. Department of Agriculture, Extension Service 1970
- Ford 1973
- Smith 1974
- Sauer 1976
- Clifford et al. 1977
- Smith et al. 1977
- Barber & Matthews 1979
- Banks et al. 1981
- Edwards & Leppla 1987
- Gray & Ibaraki 1994

Suggested Readings: Molecular Research
- Arensburger et al. 2004
- von Hagen & Kadereit 2001
- Wells et al. 2001
- Wells & Sperling 2001

1.16 Collecting Insects for Molecular Research

Collection of insect or mite specimens for molecular research requires few special techniques. In most cases, arthropods may be collected in much the same manner as discussed for general collections. Clean, contaminant-free specimens must be ensured, however. Keep in mind that some insects may carry foreign debris on their bodies, such as pollen or other potential contaminants, that may confound some studies. Some arthropods also carry parasites (either externally or internally) that may pose confusion when DNA or RNA extractions are done. Some mites commonly conceal themselves under wing covers of insects or other locations and may potentially go unnoticed.

Specific preservation techniques of the sample must follow the protocol determined by the procedure set out in Chapter 2. It is imperative to know the ultimate and specific use of the specimen beforehand, however, because the preservation techniques may affect the molecular technique being performed.

Common methods include preservation in ethanol, freezing on dry ice and then holding in an ultralow freezer, and placement in liquid nitrogen or other specific products, such as RNALater®, that helps preserve the more fragile RNA materials.

Packaging and labeling must also be done with care when specimens are meant for molecular studies. Using a soft pencil to label specimens in liquids is still the best method. Labeling specimens both inside and outside of the package or vial should be standard operating procedure because the extreme environment in which the specimens are held often causes labels, even with the best adhesive, to detach. Voucher specimens are often valuable. These may consist of properly preserved whole insects or parts thereof (e.g., wings of larger insects) to positively link the insect identification to the molecular data.

Purposes of molecular studies of insects vary widely but may also include taxonomic studies. Measuring degree of genetic relationships is valuable and is sometimes much more sensitive than traditional methods.