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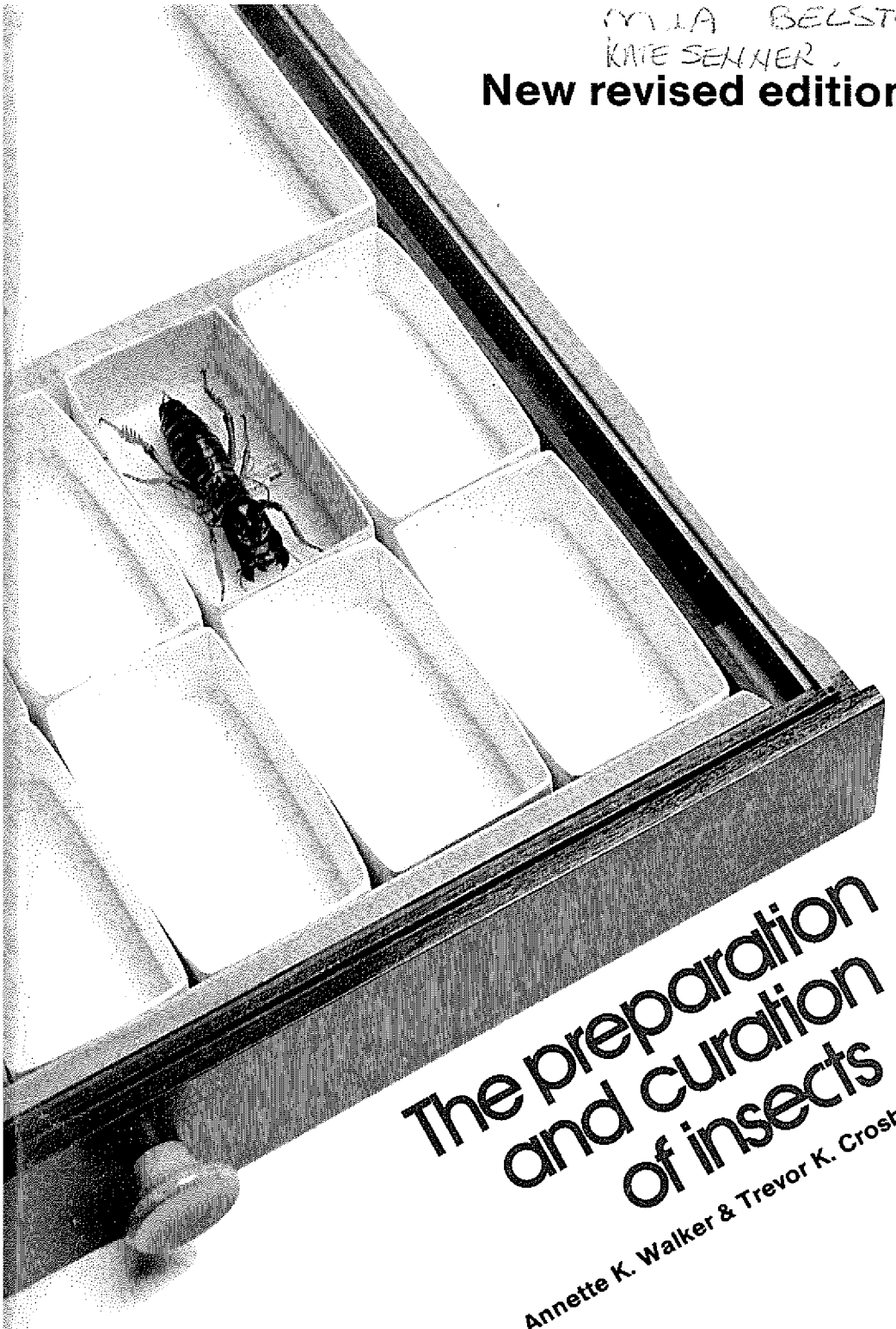

The preparation and curation of insects

Annette K. Walker & Trevor K. Crosby

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Male *Hemideina maori* (Pictet & de Saussure, 1891) (Orthoptera: Stenopelmatidae) in unit tray of a cabinet drawer.

Abstract

This handbook explains the methods and techniques used by workers of the New Zealand Arthropod Collection (NZAC) for preparing insects for its collection, and how the collection is curated and managed. Detailed information is given on the following topics: the preparation of specimens, including relaxing, pinning, card point mounting, double mounting, slide mounting, and labelling; organisation and storage of the collection; loans and the dispatching of specimens; restoration of specimens; hazardous properties of chemicals used; checklist of entomological supplies. Alternative methods and techniques have been provided for many NZAC procedures, which may be more suitable for using in other collections.

Contents

Introduction

Like most large insect collections, the New Zealand Arthropod Collection (NZAC) at Entomology Division, DSIR, Auckland, New Zealand, has accumulated specimens over many years and each year thousands more are added. Such a collection is rather like a library in that it must be well organised, with every specimen carefully labelled and easily accessible. High standards of preparation and documentation must be maintained or its scientific value will decline.

This handbook was written primarily to provide our insect preparators with a reference manual to our methods and techniques, as well as to provide a broader understanding of how the collection operates. It should ensure our collection will always be properly managed and consistent standards will be adhered to even with staff changes.

The methods and techniques do not necessarily reflect the preferences of other entomologists, but we hope that many of them will find this handbook useful and find some aspects worth adopting for their collections.

This edition has been extensively revised from the original 1979 edition. In response to requests and suggestions from colleagues, we have given more detailed explanations about our methods and techniques as well as provided alternatives for many. In addition, we have added a number of sections:

hazardous properties of chemicals used; formulae for the various solutions, fixatives, and media; and procedures for sending specimens to be identified.

Acknowledgments

Our special thanks go to the present and past long-serving insect preparators of Systematics Group, Entomology Division, DSIR, without whom this handbook would never have been initiated: Ms Jo Berry; Ms Beverley Bradshaw; Mrs Beverley Bennett; Mrs Grace Hall; Mrs Peggy Herbert; Mrs Joyce Hodges; Ms Nellie Katipa; Mr Lee Marchant; Ms Lillian Martin; Ms Heather McWilliam; Mrs Josefa Moynihan; Ms Vivienne O'Brien; Mrs Sue Paterson; and Mrs Margaret Tocker. We also thank the rest of the present and former staff of the Group for their suggestions and helpful criticisms: Dr Robin Craw; Dr Lewis Deitz (now North Carolina State University, Raleigh, U.S.A.); Mr John Dugdale; Mr Cleveland Duval; Mr Des Helmore; Dr Beverley Holloway; Dr Willy Kuschel; Dr Peter Maddison; Mrs Brenda May; Ms Clare Morales; Dr Graeme Ramsay; and Dr Charles Watt. Other useful comments and advice on techniques were received from: Dr Barbara Barratt (MAF, Invermay); Dr Stephen Foster (Entomology Division, DSIR, Auckland); the late Dr Ken Fox (Manaia); Mr Bruce Given (Blenheim); Dr Chris Green (Auckland); Mrs Olwyn Green (MAF, Lynfield); Prof. Roy Harrison (Lincoln College); Dr Allen Heath (MAF, Wallaceville); Dr Rod Macfarlane (Entomology Division, DSIR, Lincoln); Mr Ian McLellan (Westport); Mr Mike Nuttall (FRI, Rotorua); Mr Ron Ordish (National Museum, Wellington); Mr Brian Patrick (Dunedin); Prof. Bob Pilgrim (University of Canterbury, Christchurch); Dr Ken Somerfield (MAF, Levin); and Mr Peter Watts (Entomology Division, DSIR, Palmerston North). We especially thank Mr Des Helmore, illustrator, Systematics Group, for preparing the illustrations, and Science Mapping Unit, Wellington, for preparing the maps.

Killing and initial storing

Insects should be killed quickly and humanely. Specimens that are going to be stored in ethanol (ethyl alcohol) can be killed by placing them directly into 70–90% ethanol. Specimens that are going to be pinned can be killed with ethyl acetate vapour in a container (CAUTION: see hazardous chemicals p. 70).

The easiest way to kill specimens without chemicals is to place them in an airtight container in a deep freeze for at least 6 hours. Insects like large, hard beetles should be left longer.

More detailed explanations and other techniques can be found in books on collecting (e.g., Oldroyd 1958; Beirne 1962; Cogan & Smith 1974; Martin 1977; Upton & Norris 1980; Borror et al. 1981; Smithers 1981).

Field collections are usually stored in ethanol or layered between paper tissues. Specimens in ethanol can be set aside for sorting later. Layered specimens should have camphor added to each layering box to deter psocids and other insect pests, or if they have not dried out they can be placed in a plastic bag and stored in a deep freeze.

Freshly caught specimens that have not yet hardened can also be frozen and stored indefinitely in a sealed container. After thawing they will still be soft and pliable for pinning or setting.

Some specimens may need to be fixed before preserving in ethanol (formulae p. 79).

Handling specimens

Specimens kept in ethanol must not be allowed to dry out. If a sample in an open dish is to be left for a period, always cover the dish and switch off the microscope lamp.

When removing specimens from ethanol some may stick to the sides of vials. These may be rinsed out with a squeeze bottle of ethanol or removed with a fine brush.

Dried specimens should not be left uncovered, especially overnight, as they may be removed by enthusiastic cleaners, or eaten by rodents, ants, or cockroaches! In the tropics, ants might remove them.

Take particular care when handling store boxes containing dried pinned specimens as these are fragile. Open the type of store box we use as in Fig. 1. Hold it flat so any loose specimens will not fall out. Also, grip the box well away from the sides to avoid smudging any pencilled labels.

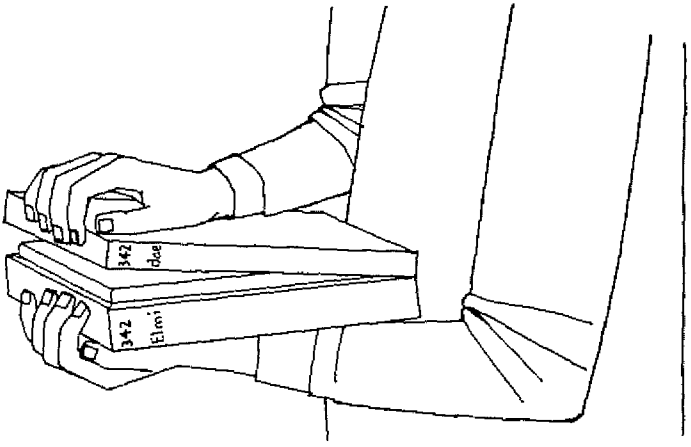


Fig. 1 *Correct way to open our store boxes.*

With glass-topped drawers, lift the glass lid off the drawer very slowly or else a rush of air may enter the drawer and blow off delicate legs, antennae, and wings.

CAUTION: loose clothing; dangling ties, hair, and jewellery; and coughing and sneezing can all damage delicate specimens. Don't smoke when handling specimens.

Implements used

Handle specimens carefully to avoid damaging legs, antennae, wings, bristles, etc. Fine, soft brushes are ideal for handling insects which are either dry or in ethanol. Sable brushes are best, but the less expensive camel-hair brushes are almost as good.

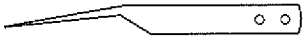


Fig. 2 *"Storkbill" forceps.*

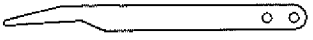


Fig. 3 *"Featherlight" forceps.*

Forceps are indispensable laboratory tools.

(1) Lightweight forceps: fine-tipped "Storkbill" for handling unmounted dry specimens, and blunt-nosed "Featherlight" for handling larvae (Fig. 2, 3).

(2) Pinning forceps: for handling pinned insects. Grip the shaft of the pin (Fig. 4), not its head. Some flat-headed pins can slip out of the grip of the forceps, which may result in irreparable damage to a specimen.

(3) Watchmaker's forceps: these have fine hard tips and are most useful for fine dissections. The pressure applied to a specimen can be cushioned considerably by placing the edge of the forefinger between the jaws (Fig. 5).

Care must be taken not to drop these forceps as the tips will bend. If necessary, damaged tips can be ground with a fine sharpening stone, but use a dissecting microscope to ensure that the tips are ground to an equal length and they close properly.

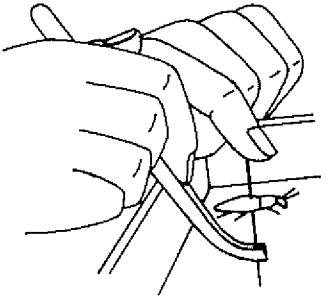


Fig. 4 *Use of pinning forceps to grip the pin and steady the specimen.*

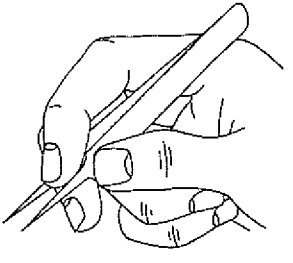


Fig. 5 *Cushioning the pressure applied to a specimen with watchmaker's forceps.*

Do not allow the container to stand in the sun as it may result in condensation forming. The ground glass edge of a desiccator should have a small amount of petroleum jelly smeared on it to ensure airtight contact with the lid; to open and close it, the lid should be slid sideways.
HINT: Containers to hold one or several small specimens in the relaxing chamber can be made of metal foil (milk bottle tops) moulded around the bottom of a vial or a small cube-shaped piece of wood, or you can use plastic lids.

IMPORTANT:

(1) Check specimens daily; they may rot or discolour if left in the humid atmosphere too long.

(2) Be sure no labels in the moisture chamber are written in ballpoint or fountain pen ink which may run and become indecipherable.

(3) There should be no direct contact between the specimens and the water.

A quick alternative method is to place specimens on a moist surface in an airtight container in an oven set at about 40°C. In a few hours specimens will be relaxed enough to handle, but they must be mounted immediately after removal from the oven for they harden in a matter of minutes. Take a few specimens at a time for mounting, and keep the remaining specimens covered.

Insect pins

The pins must be stainless steel or they will corrode and eventually ruin the specimen. Although stainless steel pins are expensive there is really no substitute. We have found the "Continental" type of pin to be the most satisfactory. The number 3 pin is a good general purpose size, suitable for direct pinning of large insects, card points, and double mounts. Other thinner sizes (0 and minuten) are necessary for more delicate insects. We do not recommend the use of 00 and 000 pins for direct pinning as they are very springy; specimens are apt to be damaged even when the pins are handled carefully. Short pins for fastening down separate identification labels (cabinet pins) do not have to be stainless steel, but it is advisable for them to be nickel-plated.

Mounting insects on pins

General points

(1) Position specimen about 3/4 of the way up the length of the pin (approx. 27 mm), depending on the thickness of the specimen. Positioning is important; sufficient room must be left above the specimen so that the shaft of the pin can be gripped with pinning forceps or fingers, and there must be room below the specimen for several labels (Fig. 7).

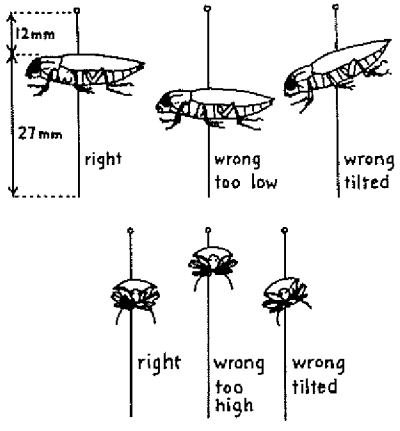


Fig. 7 *Right and wrong way of positioning a specimen on a pin (after Oman & Cushman 1946).*

(2) Use the firm number 3 pin whenever possible; the finer 0 and minuten pin sizes are for special cases only (small Hymenoptera, most calyptrate Diptera).

(3) Make sure legs are not too bunched up and that antennae are clearly visible.

Before specimens can be stored in a permanent place they must be dry. This may take a day or a week depending on specimen size. Temporarily store them in a box that has not been made airtight and where there is adequate air circulation.

Further instructions for pinning follow: Acari (p. 24), Coleoptera (p. 24), Diptera (p. 25), Hemiptera (p. 25), Hymenoptera (p. 25), Lepidoptera (p. 26), and Orthoptera (p. 30).

Berté (1979) outlines the use of acetone to improve retention of colour patterns for some groups of insects (adult Odonata, Corixidae and Notonectidae (Hemiptera), Dytiscidae (Coleoptera), and Acrididae (Orthoptera)).

Card point mounting

Making card points

Card points are punched from 3-ply Bristol board using a point punch. This card will not curl when a pin is pushed through it, yet it is thin enough for a pin to be inserted easily. Also, the tip can be bent without separating into layers; Orbit Ivory Board can be used as a substitute. Zimmerman (CSIRO, Canberra, pers. comm.) recommends a 100% linen ledger paper, as the linen fibres are of such character that the point does not easily loosen on the pin. Some manufacturers offer packets of ready-punched points; Australian Entomological Supplies (address p. 76) provide an 11 x 3.5 mm point.

To assemble a card point insert a number 3 pin 3 mm in from the wide end of the card point. (HINT: wear a finger guard to protect the index finger and to grip pins more firmly.) Do this on a firm cork base to prevent the tip curling. Push the card point 27 mm up the pin with the aid of a pinning block (Fig. 8). Make sure the card point is not loose on the pin; discard it if it is.

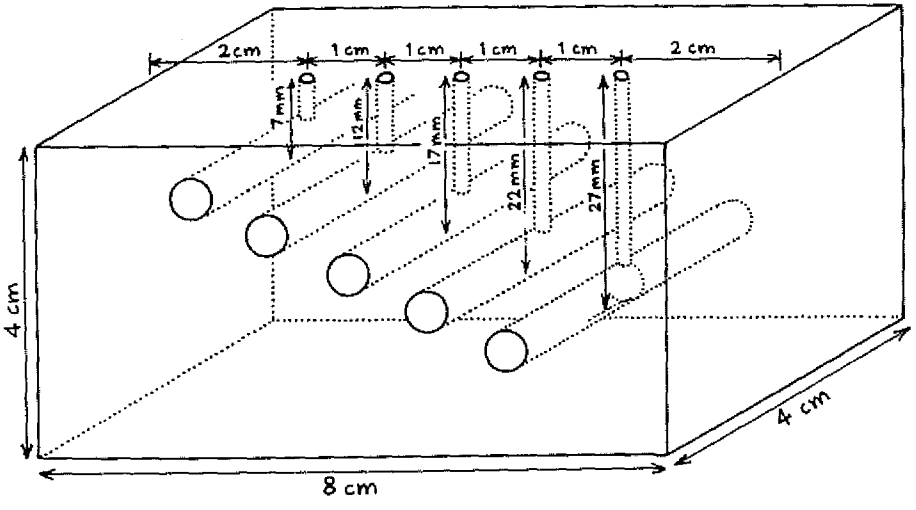


Fig. 8 *Metal pinning block. The bottom of the vertical hole corresponds with the lower surface of the transverse hole (the transverse hole is to prevent clogging of the vertical hole with paper fragments).*

General procedure for mounting specimens

(1) Place several specimens on the edge of a raised surface with their heads facing left (Fig. 9), or right if the pin is not going to be inverted (Fig. 10), so that the tip of the card point can be placed easily against the right-hand side of each specimen near the middle right leg (Fig. 10, 11).

For larger specimens, bend the final 1–2 mm tip of the card point downwards to form a right angle, either with forceps, or with the fingernails of forefinger and thumb.

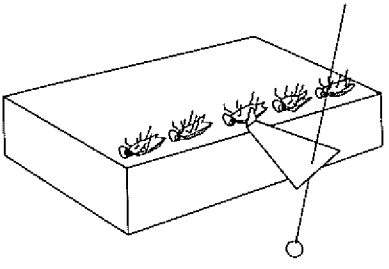


Fig. 9 *Card point mounting: specimens on backs, heads to the left, pin inverted.*

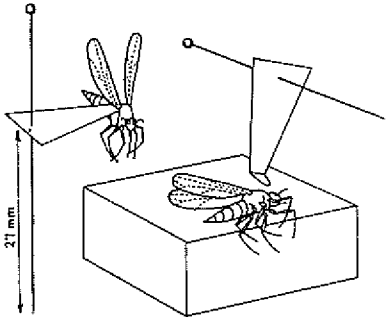


Fig. 10 *Card point mounting: specimen on its left side, head to right, pin not inverted.*

(2) Smear a small drop of glue (see p. 18) on the tip of the card point (a needle is useful for doing this).

(3) Turn the pin and press the glued tip of the card against the specimen (Fig. 11). Check that the specimen is orientated as in Fig. 10 and 12 when finished. There

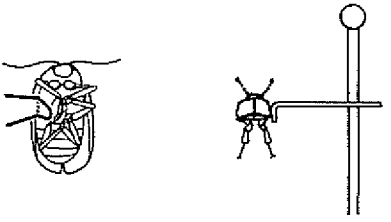


Fig. 11 *(left) Card point mounting: the tip of the card point attaches to the specimen near the middle right leg (after Beirne 1962).* ***Fig. 12*** *(right) Card point mounted specimen viewed from rear (after Borror, DeLong & Triplehorn 1981).*

must be an unobstructed view of the bottom, top, and one side of the specimen.

(4) Align any specimens which are not glued squarely by a light touch with finger or forceps.

For small specimens it is essential to use a dissecting microscope to ensure they are mounted correctly.

For larger-bodied specimens, stick the pin into Plastazote at about a 30 degree angle to the perpendicular for several minutes (Fig. 13). This means the weight of the specimen is against the card point, and allows the glue to stick to the specimen effectively.

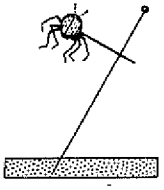


Fig. 13 *Large-bodied specimen just glued to a card point; leave for a few minutes like this while the glue dries.*

Insect-mounting glue

A water-soluble glue is recommended in case specimens may have to be removed for study. Glued specimens can then be removed from their mounts by immersing them in warm water or by gently brushing droplets of water round the specimen (p. 64).

Tillyard's insect mounting glue (Tillyard 1926: 494) has been used in our collection for over 60 years and early specimens are still adhering to their mounts satisfactorily. It is important that the glue be of the correct consistency (i.e., forms a droplet on the end of a needle). Dilute thick glue with warm distilled water otherwise it may not penetrate into the fine depressions or enclose the hairs of a specimen; consequently, a specimen may shake off a card point even under normal handling conditions. If the glue is too thin specimens will not stay on the points; leave the lid off the container for a few hours.

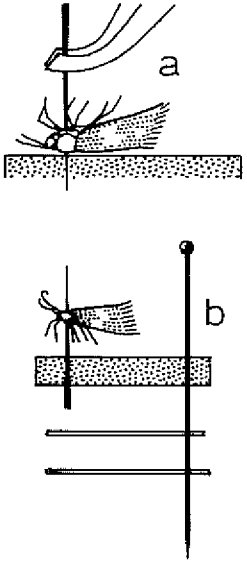


Fig. 14 *Reverse-mounted specimen (a, insert minuten tip through the specimen; b, insert minuten base through the Plastazote).*

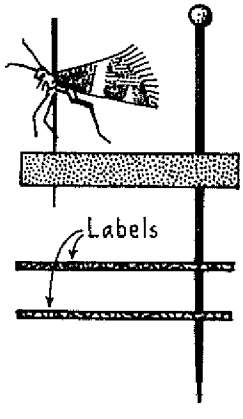


Fig. 15 *Double-mounted specimen.*

Seccotine (trade name) is an alternative water soluble glue. It does not dry as quickly as Tillyard's glue, but has the advantage of not going "stringy".

Nail varnish. In areas of high humidity it is best to use an alcohol-soluble or acetone-based adhesive such as clear nail varnish. Problems can arise with a water-soluble glue as moulds may develop or the glue may not harden. Large parasitic Hymenoptera such as ichneumonids are usually mounted on the side of a pin using a quick-drying nail varnish (p. 26).

Cardboard rectangles

These are an alternative for mounting small insects. They are favoured by specialists working with some small beetles and parasitic wasps (Noyes 1982). Advantages of this method are that the specimen is protected, and it can be mounted so it lies either flat or slightly tilted on its side to show its face and side of body (Fig. 31).

Double mounting

Small, fragile specimens, especially minute Lepidoptera, are pinned with a very fine pin called a minuten. However, this pin is not strong or large enough for labels to be added underneath, or for handling. Therefore, a double mount is used with the minuten pinned to a Plastazote (polyethylene enclosed-cell foam plastic) mount. Previously, polyporus pith (the cut up context from a bracket fungus) was used.

Cut these mounts from dense Plastazote to 18 mm long, 4 mm wide, and 5 mm deep. Insert a number 3 pin 3 mm in from one end, and position the mount so that the top is 27 mm up the pin, using the 27 mm hole of a pinning block (Fig. 8).

Double mounting is carried out as follows:

(1) Place specimens on a piece of Plastazote. Specimens under 4 mm long or with fragile legs must be placed upside down, i.e., reverse mounted (Fig. 14).

(2) Use a dissecting microscope and a pair of pinning forceps to insert a minuten (number 0.15) through the thorax between the front legs. Hint: rather than pushing a minuten down through a specimen with pinning forceps, it is often easier to push the specimen up on to the minuten by carefully lifting the Plastazote while holding the minuten still.

(3) Push the pin through the Plastazote mount, but leave enough room to avoid damaging the legs of the specimens (Fig. 15).

Insects in ethanol

Soft-bodied adults and larvae, which if kept dry would collapse and become useless for study, are stored in ethanol. Use 75% ethanol to ensure that the losses from evaporation during handling do not lower the ethanol concentration below 70%.

Ethanol is also a convenient method for storing large numbers of one species collected from the same locality. Storage of some of these specimens in ethanol saves the time and labour required for dry mounting and labelling. Caution: long-term storage in ethanol is not suitable for some insects that should be slide-mounted (e.g., aphids, mealybugs, chalcids, thrips, cecidomyiids); and storage should be out of light (p. 49).

Guidelines for dry-mounting duplicate specimens from ethanol

1–5 specimens – mount all except when further examination of internal structures might be required, or when adults are associated with larvae;

6–20 specimens – mount half and leave the rest in ethanol;

20+ specimens – mount 10 and leave the rest in ethanol.

Mount the best specimens only, leaving damaged specimens in ethanol.

A label "duplicate specimens mounted" must be included with specimens left in ethanol, and a label "duplicate specimens in ethanol" pinned to the mounted series.

Insects on microscope slides

Very small insects, and parts of insects, that will be closely examined under a compound (high-power) microscope, are mounted on microscope slides. The specimens are protected with glass coverslips which lie over the specimen in the mountant. Coverslips come in various sizes to suit specimen size.

Slide making should be carried out in a room with adequate ventilation, as some solvents are hazardous, e.g., xylene.

There are three main types of microscope slides:

(1) Glass slides. Standard 76 x 25 mm slides, 0.8/1.0 mm thick, with ground edges.

(2) Cobb aluminium double-coverslip slides. These slides allow specimens to be viewed from both sides. The frame has a hole cut in the bottom over which a 25 mm square coverslip is positioned. Specimens are mounted on this coverslip in the normal way and

covered with a further coverslip. Thick card is then positioned each side of the square coverslip (Fig. 16) and the slide frame margins crimped down to hold the card.

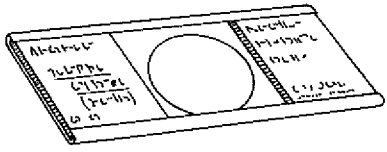


Fig. 16 *Cobb aluminium double-coverslip slide.*

These slides are light and can be stacked without damage to the specimen, but their cost is slightly higher than conventional glass slides, and their preparation takes longer.

(3) Cavity slides. These are the standard 76 x 25 mm size with a shallow depression in the middle. They are useful for mounting larger specimens which cannot be accommodated on normal slides.

Procedures for making a slide

Mounting a specimen on a slide is the final step in a long procedure. Each insect group requires certain particular methods of clearing, staining, and dehydration which are too specialised to deal with here. However, the techniques for slide mounting are standard, and are outlined below.

Usually a slide mount is made by placing a drop of mounting medium on to the slide, carefully arranging the specimen in the medium, and then gently lowering a coverslip over the specimen and medium. Lower a coverslip by placing one edge on a slide, then slowly lower the coverslip to meet the mounting medium and slide; the edge of the coverslip which is held should be the last part to meet the slide.

Winged specimens, or specimens which require careful arrangement are more easily arranged upside-down on a coverslip. A cardboard holder with a shallow depression is used to hold the coverslip in place (Fig. 17). For

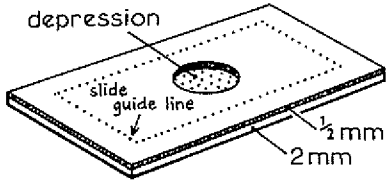


Fig. 17 *Cardboard holder for coverslip mounting.*

convenience, the holder is slightly larger than a standard slide.

To mount a specimen on a coverslip;

(1) Place coverslip in depression in cardboard holder;

(2) Put a drop of mounting medium in the centre of coverslip;

(3) Arrange the specimen upside-down on coverslip;

(4) Gently lower slide on to coverslip;

(5) Invert slide. If further arrangement of specimen necessary, lightly press on coverslip.

General points on slide making

(1) Make sure all glass slides and coverslips are thoroughly clean (clean with ethanol on a tissue or rag).

(2) Generally only one specimen or species should be mounted on each slide.

(3) Keep the specimen and coverslip as near as possible to the middle of the slide.

(4) Lower the coverslip gently on to the mounting medium to avoid trapping air bubbles or squashing the specimen.

(5) Use the standard label format, as described on p. 37. Make sure the slide label is in the reverse position to the specimen as in Fig. 45 (i.e., the head of the specimen points towards the bottom of the label) so that its image is the right way up under the compound microscope (i.e., the head points towards the top of the label).

Mounting media

Gatenby & Beams (1950), Gray (1954), Pantin (1959), and Peacock (1973) give excellent summaries of the various mounting media and should be consulted for further information. In morphometric studies use only one type of mountant, as specimens may alter in size because of different processing methods and reagents used for different mountants (Mound & Palmer 1981).

(1) Resin-based media. These have excellent transparency, but are not soluble in water, so specimens must first be dehydrated in increasing concentrations of ethanol. Canada Balsam is particularly recommended as its refractive index (1.53) is very close to that of glass, and, as it has been in use longer than any other mounting medium, its stability is well proven. It is soluble in xylene. Palma (1978) gives a detailed account of the method, and Mound & Walker (1982) outline the method for mounting thrips.

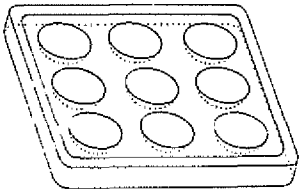


Fig. 18 *Transparent Pyrex spot plate.*

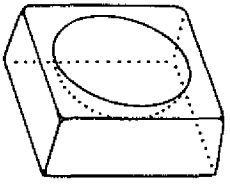


Fig. 19 *Glass staining well.*

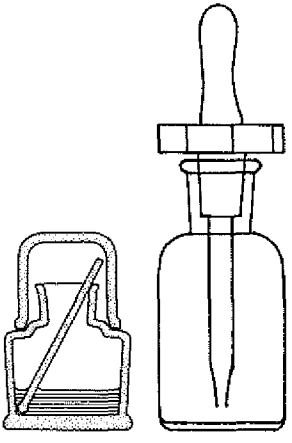


Fig. 20 *(left) Balsam bottle.* ***Fig. 21*** *(right) Dropping bottle.*

If it is an advantage to mount direct from absolute ethanol, or to avoid xylene, Euparal can be used. Its refractive index is slightly lower (1.48).

Specimens mounted in resin-based media must be dried slowly in an oven at 40–45°C for about 6 weeks. Quick drying can be achieved with a microwave oven but care is required: dry for about 1 minute. This may be useful when an identification is wanted quickly.

(2) Water-based media. Gum chloral media are the most widely used; Hoyer's, Berlese's, de Faure's, and Andre's media (p. 79) all have different amounts of gum arabic, chloral hydrate, glycerol, and other additives. Varying the proportions of the ingredients allows the best refractive index to be obtained for the particular requirements of the group being examined.

Specimens can be mounted directly from water or any concentration of ethanol and will clear within a few days. Specimens can be remounted by dissolving the medium in water. The slide must be hardened for 2 or 3 weeks at about 30°C, then the coverslip should be ringed with a preparation such as Glyptal. Wu (1986) outlines a quick method for ringing.

(3) Other mounting media sometimes used include various formulations of polyvinyl alcohol (PVA) which have the advantage of clearing most specimens within 24 hours (see Salmon 1947, 1954). Although PVA has good optical and clearing qualities, the preparations shrink on drying, and it is difficult to recover specimens. We do not recommend their use for permanent slides.

Other media based on methyl cellulose ("Methocellulose") and polyethylene glycol (carbowax) may also be used.

Useful equipment for slide making

(1) Transparent Pyrex spot plates (Fig. 18) are useful for processing very small specimens during clearing, staining, and dehydration, especially if specimens need to be heated.

(2) Staining wells (cavity dishes) (Fig. 19) are useful for processing individual specimens.

(3) Balsam bottles (Fig. 20) are useful for dispensing mounting media. Keep only a small quantity of medium in a bottle to ensure only a small drop forms on the glass dropping rod. If the medium becomes too thick, dilute with the appropriate solvent.

(4) Dropping or pipette bottles (Fig. 21) are used for dispensing small amounts of ethanol, clearing, and staining agents.

Fig. 22 Lifter made from modified hypodermic syringe needle.

(5) Lifters. We use modified hypodermic syringe needles (Fig. 22) for lifting specimens from one solution to another. These are made up as follows:

(a) Grind about 5 mm from one side at the end of a hypodermic needle so that slightly less than half the diameter remains;

(b) Bend the ground end inwards at right angles to form a spatula shape at the tip;

(c) Mount the needle in any convenient handle.

The eye of a sewing needle can be used as an acceptable alternative.

(6) Dissecting probes. Strong and extremely sharp probes can be made from tungsten wire as described by Galbreath & Galbreath (1977).

General points on preparing and preserving major orders

Acari (mites)

Mites of the same species and from the same locality can be bulk stored in a mini-vial. The mites are cleared by heating in lactic acid and stored permanently in polyethylene glycol (PEG 300). The mini-vial is pinned, labelled, and stored in the same way as a dried specimen (Fig. 55). Specimens have been kept successfully 12 years by this method.

Specimens requiring detailed examination are usually mounted on Cobb aluminium frame slides (p. 21). Some mounting methods are given by Foulkes (1983), and Fain (1980) gives a procedure for remounting mites in water-based mountants without disturbing specimens.

Coleoptera (beetles)

(1) Store most Coleoptera less than 1 mm long in ethanol.

(2) Larger Coleoptera, 1–10 mm long.

(a) Pin with a number 3 pin those that can be mounted without damage (Fig. 23, 24); usually these are wider-bodied species over 7 mm long.

(b) Glue specimens under 7 mm long, or those with long slender bodies, to a card point (see Card mounting, p. 17, and Fig. 11, 12).

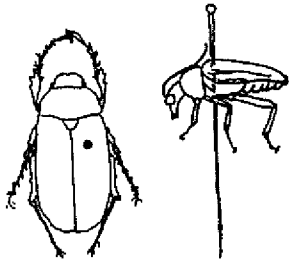


Fig. 23 (left) Pin position for Coleoptera (after Borror, DeLong & Triplehorn 1981). Fig. 24 (right) Position of pinned specimen (after Beirne 1962).

(3) Pin Coleoptera longer than 10 mm. Hold the beetle in one hand between the thumb and forefinger, and insert a number 3 pin through the right elytron (Fig. 23). The pin must be at right angles to the body of the beetle (Fig. 24).

If there are larvae associated with adults keep at least one adult with them in ethanol. A label "associated larvae" must be pinned to some of the

mounted specimens, and a label "associated adults mounted" added to the vial with the larvae.

Diptera (flies)

Freshly killed specimens may be pinned before they harden or layered (stored between layers of paper tissue). They can also be placed directly into ethanol, but only very rarely should flies be subsequently removed from ethanol and allowed to air-dry (see Technique, p. 62). However, it is possible to critical point dry specimens from ethanol and achieve superior results to ordinary air-dried pinned specimens (p. 51).

Layered specimens must be relaxed before handling (see Relaxing, p. 13).

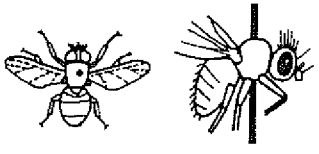


Fig. 25 *(left) Pin position for Diptera.* ***Fig. 26*** *(right) Side view of pinned specimen (both after Beirne 1962).*

(1) Small Diptera. Glue to a card point (p. 17, and Fig. 10) or double mount (p. 19).

(2) Large Diptera (e.g., blowflies, tachinids). Insert a number 0 pin through the thorax slightly to the right of centre to avoid damaging delicate bristles used for identification. Spread the wings apart (Fig. 25, 26).

Hemiptera (leafhoppers, scales, aphids)

Field collections of Hemiptera are either stored in ethanol or kept dry (layered). All immature stages remain in ethanol (exception: large leafhopper nymphs can be mounted). Small delicate adults like aphids, mealybugs, and scales should be stored in ethanol only for a short time as they deteriorate with long-term ethanol storage; ideally they should be slide-mounted as soon as possible. Aphids can be stored dry for later slide mounting.

(1) If the scutellum is large enough to take a number 3 pin, hold the bug in one hand between thumb and forefinger, and insert the pin slightly to the right of the middle of the scutellum (Fig. 27).

(2) Card mount Hemiptera with a scutellum too small to take a number 3 pin (p. 17).

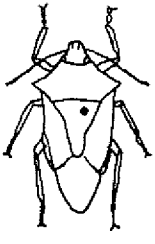


Fig. 27 *Pin position for Hemiptera (after Borror, DeLong & Triplehorn 1981).*

Many large adult bugs are very hard, and can be removed from ethanol, dried, and pinned. If there are immature stages associated with the adults keep at least one adult with them. A label "associated nymphs" must be pinned to some of the mounted specimens, and a label "associated adults mounted" added to the vial with the nymphs.

Hymenoptera (wasps, bees, ants)

Field collections of Hymenoptera are usually stored between layers of paper tissue or in ethanol. Relax layered specimens before mounting (p. 13). Don't transfer dried specimens to ethanol.

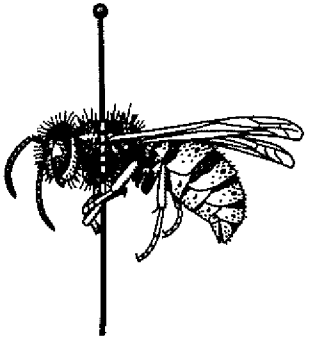


Fig. 28 *Pin through thorax of large roundish hymenopteran.*

(1) Large Hymenoptera. For roundish specimens like bees and wasps, insert a number 3 or 0 pin through the thorax slightly to the right of centre, and push the specimen about 3/4 the way up the pin (Fig. 28).

Laterally compressed specimens, like ichneumonids and large braconids which have their wings raised vertically up above their bodies, can be glued to the side of a pin with a quick drying glue like nail varnish (Fig. 29).

(2) Small Hymenoptera. Mount on card points (p. 17). Glue a specimen to a card point in the middle of the thorax using as little glue as possible; ensure the head, legs, and wings are free from glue. Those which have their wings raised vertically above their bodies are mounted on the right hand side of their thorax on the top of the card point with Seccotine glue; the wings extend out flat from the tip of the card point (Fig. 30). If the wings are spread out from the body, ensure the specimen is only attached to the card point on one side of the thorax.

Chalcid mounting. Mount on card rectangles (p. 19) using seccotine glue. Spread the wings out at right angles to the body, and tilt the specimen slightly on its side to show its face and one side of its body (Fig. 31; Noyes 1982).

When a parasite has been reared from a host, mount the host remains, e.g., pupal skin, together with the parasite, either on the small point or on an additional point below the specimen. If large numbers of a parasite emerge from a host, these can be stored dry in a mini-vial (p. 50).

Specimens can be air-dry mounted from ethanol using the following procedure:

(1) Remove a specimen from ethanol with watchmakers forceps and lie it flat on a slow drying, smooth surfaced paper (we find library index cards very suitable).

(2) With the excess ethanol gathered in the watchmakers forceps, make a puddle of ethanol around the specimen and brush the wings out flat.

(3) Allow the specimen to dry slowly at room temperature, then mount.

Critical point drying of specimens from ethanol gives excellent results (p. 51).

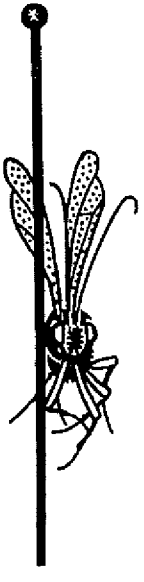


Fig. 29 *Laterally compressed type of hymenopteran glued to the side of a pin.*

Lepidoptera (butterflies, moths)

Ideally, specimens should be pinned and their wings spread straight after killing while they are still relaxed. They may be temporarily stored between layers of soft tissue paper

Fig. 30 *Card point mounted hymenopteran.*

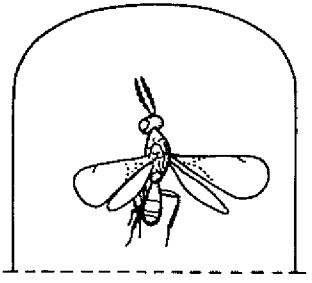


Fig. 31 *Chalcid mounted on card rectangle (after Noyes 1982).*

Fig. 32 *(left) Pin position for Lepidoptera (after Beirne 1962).* ***Fig. 33*** *(right) Side view of pinned specimen.*

and relaxed (p. 13) before handling in the laboratory. If freshly killed specimens are stored in a deep freeze they can be pinned and set as soon as they thaw. Never store adult Lepidoptera in ethanol. Caterpillars lose their coloration when stored in ethanol, but maintain some coloration if they are critical point dried.

Insert a pin through the middle of the thorax until the specimen is about 3/4 of the way up the length of the pin (Fig. 32, 33), then spread the wings (Fig. 34).

(1) Large moths and butterflies: number 3 pin.

(2) Smaller moths: number 0 pin.

(3) Moths under 10 mm long: mount on a minuten pin to avoid direct handling (Fig. 15).

(4) Moths under 4 mm long: reverse mount on a minuten (Fig. 14).

Setting (spreading of wings)

Ideally some specimens from a series should be carefully mounted to show their markings. Don't be disappointed if your first attempts are not perfect – practice and a great deal of skill are needed to set specimens perfectly for study or display. Long spreading boards are preferred as they are safer and more convenient for setting large numbers of specimens (Fig. 34).

Mould sometimes is a problem when drying out specimens on a board – 10% chlorocresol soaked and dried into the board's surface prevents this.

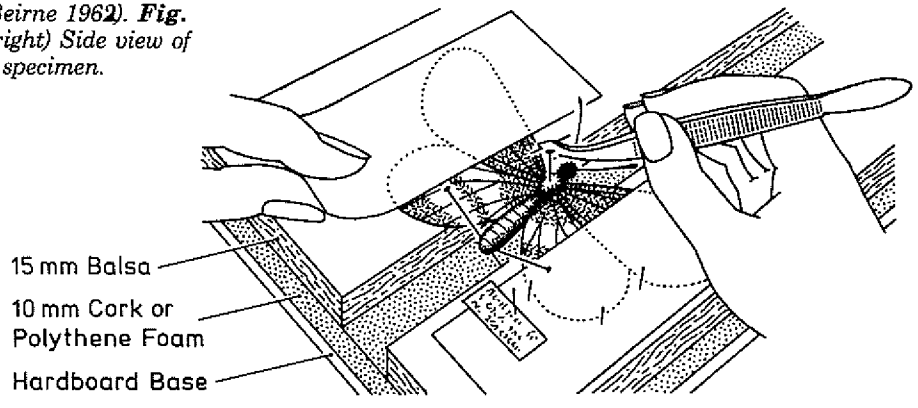


Fig. 34 *Setting Lepidoptera (after Upton & Norris 1980).*

Before setting specimens on a board, run the handle of a scapel along the pinning surfaces to smooth down the rough edges of the pinholes left from previous use. A smooth surface ensures that the wings can be spread easily and without loss of scales. Use new paper strips each time for the same reasons.

HINT: As an alternative to traditional balsa or cork pinning boards, pieces of Plastazote can be glued together. Grooves of varying depths can be cut to suit different specimen sizes. Handle Plastazote boards carefully as they can bend and ruin your setting.

Field setting

Ideally, specimens should be pinned straight after killing and their wings spread while they are still relaxed. However, even partial wing spreading while pinning in the field makes final setting much easier. Field pinning and setting can be managed in the following way:

(1) Large moths and butterflies: Pin so the body is on the Plastazote surface of the field store box, and spread the wings as far as practical (Fig. 35). Later relax the specimen, push it 3/4 of the way up the length of the pin, and spread the wings on the setting board to their final position as detailed on next page.

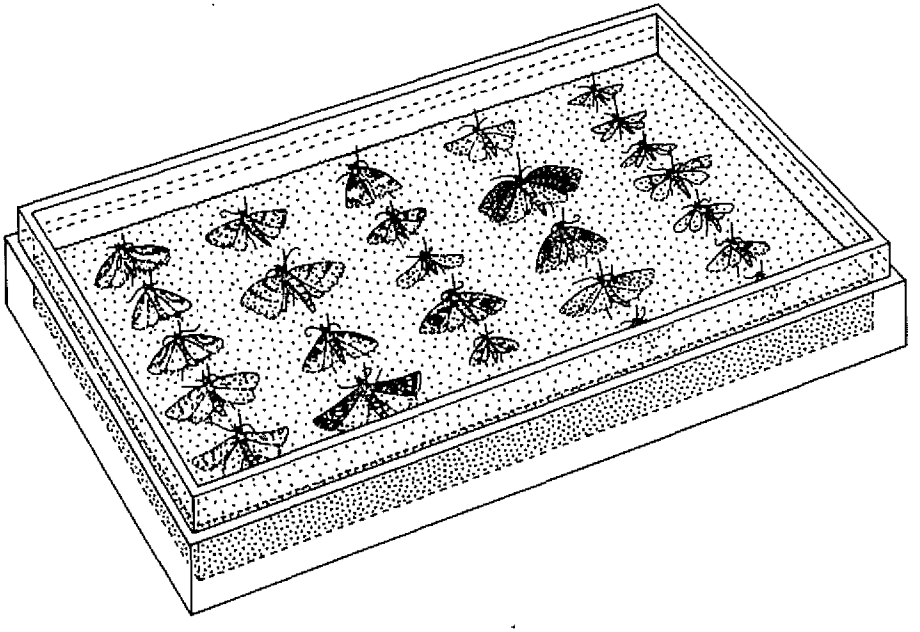


Fig. 35 *Large Lepidoptera pinned in the field, and their wings partly spread.*

(2) Small moths which require double mounting: Pin with a minuten so the body is on the Plastazote surface of a shallow plastic box, and spread the wings as far as practical. Later relax the specimen, spread the wings to their final position as detailed below, and double mount.

Final setting

(1) Push the pin on which the specimen is mounted into the bottom of the channel of the spreading block or board until the wing hinges are level with the pinning surface. Ensure that there is room on the setting board for the wings to be spread forward, and the body fits snugly into the groove.

(2) Prop up the abdomen with a small wad of cottonwool or with pins.

(3) Gently hold down the pair of wings on one side with soft-nosed forceps and fasten them down with paper strips. Repeat with the other side. Small moths must be pinned down with lightweight paper or cellophane strips. Individual "wing holders" can be made using stiff, shirt-box cellophane which are kept in position with a pin.

(4) Place the point of a fine pin behind one of the main wing veins of the forewing with pinning forceps (Fig. 35) and gently ease the forewing forward under the paper strips. Forewings should be spread only until the hind edge is at right angles to the thorax. Hindwings should then be spread so that the folded wing base is opened. If either wing is held too tightly by the paper strip, slip a pin between the block and the strip to relieve the pressure.

(5) Fasten the wings down with additional paper, ensuring that the tips of the wings are covered as these tend to curl when drying. Pins fastening the paper can be angled out to increase the pressure if necessary.

Make sure the collection data are kept with the specimen. Dry overnight at 30°C, or 2–6 weeks at room temperature.

Small Lepidoptera are set on a balsa board, in a groove 4 mm deep.

HINT: Freshly killed specimens can be easily spread by placing them on a piece of Plastazote after it has been rubbed against cotton cloth to give it an electrostatic charge. Also, the slightly textured surface of Plastazote "grips" the wings.

Orthoptera (wetas, grasshoppers) and Phasmatodea (stick insects)

Inject with Bouin's fixative (p. 79) and store in ethanol.

If a very large specimen is to be pinned for display, slit the underside of the abdomen open, remove the gut contents, and fill with sufficient cottonwool to maintain original body size and shape (be careful not to rub the colour off stick insects).

Specimen labels

General points on labelling

Because labels must last as long as the specimens, it is important to take great care to write legibly using good quality paper and durable ink.

Label paper must have a high standard of pH neutrality, colour fastness, strength, and smoothness of writing surface. The importance of a good quality paper can be easily underestimated. 50 years may pass before faults begin to appear. We have found Goatskin Parchment Paper to be very suitable for labelling both dried insects and specimens in ethanol.

Ink. Use black, waterproof, quick-drying ink. Allow labels to dry for at least 5 minutes before placing them in ethanol.

Pen. A drawing pen with an 0.18 nib is most satisfactory for labelling. Such pens must be handled carefully; they are not like disposable ballpoints. They should be kept clean. When not in use, the pen must be capped to protect the nib and to prevent the ink from drying and clogging the nib.

Labels must be printed in a small, neat hand. For dried specimens a label should be no larger than 12 x 8 mm; for specimens in ethanol it can be up to 35 x 12 mm.

Take care when writing labels from field data not to alter the order of words; for example, "Magnetic Cove Station" is not the same as "Magnetic Station Cove". Also, if words are left out of the original collecting data, information may be lost, e.g., "beaten from *Melicytus ramiflorus*" must not become "*Melicytus ramiflorus*", which gives no information on the relationship between the specimens and *Melicytus ramiflorus*.

When a common name for a host plant or animal has been used on the temporary field label, this must be changed to the scientific name on the permanent specimen label. The scientific name should be underlined if possible. All altitudes recorded in imperial feet must be converted to their metric equivalents (p. 91). Ensure the year part of a date is written in full with the century, i.e., "1985", not "85".

Aids to check queries about localities

We keep a diary to record field trips. We include the dates of trips, who went, the localities collected in, and information about the collecting conditions. This can be consulted if there is any doubt about hard-to-read or incomplete field labels.

To check localities and their spelling we hold a reference set of current inch-to-the-mile maps (NZMS 1) and 1:50 000 metric series maps (NZMS 260), and the "Gazetteer of New Zealand Place Names". These are published by the New

Zealand Department of Survey and Land Information. We also hold a set of the maps which can be taken into the field.

We use a standard card to record information about bulk collections of specimens (Fig. 36). The card index can be consulted if there are any queries arising about localities or how specimens were collected.

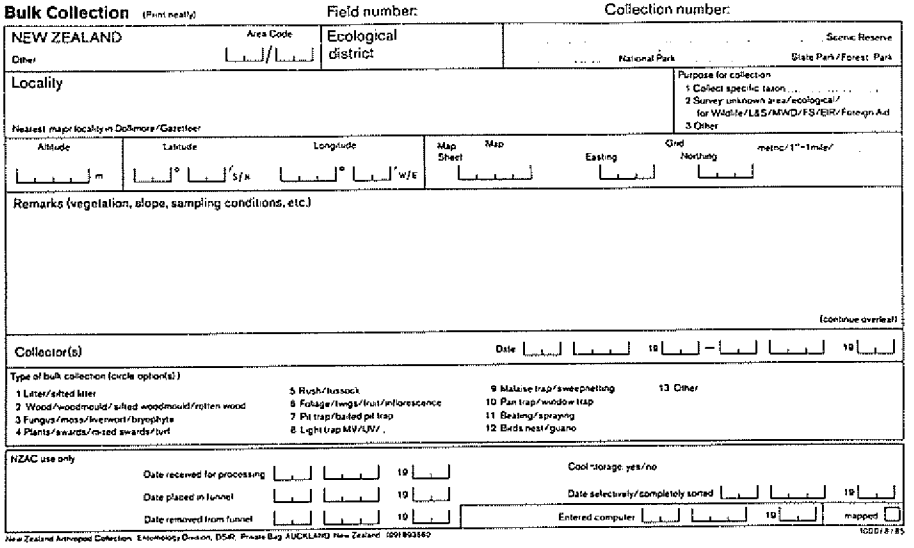


Fig. 36 *Form to record information about bulk collections of specimens.*

Handwritten labels

Labels are written by hand unless more than five with the same wording are required. To keep within the standard 12 x 8 mm size limit of labels for pinned insects, place a sheet printed with dark guide lines under the label paper. Note: the 12 x 8 mm label size is the standard for the N.Z. Arthropod Collection; in other collections where more lines need to be handwritten on labels than in ours a slightly larger 15 x 8 mm label may be more practical, resulting in neater labels and the avoidance of second labels.

Handwritten labels in ethanol need not have such small writing. The standard size of a label in ethanol should be about 35 x 12 mm, which allows four lines of writing and is about 3/4 the length of a standard 50 mm vial. A label must be placed in a vial so that it can be read from left to right when the closure is at the right-hand end. Before inserting a label bend it along its long axis so that it will be close to the side of the vial; bend it along your index finger, or along a pencil.

When inserting a label into a vial containing specimens, gently ease the label down the side of the vial to prevent any specimens from being caught between the label and the side of the vial, where they can be damaged or obscure the data. Ensure the label is not too long, otherwise it may cut specimens resting on the bottom of the vial when the closure is pushed in.

Never fold labels, as specimens can be easily trapped, squashed, or lost during removal for examination.

Photographic paper labels can be used in ethanol, but any additional writing in Indian ink will come off resin-coated paper if the surface is rubbed.

Printed labels

When more than five labels are required with the same data, they can be reproduced photographically, by offset multilithing, or from set type. Some entomological supply houses can provide labels, e.g., Australian Entomological Supplies (address p. 76).

We use special forms to order and to assist with the layout of printed labels (Fig. 37, 38). Each dry label can be

DRY LABEL (12 × 8 mm) ORDER FORM (Print neatly)

Name: Date:

Number required: Urgent: yes / no

1

2

3

4

5

< = start italics > = end italics

Date processed:

Date returned:

Fig. 37 *Form for ordering printed labels for pinned specimens.*

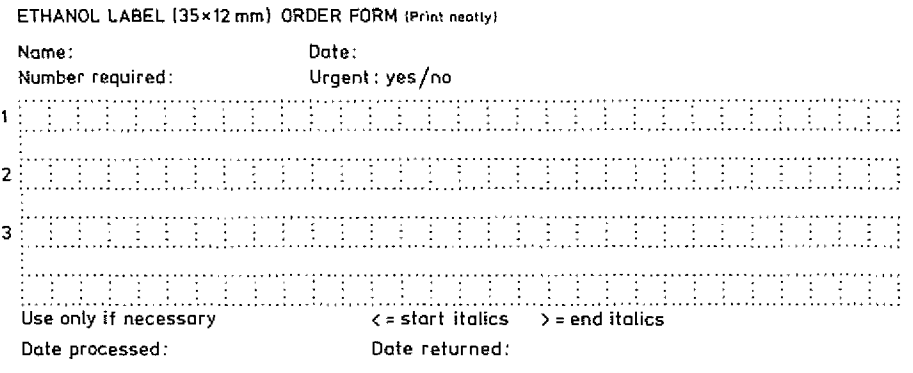


Fig. 38 *Form for ordering printed labels for ethanol-stored specimens.*

up to five lines deep, with each line up to 15 characters long (including spaces). Each ethanol label can be up to four lines deep, with each line up to 30 characters long (including spaces).

We use the following system for preparing label master sheets to be printed photographically or by offset multilith printing.

Labels are typed into a portable computer using a specially written "Label" program which permits users to correct labels as they go. They are typed from either the order forms (Fig. 37, 38), or direct from the field labels with specimens. The computer can be used in any part of the collection room where there are specimens to be labelled. The labels are transferred on to a floppy disk (formatted IBM-PC compatible), and computer typeset by a commercial firm. The dry labels are typeset at twice their final size, and reduced by the firm to the required size.

A master sheet is prepared by using the reduced labels laid out and glued to A4 paper. This master sheet and the label paper are then given to a printer for offset multilith reproduction, or the master sheet alone is given to a photographer for photographic reproduction.

Formerly labels were typed, rather than typeset, using an IBM Selectric II typewriter set to 12 pitch (12 characters per inch) and with black carbon ribbon. The type head used was "Adjutant", but for italics "Courier Italic 12" was used. The labels were photographically reduced to their final size, and a master sheet prepared for offset multilith printing.

Photographic reproduction

We use a resin-coated photographic paper. It is quick to process, is more permanent than other photographic papers because of its resin-coated surface, and has good firmness

for pinning. If used in ethanol, any additional writing in Indian ink will come off if the surface is rubbed.

This method is used for obtaining our colour coded labels (p. 42).

Offset multilith printing

The advantage of this method is that labels are printed on high quality label paper which we supply to the printer. Reproduction is excellent as long as a high quality metal plate is used.

We have 50 copies of each master sheet printed. Although more labels may be printed than required in many cases, this method saves time, gives consistently high quality labels, and is cheaper than hand labelling large numbers of specimens.

In ethanol a slight amount of dye from the printing ink may leach out, but causes no legibility problems.

Printing presses

Small, hand-operated printed presses with small type founts are available at reasonable cost (e.g., from The Kelsey Co., Box 941, Meriden, Connecticut 06450, U.S.A.). Experience is needed in setting the type for labels and inking the platen.

Computer printers

Personal computers with quality printers can be used to produce labels of an acceptable standard for purposes such as determination labels. Top quality printers, such as the Apple Laserwriter Plus, can produce labels on Goatskin Parchment Paper of near computer-typeset quality. However, these labels are not suitable for use in liquid preservatives as the printing disintegrates the same way as photocopied labels do.

Slide labels

Because permanence is of primary concern, labels should be made of a high quality paper and with a lasting adhesive suitable for use on glass. They must be able to withstand heating in a drying oven. When ordering slide labels make sure you specify they are to have a barrier layer between the glue and the paper to prevent the glue from seeping through. Glues with a natural rubber base deteriorate, especially when exposed to light or heat.

Our self-adhesive, 23 mm square slide labels are supplied on sheets. For little extra cost information can be multilithed on the labels.

Label format

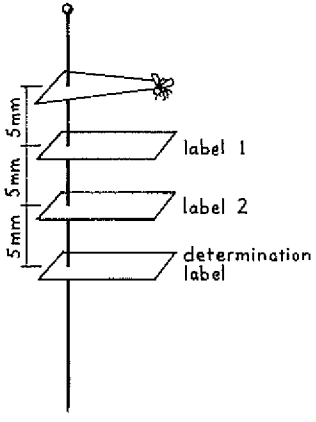


Fig. 39 *Positioning of labels on a pin.*

(1) Pinned insects

A standard format for labels is followed throughout the world: locality, date, and collector are recorded in that order. Other useful information can be given on supplementary labels added underneath, e.g., expedition, voucher specimen, determination, and type specimen labels.

Each label added to a pinned specimen must have at least a 5 mm gap above it so that it can be read without having to move it down the pin (Fig. 39).

The first two labels follow a strict format:

LABEL 1 (Fig. 40, 41). This gives the main information on where and when the specimen was collected.

1st line: Country, written in capital letters. For NEW ZEALAND, this is followed by the collection area abbreviation (p. 39, Fig. 48, 49).

If a specimen comes from the Kermadec Islands or subantarctic islands, the name of the major island group is written in capitals (Fig. 41). For the Chatham Islands and Three Kings Islands, the name of the major island group is written in capitals, followed by "NZ".

2nd line: General locality. Islands close to the main islands of mainland New Zealand come on this line (e.g., Noises Is in AK; Great Barrier I. and Little Barrier I. in CL). Also islands of major island groups are included on this line (e.g., Raoul I. from the Kermadec Islands).

3rd line: Specific locality, and altitude in metres.

4th line: Date – day, month (abbreviated to first three letters), and year. In handwritten labels "June" may be written in full to avoid confusion with "Jan". Always include the century as part of the year – "1985", not "85".

5th line: collector (initials and full surname).

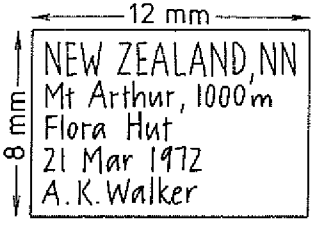


Fig. 40 *Label 1 format for New Zealand collecting areas.*

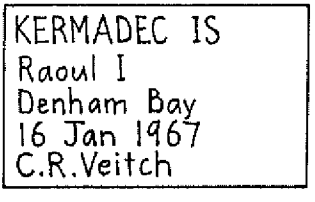


Fig. 41 *Label 1 format for a specimen from the Kermadec Islands or subantarctic islands.*

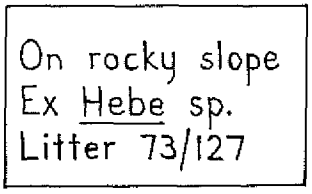


Fig. 42 *Label 2 format.*

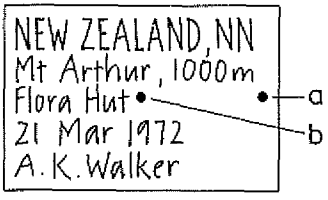


Fig. 43 *Where to position a pin through a label.*

Fig. 44 *Label for ethanol-stored specimens.*

The collector's name can be placed on label 2 if there is insufficient space on label 1. The altitude may also be fitted in a more convenient space.

Never put only part of a line or piece of information on label 1 and the rest on label 2. Never put information on the underside of a label, as this is likely to be overlooked.

LABEL 2 (Fig. 42). Additional information on where the specimen was collected, its host plant, collecting method, etc., and any litter or moss sample number (p. 42).

Pin position on labels

Pins should not go through any data on the label.

For directly pinned specimens, the label should be pinned about the middle (position a, Fig. 43).

For card-point or double-mounted specimens the label should be pinned near the edge on the right-hand side (position b, Fig. 43).

(2) Specimens in ethanol

The same sequence of information is used as for pinned specimens, but as the label is considerably larger (35 x 12 mm), the data can usually be written on just one label (Fig. 44).

If more than one label is required it is helpful to place this second label so that it backs on to the first. Ensure specimens are not trapped between the labels.

(3) Specimens on slides

Collection data are printed on the right-hand label and identification data on the left (Fig. 45). Waltz & McCafferty (1984) recommend adding the type of mounting medium as well (Fig. 45, c/b = Canada Balsam).

When recording the locality of parasitic insects such as Mallophaga (biting lice), Anoplura (sucking lice), and Siphonaptera (fleas), the scientific name of the host animal is given first, followed by the locality in which it was collected.

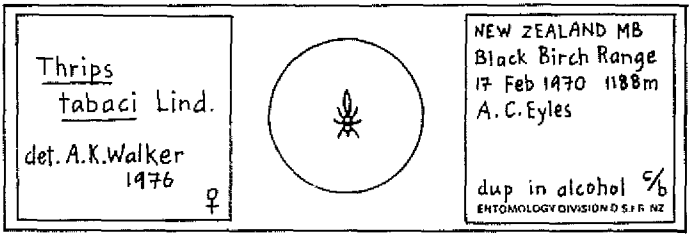


Fig. 45 *Format for slide labels.*

Austrosimulium ♀
australense (Schiner, 1868)
det. T.K. Crosby 1985

Fig. 46 *Determination label.*

Barea confusella
sens. auct. nec Walker
det. J.S. Dugdale 1986

Fig. 47 *Determination label with abbreviations.*

(4) Determination (identification) labels

The sequence of information is genus, species, author of species name, the abbreviation "det." followed by determiner (initials with surname in full), and year of determination (Fig. 45 (left-hand label), 46, 47). The sex of a specimen may be placed at a convenient place on the label. The year a species was described may be placed after the author name. The main abbreviations used are:

aff. = having affinity with (another species).
auct., sens. auct. = of authors; species name used in the misidentified sense of subsequent author(s), not in the sense of the authority who described it.
cf = compared with (close to another species but not identical; an unnamed taxon).
det. = determined by.
m. = (mihi) belonging to me, as a new species.
nec = not.
nob. = belonging to me, as a new species.
non = not.
s.l., s. lat., sens. lat. = sensu lato, in the wide sense.
s.s., s. str., sens. str. = sensu stricto, in the strict sense.
sp. = species (specimen cannot be named to species).
sp. indet. = indeterminate species (because the specimen is damaged, immature, or the opposite sex required).
( ) = parentheses around an author's name indicates that the species is now placed in a different genus to that in which the author originally described it.

Abbreviations

Although abbreviations may be used to provide the maximum amount of information on labels, it is important that they are consistent and not confusing.

The following abbreviations are used on labels in the N.Z. Arthropod Collection:

Jan Feb Mar Apr May Jun Jul Aug Sep Oct Nov Dec	January, February, March, etc.
N S E W	North, South, East, West
♀	female
♂	male
assoc	associated with
Ck	Creek
cnr	corner
dup	duplicate
em	emerged
ex	out of (NOT to be used for "beaten from")
Exped	Expedition
FP	Forest Park (after name of Forest Park)

Gt	Great
HWM	High Water Mark
I	Island
Is	Islands
km	kilometre(s)
L	Lake
leg	collected by (followed by name of collector)
loc	location
m	metre(s)
Mt	Mount, Mountain
NP	National Park (after name of park)
nr	near
Pen	Peninsula
Pk	Peak
PQS	Port Quarantine Service
Pt	Point
R	River
Ra	Range
Rd	Road
Res	Reserve
Sdle	Saddle
SF	State Forest (after name of forest)
SH	State Highway (followed by highway number)
SL	Sea Level
Smt	Summit
Stm	Stream
Stn	Station
V	Valley

(Note: full stops are not normally used after the abbreviations, but a space must be left before the next word. On handwritten labels when the printing is cramped it may be necessary to use full stops.)

New Zealand collecting areas

We use a standard form of area code (Crosby et al. 1976). Each label has in addition to "NEW ZEALAND" a two-letter abbreviation giving the general region of a collecting locality (p. 40, Fig. 48, 49). This code can be used for computer data retrieval, and in publications as a convenient way of ordering collection data of specimens.

Codes

As only a limited amount of information can be written on specimen labels, a code number is a useful way to indicate additional information catalogued elsewhere.

Careful consideration must be given before any coding system is adopted. Once the code is printed on labels it is very hard to alter. Care must also be taken not to defeat the purpose of codes by using so many of them as to be

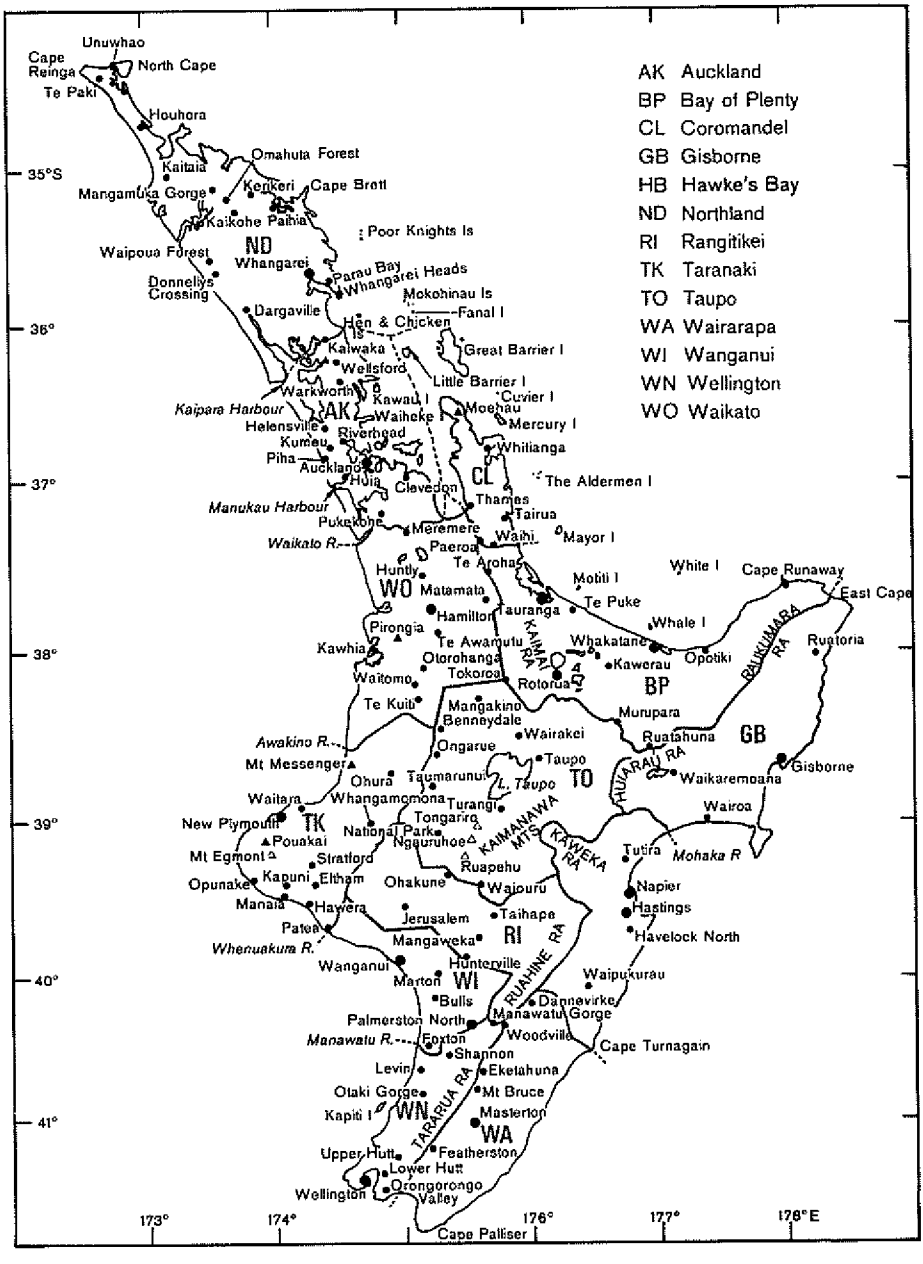


Fig. 48 *Abbreviations for North Island collecting areas.*

Fig. 49 *Abbreviations for South Island collecting areas.*

confusing. Remember that people must know what the code means in 50 years time; otherwise information about specimens may be lost. We photocopy our catalogues and file the copy in our archives.

Code numbers

Two code numbers are used in our collection:

(1) Litter (or moss) sample code

As soon as litter or moss samples are received they are given code numbers on a bulk collection card (p. 32). These are catalogued in chronological order together with all the field information the collector has written. Different samples with the same collection site are filed together in the card catalogue, which makes for easier reference later on.

The code gives the year first, followed by the collection number of the particular sample, e.g., 74/196. This code is added to a label.

(2) Rearing code

Each sample containing larvae to be reared is given a code number as soon as it is placed in a rearing container. For instance, W74/84 indicates that a specimen had been reared from wood in 1974 and is Sample 84 of that year. Additional information, such as host and biological data, is entered in a catalogue. As specimens emerge and are processed, code numbers are printed on an orange label, together with the emergence and host plant data, and added beneath the other labels.

Although the type of numbering system is the same for both codes, any possibility of numbers being confused is avoided by giving the additional information with the code, and by printing the rearing code on orange label paper.

Colour codes

Any colour code system must be well thought out before use. A few distinctively coloured labels facilitate easy retrieval of specimens, but too many different colours scattered throughout a collection are just as confusing as none at all. No coloured label should be used without additional printing to specify its purpose.

We use coloured photographic paper which has the same processing requirements as ordinary photographic paper. Only the red, blue, and gold retain their colour in ethanol.

(1) Specimens sent on loan—ownership labels

Every specimen or vial of specimens sent on loan to another institution or specialist has a gold ownership label added. A

distinctive colour is very helpful when a specialist has completed a revision, and must sort and return material borrowed from many collections. Our label has the wording:

N.Z. Arthropod Collection,
NZAC Entomology Div.
DSIR, Auckland
NEW ZEALAND

(2) Specimens reared

Orange photographic paper is used, with the word "reared" and the rearing number printed on each label (p. 42). Unfortunately, the orange paper is not colour-fast in ethanol. Specimens kept in ethanol must have the information printed on white label paper.

(3) Illustrated specimens

Any specimen which has been illustrated or photographed is given a dark green label. The date and name of the illustrator or photographer are printed on the label.

(4) (a) Voucher specimens of species released for biological control
(b) Voucher specimens of species imported and reared in quarantine

These labels are printed in white on a black photographic paper.

(5) Type specimens

Type specimens are unique, and require a distinctive label on each specimen. We have chosen red for ***holotype*** and ***lectotype***, blue for ***paratypes*** and ***paralectotypes***, and light green for ***syntypes***. These can be made available to systematists who are going to deposit type specimens in the N.Z. Arthropod Collection.

(6) Compared with type

Any specimen which has been compared with a primary type specimen has a yellow label added stating the person who made the comparison and the year. This label is used when a type specimen deposited in another institution has been seen by a specialist and taxonomic identities have been established with certainty.

Organisation of the collection

Safety and security

Fire and vandalism are major hazards in any collection. We have fireproof doors at either end of the collection room in addition to fire extinguishers. We do not permit smoking in the collection areas or adjoining offices. We do not have gas in offices to heat specimens being macerated or otherwise prepared for examination; all heating is supplied by thermostatically controlled electric hotplates.

The main doors to the collection are kept locked outside normal working hours to minimise the risk of vandalism.

Arrangement of the collections

The general New Zealand collection is arranged according to order, and within orders according to family; the family sequence is that of CSIRO (1970). As a rule, only one family is kept in each store box or jar. The identified specimens of a family precede the unidentified specimens.

The exotic collection is small compared to the general New Zealand collection. In most orders it is stored separately, in families, and kept on the top shelves of the storage bays above the New Zealand material of the same family.

Holotypes, lectotypes, and some paratypes are kept separate from the main collection. The pinned specimens are housed in unit trays in insect cabinets (p. 48).

The microscope slide collection is small, and many specialists keep boxes in their rooms. The remaining boxes are stacked in a general use room.

Acquisitions

Any specimen or collections acquired are incorporated into the main collection, and a label with the name of the collection and the year of acquisition is added to each specimen. We do not use acquisition numbers, although this is under review for primary type specimens.

We encourage workers to deposit voucher specimens, of species listed in papers and reports, into organisations with research collections of insects. Make sure the specimens are well prepared and correctly labelled (Deitz 1979a); a few well-prepared specimens of a species are of much more scientific value and more warmly received than many ill-prepared specimens. We place a voucher label with each specimen or vial stating the author, date, and publication details before incorporating them in the collection.

It is worth mentioning here that private collectors should make arrangements for their collections to be passed on to another entomologist or reputable institution in the

event of their death. Many important collections have been lost, or had their scientific value lessened, because of deterioration through subsequent inadequate curation.

Storage

Pinned insects

Pinned insects must be stored in close-fitting boxes or drawers away from direct sunlight.

(1) Store boxes

We have found store boxes to be the most useful and practicable means of storing most pinned insects. One can liken a collection of store boxes to library books on shelves. Later this system can be easily adapted to a Compactor-type storage system (ASC 1976), or to a system of storing boxes flat in shelves.

We use 30 x 21.5 x 6.5 cm plywood store boxes (see specifications p. 46). Their size is convenient for use beside a microscope, and two boxes with packing material will pack safely inside the largest postal container allowed by the N.Z. Post Office (p. 57).

On the outside of some boxes we add coloured self-adhesive labels.

(1) White: specimens need to be labelled or remounted, or box requires attention.

(2) Black: specimens on loan from other institutions in the box.

(3) Red: label information for specimens in the box has been entered on the computer in readiness for typesetting. The date this was done is noted on the label.

(4) Green: labels have been received back from the printer, and need to be added to the specimens in the box.

(5) Yellow: specimens from the Pacific Islands.

A pinning base of soft, 9–10 mm thick Plastazote is glued to the bottom of a store box. A small cardboard container is glued to the bottom right-hand corner to hold a standard 30 x 30 mm (1/4 oz) camphor block.

CAUTION: avoid using contact cements to glue down the Plastazote and camphor box as they may not penetrate into Plastazote or stick where there is a concentration of camphor. Use a gelatine-based glue which will penetrate into the Plastazote.

Layout of specimens in a store box

Identification labels are pinned along the left-hand margin. Specimens are arranged from left to right, and in rows from top to bottom (Fig. 50).

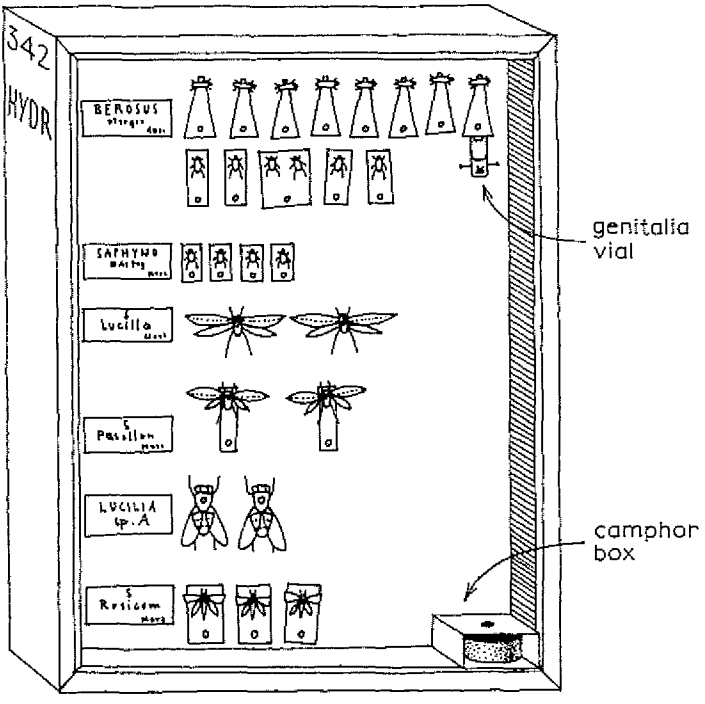


Fig. 50 *A sample layout of specimens in a store box.*

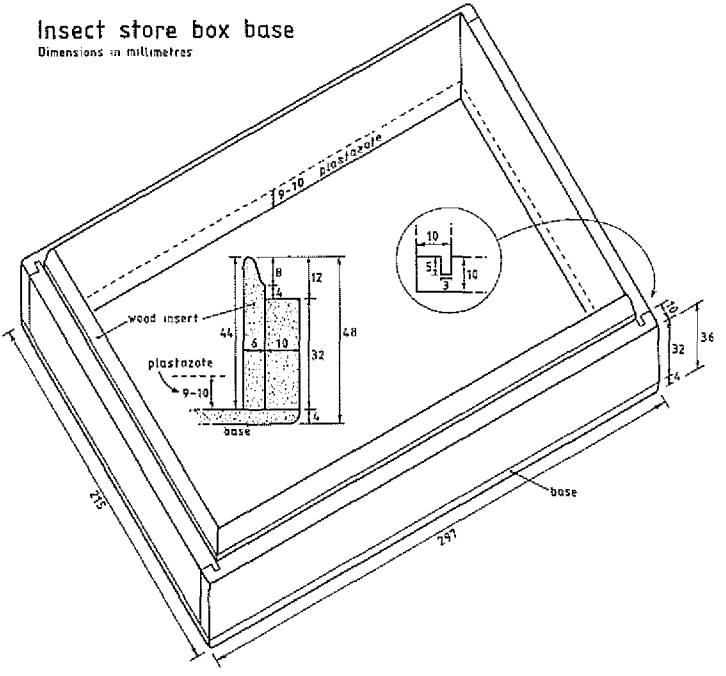
If a genitalia vial (p. 50) is added to a specimen, it must be mounted on its pin so that it is vertical when the store box is upright (Fig. 50; Deitz 1979b). Genitalia vials should be secured in position with extra pins on either side, to prevent them rotating and damaging adjacent specimens when store boxes are handled.

Specifications for insect store boxes
Box
(1) Dimensions as in Fig. 51 and 52.
(2) Construct with seasoned wood and plywood.
(3) All surfaces to be smooth and flush, with edges rounded.
(4) Top to be close-fitting.
(5) Boxes to be clean.
(6) Finish with light-coloured matt varnish.

Figs 51, 52(Opposite). *Specifications for manufacturing insect store boxes.*

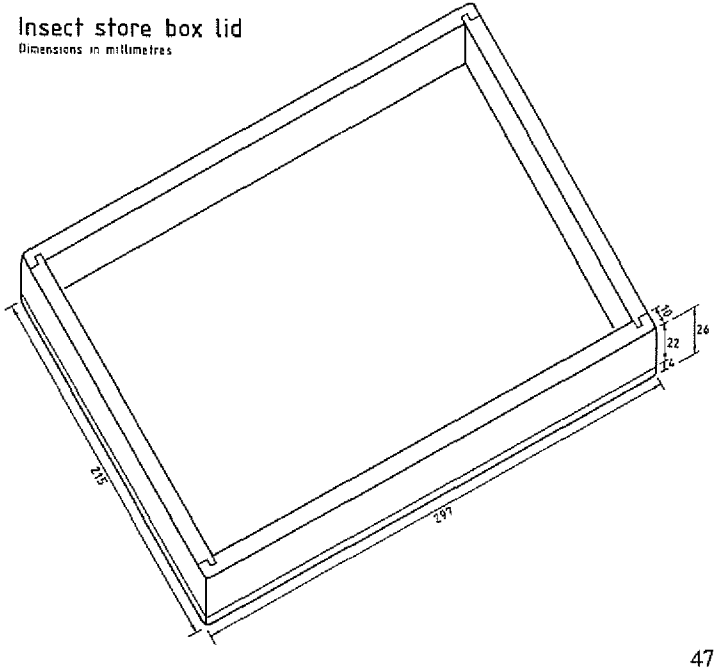
Insect store box base

Dimensions in millimetres



Insect store box lid

Dimensions in millimetres



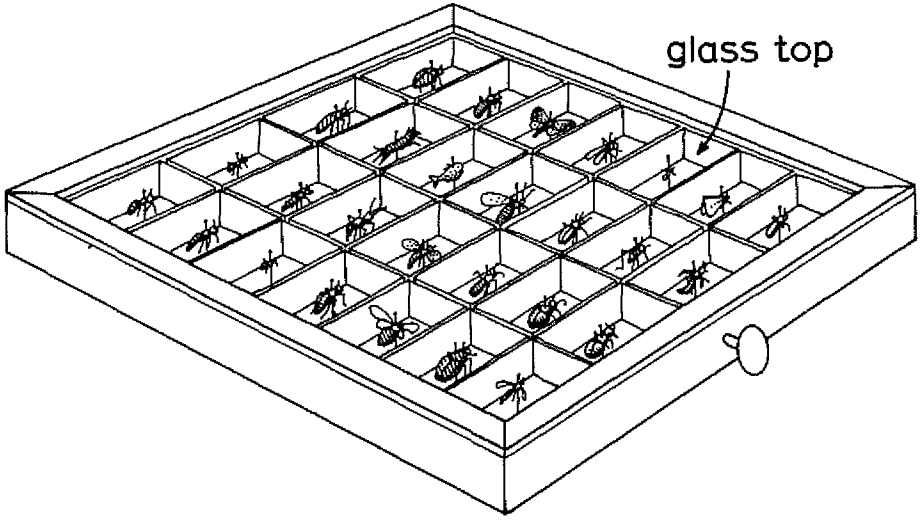


Fig. 53 *Storage of type specimens in unit trays.*

Plastazote

(1) White, 9–10 mm thick.

(2) Guillotine up to 2 mm wider than inner dimensions.

(3) Glue firmly to box bottom using appropriate adhesive (e.g., Gripit); glue to be spread in a band along each side and across the two diagonals; a weight to be placed on the Plastazote until the glue is set.

Cabinet drawers with unit trays

Pinned insects can also be stored in unit trays (small cardboard boxes with Plastazote in the bottom). We have adopted this system for storing type specimens. Twenty-eight unit trays fit in the glass-topped drawers we use (Fig. 53). Specimens can either be examined without removing the lid or an individual type and its unit tray can be taken out for examination. This keeps the handling of type specimens to a minimum.

Heavy-bodied insects which are too large to be easily stored in store boxes (e.g., wetas, stick insects) are kept in unit trays in glass-topped drawers (see cover).

Insects in ethanol

The ethanol collection is stored in a darkened room separated from the remainder of the collection (see Jocqué (1983) for his experience with spiders bleached by light). The room is well ventilated, and a DANGER, ETHANOL STORAGE, NO SMOKING sign is clearly displayed. Precautions are taken against any risk of fire—for instance, gas taps were taped over or removed.

Storage jars

All specimens in ethanol are stored in glass vials, which are packed in larger containers to prevent evaporation. When choosing these containers several points should be considered. Clear glass is preferable for easy viewing of the contents. The container should be at least 40 mm higher than the vials, to allow for a cottonwool pad under the vials, and enough room for ethanol to cover the vials. Plastic screwtop lids will outlast metal lids which, even with inserts, may corrode after a few years. The lid should be airtight to minimise evaporation of the ethanol.

We use 0.6 litre preserving jars with plastic screwtop lids. These jars hold up to 42 vials. A paper label with the family name in large capitals is placed inside the jar, and the generic and specific names can be added underneath. When several identified species are stored in one jar, a Plastazote strip can be used to make a division inside the jar (rubber bands deteriorate in ethanol). A 85 x 40 x 2 mm plastic strip (polyethylene or polypropylene) makes an ideal divider.

Jars are filled with 75% ethanol, checked every 2 years, and topped up with 95% ethanol if required. The date of ethanol topping is recorded on a sheet displayed in the ethanol collection room.

Glass vials

These should have a flat bottom so that they will stand up on their own and should be made of strong glass (it can be very dangerous if the rim breaks while pushing in a closure; if you are uncertain about the strength of the rim, use a towel to push the closure in). The closure must be leak-proof – ethanol soon evaporates if the closure is faulty.

CAUTION: Make sure the closure is made from polyethylene or polypropylene, and not PVC (polyvinyl chloride), which will break down within 2 or 3 years when submerged in ethanol.

We use 50 x 12 mm vials for the majority of New Zealand insects. Larger insects (e.g., stick insects) are stored in vials 25 mm diameter by up to 150 mm long.

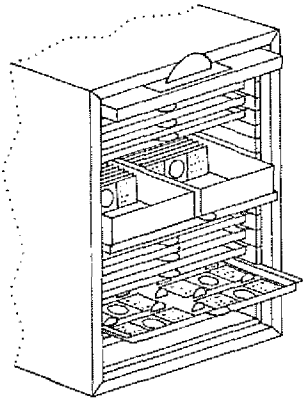


Fig. 54 *Storage system for microscope slides.*

Microscope slides

Slides can be stored either vertically in boxes with up to 100 divisions, or laid flat on trays.

We use 290 x 185 mm aluminium trays holding 20 slides which are kept in wooden slide cabinets each holding 24 trays (Fig. 54).

If there are large numbers of the same species, the slides are packed vertically in 139 x 78 x 21 mm cardboard boxes, similar in size to a 100-pack slide box. Two thick pieces of card—about the same size as the slide label and thicker than the coverslip—are slotted in between each slide on each side of the coverslip to avoid the slides crushing each other or sticking together. Four boxes, each holding about 70 slides, sit comfortably on a slide tray (Fig. 54).

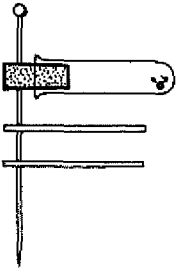


Fig. 55 *Storage of specimens in a mini-vial.*

Mini-vials

Mini-vial storage is used for large numbers of very small specimens of the same species (e.g., Acari, Hymenoptera).

The vials used are 27 x 6 mm Durham fermentation tubes plugged with a Plastazote closure (Fig. 55). A pin is inserted through the Plastazote plug in the same manner as a dried specimen. They are stored vertically in a store box. These mini-vials are not suitable for genitalia vials.

Genitalia vials

Two types of vial are available — a traditional glass vial with a cork closure, or an "arthropod microvial" made of semi-transparent polyethylene with a silicon rubber closure. Glass vials with cork closures must be stored so that the glycerol does not come into contact with the cork (Deitz 1979b). Andersson (1976a,b) describes as another alternative an inexpensive, convenient method of making genitalia vials from plastic tubing.

Deep freeze storage

Freshly caught specimens which are to be pinned can be sealed in field layering boxes or other suitable containers and stored in a deep freeze. Microlepidoptera can be stored in their temporary shallow pinning boxes (p. 29). Specimens should not be dried out before placing them in the deep freeze. Cardboard containers must be placed in plastic bags and sealed to prevent moisture leaving and the specimens becoming freeze-dried, and to stop the cardboard sticking to the deep freeze.

We have stored specimens in a deep freeze for 4 years without problems, and know of specimens stored for 12 years without spoiling.

Critical point dried specimens

This technique allows small insects stored or collected into ethanol to be dried without the body collapsing or losing further colour. The body appendages remain more flexible than air-dried specimens, and therefore are less likely to be damaged. Colours are slightly dulled compared with air-dried specimens (Craig & Craig 1987). Gordh & Hall (1979) give an account of the technique.

Critical point drying is required for specimens to be examined with a scanning electron microscope (SEM). It also produces excellent specimens for display purposes.

Dried specimens are kept in small boxes in drawers. The drawers have silica gel in them to ensure the moisture content remains low.

Plant material associated with insects

We hold an extensive collection of leaf mines, galls, wood workings, and dried scale insects together with their hosts.

Leaf mines

The plant is pressed and attached to a standard herbarium sheet, with details of plant name, locality, date, and collector. The sheets are placed in a manila folder and filed in a standard filing cabinet. Small pressed plant specimens are placed in clear cellophane packets.

Galls

These are placed in paper packets, data added, and stored in herbarium cardboard boxes.

Wood borings

Wood specimens are split to show the typical workings of a particular species. Details of tree name, locality, date, and collector are written in Indian ink on the wood. The collection is stored in herbarium cardboard boxes.

Scale insects and whitefly pupae

Dry host plants with their associated unmounted specimens are stored in envelopes with locality, date, collector, and host plant written on the outside. The envelopes are filed in herbarium cardboard boxes.

Photographic slides and prints

Labelled slides and prints of insects, insect damage, and field expeditions are kept in a standard filing cabinet. The slides are stored in transparent slide holders for easy viewing and checking. CAUTION: ensure plastic holders are of proven archival quality.

Archival records

Records of archival value, including unpublished manuscript notes of some early workers, are kept either in a standard filing cabinet or in herbarium cardboard boxes.

Precautions against insect pests

Deterrents

Camphor is used in store boxes to deter insect pests (e.g., dermestids, psocids, and silverfish), but remember that it is only a deterrent, not a cure. (HINT: unrefined camphor is more satisfactory than refined brands, which tend to stick together and crumble when separated). Because our collection is stored in an air-conditioned room (20°C, 60% relative humidity) it is necessary to replace the camphor after 6 months in new store boxes and after 1 year in all others. The date of camphor changing is recorded on a sheet displayed in the collection room. The rate at which camphor vaporises in other collections may be quite different. In the past, naphthalene and para-dichlorobenzene were used as deterrents in the insect collection, but camphor was substituted when the potential health hazards of these chemicals (Sax 1963) were recognised to be higher than camphor (also see deterrents p. 67).

Mould can be a major problem in hot, humid climates. Unless the collection is stored in an air-conditioned, humidity-controlled room, specimens will deteriorate. In temperate climates chlorocresol can be used to inhibit the growth of moulds.

Fumigants

Store boxes infested with insect pests must be removed and fumigated immediately. If the infestation is restricted to individual boxes, these can be sealed in a plastic bag with a large wad of cottonwool soaked in ethyl acetate (CAUTION: do this in a fume cupboard, as ethyl acetate is hazardous, p. 70).

HINT: fine dust gathering underneath a specimen and labels in flat-stored boxes, or at the base of upright-stored boxes, indicates that dermestids, psocids, or mites are present.

The room in which a collection is stored must also be properly protected against insect pests. Ideally the collection and adjoining rooms should be fumigated once a year. Our collection is fumigated by a commercial firm using either Dichlorvos/Vapona (a short-life, organophosphorus insecticide), or a synthetic pyrethroid. CAUTION: remove all plastic rearing containers from areas to be fumigated. Dichlorvos vapour reacts with plastic, and the fumes given off over the subsequent weeks can kill insects being reared in the containers; insects are most sensitive to the vapour during the moult from pupa to adult.

Loans

Most institutions holding an insect collection operate a loan recording system, both to keep track of material out on loan and to provide an official and legal basis for its return. Far too often in the past institutions have lost specimens because of inadequate records, e.g., loan details only known to researchers who have left an institution or have died.

Filled-in loan (or dispatch) forms are to include the Order, Family, and name of the taxonomic group to which the specimens belong, the number of specimens, vials, or microscope slides dispatched, and the geographic collection area (e.g., New Zealand, subantarctic islands, Pacific islands, Chatham Islands). When microscope slides are dispatched, make a photocopy of them to keep as a record with the dispatch form. Under the Antiquities Act 1975, if holotypes or lectotypes of New Zealand species are being dispatched overseas, a Temporary Export Certificate must be obtained from the Department of Internal Affairs, Private Bag, Wellington before they are sent.

Our dispatch forms (Fig. 56) are numbered consecutively. They are made out in triplicate; two copies (white and green) are sent to the person receiving the dispatch, and the third copy (yellow) is filed in a "Current dispatches" file. It is important to keep a copy until receipt of the dispatch is acknowledged, otherwise there is no record of the dispatch details. When the dispatch is received the receiver signs and returns the green form, which we file in the "Current dispatches" file in place of the unsigned yellow one. When the dispatch is returned, a printed postcard or letter is sent acknowledging its return. The date of return and the person receiving the dispatch are added to our copy of the dispatch form before it is transferred to the "Completed dispatches" file.

We usually send one dispatch form separately from the specimens, so that the borrower is aware of their dispatch and their expected date of arrival.

A cross-reference card index is kept also. The cards are filed according to the name of the person requesting the dispatch, and bear the dispatch number, dates requested and returned, and a brief description of the dispatch. The cards allow us to check very quickly what material any person has on loan. These records are now being computerised.

Fig. 56(Opposite). *Dispatch form used by NZAC.*

Collection abbreviation: NZAC

Dispatch Number .....

NEW ZEALAND ARTHROPOD COLLECTION

RECEIPT FOR SPECIMENS

To:
.....
.....
.....
.....
.....

Dispatch approved by .....
Packaged by .....
Dispatched by registered airmail/surface mail in
.....package(s) on...../19....
in good condition/condition as stated below.

☐ loan at your request ☐ gift
☐ examination at our request ☐ exchange
☐ return of your specimens .....

Specimens from:
☐ New Zealand ☐ New Zealand Offshore Island
☐ Pacific ☐ .....

ORDER FAMILY

Number (pinned, vials, slides)	Details of specimens (taxon, locality, condition, type status, etc.)

Acknowledgement of return of specimens to NZAC
Received: ..... 19 .... Signature: .....

Temporary Export Certificate Number
(required for N.Z. primary types sent overseas, antiquities Act 1975).....

Conditions governing loan of specimens

Primary types designated from these specimens and unique adult and immature specimens must be returned.
Retention of other specimens is permitted only by written agreement with the Curator. All parcels are to be sent by registered mail, and addressed to the Curator.
Loans of primary type specimens are for a maximum of 3 months; loans of other specimens are normally for 2 years.
I agree to abide by the above conditions, and to return the specimens forthwith if requested to do so.

I will return the specimens in ☐ 3 months ☐ 6 months ☐ 1 year ☐ 2 years ☐ 3 years ☐ 4 years ☐ 5 years
Received: ..... 19 .... in condition as stated/other (please specify below)

Signature: ..... Condition: .....

Please sign and return green copy to the Curator, New Zealand Arthropod Collection, when specimens are received.

Postal Address: Entomology Division, DSIR, Private Bag, Auckland, New Zealand.
Location: 120 Mt Albert Rd, Mt Albert, Auckland, New Zealand.
Telephone: (09) 893-660 Telegraphic Address: PLANTLAB or ENTLAB Auckland Telex: NZ21623

Packaging and posting specimens

Packing specimens in containers

Never put ethanol-stored specimens and pinned specimens in the same container; they must always be separate when sent in the same package. If a vial becomes loose, pinned specimens may be destroyed; if a vial closure becomes loose because of reduced air pressure in aircraft, ethanol will spill everywhere and affect the pinned specimens.

Similarly, never put microscope slides and pinned specimens in the same container.

Specimens on pins

Add an ownership label (p. 42) to the pin underneath each specimen. Pin each specimen firmly in the specimen box (boxes of different sizes are used depending on the number and size of specimens being sent). Be sure there are no gaps between the pinning base and the walls of the box. Brace with extra pins any insects with a long abdomen or long legs, those on old-type double mounts, and those with genitalia vials (Fig. 57). Ensure bracing pins are not pushed through labels. Pin a small, fluffy piece of cottonwool in a corner to catch any fragments that may become detached during transit. If there is a camphor container, remove the camphor and replace it with cottonwool. Wrap a sheet of fine plastic ("Gladwrap") over the open box and tape it down so that if the parcel is opened by Customs Officers for inspection the specimens are protected but can be seen, and any loose fragments detached from specimens will not drop out.

Individual pinned specimens can be posted in an unbreakable plastic (polycarbonate) vial rather than a box. Pin the specimen to the inside face of a cork closure and carefully insert it into the vial.

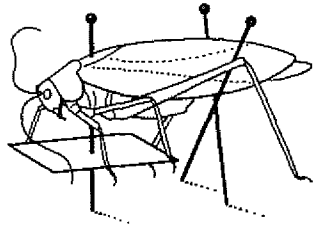


Fig. 57 *Specimen braced with extra pins.*

Specimens in ethanol

Add an ownership label (p. 42) to each vial. If the vial is to be sent any distance and the specimens inside are large, force a plug of cottonwool down in the ethanol to squeeze out air bubbles and to reduce movement of specimens during transit. Also, if there are loose labels in a vial with small soft-bodied specimens use cottonwool to wedge the labels firmly

to the side of the vial, otherwise they may act as battering rams against specimens. Wrap each vial in tissue paper (toilet paper is most suitable). Put up to 10 vials in a plastic bag and seal it.

Bulk trap collections in ethanol

The contents of a Malaise or pitfall trap can be contained in a gauze bag which is then sealed in a plastic bag with a small amount of ethanol. This can be packed in any suitable mailing container; we use cardboard mailing tubes 150 mm long and 65 mm diameter. The package must be sent airmail to ensure quick delivery.

Microscope slides

Photocopy the slides and keep a copy with the retained dispatch form. Pack slides in any convenient slide holder. Most slides already have an ownership label on them, but if not, add one. Be sure to pack slides firmly in their holders using tissue paper so they do not rattle.

Type specimens

Send no more than two holotypes or lectotypes together in the same parcel, and post parcels several days apart. The loan of New Zealand holotypes and lectotypes from New Zealand institutions to overseas borrowers must be made according to the requirements of the Antiquities Act (1975).

Packaging and parcel size

Pack at least 8 cm of wood-wool padding (for surface mail) or polystyrene chips (for airmail) between the specimen container and the outer container. Do not pack it in too tightly, otherwise its protective, cushioning effect for the specimen container will be lost and specimens will receive every knock and jar the parcel receives during transit. Wrap with heavy-duty brown paper, seal with adhesive tape, and tie with string.

One of our wooden insect store box and its layer of packing fit snugly into a number 6 carton (43 x 33 x 25 cm) and two wooden store boxes plus packing into a number 8 carton (45 x 45 x 35 cm), the largest container allowed by international mail. We now use lighter cardboard store boxes for posting; two will fit snugly into a number 6 carton.

In New Zealand the combined length and girth of a parcel is not to exceed 2 m, and the greatest length of any one side is not to exceed 1.05 m. The maximum weight is 10 kg.

Posting

We post all parcels of specimens by registered mail. We record the date, address, number of parcels, and whether sent by surface or airmail in a posting book. We ensure that

the mailing centre has a record of the actual registration numbers of parcels being sent; this is essential for tracing any parcels lost in transit, especially if several registered parcels are posted on the same day to different parts of the world. If a large consignment is to be sent, we divide it in half and send as two parcels several days apart to reduce the risk of loss of the whole loan. We send nearly all overseas dispatches by airmail to minimise the risk of damage to specimens.

Remember to check that a completed dispatch form has been filled out and sent separately for every consignment. Every parcel of specimens sent to another country must have an accompanying customs declaration form. Under the heading "Detailed Description of Contents", write "dead insects for scientific study". No parcels are marked as "gifts"; all are marked as "no commercial value".

Sending specimens to specialists

Before submitting any specimens for identification, contact the specialist.

(1) Explain the reasons the identifications are required, e.g., for a research project, survey, or thesis, and when the specimens would be sent.

(2) Indicate the taxonomic group(s) involved.

(3) Give the approximate number of specimens to be identified, and whether they are immature stages or adults.

(4) Indicate the level of identification required, e.g., to species, genus, or family.

(5) State the date the identifications are required by.

Specialists can then tell you if they are willing to make the identifications, and what charges may be involved for their services. They can also tell you how specimens should be prepared if their requirements differ from general preparation techniques.

If you want identifications done for a major project, you should write and seek cooperation at the planning stage. If you delay seeking cooperation until your project is being written up, it is unlikely your request will be treated sympathetically and it may be declined because of other commitments.

Preparing the specimens

(1) Specimens should be prepared and labelled to proper standards. Faster service is achieved by relieving specialists of most technical work associated with identifications; poorly mounted or damaged specimens are time-consuming to examine, and often cannot be identified at all. Full data labels and any other ecological information must be with the specimens; field labels or coded labels are inadequate.

Specimens submitted to systematists at the N.Z. Arthropod Collection must be prepared and labelled according to the standards outlined in this book.

(2) Sort specimens to species (matching like with like) as far as you are able. When submitting specimens in vials, do not submit two or more obviously different taxa together in the same vial.

(3) Rear some immature specimens to adults so that all stages are available for examination rather than submitting immature specimens alone.

(4) Package the specimens for posting to the standards outlined in this book.

Identification of specimens

The NZAC provides an "Identification Report" about specimens submitted (Fig. 58) and will usually add a determination label before returning them. Some specialists may wish to retain taxonomically important specimens which represent new taxa, new host records, or new distribution records. They may also wish to retain specimens of other species to add to the collection.

When an identification is quoted, the abbreviations and qualifying words given as part of the scientific name by the specialist must not be omitted from the scientific name as they are essential parts of the identification.

An appropriate acknowledgment to the specialist should be made in any documents or publications in which an identification is cited.

Ramsay & Singh (1982) provide a list of New Zealand taxonomists and the groups they are prepared to identify. If you wish to identify the specimens yourself using the resources of a collection, the appropriate Curators should be contacted first. Bench fees may be payable, especially if you are doing the identifications for paid contract work.

Fig. 58(Opposite). *Identification report form used by NZAC.*

NEW ZEALAND ARTHROPOD COLLECTION

Postal Address: Entomology Division, DSIR, Private Bag, Auckland, New Zealand
Location: 120 Mt Albert Rd, Mt Albert, Auckland, New Zealand
Telephone: (09) 893-660 Telex: NZ21623

Report Number .....

IDENTIFICATION REPORT

To:
.....
.....
.....
.....
.....

Date: .....
Your reference number: .....
Identified by: (1) .....
(2) .....
(3) .....

Number of specimens	ORDER / FAMILY / Identification and remarks	Identified by:	Number of specimens retained

The person making the identification should be acknowledged in any publication in which these determinations are used.

Restoration of specimens

Removing specimens from ethanol

Removing Hymenoptera from ethanol has been covered on p. 26, but valuable specimens of adult Lepidoptera and some calyptrate Diptera which have been inadvertently stored in ethanol can also be recovered by removing and drying using the following technique.

(1) Take specimen from ethanol, drain, and soak in acetone or petroleum ether for 1 to 3 hours depending on the size of the specimen. Change solvent frequently.

(2) Dry under moderate heat, e.g., desk lamp. For Lepidoptera, gently tease the head, thorax, and wing fringe scales as the specimen is drying.

(3) Pin the specimen (do not attempt to relax it). Keep important old labels on the pin with the specimen.

CAUTION: Petroleum ether and acetone are very inflammable—keep away from naked flames. Carry out the procedure in a fume cupboard or a well ventilated room.

Other techniques use cellosolve (Sabrosky 1966) or Barber's Fluid (May 1958).

The specimens may also be critical point dried (p. 51).

Removing verdigris from pinned specimens

Insects pinned with non-stainless steel pins often become coated around the pinhole with a green substance, "verdigris", formed by organic acids from the insect's body reacting with copper from pins to give a copper oxide. This reaction is difficult to control.

Brush an excess of acetone or petroleum ether onto the affected area, allow it to soak in, then gently remove the verdigris with a fine brush. The process has to be repeated after a year or two as the verdigris will gradually return.

CAUTION: Petroleum ether and acetone are very inflammable—keep away from naked flames. Carry out the procedure in a fume cupboard or a well ventilated room.

Restoring old shrivelled insects

The following technique is satisfactory for restoring large, dry specimens needing repinning or cleaning, and specimens which have become shrivelled or desiccated and will later be stored in ethanol.

(1) Soak specimen in 1 part ordinary household detergent to 3 parts warm water for no longer than 15 minutes. (Note: the proportion of detergent given is only a guide, as much depends on the detergent used and the state of the specimen.)

(2) If the specimen is not sufficiently relaxed in the detergent solution after 15 minutes, remove it from the

solution and place in a relaxing chamber (p. 13) for up to 2 hours. This should be sufficient time to soften it completely.

(3) If the specimen is to be pinned, rinse in acetone, arrange legs, and pin immediately. If the specimen is to be preserved in ethanol, rinse in ethanol before storing.

An alternative method is to use a 1% solution of trisodium phosphate (Na_3PO_4) in cold water. Sink the specimen with a drop of detergent, leave for 24 hours, and then wash it in ethanol.

Upton & Norris (1980) recommend soaking specimens in Decon 90 (a detergent used in laboratories) for 16 hours, then thoroughly rinsing them and immersing them in water until restoration is complete.

CAUTION: Do not put the specimen labels in the solutions.

Replacing pins and double mounting old insects

CAUTION: Original labels, although difficult to read and incomplete, must be kept with their specimens, as often the handwriting is the only clue to the history of the specimen or the identity of its collector.

Old specimens are usually pinned with non-stainless steel pins. The pins may be weak or corroded, and often verdigris is a problem (p. 62). Various methods for removing pins have been discussed in the literature, such as Barber's fluid (May 1958) or using an electrical current to heat the pin (Upton & Norris 1980). However, in most instances simple methods and patience are adequate.

CAUTION: remounting valuable material (i.e., type specimens) must be left to specialists.

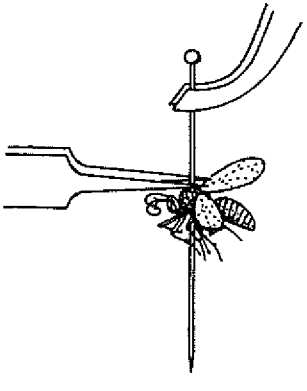


Fig. 59 *Removing an old pin from a specimen.*

Large specimens

Relax the specimen, by soaking it in a solution of warm water and detergent (p. 62), or by placing it in a relaxing chamber until soft. Hold the specimen between thumb and forefinger, grip the pin with pinning forceps in the other hand, and gently ease the pin out by twisting the pin back and forth.

If the legs and wings are spread, and the specimen is difficult to hold with thumb and forefinger, with one hand hold watchmaker's forceps around the pin against the insect and with the other hand use pinning forceps to gently twist and ease the old pin out (Fig. 59). (HINT: the lower part of the body can rest on Plastazote to steady the specimen.)

If the old hole is too large for the replacement pin, place a small drop of glue on the pin shaft slightly below where the specimen will rest. Then carefully repin the specimen by moving it up over the glue.

Small specimens

It is best with small specimens to leave the old pin and pin it to a double mount (p. 19). If the old pin is too long use a pair of fine wirecutters to cut it to a length suitable for double mounting. CAUTION: this requires great care, and can be a difficult operation if the pin is tough to cut, as violent jarring by wirecutters can easily damage a delicate specimen. If the old pin has broken just below the insect, the specimen may be glued to the double mount.

Soaking old specimens off card mounts

Formerly, specimens were often glued flat onto pieces of card. These specimens can easily be removed for study by soaking the specimen and card in warm water. Alternatively, drops of warm water can be placed around the specimen (important if the card has label information on its lower side!). If the glue is not water-soluble, acetone or ether may act as solvents.

Ultrasonic cleaning

Soil-inhabiting insects gather a lot of dirt on their bodies which can be removed satisfactorily with an ultrasonic cleaner. Specimens can be cleaned in their ethanol storage fluid, but best results are obtained by placing the specimen in a warm solution of detergent and water in a stoppered vial which is dipped into water in the tank of the ultrasonic cleaner. Alternatively, place specimens in a basket and immerse them directly in a detergent solution in the tank. Tests should be made to ensure that the power of the ultrasonic cleaner is not so great as to damage or destroy the specimens. Best results are achieved when all the specimens are of similar size and robustness. The time required for cleaning will depend on how large and how dirty specimens are, but usually 2 minutes is quite sufficient for most specimens. We have also found this technique useful for cleaning litter samples before sorting.

Shear & Levi (1970) also give useful hints for ultrasonic cleaning of spiders and myriapods.

CAUTION: Do not stay in the room while the cleaner is operating. The high intensity vibrations produced is capable of destroying human tissue, and repeated exposure can lead to permanent loss of hearing. We have a wooden cover to go over our small cleaner while it is operating.

Note: Freeze-drying and air-drying techniques are applicable only to specimens for display; these specimens are not for taxonomic study. Critical point dried specimens (p. 51) are suitable for taxonomic study as well as for display.

Freeze-drying insect larvae

This technique has become increasingly popular as commercial freeze- drying machines have become available. Excellent results can be obtained with live larvae, and colour retention is quite outstanding. However, specimens freeze-dried after storage in ethanol discolour, often split, and do not look lifelike. Larvae less than 10 mm long are difficult to freeze-dry unless special precautions are taken to keep them below freezing point throughout the procedure; for such specimens use critical point drying (p. 51).

(1) Place the freeze-drying container in a deep freeze to cool it while preparing specimens.

(2) Lower the specimens into liquid nitrogen. Larvae which tend to react violently when handled with forceps can be lowered into the liquid nitrogen while they are still feeding on their host (which can also be freeze-dried).

(3) After the vigorous boiling reaction has slowed down, remove the specimens with soft-nosed forceps. Transfer specimens quickly into the cold freeze-drying container.

(4) Attach the container to the freeze-drying machine as soon as possible or the specimens will start to thaw. The time required for complete drying depends on the size of the insect and must be determined by practice.

Specimens can be freeze-dried in a deep freeze, but not as well as using a machine. Place specimens in an unsealed container, and leave them for a month or so to freeze-dry.

CAUTION: freeze-dried specimens are brittle—handle them with care.

Air-drying insect larvae

This technique is limited in its application but it can be used when no freeze-drying facilities are available. Oldroyd (1958) outlines a suitable method.

Embedding insects in plastic

Burgmans (1968), Ralph & Hoverd (1968), and Moulds (1975) all describe techniques of embedding insects in resin. Embedding resin is available in New Zealand from most hardware stores. Australian Entomological Supplies (address p. 76) sells an embedding kit.

Basic use of a dissecting microscope

Although a large reading lens or hand-lens is useful in examining small specimens in a store box, as well as for preliminary sorting of field samples, accurate sorting and identification usually requires the use of a microscope.

Points to remember for using a dissecting (low-powered) microscope effectively and without strain are:

(1) The height of the chair must be correct in relation to the eyepieces of the microscope, otherwise you will have an aching back by the end of the day. You should not need to hunch up or stretch to look through the eyepieces.

(2) A good light source is essential (a 100 watt desk lamp may be suitable if a microscope light and transformer are not available).

(3) Eyepieces should be clean. Clean with lens tissue. (Do not use solvents, or try to remove any lens for cleaning; this must be done by an expert to avoid marring the lens surface and to ensure lenses are properly aligned).

(4) Focusing. Before using a microscope be sure the eyepieces are in focus to suit your eyes. Usually only one of the eyepieces of a dissecting microscope can be adjusted. Look through the non-adjustable eyepiece and focus sharply on something in the centre of the field of view (such as the fibres on the edge of a piece of paper). Then look through the adjustable eyepiece at the same object and turn the eyepiece adjuster until the image is sharp. When the microscope is properly adjusted the images seen by each eye should merge into a clear three-dimensional image. Check the focusing at the same time as you clean the eyepieces.

A light blue, green, or black background underneath a clear glass microscope stage may be better than clear glass alone for examining specimens using above-stage lighting.

CAUTION: The life of a microscope bulb will be reduced considerably unless the lamp is turned on gradually from its transformer to avoid a surge of current, or if the light is run at high voltage (usually indicated by a red sector on the transformer scale) for any length of time. The brightness of a cold-light source (Schott) is greatly improved with the use of the magnifier attachment on the swan-neck.

Turn the microscope light off at the transformer, and cover the microscope after use.

A compound microscope requires finer adjustments (see Bradbury (1984) for a comprehensive account).

CAUTION: Prolonged use with intense light can damage your eyes.

We have our microscopes cleaned and serviced every 2 years by a commercial firm.

Dissecting specimens

Often parts of insects must be dissected for detailed examination. For this, fine dissecting scissors, fine probes (p. 24), and watchmaker's forceps are useful. Any dissection of insect parts should be done under a dissecting microscope.

Parts which have been dissected and examined are either stored in genitalia vials kept with the dissected specimens, or mounted on a slide (slides must have labels associating them with the rest of the specimen). Some workers mount parts in Canada balsam on a flat piece of clear plastic pinned beneath the dissected specimen; however, if this method is used, make sure that the plastic used is not one which will be affected by camphor fumes.

For further information on dissection techniques see Upton & Norris (1980; general), Robinson (1976; Lepidoptera genitalia), and May (1977; Coleoptera larvae).

Hazardous properties of deterrents

(based on Bretherick 1981)

Threshold limit values (TLV) are expressed as parts of gas or vapour per million parts of contaminated air at 25°C and 1023 mbars, and as mg/m^3 for dusts. They "represent conditions which it is believed that nearly all workers may be repeatedly exposed day after day [8-hour day], without adverse affect" (the higher the figure the safer the product). The N.Z. Health Department can measure the level of a deterrent in a collections area.

Camphor

Synthetic camphor TLV 2 ppm (12 mg/m^3). No TLV for natural camphor.

Synthetic camphor can be made from two hazardous chemicals—vinyl chloride and cyclopentadiene.

Para-dichlorobenzene

TLV 75 ppm (450 mg/m^3).

Inhalation of vapours may cause drowsiness, and irritation of nose and eyes. Long exposure may result in liver damage.

Naphthalene

TLV 10 ppm (50 mg/m^3).

Hazardous chemicals

(based on Bretherick 1981)

Many chemicals used are hazardous because they have toxic effects if misused, or are fire hazards if spilt. Be aware of their dangers, and never neglect safety precautions when using them. Use a fume cupboard when transferring from one container to another; wear gloves, mask, and safety glasses as appropriate.

Spillage disposal (general instructions). Shut off all possible sources of ignition. Wear gloves and face shield. Mop up with plenty of water and run to waste diluting greatly with running water. Ventilate area well to evaporate remaining liquid and dispel vapour.

Acetic acid

A colourless liquid with a pungent acrid odour. Glacial acetic acid freezes to a crystalline solid in cool weather. Miscible with water.

FLAMMABLE. CAUSES SEVERE BURNS.

The vapour irritates the respiratory system. The vapour irritates and the liquid burns the eyes severely. The liquid is very irritating to the skin and can cause burns and ulcers. If taken by mouth, it causes internal irritation and damage.

Fire hazard: flash point 43°C; explosive limits 4–16%; ignition temperature 426°C. Extinguish fire with water spray, dry powder, carbon dioxide, or vaporising liquids.

Acetone

A colourless, mobile liquid with characteristic odour. Miscible with water.

HIGHLY FLAMMABLE.

Inhalation of vapour may cause dizziness, narcosis, and coma. The liquid irritates the eyes and may cause severe damage. If swallowed it may cause gastric irritation, narcosis, and coma.

Fire hazard: flash point –18°C; explosive limits 3–13%; ignition temperature 538°C. Extinguish fire with water spray, dry powder, carbon dioxide, or vapourising liquids.

Ammonia (gas)

Colourless gas with pungent odour.

FLAMMABLE. TOXIC BY INHALATION.

The gas irritates all parts of the respiratory system. The gas irritates the eyes severely.

Ammonia (solutions)

Commonly supplied as 35% solution in water. In warm weather the strong solution develops pressure in the bottle and the cap must be removed with care.

CAUSES BURNS. IRRITATING TO EYES, RESPIRATORY SYSTEM, AND SKIN.

The vapour irritates all parts of the respiratory system. The solution causes severe eye burns. The solution burns the skin. If swallowed, the solution causes severe internal damage.

Benzene (Benzol, coal naphtha)

Colourless, volatile liquid with characteristic odour. Immiscible with water.

HIGHLY FLAMMABLE. TOXIC BY INHALATION AND CONTACT WITH SKIN. DANGER OF VERY SERIOUS IRREVERSIBLE EFFECTS.

Inhalation of the vapour causes dizziness, headache, and excitment; high concentrations may cause unconsciousness. The vapour irritates the eyes and mucous membranes. The liquid is absorbed through the skin and poisoning may result from this. Assumed to be very poisonous if taken by mouth. CHRONIC EFFECTS. Repeated inhalation of low concentrations over a considerable period may cause severe, even fatal, blood disease (leukaemia).

Fire hazard: Flash point −11°C; explosive limits 1.4–8%; ignition temperature 562°C. Extinguish fire with foam, dry powder, or vaporising liquids.

Spillage disposal. Wear breathing apparatus and gloves. Either apply non-flammable dispersing agent, e.g., BDH "Slix", work to emulsion with brush and water, and run to waste diluting greatly with running water; or absorb on sand, shovel into bucket(s), and transport to safe, open area for atmospheric evaporation.

Chloral hydrate

Colourless crystals with acrid odour and bitter taste; soluble in water.

TOXIC IF SWALLOWED. IRRITATING TO EYES AND SKIN.

Irritates the skin and eyes. If taken by mouth it may cause nausea, vomiting, coldness of extremities, and unconsciousness.

Chloroform (trichloromethane)

Colourless volatile liquid with a characteristic odour; immiscible in water.

HARMFUL BY INHALATION.

The vapour has anaesthetic properties, causing drowsiness, giddiness, headache, nausea, vomiting, and

unconsciousness. The vapour and liquid irritate the eyes causing conjunctivitis. The liquid is poisonous if taken by mouth. Suspected carcinogen.

Spillage disposal. Wear breathing apparatus and gloves. Either apply dispersing agent, work to an emulsion, and run to waste diluting greatly with running water; or absorb on sand, shovel in bucket(s), and transport to safe, open area for atmospheric evaporation.

Ethanol (Ethyl alcohol)

Colourless, mobile liquid with characteristic smell; miscible with water.

HIGHLY FLAMMABLE.

Avoid breathing vapour in high concentrations because of its intoxicating qualities.

Fire hazard: Flash point 12°C; explosive limits 3.3–19%; ignition temperature 423°C. Extinguish fire with water spray, dry powder, carbon dioxide, or vapourising liquid.

Ethyl acetate

Colourless, volatile liquid with fragrant odour; 1 part soluble in about 35 parts water at 25°C.

HIGHLY FLAMMABLE.

The vapour may irritate the eyes and respiratory system. The liquid irritates the eyes and mucous surfaces. Prolonged inhalation may cause kidney and liver damage.

Fire hazard: Flash point –4.4°C; explosive limits 2.5–11.5%; ignition temperature 427°C. Extinguish fire with water spray, foam, dry powder, carbon dioxide, or vaporising liquid.

Spillage disposal. Wear face shield or goggles, and gloves. Either apply non-flammable dispersing agent, e.g., BDH "Slix", work to an emulsion, and run to waste diluting greatly with running water; or absorb on sand, shovel into bucket(s), and transport to safe, open area for atmospheric evaporation.

Formaldehyde solution (formalin)

Colourless, sometimes milky solution with pungent odour; miscible with water; the solution generally contains 37–41% formaldehyde and 11–14% methanol.

TOXIC BY INHALATION, IN CONTACT WITH SKIN, AND IF SWALLOWED.

Avoid storage in room with hydrochloric acid and household chlorine bleach, as mixed vapours form the potent carcinogen bis(chloromethyl) ether.

The vapour irritates all parts of the respiratory system. The liquid and vapour irritate the eyes severely. The liquid

in contact with the skin has a hardening or tanning effect and causes irritation. Severe abdominal pains with nausea and vomiting and possibly loss of consciousness follow ingestion. CHRONIC EFFECTS. High concentration of vapour inhaled for long periods can cause laryngitis, bronchitis, or bronchial pneumonia; prolonged exposure may cause conjunctivitis; in contact with skin for long periods will cause cracking of skin and ulceration, particularly around fingernails. Nasal tumours have recently been reported.

Fire hazard: Flash point 50°C. Extinguish fire with water spray, dry powder, carbon dioxide, or vaporising liquid.

Lactic acid

Colourless or slightly yellow, syrupy, hygroscopic liquid; miscible with water.

IRRITATING TO EYES AND SKIN.

Irritates and may burn the eyes and skin. Irritant if taken by mouth.

Spillage disposal. In addition to general instructions on p. 68, first spread soda ash liberally over the spillage.

Petroleum spirit (petroleum ether)

HIGHLY FLAMMABLE.

Inhalation of high concentrations of the vapour, particularly of the lower boiling fractions, can cause intoxication, headache, nausea, and coma. The liquids irritate the eyes, and skin contact results in defatting of the area of contact, increasing the risk of dermatitis from other agents. If taken by mouth they may cause burning sensation, vomiting, diarrhoea, and drowsiness.

Fire hazard: Flash point (lower fractions) below –17°C; explosive limits approx. 1–6%; ignition temperatures range from about 250°C. Extinguish fire with foam, dry powder, carbon dioxide, or vaporising liquid.

Spillage disposal. Wear face shield or goggles and gloves. Either apply non-flammable dispersing agent, e.g., BDH "Slix", work to emulsion, and run to waste diluting greatly with running water; or absorb on sand, shovel into bucket(s), and transport to safe, open area for atmospheric evaporation.

Phenol (carbolic acid)

Colourless to pink crystalline substance with distinctive odour; somewhat soluble in water.

TOXIC IN CONTACT WITH SKIN AND IF SWALLOWED CAUSES BURNS.

The vapour irritates the respiratory system and eyes.

Skin contact causes softening and whitening followed by the development of painful burns; its rapid absorption through the skin may cause headache, dizziness, rapid and difficult breathing, weakness, and collapse. If taken by mouth it causes severe burns, abdominal pain, nausea, vomiting, and internal damage. CHRONIC EFFECTS. The inhalation of vapour over a long period may cause digestive disturbances, nervous disorders, skin eruptions, and damage to the liver and kidneys; dermatitis may result from prolonged contact with weak solutions.

Spillage disposal. Wear face-shield or goggles. Mix with sand and transport to a safe, open area for burial. Site of spillage must be washed thoroughly with water and detergent.

Picric acid

Picric acid dangers are well covered by Pilgrim (1957).

Picric acid (yellow crystals) should be kept moist with not less than half its own weight of water.
RISK OF EXPLOSION BY SHOCK, FRICTION, FIRE, OR OTHER SOURCES OF IGNITION. FORMS VERY SENSITIVE EXPLOSIVE METALLIC COMPOUNDS. TOXIC BY INHALATION, IN CONTACT WITH SKIN, AND IF SWALLOWED.

Skin contact may result in dermatitis. Poisonous if taken by mouth. CHRONIC EFFECTS. Absorption through the skin or inhalation of dust over a long period may result in skin eruptions, headache, nausea, vomiting, or diarrhoea; the skin may become yellow.

Spillage disposal. Wear face shield or goggles and gloves. Moisten well with water and mix with sand. Transport to isolated area for burial. Site of spillage must be washed thoroughly with water and detergent.

Potassium hydroxide (caustic potash)

Colourless sticks, flakes, powder, or pellets soluble in water.
CAUSES SEVERE BURNS.

The solid and its solutions severely irritate and burn the eyes and skin. If taken by mouth there would be severe internal irritation and damage.

Spillage disposal. Shovel into large volumes of water in an enamel or polythene vessel and stir to dissolve; run the solution to waste diluting greatly with running water.

Potassium nitrate

Colourless crystals, soluble in water.
EXPLOSIVE WHEN MIXED WITH COMBUSTIBLE MATERIALS.

Spillage disposal. Shovel into bucket of water and run solution to waste, diluting greatly with running water. Site of spillage should be washed thoroughly to remove all oxidant, which is liable to make any organic matter with which it comes into contact dangerously combustible when dry (particularly wood, paper, and textiles). Clothing wetted with the solution should be washed thoroughly.

Checklist of supplies

This checklist is used by the N.Z. Arthropod Collection, and is included because there is no specialised entomological supply house in New Zealand; consequently there are difficulties at times in obtaining items. Mention of a proprietary name does not imply endorsement of the product. We constantly check the suitability of other products and suppliers, and may change to them if it is to our advantage.

Abbreviations used for suppliers are listed at the end of the checklist.

Item	Manufacturer	Supplier
Alcohol see ethanol		
Balsa board	–	Toy and model shops
Balsam bottles	–	SSB
Bristol board (3-ply)	De la Rue	–
Brushes	–	Art suppliers
Cabinet drawers	Russell Mfg	RM
Camphor blocks, 1/4-oz, ("Pentagon")	Hong Kong	NZD
Camphor box (die 2447)	UEB Box Division	UEB
Canada Balsam in xylene	BDH	SSB
Carbowax see polyethylene glycol		
Cardboard rectangles	–	AES
Cartons (for packing store boxes)	UEB Box Division	UEB
Cavity dishes	–	LP
Cavity slides	–	NZFE
Cellosolve	BDH	SSB
Cellulose wadding	–	KMS
Clove oil	–	LP
Cobb aluminium slides (for double coverslip method)	Technical & Physical Engineering Research Institute, Holland	PL
Coloured photographic see photographic paper		
Cottonwool	–	KMS
Coverslips, square and round	–	SS
Dissecting scissors	John Weiss	JW, AES
Embedding plastic	Consolidated Chemical	Hardware stores, AES
Embro	–	AES
Ethanol (95%, rectified spirit)	–	KMS, SSB
Ethyl acetate	BDH	SSB
Euparal	BDH 36122, GBI Laboratories	SSB
Forceps: Featherlight	Griffin DKC-620-X	SSB, AES
Storkbill	" DKC-700-4	"
Pinning	" DKC-590-T	"
Watchmaker's (type INOX 5)	" DKC-770-R	"
Genitalia vials	–	A, BP, TD
Glass slides	Sail	NZFE
Glue ("Gripit") for glueing Plastazote	Davis Gelatine (N.Z.) Ltd	Hardware stores
Glue for mounting insects	Seccotine	AES
Glyptal	–	SSB
Goatskin Parchment Paper see label paper		
Gum arabic ("photo purified")	Mallinkrodt	SSB

Item	Manufacturer	Supplier
Hand lens: large	–	Optical supplies
small, x 10 Compound	–	AES
Ivory board (Orbit, 400 gsm)	Wiggins Teape (importer)	Art suppliers
Jars (0.6 l storage)	N.Z. Glass Manufacturers	Hardware stores
Label paper (Goatskin, 100 gsm, blue-white weave)	Wiggins Teape (importer)	Art suppliers
Methyl cellulose ("Methocellulose")	BDH	SSB
Microscopes and accessories	see Ramsay & Singh (1982)	
Mini-vials (Durham fermentation tubes)	Volac	SSB
Paper roll for wrapping parcels (brown, 900 mm wide, 110 gsm)	–	RP
Pen (for labelling, Rotring rapidograph Iso with ink cartridges)	Rotring	HN, OG
Photographic paper (resin-coated)	Tura or Kentmore	L
Pins: insect (anticorro, stainless, nylon head, white)	Emil Arlt	EA, AES
minuten	Emil Arlt	EA, AES
cabinet	Lill	WD
Pipette (dropping) bottles	–	SSB
1-oz polystop	–	SSB
Plastazote (polyethylene enclosed-cell foam plastic)	–	S, trampers supply shops as sleeping mats
Point punch (style A, straight base, blunt point)	Wards	SSB, AES
Polycarbonate vials	–	SSB
Polyethylene glycol:		
"Peg 300" for mini-vials	–	SSB
4000, for slide mounting	BDH	SSB
Pyrex spot plates	Arthur Thomas	SSB
Setting board	–	AES
Slide boxes: Tray storage	Russell Mfg	RM, AES
Slide storage	UEB Box Division	UEB
Slide labels (glue quality HP5)	Quik Stik	QS
Slide Oven ("Watvic")	Watson Victor	WV
Slide trays (aluminium)	F.G.P. Pty Ltd	F
Staining well (Embryological watch glass)	Arthur Thomas	SSB
Store boxes	Russell Mfg, or local maker	RM, local maker, AES
Ultrasonic cleaner	–	WV
Vials (50 x 12 mm, glass) and polyethylene closures	Glass Wholesalers Supplies (GWS)	SSB

Abbreviations and addresses of suppliers

A Arthropod Specialities Co.
Box 1973, Sacramento,
California 95809, U.S.A.

AES Australian Entomological Supplies
Box 314, Miranda,
N.S.W. 2228, Australia

BP Bioquip Products Ltd
Box 61, Santa Monica,
California 90406, U.S.A.

EA Emil Arlt
Box 37,
A-1125 Wien, Austria

F F.G.P. Co. Pty Ltd
21-31 Branson Ave,
Clearview, S.A. 5085, Australia

HN Henry B. Norcross Ltd
Box 21102, Henderson,
Auckland 8

JW John Weiss & Son Ltd
11 Wigmore St,
London W1H 0DN, England

KMS Kempthorne Medical Supplies
Box 1234, Auckland

L E.C. Lackland Ltd
Box 56036,
Auckland

LP Labsupply Pierce (N.Z.) Ltd
Box 32234, Birkenhead,
Auckland

NZD New Zealand Distributors
Box 9869, Newmarket,
Auckland

NZFE New Zealand & Far East Ltd
Box 9291,
Auckland

OG Office Graphics Ltd
Box 77006, Mt Albert,
Auckland

PL Plessey Fabrieken N.V.
Box 46, Noordwijk,
The Netherlands

QS Quik Stik International Ltd
Box 13031,
Auckland

RM Russell Manufacturing Co. Ltd
Bolt Rd,
Tahunanui, Nelson

RP Ransons Packaging and Display Ltd
Box 8745, Symond St,
Auckland

S Sheppard Industries
Box 62067, Mt Wellington,
Auckland

SS Scientific Supplies
Box 14454, Panmure,
Auckland

SSB Salmond Smith Biolab Ltd
Private Bag, Northcote,
Auckland

TD T.P. Drewitt
104 Marlborough Rd,
Chingford,
London E4 9AL, England

UEB UEB Industries Ltd
Camphor box:
Set-up Division,
Box 14109, Auckland

Packing box:
Box 57012,
Mangere Bridge, Auckland

WD Watkins & Doncaster
Four Throws, Hawkhurst,
Kent, England

WV Watson Victor Nicholas Ltd
Box 1216,
Auckland

Entomological catalogues

(other suppliers listed in Ramsay & Singh (1982))

ARTHUR H. THOMAS
Salmond Smith Biolab Ltd,
Private Bag, Northcote, Auckland

AUSTRALIAN ENTOMOLOGICAL SUPPLIES
Box 314, Miranda, N.S.W. 2228,
Australia (no N.Z. agent)

BIOQUIP PRODUCTS
Box 61,
Santa Monica CA 90406,
U.S.A. (no N.Z. agent)

GRIFFIN
Kempthorne Medical Supplies Ltd,
Box 1234, Auckland

TURTOX/CAMBOSCO
Salmond Smith Biolab Ltd,
Private Bag, Northcote,
Auckland

WARDS
Salmond Smith Biolab Ltd,
Private Bag, Northcote,
Auckland

WATKINS & DONCASTER
Four Throws, Hawkhurst,
Kent, England (no N.Z. agent)

Formulae

(fixatives, fluids, preservatives, mounting media, and glue)

Ethanol (general preservative)

	required strength				
	60%	70%	75%	80%	90%
Ethanol, commercial grade 95%	6	7	7.5	8	9 parts
Water	3.5	2.5	2	1.5	0.5 parts

Acetoalcohol mixture (fixative)

Ethanol, commercial grade 95%	50 ml
Distilled water	45 ml
Glacial acetic acid	20 ml

AGA solution (thrips preservative)

Ethanol, 60%	10 parts
Glycerol	1 part
Glacial acetic acid	1 part

Andre's slide mounting medium

Glacial acetic acid	50 ml
Chloral hydrate	50 g
Water, distilled	50 ml

Barber's fluid (beetle relaxing fluid)

Ethanol, commercial grade 95%	53 parts
Water	49 parts
Ethyl acetate	19 parts
Benzol	7 parts

If the solution is cloudy, add a few drops of 95% ethanol and shake.

Berlese's slide mounting medium

Chloral hydrate	160 g
Gum arabic (clear crystals)	15 g
Glucose syrup	10 g
Glacial acetic acid	5 g
Water, distilled	20 ml

Bouin's fixative (for injecting Orthoptera to fix and distend them)

Picric acid, saturated aqueous solution	75 ml
Formalin, commercial grade 40% formaldehyde	25 ml
Glacial acetic acid	5 ml

Fix specimens for 12 h or longer, and transfer direct to 75% ethanol. Specimens may be left in Bouin's indefinitely.
CAUTION: see Hazardous chemicals (p. 72).

Carnoy's fixative (for fixing material for chromosome studies)

Ethanol, commercial grade 95%	3 parts
Glacial acetic acid	1 part

The fixative should be made up shortly before use. Remove surface water from specimens before putting them live in the fixative.

Carnoy's fluid (modified; for fixing Lepidoptera larvae)

Ethanol, commercial grade 95%	6 parts
Chloroform	3 parts
Glacial acetic acid	1 part

Essig's slide mounting medium (for soft-bodied Homoptera)

Lactic acid (reagent grade 85%)	20 parts
Phenol (saturated in distilled water)	2 parts
Glacial acetic acid	4 parts
Water, distilled	1 part

Heat at 56–60°C for 30–60 minutes. Store in a brown bottle.

FAA fixative

Formalin, commercial grade 40% formaldehyde	10 parts
Ethanol, commercial grade 95%	50 parts
Glacial acetic acid	1 part
Water	40 parts

de Faure's slide mounting medium

Gum arabic	30 g
Chloral hydrate	50 g
Glycerol	20 ml
Water, distilled	50 ml

Mix and filter.

Gault's solution (for pitfall traps)

Sodium chloride (salt)	50 g
Chloral hydrate	10 g
Potassium nitrate	10 g
Water	1000 ml

For field use, make up the dry parts and leave in the bottom of a 1 litre container. When ready for use, fill the container with clean water.

CAUTION: see potassium nitrate in Hazardous chemicals (p. 72).

Hoyer's slide mounting medium (for mites and thrips)

Gum arabic (clear crystals)	30 g
Water, distilled	50 ml
Chloral hydrate	200 g
Glycerol	20 g

Dissolve the gum arabic in the water at room temperature. Add the chloral hydrate and leave a day or two until all solids have dissolved. Add the glycerol. Filter through glass wool. Store in a bottle with a glass stopper.

KAA solution (for killing soft-bodied larvae)

Kerosine	1 part
Ethanol, commercial grade 95%	10 parts
Glacial acetic acid	2 parts

Add 1 part Dioxane for KAAD solution.

For very soft-bodied larvae, use half as much kerosine or less. The solution is excellent for killing larvae to be photographed, particularly scarabs. Transfer to 95% ethanol after 24 hours for long-term storage.

KOH solution, 10%
(for dissolving soft tissues from sclerotised structures)

Potassium hydroxide (KOH) pellets	25 g
Water, distilled	250 ml

Mix in a beaker, and transfer to a bottle once the solution has cooled. Leave the top off until the solution has come to room temperature.

Kahle's fixative

Ethanol, commercial grade 95%	15 parts
Water, distilled	30 parts
Formalin, commercial grade 40% formaldehyde	6 parts
Glacial acetic acid	1 part

Specimens can be kept in this fixative indefinitely.

Pampel's fixative (suitable for spiders)

Ethanol, commercial grade 95%	15 parts
Water, distilled	30 parts
Formalin, commercial grade 40% formaldehyde	6 parts
Glacial acetic acid	4 parts

Gibbs (1981) explains how to use the fixative for spiders.

PEA (fixative for beetle larvae)

Petroleum ether	1 part
Ethanol, commercial grade 95%	7–10 parts
Glacial acetic acid	2 parts

Keep specimens in PEA overnight, remove, and store in 75% ethanol. Specimens can be kept in PEA for longer periods without damage.

Tillyard's glue (water-soluble), for mounting insects on points and card

Gum arabic	60 g
Sugar	30 g
Carbolic acid	2 ml
Ethanol, commercial grade 95%	8 ml
Water, distilled (warm)	45 ml

Carbolic acid is made from 5% phenol crystals dissolved in distilled water. Dissolve the gum arabic in the warm distilled water. Add the sugar, phenol, and ethanol. Filter through glass wool if necessary.

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Further reading

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Index

Conversion from feet to metres

feet	0	1	2	3	4	5	6	7	8	9
					metres					
0		0.31	0.61	0.91	1.22	1.52	1.83	2.13	2.44	2.74
10	3.05	3.35	3.66	3.96	4.27	4.57	4.88	5.18	5.49	5.79
20	6.10	6.40	6.71	7.01	7.32	7.62	7.93	8.23	9.53	8.84
30	9.14	9.45	9.75	10.06	10.36	10.67	10.97	11.28	11.58	11.89
40	12.19	12.50	12.80	13.10	13.41	13.72	14.02	14.33	14.63	14.94
50	15.24	15.55	15.85	16.15	16.46	16.76	17.07	17.37	17.68	17.98
60	18.29	18.59	18.90	19.20	19.51	19.81	20.12	20.42	20.73	21.03
70	21.34	21.64	21.95	22.25	22.56	22.86	23.17	23.47	23.77	24.08
80	24.38	24.69	24.99	25.30	25.60	25.91	26.21	26.52	26.82	27.13
90	27.43	27.74	28.04	28.35	28.65	28.96	29.26	29.57	29.87	30.18
100	30.48	30.79	31.09	31.39	31.70	32.00	32.31	32.61	32.92	33.22

The Entomological Society of New Zealand (Incorporated)

The Society was founded in 1951 to provide a common meeting ground for everyone interested in entomology in New Zealand. It publishes an annual journal ("New Zealand entomologist"), a Scientific Supplement ("The Weta"), and occasional bulletins, and holds a conference each year. A computerised bibliography of New Zealand entomology has been prepared. A 21st Anniversary Research Fund was set up in 1973; applications for grants can be made by both members and non-members and societies or groups. There are a number of local Branches which hold regular meetings.

Anyone interested in joining the Society or wanting further information should contact:

The Secretary
Entomological Society of N.Z. (Inc.)
8 Maymorn Road
Te Marua
Upper Hutt

Entomology Division
Department of Scientific and Industrial Research

Office address: Entomology Division, DSIR
Mt Albert Research Centre
120 Mt Albert Road
Mt Albert
Auckland 3

Postal address: Entomology Division, DSIR
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Telephone:	National	(09) 893 660
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The Authors

Annette K. Walker worked with the New Zealand Arthropod Collection for 15 years; for the last 2 years she has worked as a hymenopterist with the Commonwealth Agricultural Bureaux, International Institute of Entomology, London. Her 25 publications include 2 co-authored contributions on thrips in the "Fauna of New Zealand" series, 2 popular insect books in the "Mobil New Zealand Nature Series", and a revision of David Miller's book "Common Insects in New Zealand".

Trevor Crosby has been curator of the New Zealand Arthropod Collection for 12 years; his research concentrates on blackflies (sandflies). Dr Crosby has written some 40 scientific publications; a recent paper he co-authored was awarded the Philip Allen Memorial Award for 1987 by the Forensic Society of Great Britain.

This handbook sets out methods and techniques, used by the New Zealand Arthropod Collection, (NZAC), for preparing insects for its collection and how the collection is curated and managed. The NZAC is part of Entomology Division, New Zealand Department of Scientific and Industrial Research.