JOINT NATURE CONSERVATION COMMITTEE

Institute of Terrestrial Ecology

BWARS

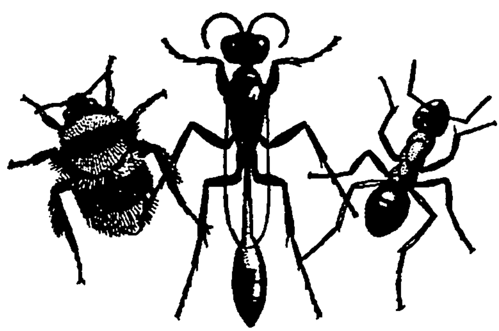
Bees, Wasps and Ants Recording Society

Starter Pack

Centre for Ecology and Hydrology
Natural Environment Research Council

BWARS

**Bees, Wasps and Ants
Recording Society**



Starter Pack

Second edition. Revised for BWARS by Robin Edwards (1995)

Published by the Biological Records Centre,
Environmental Information Centre
NERC Institute of Terrestrial Ecology
Monks Wood
Huntingdon
Cambs PE17 2LS

Printed in 1996 by RAM Technical Print, Derby

ISBN 1 870393 26 0

BWARS is indebted to the Biological Records Centre for arranging the printing, and for providing the finance, for the publication and initial distribution of this Starter Pack.

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About BWARS

There is an urgent need for proper, well-coordinated data on distribution and habits for many animals to support conservation programmes, ecological research, and to promote effective conservation strategies on a national basis (the intended function of the National Conservation Agencies). Such data are not only useful to monitor individual populations or species, but can be used to great effect to predict or diagnose the condition of areas of natural habitat. This is possible because of the sensitivity of many plants and animals, especially insects (because of their great diversity and sheer numbers), to otherwise unobtrusive environmental changes, all too frequently detrimental and man-made.

To collate, sort and store these data, the Biological Records Centre (BRC) was set up in 1964 at Monks Wood, and since 1991 has been jointly funded by the Natural Environment Research Council (NERC) and the Joint Nature Conservation Committee (JNCC). Information is stored on computer for easy access and comprises data on distribution (principally) and (among others) feeding habits, host records, general behaviour and activity. BRC publishes much of the data from recording schemes as distribution maps.

BRC helped to set up a number of recording schemes, including those for bumble bees, wasps and ants which were later amalgamated into the Bees, Wasps and Ants Recording Scheme (BWARS). BWARS was started in 1978 with an initiative from the International Bee Research Association. George Else (Natural History Museum, London) ran the whole scheme virtually single-handed until 1986. During this period over 150 members received the Bulletins compiled by George, and a number of field meetings were arranged with the help of Ian McLean and Alan Stubbs (Nature Conservancy Council; now English Nature). The first committee emerged from a public meeting held in May 1986.

In January 1995, BWARS changed to a formally constituted Society with elected officers, and is now run by a steering group of several volunteer entomologists who meet annually. The acronym did not change and remains 'BWARS'. The Society is still sending data to BRC for storage, and the latter assists with the publication of this Starter Pack and new Provisional Atlases.

BWARS publishes a bi-annual Newsletter containing a list of the current officers; information on meetings; profiles of aculeates and Society members; notes on sites; observations of interest; and other contributions from members. We look forward to your involvement in the recording scheme!

Introduction

Some guidance on the collection and identification of Hymenoptera is essential for the raw recruit. It is hoped that this pack will help you to get started, or to direct your interests to species which may be unfamiliar. The Society is involved with the three groups of insects that are known collectively as the 'Aculeata' – the stinging Hymenoptera, as opposed to the 'Parasitica' – parasitic wasps, and the Symphyta – sawflies. Most novice entomologists, and many who are experienced but unfamiliar with aculeates, will not know the techniques available for observing and collecting these insects, or how to identify them.

Section 2 of the Pack is devoted to the collection and preservation of specimens. Please remember that aculeates should only be collected with restraint as few species are common. Collecting one or two specimens of a species is usually vital for identification and recording purposes, and is likely to do no harm, but with species which may be locally rare it is best to take only a male rather than a female. There is, of course, much valuable work still to be carried out on the natural history of aculeates which involves observation rather than collecting. However, if site conservation and knowledge of the wider status of aculeates is to be put on a firm basis, the identification of voucher specimens (which involves collecting) is usually essential.

In addition, it is increasingly important to be aware of the need to conserve our natural fauna, and this is as true for aculeates as for other animals. Thus, in Section 1 we give a very broad overview of this aspect, and in Section 5 we show how some aculeates can be encouraged to nest in gardens, which can be vital oases for wildlife in urban areas.

The Society provides the common link for the activities mentioned above. However, the principal aim of the Society's work is to record where the various species occur. This important part of the scheme will be explained in Section 4: selection of 'target species', how to make a record, and completing and submitting computerised records or record cards.

Maps including your records will be published in a series of Provisional Atlases, and these will also show areas where searches could usefully be made. Most maps published in the Atlases will be accompanied by 'species profiles' containing details of the ecology of the insects concerned.

SECTION 1

THE CONSERVATION OF BEES, WASPS AND ANTS

George R. Else, John Felton and Alan Stubbs

Introduction

The vast majority of our wild bees and wasps are solitary species whose presence attracts little attention. Some of these unobtrusive species may easily be mistaken for hive bees or bumble bees: others share the yellow and black markings of social wasps. However, most of the species are small and merge into the background of the living countryside and even towns. But the bees, wasps and ants are a fascinating and important group of insects which deserve positive recognition by the conservation movement. They are often relatively rare species with highly specialised requirements.

Conservation value

It is very easy to overlook the presence and role of many insects in our countryside. With aculeates one can point to a direct beneficial role as well as many other exceptional qualities.

- Bees, and indeed to some extent wasps, are valuable in the cross-pollination of cultivated flowering crops, clover and fruit trees being prime examples. The serious consequences of the loss of common wild pollinators has been recognised in some fruit-growing districts because it is expensive and not always practical to import bee hives. In addition, the clovers have specialised flowers which are most efficiently pollinated by certain species of bees. The loss of wild bees may be particularly attributed to the use of toxic sprays, the loss of marginal land containing nesting sites and flowers, and to the mowing of roadside verges. Wild bees will do the job free, but, whilst fruit trees may flower for a week or two, wild flowers are needed to support species with a longer flight period.

- The pollination of flowers should also be of direct concern to those managing areas of natural and semi-natural vegetation. Most species of aculeates show preferences for certain types of flowers, notably those adapted to bee pollination, and some bees are dependent on certain flower species. The health of the pollinators has a direct bearing on the seed production of plants so that, at least in theory, the vegetation composition of an area should be affected by the presence or absence of certain pollinators.

- Wasps have a natural role as predators of other insects. One may argue for the benefit of social wasps in serving a useful role in killing numerous harmful and nuisance insects – they just happen also to like jam (substitute flowers) and lose their peaceful nature when their nests are disturbed. The solitary species are serving the same function in a more unobtrusive fashion and most of them cannot sting people even if molested. They are often fairly specific about what they take as prey so they may be of local ecological importance. Such prey variously includes caterpillars, flies, beetles, aphids and other bugs.

- Ants also play an important role in many ecosystems. In habitats where they are abundant, they may be major predators and scavengers of other organisms. Their presence can often have a beneficial effect on the populations of invertebrates with which they are closely associated. However, others (eg wood ants) are by no means beneficial in some areas!

- The aculeates can form an important component of the girder system within ecosystems (inter-relationship between community units, eg as pollinators and in having a role in several different micro-habitats).

- Solitary wasps stand in the same relation to their prey as do the birds of prey to theirs. Monitoring of sphecid populations could provide a sensitive environmental indicator.

- Both social and solitary species provide opportunities to study a range of very advanced patterns of behaviour. In this respect, aculeates are much more important than most other insects. They are of considerable interest in many fields of study, including evolution and mechanisms of behaviour and learning.

- There are many examples of parasitism and cuckoo relationships whose evolutionary and behavioural aspects are of interest. The inter-relationship between species is often highly specific.

- There are some good examples of local races and of the British forms differing from those on the continent. These are of genetic and zoogeographical interest.

- A rich variety of insects is dependent on bees, wasps and ants either as parasites or as scavengers and inquilines in nests. The beetles associated with hornet nests are a notable example. Thus, the conservation perspective extends on a broader front than one may assume at first sight.

- The aculeates are particularly sensitive animals which react rapidly to change. They therefore provide a good group for environmental monitoring. There are some very thorough local and old county lists which could be used as a basis for monitoring selected areas.

- Some species pairs have ranges which are mutually exclusive, and with a fairly well-defined line dividing them. The separation is thought to be due to climatic factors so that there may here be a sensitive means of monitoring response to climatic fluctuations. Some species which are on the edge of their range have possibly become extinct since the study of Hymenoptera began two centuries ago.

AN OUTLINE OF THE BRITISH FAUNA

The aculeate fauna of Britain and Ireland comprises some 600 species, of which just over one third are solitary bees, one third are solitary wasps, and the remainder are ants, social bees and social wasps.

Solitary bees The British bee fauna contains some attractive species as well as many obscure ones. Some are large and might be mistaken for a hive bee or worker bumble bee, whilst the range goes down to very small species only a few millimetres long. The fauna is dominated by a few large genera, such as *Andrena* with about 60 species.

Solitary species have one or sometimes two generations a year. Most species have fairly specific requirements for nest sites. Many like to burrow in quarry faces, bare banks or on bare flat ground – even heavily trampled areas in a public park often have some species. Sandy ground provides the most favourable soil, though clay is often used providing it remains fairly dry throughout the year. Though solitary, many species favour very restricted areas as nesting sites so that large colonies may be formed. Nevertheless, each bee makes its own burrow; there is no sharing as in social species.

There are some genera of bees which have the yellow and black pattern of wasps or a red and black pattern, and these are cuckoos of the pollen- and nectar-gathering bees; the females can be found round the entrances to the burrows of their specific hosts. Another group of bees resembles small black wasps and some of them share the habit of nesting in the broken ends of old bramble *(Rubus fruticosus)* stems and similar situations. Among the more unlikely nest sites are empty snail shells.

Many species are particularly associated with certain flowers. Where there are strong colonies of bees often running into hundreds and sometimes thousands, they must have a marked role in pollination. Some species of bee are very largely confined to a single flower species or genus such as *Macropis europaea* on yellow loosestrife *(Lysimachia vulgaris)*, *Andrena florea* on white bryony *(Bryonia dioica)* and *Melitta haemorrhoidalis* on bellflower *(Campanula)* species.

Solitary bees have many parasites. In addition to parasitic solitary bees, these include flies which lurk around the entrances to their burrows. Bees with a distorted abdomen usually harbour one or more Strepsiptera, highly specialised parasitic insects apparently related to beetles.

Solitary wasps This term covers a wide range of very different types of wasp and it is here that the enormously developed patterns of behaviour are perhaps most remarkable.

All are predatory, each group or species having its own choice of prey. The spider-hunting wasps have a dangerous task, made easier by the innate fear of some spiders on the approach of these wasps – the most remarkable is *Aporus unicolor* which attacks *Atypus affinis* in its purse-web, a spider with huge fangs. The prey of various other wasps includes caterpillars, flies, aphids and plant bugs.

The prey is usually placed in a specially dug burrow and, as with bees, a sandy soil is very favourable. Other species burrow in the ends of dead bramble stems and other pithy plant shoots. Among the more bizarre types of nests are those of the potter wasp which makes a little vase-like cell out of mud on heather *(Calluna vulgaris)* shoots. A number of species nest in old beetle burrows in dead wood or even nail holes in fence posts. Though the solitary wasps are essentially predators, they visit flowers to various degrees and play a not insignificant part in pollination.

Two interesting, though rarely studied, groups are the Dryinidae and Bethylidae, with about 50 British species. Females may be fully winged, brachypterous or apterous (when they appear ant-like). In the first family, most females have modified, chelate, fore tarsi which are used to grip their homopterous (leafhopper) prey. Female *Aphelopus*, however, lack this claw and instead grip their prey with the fore- and mid-legs.

Dryinid larvae feed externally on the host, living in a sac (the remains of the first larval exuvium). Bethylid larvae are predacious on beetle and moth larvae, sometimes those associated with stored cereals. Females often drag paralysed prey into a sheltered place before ovipositing on them: some have been seen to stay with, and 'guard', the developing larvae, and some may even mate with their own progeny.

Other wasp groups are parasites, such as the beautiful metallic chrysidid wasps, and the ant-like wingless female tiphiid and mutillid wasps that attack tiger beetle larvae and bumble bee larvae and pupae.

Ants All ants are social; there are no solitary species anywhere in the world. The 50 or so species in Britain display much of the interesting biology and behaviour which make ants a fascinating group to study.

Colonies may range in size from a few dozen individuals in genera such as *Leptothorax* to a few hundred thousand in the *Formica rufa* group (wood ants). Nests can be found in the soil, under stones, in twigs, tree stumps, rock crevices, and many other situations. The nest itself often goes unnoticed, although a few species of *Lasius* and *Formica* raise more obvious mounds of vegetable material. Some species are associated with certain habitats, such as woodland, heather moor, sandy heath or permanent pasture. Others are more adaptable (eg the common black garden ant, *Lasius niger*) and may be found nesting in various habitats including urban environments.

An ant colony usually consists of one or more fertile queens and a large number of smaller workers which are female but without a reproductive capacity. At certain times of the year, mature colonies may also produce unmated females (gynes) and males. This sexual generation is often winged and flies from nests to mate: the males then die and the newly mated queens break off their wings and search for somewhere to start a new colony. Colonies survive the winter by hibernating, usually deep underground; in Britain most are active again by March or early April. Survival depends on the presence of a fertile queen, which may live for 15-20 years; by accepting new queens, the colonies of some species can last much longer.

Most ants are predatory or scavenge dead insects, but some form close associations with sap-sucking Homoptera (eg aphids and coccids) in order to obtain their sweet excreta (honeydew). Many species visit the floral and extrafloral nectaries of low-growing plants, and may play a role in transporting the seed of others such as violets *(Viola)* and fumitory *(Fumaria)*.

Ant nests may be home to a large variety of other organisms ('myrmecophiles') which live either as commensals or parasites. For example, larvae of the large blue butterfly (*Maculinea arion*) live as predators in the nests of *Myrmica sabuleti*. Some ant species are true social parasites.

Social bees Apart from the honey bee, there are about 18 species of bumble bees and six species of cuckoo bumble bees. They may have a number of races, either within the British range or between Britain and the continent.

The social bees have an annual life cycle. The queen bumble bee hibernates (eg in dead wood, leaf litter or underground) and forms a new colony in the spring consisting of workers. In summer, males and females are produced which mate. Only the newly fertilized females (queens) will live through to the next year. The workers are much smaller than the queens.

Cuckoo bumble bees have queens which appear after hibernation in early summer, when their host bumble bee nests have already built-up strong colonies of workers. The cuckoo sometimes kills the rightful queen, and the workers then rear the brood of the cuckoo which produces only males and queens.

Nests are either underground or in litter, especially where abandoned rodent nests provide a ball of fine grasses which can be used to surround the bees' nests. There are so many parasites and other hazards that it is surprising bumble bees survive; for instance, the adult bumble bee may be attacked by conopid flies whose larvae live inside the bee. The nests often contain a good variety of harmless scavenging and commensal insects, including larvae of beetles and flies.

Individual species generally visit a fairly select range of flowers, tongue length being one constraint. All species require a suitable range of flowers throughout the season, though some species have marked plant preferences, such as *Bombus monticola* for bilberry *(Vaccinium myrtillus)*.

Social wasps The life cycle is very similar to that outlined for bumble bees, even so far as one species being a cuckoo.

The nests of the various species may be underground, hung from the branch of a bush, in a hollow tree, or in man-made structures. It would seem there are fewer parasites, but the range of nest scavengers is large, those of the hornet being especially interesting.

Certain flowers are well known as being specifically wasp-attractive for pollination, as with figwort *(Scrophularia)*, but social wasps also play a minor role in pollinating other non-specific flowers, eg *Cotoneaster*.

The hornet is of particular interest since it became markedly scarce over a period of some 40 years until the late 1980s. In the following decade, however, a noticeable increase in numbers occurred. It is difficult to specify the reason for the original decline, although the loss of old, large trees with their ideal nest holes may have been a contributory factor. The subsequent resurgence could be due to changes in general weather patterns in Britain, with longer, colder springs and longer, warmer autumns.

Two mainland European species of *Dolichovespula*, *D. media* and *D. saxonica*, became established in the south of England in the 1980s, and quickly dispersed to the west and north of England. Why did they not travel the few miles across the English Channel many years ago? Again, perhaps changing weather patterns were responsible.

HABITAT REQUIREMENTS

Broad criteria

- The majority of species require sites with a warm, sunny aspect. A sunny south-facing bank often provides a suitable site, especially where sheltered and subject to high temperatures.

- Sandy soils are especially favourable (eg heaths and dunes) but dry clay soils may also be suitable, especially in coastal districts. Some species favour chalk and limestone. Only a few specialists are adapted to fens or open woodland.

- There must be suitable conditions for nesting, as well as for gathering food and prey.

Underground nesting sites

- Suitable ground nesting sites are essential. Though often nesting in isolation, many species of solitary wasps and bees form dense aggregations in very restricted areas, and these colonies may use the same site for many years. Some species require a vegetation-free vertical face, a sloping bank or a path; others simply require an open structured vegetation allowing easy access to the soil. The continuity of the nesting areas is crucial as it cannot be assumed the insects will be able to adopt an alternative 'suitable' site. Sympathetic management in preventing these areas becoming overgrown may be required, and nesting areas could be deliberately created. Road cuttings, quarries, dune systems and pits in soft sandy rocks often provide suitable nesting localities. Naturally occurring exposed patches of ground and banks can often be found on the coast along cliffs, especially landslip areas.

- Because of the preference for bare ground, thought should be given before embarking upon a programme of restoring trampled areas and erosion points. It may be that a certain amount of trampling can be used as a management tool in such areas as heathland. Checks at about one month intervals between late March and September should reveal the presence of burrows or nests, and on a sunny day the insects, if the site is occupied.

- In woodland or other areas where trees are present, gale blow may fell a tree so that a vertical bank of soil is held between the upturned roots. If this bank has a southerly aspect in a sunny situation, it may provide an important nesting site in an area where opportunities are otherwise few.

Surface nesting sites

- Woodland rides, glades and margins are especially useful habitats, but this group largely avoids the inside of a wood. Hedgerows can sometimes be suitable if not over-managed.

- For those solitary species nesting in bramble, this plant must be retained on the sunny aspect of hedges and similar situations. The presence of broken ends of old dead stems is required; thus, light trimming can be beneficial but should not involve drastic removal of existing broken ends with occupied burrows. Some species will readily adopt lengths of stem about 30 cm long placed on short turf near woodland edges – but it is essential not to move the stems whilst the wasps are active as they learn the exact position of their nest burrow.

- Dead trees with beetle burrows, old fence posts, gate posts and any other wooden structure with small cylindrical holes may provide useful nest sites. Even half-burnt birch *(Betula)* trunks on a heath may be occupied, and thought should be given before clearing. Exposed sunny situations are favourable for these insects (but not necessarily for other dead wood fauna).

- Other nesting sites include empty snail shells (some *Osmia* and a *Hoplitis*), old cigar galls of *Lipara lucens* on reed *(Phragmites)* stems (*Hylaeus pectoralis*), hollow trees (*Vespa crabro*), rotting twigs or galls (some *Leptothorax* species), hollow dry dock (*Rumex* spp.) stems and cut ends of dead pithy stems such as elder *(Alnus glutinosa)*.

Requirements away from nesting sites

- All nectar-feeding species are dependent to varying degrees on flowers and often on particular types. Apiaceae (especially *Angelica*, hogweed *(Heracleum)* and parsnip *(Pastinaca)*) and Asteraceae are especially good, whilst sallow (*Salix*) is favoured in the spring. One needs to manage vegetation so that a succession of suitable flowers is available through the season, especially during the main flight of the species present in a locality.

- It is important to recognise that the flowers of casual plants, including so-called 'weeds', can be invaluable, as on the fringe areas of heathland and in artificial sites such as old pits.

- Aculeates often have very different habitat requirements for nesting and for gathering food. For instance, a bee may nest in burrows in a bare sandy path which in terms of management considerations has a very different perspective to the equal requirement of certain types of flowers in adjacent, or even distant, untrampled herbage. The ecological girder effect becomes more complex with a wasp which may nest in vacant beetle burrows in a fence post and catch leaf hoppers as prey from birch bushes, such bushes perhaps being regarded as a nuisance value because they invade a vegetation community demanding quite different considerations. One might envisage a situation where the wasp became extinct because the birch became of the wrong age class to support its leaf hopper prey. Individual ant colonies can also become highly dependent on aphid honeydew from quite specific types of vegetation.

Urban areas and buildings

- Many species can thrive in the open sunny conditions provided by urban areas. Nesting sites may be present in trampled areas in parks, old fences, walls and tree trunks, neglected corners with bramble, or the cut ends of garden shrubs such as buddleia, whilst waste ground weeds and horticultural plants may provide an abundance of flowers.

- Old walls with a sunny aspect can provide good nesting sites if the cement is soft enough for burrowing. It is suggested that a survey be made of old walls, such as those in the grounds of old estates, as there may be relict populations of species which have lost their natural nesting sites in various districts. Obviously pointing of the cement work could obliterate the discrete nesting areas and for this reason there is an early need to locate any key section of wall. Even relatively recent walls can be suitable given the right condition of cement work.

FACTORS AGAINST SURVIVAL

The factors against the survival of aculeates are many but in part may be deduced from the habitat requirements given above. Among the chief adverse factors are:

- the use of toxic sprays on flowers;

- prevention of herbage flowering, eg by mowing during the flowering season, over-grazing and the use of herbicides;
- the removal of flowering shrubs or cutting in a fashion which prevents flowering.
- removal or too frequent cutting of bramble which provides nesting sites for aculeates (especially where there is a sunny aspect);
- the loss of bare banks and tracks used as nest sites ;
- over-intensive use of sandy tracks by horses which churn up breeding sites, and over-grazing of certain habitats by livestock;
- the removal of dead wood, wooden posts and fence palings, etc, used as nesting sites. Old walls are occasionally also important;
- drastic disturbances of breeding sites in winter when it is easiest to overlook the implications of management on aculeates;
- general loss of natural and semi-natural habitat (such as heaths) through development or mismanagement;
- the decline of traditional land management practices, such as light grazing which keep sites open and suitable for aculeates.

CONSERVATION AREAS

The aculeates are essentially a warmth-loving group, with many British species at the edge of their range. Thus, areas with high insolation such as heathland, coastal dunes and sandy cliffs are especially favoured. The greatest diversity and numerical abundance are in the south of Britain and, more especially, the south coast and neighbouring heathland areas. There is a rapid decline in diversity towards the north Midlands but this is compensated by the occurrence of a number of northern species moving southwards from Scotland. There are also extremely good examples of eastern and western distributions.

Small areas within a large site may be of particular local importance: whilst a species may be abundant at a nesting site a few square metres in area, it cannot be emphasised too strongly that many of the species are rare in the countryside as a whole. Their survival in these low numbers would seem to centre around very fragile nuclei. Therefore, it is well worth identifying the occurrence of the scarcer species against the inevitable background of common species.

This article is a revised version of the booklet entitled *The conservation of bees and wasps*, published in 1979 by the (then) Nature Conservancy Council. Reproduced by kind permission of English Nature (1995). Additional material by John Burn, Robin Edwards and Simon Hoy (1995).

SECTION 2

COLLECTING AND PRESERVING ACULEATE HYMENOPTERA

A. COLLECTING ACULEATES

(excluding bethylids, dryinids and embolemids)

George R. Else

Introduction

The accurate identification of specimens is clearly of paramount importance to the success of any recording scheme. It is anticipated that some participants in BWARS will already have acquired sufficient expertise to determine with confidence most of their own material. However, it is also recognised that many others will be novices: their lack of experience, however, should not be a handicap to their involvement in the Society. Indeed, we hope to encourage beginners in a variety of ways, and this section of the Starter Pack is primarily intended to help newcomers.

It must be stressed that this is a personal account of techniques which have come to be accepted, after many years' experience, as being perfectly adequate. Nevertheless, new collecting and preservation methods are constantly being devised and old ones refined. The Society's Newsletter is the perfect forum for presenting such innovations, and it is hoped that other hymenopterists will describe their own preferences in future issues.

The Aculeata comprise only a small fraction of the total number of species within the order Hymenoptera. They include both flying and wingless forms. Many will be encountered visiting flowers for nectar or pollen, whereas others may be attracted to holes in woodwork, or to exposed soil in their search for suitable nest sites. The various techniques employed to collect these insects are almost as diverse as the life styles of the creatures themselves. Only a few of the more basic collecting devices are described here; more specialised ones, including such items as malaise, pitfall and yellow pan traps are detailed by Jeremy Field later in this Section, and in Gauld and Bolton (1988), and Noyes (1982) (see Bibliography).

Nets and their use

For winged species the most important prerequisite is sunshine; very few species are active on cool, overcast days. As a result, aculeate collectors are more dependent on weather forecasts than most other entomologists. The most essential item of equipment for the collector is a kite net. The net bag can be of cotton (preferred) or nylon, and with a mesh gauge as fine as possible – many species are so minute that they will simply pass straight through coarser ones. These bags are generally black or white, according to individual preference. Usually the net frame can be attached to a long handle, a great advantage when collecting from high tree blossom (sallow catkins for instance), which would otherwise be out of reach. It is good insurance to pack a spare net bag in case one is ripped by thorns. In an emergency, temporary repairs can be made using strips of 'Micropore' surgical tape.

Sweeping mixed herbage with a kite net can be most productive, although the net hem (around the frame) will soon suffer. Unfortunately, the use of a tougher, non-transparent sweep net (so useful for some other insect orders) is not advisable for such active insects, as these will quickly escape as soon as you peer into the net.

Netted specimens can be transferred direct to a killing jar, but there are some exceptions. For example, mated pairs are best kept separate from the general catch, so they do not lose their significance. This, of course, is rarely necessary in the British Isles, but is sometimes of vital importance when collecting overseas (where the correct association of the sexes of a particular species may be unknown). For the same reason, wasps with their prey should also be isolated from the main catch.

Collecting bees

Pollen-laden bees must always be kept apart from the catch; otherwise they are apt to deposit copious amounts of pollen on to other specimens, frequently staining the latter's wings. It is often useful to retain some female bees complete with such loads for later microscopic examination of the grains, in an attempt to identify the pollen sources of the bees in question. If this is not necessary, then the female should be imprisoned with a slip of tissue in a glass tube (one about 7.5 by 2.5 cm will do for most species) with a plastic stopper. Many aculeates can gnaw into a cork stopper: bees, for instance, covering their hairs with a fine, unsightly brown dust. Do not use too much tissue: the bee must be able to crawl around within the tube. Keep the latter in cool and dark conditions (the inside of a shaded collecting bag is ideal) for about an hour – more if the specimen can stand it. Most females will shed (mainly by grooming) their pollen, although those loaded with vast quantities (some large Andrenas and *Dasypoda altercator*) may have to be left for a longer period and the tissue replaced once or twice. Some females are very loathe to part with their pollen (this is particularly true of megachilids).

Some female bees will forage for pollen from a very wide spectrum of unrelated plants, sometimes flying to almost any flower which happens to be within the vicinity of their nest. These bees are known as polylectic species. Others, however, are more specialised, visiting only a small number of closely related species (oligolectic) or even a single species (monolectic). Males are normally not so particular, as they do not collect pollen. Females of oligolectic and monolectic species will frequently visit other, non-related plant groups, for nectar. As it is not always possible to observe whether a bee is collecting pollen, or just imbibing nectar, the complete range of pollen sources of most of our species remains unknown. This is a field where amateur involvement could make a great contribution to our knowledge of this subject. Identification of these sources from pollens removed from provisioned cells holds the greatest promise of providing such information.

Collecting ants

Ants present different problems from most other aculeates in terms of collecting, and so an alternative approach is required. The collecting bag should include a pooter, a set of glass tubes containing 80–90% alcohol, a sheath knife (ie a knife with a fixed blade) and a pair of forceps. Black polythene bags, a white sheet and a hand trowel are also necessary if litter samples are to be collected. A hand lens, preferably with a magnification of ×20, is required to identify species in the field.

Many ants are found abundantly in dry, sunny locations on light soils, and such places can be very rewarding, but a full survey requires a search of all niches in the locality as many species have very specific habitat preferences. *Formica candica* (*F. transkaucasica*), for example, is unusual in occurring only in lowland bogs in southern England and a site in south Wales. A few species are even found in deep woodland shade (for example, *Stenamma* species) and so often go unnoticed; searching such darkened areas is often more successful in early spring before the trees are in full leaf. A cursory survey of a site will usually reveal the dominant species, as their workers go about their business in the open. Several ants like to nest under stones (particularly *Myrmica* species) and many others under or in rotten logs. Species with small colonies are often attracted to vacated plant galls, or secrete their nests beneath bark (*Leptothorax acervorum*, for instance). Likely pieces of loose bark can be prised away from the underlying wood by using the knife, as can flakes of rock on exposed cliff sites. Clumps of earth and grassy tussocks should also be investigated, but try to keep habitat disturbance to a minimum by replacing sods, stones and wood afterwards. Some species are arboreal in their foraging habits (*Formica rufa*, *Lasius fuliginosus* and *L. brunneus*) and are usually encountered as they pass up and down tree trunks or as they wander amongst the foliage. During the warmer months of the year nests will often contain both sexes of the winged, reproductive forms prior to their mating flights. Nests may also contain 'guest' ants, ie those which are social parasites or 'slaves': many of these parasites are rarely seen and are worth looking for.

Ants are best collected by one of the following methods. Small specimens can be picked up with fine forceps or an alcohol-laden brush or, with large species, the fingers. Small ants stick quite well to a licked finger tip. A pooter can sometimes be used to aspirate non-formicines, ie ants which sting rather than spray formic acid (inhaling formic acid vapour is not recommended!). The last technique is useful when collecting off vertical surfaces or for collecting small colonies to set up in formicaria for studying at home (you will need to find a queen to keep it viable and the colony should be returned once finished with).

Specimens can be quickly killed by placing them in tubes of alcohol for carriage home. Do not forget to add the locality and date to tubes (use a pencil or waterproof ink). The more secretive, subterranean species are best obtained by collecting leaf litter from promising areas and taking this back to base in sealed black, polythene bags. The contents of the bags are then emptied into Winkler Bags or Berlese Funnels. A light is suspended above the Berlese Funnel, which has the effect of drying out the leaf litter from the surface downwards. The ants respond by retreating downwards into the moister lower layers, eventually falling into a tube of alcohol at the base of the bag via a funnel. The Winkler Bag works in the same manner but without a power source. These methods are slow and laborious; an alternative is to spread the leaf litter or nest material over a white sheet or tray, and then sort it by hand.

Where to look for nests and immature stages of bees, wasps and ants

The flight season for bees and wasps extends from March to October. During this period nests can be discovered in the soil (some species nesting in densely crowded aggregations, which often attract attention), in suitable burrows in timber, in hollow stems and trunks, in vacated galls, empty snail shells and in other, more unusual situations.

During the summer, nests cannot usually be safely opened for fear of disturbing or damaging the immature stages developing within. However, nests constructed deep in the soil are most easily located whilst being provisioned, but they are often very difficult to excavate satisfactorily, owing to the problem of keeping in contact with the main burrow and locating the lateral burrows (these are sometimes filled in with spoil once the cells have been provisioned and sealed). It is, therefore, not surprising that the nest architecture and immature stages of many of our fossorial aculeates are so poorly known. This is yet another example of a project to which the amateur could have much to contribute.

Winter is the best time to seek aerial nests: there is nothing quite like wading around in the freezing waters of a reed bed in search of the old, vacated, weathered, cigar-shaped galls of the chloropid fly *Lipara lucens*. It is within these galls (not last season's still-green galls) that the little colletid bee *Hylaeus pectoralis* nests. This species was formerly considered to be an East Anglian fenland speciality, but in recent years it has been collected from Essex, Surrey and from west Sussex to west Dorset.

'Brambling' expeditions are also a worthwhile winter undertaking. The stems must be dead and broken so exposing the pith. Stems which are possibly occupied are recognised by a round, terminal hole in the pith. They must also be in a sunlit situation. Some species excavate the pith (*Hoplitis claviventris* and *Ceratina cyanea*, for example), while others occupy existing borings (an example being *Trypoxylon attenuatum*). Stems are best gathered from the New Year onwards, as it is necessary for them to be subjected to very low temperatures, thus breaking the winter diapause; those collected at the end of the summer and kept in a warm room will rarely produce anything.

Much can be learned from dissecting these stems. Many will be empty, but others will contain rows of sequentially arranged cells, mostly occupied during the winter by prepupae. For convenience the contents from these cells can be removed and transferred to numbered gelatine capsules. These capsules come in various sizes and are available from Davcaps (see Section 6 for address). If you collect and open stems in January, it is possible to rear the contents by late February, providing the contents are placed in a warm situation: an airing cupboard for instance, **not** on a window sill. By numbering the capsules it is possible to determine the sequence of sexes within the stem; male cells tend to be located nearest to the nest entrance. Stems with very small entrance burrows (pinhole size) invariably contain the nests of *Spilomena* species (usually *S. enslini*). Being so small these sphecid wasps are rarely encountered in the field, but they can be reared in quantity from stems (females unusually outnumbering the males). Stem-rearing is also an excellent method of obtaining cleptoparasites and parasitoids, with of course the added satisfaction of confirming the correct host association.

If you clip off one stem-nest, then a new nest site is created. By removing and laying a length of bramble stem on open, short turf, then three such nesting sites are created: one at either end of the loose section, the other at the end of the attached one. Habitually visiting a productive locality, doing the same walk around it each winter when suspected nests are collected and attractive stems cut, should provide a greater chance of finding further occupied nests in future seasons. All you need is a pair of secateurs, stout gloves and some self-sealing polythene bags. Don't forget to slip a piece of paper with the locality and date into each bag. You may be lucky enough to find hibernacula (burrows without partitions) of the little blue carpenter bee, *Ceratina cyanea*. This species is locally distributed in south-eastern England. During the winter, a stem may contain a series of both sexes of adults of this species. These will emerge in the spring, the females seeking out fresh stems as nest sites. Danks (1971) provides a very useful summary of the biology of British aculeates which nest in bramble stems. The work also contains a key to these nests and is essential reading for anyone taking up this interest.

Dead wood will also contain nests, but finding the latter and removing the contents of the cells can cause great damage to an important biotope. Demolishing a tree or removing an attractive fence post is not going to make you popular. Nevertheless, a few nests can be collected with little effort. An example is that of *Passaloecus eremita*. This small sphecid wasp was only added to the British list in 1982, from adults and nests found in Kent, east and west Sussex. Since then it has also been recorded from several other counties in southern England. British nests of this wasp have mostly been found in the exit holes of small beetles in pine bark on standing trees and in wooden fence posts. Completed nests are sealed with white pine resin, with a very characteristic, outer complete or interrupted annulus of resin (the incomplete one being reduced to a series of resin droplets). As far as is known no other British *Passaloecus* produces such a ring. These nest entrances stand out on a burnt tree bole, less obviously in a fence post. Normally several nests of this species will be found together. It has been discovered that most nests of this wasp occurring in tree bark are unicellular, indicating that only quite small sections of bark need to be cut away from the trunk, rather than substantial sheets. This causes a minimum of damage, preserving the remaining surface as a nest site for subsequent seasons. Chunks of bark containing nests can be placed in sealed polythene bags to await emergence of the bees or wasps.

Many aculeates will readily accept trap nests. These are usually blocks of wood which have been bored out to various diameters to attract species of different sizes. The blocks are suspended horizontally and protected from rain, in some aerial, sun-lit position. They are retrieved at the end of the flight season, opened and the contents transferred to gelatine capsules. The manufacture of such traps is described by Krombein (1967) and Gauld and Bolton (1988). Bundles of dead, dry bramble or elder stems, and even drinking straws can be similarly used (see the article by Chris O'Toole in Section 5).

Nests of ants (according to species) can be sought for beneath large stones, in burrows in the soil, in rotten wood, and in mounds of leaf litter (wood ants). See also notes in 'Collecting ants', above. In addition, some ants may be found associated with buildings: the most common is the black garden ant, *Lasius niger*, well known for its large colonies under paving slabs, etc. However, some nests of this species may be found **under** house foundations, or even behind skirting boards, while others may be in cavity walls filled with insulating foam; all these sites are a fairly recent (1980s) departure from normal nesting behaviour. *Hypoponera punctatissima*, a rare species outdoors in Britain, is sometimes found in commercial premises, where their nests may be behind broken tiles on walls, or in cracks round drain gratings. Pharaoh's ants, *Monomorium pharaonis*, can be serious pests in hospitals, where huge colonies occur. They feed on food in patients' lockers and in canteens, and they have even been found in open wounds during operations!

Killing agents and killing jars

The most suitable killing agent for aculeates is undoubtedly ethyl acetate. It has great advantages over a number of alternatives. For example, it is not as dangerous as cyanide, does not affect the colours of integumental pigmentation on prolonged exposure (cyanide often turns yellows red), and leaves freshly killed specimens fully relaxed, invariably with the mouthparts fully extended. It does have one major disadvantage: it is highly inflammable. Insects are not killed instantaneously, although a newly charged killing jar will stupefy insects within a few minutes. At least one hour will be required to ensure that small and medium-sized aculeates are dead, while specimens the size of a large bumble bee will need to be kept in a sealed jar for several hours (preferably overnight), before they can safely be assumed to be dead. It is very disheartening to pin a specimen and on returning some hours later discover the unfortunate insect treading air.

The simplest and cheapest killing jar consists of an empty, screw-top jam jar. To this add two or three small balls of tissue which have been immersed in ethyl acetate. A wad of tissue, occupying about a quarter of the height of the jar, is next added to form a basal layer. After a few minutes enough of the reagent should have evaporated to enable the jar to be ready for use.

There are some important do's and dont's when using such a killing jar. First, ethyl acetate is a liquid, so do ensure that specimens (particularly densely hairy ones) do not come into contact with it, as this will cause the hairs to mat. Because the reagent evaporates quite quickly and is thus lost every time the jar is opened, it will require recharging periodically. The commonest mistake when charging an ethyl acetate killing bottle is to use too much of the liquid. The very high concentration of the damp vapour and the wet tissue will also cause the hairs of most species to mat and the abdominal bands of white tomentum of female *Colletes* to assume a dark, oily appearance, a condition which alas seems to be irreversible.

The easiest way to recharge a jar is to take another small piece of rolled tissue paper (about the size of a large grapefruit pip), wet it with ethyl acetate and, by using a long pair of pointed forceps, introduce the moistened tissue between the glass of the jar and its wad of tissue paper. Do not leave specimens in the jar for too long, as they soon dry out. Do not overcrowd a jar. Do not mix different insect orders together in the same jar (beetles for instance will take much longer to die and commonly defecate in the jar, or emit copious quantities of an obnoxious brown liquid from the mouth), and **never** add Lepidoptera to an aculeate killing jar, as the Hymenoptera will soon become covered in scales. Sawflies and other predacious insects may tear aculeates apart in the jar. Bumble bees frequently regurgitate honey while in their death throes, and this is commonly smeared on to other insects in the same jar. For this reason, it is better to use a separate, larger container for these bees.

B. COLLECTING AND SAMPLING WITH TRAPS

Jeremy Field

Trapping and net collecting are complementary methods of recording the aculeates present at a site. Some species are more likely to be caught with a net than a trap, and *vice versa*; also, species which are rarely seen by the net-collector can be extremely common in trap catches. Thus, a combination of net collecting and trapping will produce the most complete species list.

Before starting a trapping scheme, it is important to remember that **large numbers of insects of all kinds, common and rare**, are likely to be killed. Thus, we urge members to consider fully their purposes for any such trapping. Collection of large numbers of any species of insect solely to add to a collection or to 'see what's there' cannot always be justified. It is appreciated, however, that many aculeates are too small to identify, or just too difficult to identify, without killing them. Therefore, some trapping may inevitably be required. In these cases, a malaise trap or pitfall trap is passive (ie they do not attract insects) and will sample small areas without seriously depleting the local fauna. If in doubt, please consult one of the Newsletter editors or one of the other experienced BWARS members. In addition, **always** obtain permission from the land-owner before erecting traps on private property.

Two important advantages of traps are that they can be left in position and checked only intermittently, and they provide quantitative information on seasonal variations in abundance and species composition. A collector might feel that a certain species is much more common in June than August. A trap left in one place throughout the summer might allow the more precise statement to be made that it is '2.5 times more common in June than August (in that particular year)'.

Malaise traps and water traps ('yellow-pan traps') are probably most successful for sampling aculeates, although other traps can be better for certain species. Small pitfall traps, for example, catch relatively few aculeates but produce more wingless mutillid females (eg *Smicromyrme*) than malaise and water traps. Trap-nesting, in which aculeates are induced to utilize artificial nest sites, is described in Section 5.

Malaise traps

Malaise traps are large, tent-like structures of fine-mesh terylene netting held up with wooden posts and guy ropes (Figure 1). They can be obtained from suppliers (see Section 6 for addresses) or put together from raw materials. Many different designs are possible. The 'roof' of the tent generally slopes up at one end to the

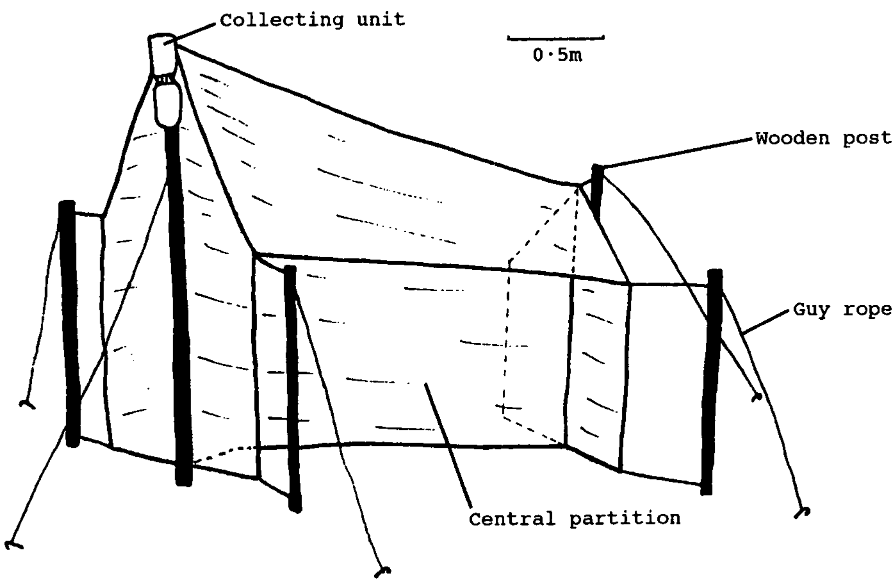


Figure 1. Sketch of a malaise trap

top corner where there is a hole leading to a collection unit which can be partly filled with 70% alcohol. Insects which enter the tent move up towards the light and enter the collection unit where they are preserved in perfect condition. The trap can be erected in about 30 minutes with practice and left for up to two weeks before the collecting unit needs changing (though it may dry out more quickly in very hot weather). Insects can then be sorted in petri dishes, dried for a few minutes on tissues under a lamp, then pinned.

At a sandy heathland site in southern England a malaise trap might catch an average of about 100 aculeates in a ten-day period during the summer. The size of the catch will greatly depend on the siting of the trap. Ideally it should be placed across an aculeate flight line (eg across a path or at the edge of a wood) with the collecting unit towards the light, though traps placed in the open can be surprisingly successful; some trial and error is necessary. A major problem is that traps may blow over in high winds, especially in the exposed situations often favoured by aculeates. Secure moorings and a sheltered siting will thus reduce the likelihood of collapse. Aculeates form a very small proportion of the total catch and sorting them out is very time-consuming.

Water traps

These are simply plastic containers partly filled with water and left in the field. Bowls or pans 15–30 cm in diameter are most often used (eg washing-up bowls, plastic plant pots): white and yellow bowls are probably the most attractive to aculeates. A drop or two of detergent breaks the surface film, greatly increasing the size of the catch, and a tablespoon of salt prevents excessive swelling of insects which cannot be immediately removed. The catch can be separated by pouring the contents of the trap through a piece of netting or a tea strainer. The insects are then placed in tubes of 70% alcohol for sorting later.

Again, the trap site is critical. A white washing-up bowl placed on open sand might capture an average of 30 aculeates in ten days, but in taller vegetation a trap placed on the ground is ineffective. Instead, it should be raised on a pole to a level just above the top of the vegetation. The bottom of the bowl bears a screw-fitting or rests in a metal dish nailed to the top of the pole. A water trap catches fewer aculeates than a malaise trap but is much easier to transport and set up, though large numbers of traps require a lot of water. Also, aculeates form a much larger proportion of the total catch, reducing sorting time. The major problems are evaporation in hot weather and overflowing in heavy rain. Left for too long in hot weather, some of the insects will start to decompose. All aculeates can usually still be identified after a week but not all will be in perfect condition.

Trapping success

This will be greatly affected by weather during the trapping period and by more subtle factors like the degree of exposure of the trap. A malaise trap and a water trap placed next to each other, however, will catch very different species. Deciding on which trap to use will depend on the group(s) of primary interest to the collector. In general, malaise traps are most successful for sampling free-flying wasps (eg most sphecids and vespids), which usually make up over half of the catch. Water traps are more successful for low-flying or ambulatory species (eg pompilids) and bees, particularly social bees. The general collector should ideally use both, together with net collecting and trap nesting.

There is a lot of scope for experimentation. Little is known, for example, of the effect of the colour of water traps on the catch of aculeates (see Kirk (1984) for effects of colour on catches of thrips and dipterans). Disney *et al.* (1982) show that malaise, water and pitfall traps each capture very different dipterans. As traps require little attention once set up, such experiments can be carried out by the amateur who is free only at weekends. See also Southwood (1978).

C. PRESERVING ACULEATES

George Else

Papering and relaxing

Whenever possible freshly killed material should be mounted as soon as possible. However, this is not always convenient, particularly on collecting expeditions overseas. In such circumstances, specimens can be papered and put to one side for later attention. Papering involves the use of triangular or rectangular packets. The best sort are those obtainable from philatelist dealers. These are rectangular envelopes, manufactured from thin translucent paper and having adhesive flaps. A recommended size is 10.5 cm by 9.0 cm. Specimens are removed from the killing jar and placed on a flat surface for initial sorting. A sheet of absorbent tissue is then cut to a length just a little shorter than the envelope, and folded over, so that it is now almost as wide as the packet. It is then opened and the specimens carefully spread over one side of the tissue. The opposite side is then folded over the layer of specimens and the tissue slipped into the packet. It is a good idea to seal the latter, so helping to prevent the entry of pests.

Remember to write the locality and date on the envelope before the specimens are placed inside. Never use an ink that will run when the packet is introduced to a relaxing box. Full packets are best stored in boxes (containing a pest deterrent such as paradichlorbenzene – **avoid excessive inhalation of the vapour**) and placed in self-seal polythene bags until required. On collecting expeditions in the tropics it is also important to add thymol, and perhaps silica gel to boxes containing papered material, in order to protect the contents from mould and moisture which are ever-present threats in places of high humidity. The tissue tends to grip the specimens, so preventing them from being shaken around and possibly being damaged.

Relaxing. Dry, papered material obviously needs to be relaxed prior to mounting. Also, it is often necessary to dissect a long-dead, pinned specimen, and this too needs to be first relaxed. A relaxing box consists of an air-tight, clear plastic box: a sandwich box is ideal. Add a few crystals of thymol (to protect against mould), and above this place a close-fitting layer of either tissue or cellulose wadding (not cotton wool, which becomes tangled with insect claws). The tissue is slightly dampened, **not drenched**, with water. The pinned material and the packets containing dry specimens are finally added. These can be raised a little above the damp tissue by placing them on a sheet of plastazote, thus preventing them from taking up excess water.

The time taken to relax a specimen varies according to its size and age (one about 100 years old will always cause problems). Generally, small to medium-sized aculeates which have been collected quite recently will take about 24 hours. When the envelopes containing the relaxed specimens have been taken out of the box and cut open, check to see if the material is sufficiently pliable for mounting. If in doubt replace in the relaxer and leave for a further 24 hours. Great care is required when dealing with some groups. For example, if the white abdominal hair bands of some bees become too damp, they become black, and are irreversibly destroyed, thereby ruining the appearance of the specimens.

Pins and pinning

All aculeates should be pinned, except dryinids, bethylids, ants and very small wasps, which should be mounted on card points. The size of specimen determines the choice of pin used. Thus, a wasp or bee about 2.0–5.0 mm in body length should be pinned with a stainless steel micropin, whereas larger insects should always be mounted on a Continental pin of size 0 to 5. On no account should a 00 pin be employed as this size is so fine that it easily bends and cannot be fully straightened out again. It is also more likely to 'twang', with disastrous results for the specimen. The larger the insect, the stouter the pin. For example, a small halictid bee such as *Lasioglossum villosulum* will require a 0 pin; on the other hand, bumble bees (depending on size and caste) will require a pin size between 2 and 4.

Continental pins, irrespective of size (ie diameter), are all 38 mm long, usually have nylon heads, and are manufactured from stainless steel. They are produced (in packets of 100) in Austria, but can be purchased in the UK from Watkins and Doncaster (address in Section 6). Shorter, 'English' pins should not be used. These are invariably made of brass (often painted black) and, in time, will usually react with the fat content of the thorax. This slow reaction causes verdigris, easily identified by the green filaments which sprout from the pin as it emerges from both the dorsum and venter of the thorax. This may be preceded by a layer of small green flakes which coat the pin above and below the specimen. Eventually the reaction will destroy the pin, frequently causing it to snap within the thorax.

When pinning, steady the specimen firmly against a sheet of plastazote or other soft, firm material with a pair of fine, pointed forceps, ensuring that both pin and specimen are in the same vertical axis. The pin should go through the thorax (mesonotum), slightly to one side of the mid-line, so as not to destroy the punctation or surface sculpture on the disc (these are often important identification characters). Sufficient room should be left above the specimen so that the pin can be handled safely. A pinning stage for Continental pins is recommended as this will ensure that a series of specimens are all at the same level on their pins (the stage is also used in the same manner for attaching labels to pins. When micro-pinning, carry out the operation under the low power of a binocular microscope.

Slight downward pressure, from a pair of curved pinning forceps, will generally result in the wings flicking upwards. Do not try to force them to do so by pressing too hard on the thorax, as this will cause damage. Legs and other parts of the body can be arranged in position by the use of a pair of fine-pointed forceps. Legs should be kept close to the sides of the body and extended downwards. Thus, with the wings pointing upwards and the legs down, the entire lateral surface of the head and body will be fully visible. This is vital in some species, as important identification characters occur in this region. The mandibles should be opened to display their dentition (and the complete labrum); the tongue and associated palps should be fully extended so that they are entirely visible; the antennae held outwards; and, if the specimen is a male, the genital capsule should be exerted (but not severed from its retaining basal membranes). The capsule is drawn out by carefully inserting a pin between the terminal tergum and sternum, engaging it behind the base and thus easing it out. The abdomen can be supported in position by a pair of cross-pins and the appendages held in place by more pins until dry. It will generally take at least a week before the specimen is dry enough to label and incorporate in the collection.

All this, of course, takes time, but it is not a wasted exercise, as it will enable the specimen to be identified later with the minimum of trouble. Setting aculeates in the manner of Lepidoptera (ie with wings and legs all in the same flat plane) is to be discouraged. The effect may be aesthetically pleasing, but in many instances the specimen will be very difficult to identify subsequently, as lateral body characters may be all but impossible to see, being effectively masked by femora and the slightly drooping wings. An even worse habit is to set a largish aculeate (including queen bumble bees) in the same style, using a stout micropin, and then to stage the insect on a length of *Polyporus*. I have never understood the logic of this technique as, apart from the very real threat of damage, the final effect is often preposterous. The legs and antennae of a specimen mounted in this fashion become extremely vulnerable to damage, as the *Polyporus* stage is likely to rotate on its main pin (this is almost bound to happen if the stage is not cross-pinned, should the insect be sent through the post). Handling such a specimen is also dangerous, particularly when it becomes necessary to study it under a microscope.

A small micro-pinned aculeate should ideally be pinned into one end of a 6 mm length of *Polyporus* or plastazote. The strip is then impaled on a Continental no. 5 pin. The specimen is placed across the strip, its main body axis at right angles to the axis of the stage. Never mount more than one specimen per stage (collections sometimes contain examples of such a multiple arrangement: occasionally the series may be of mixed species!). The prey taken with wasps, however, should be pinned alongside their captors on the stage, or beneath each wasp when these have been mounted on Continental pins. If the prey is preserved in spirit, then it should be carefully cross-referenced with its pinned captor. Pairs taken in copula should also be pinned together in the same fashion.

Often specimens need to be cleaned, as regurgitated honey, pollen grains, or tissue fibres may adhere to individuals in the killing jar. Sometimes a dull deposit of unknown origin coats the more basal tergites of the gaster, interfering with the view of the sclerite's punctation and surface microsculpture. This substance is easily removed by rubbing the affected surfaces with a fine paint brush dipped in xylene. The brush is cleaned by wiping on a piece of paper. The xylene rapidly evaporates, **but avoid inhaling the vapour**. This method also helps in cleaning a specimen covered with pollen grains, dust, etc.

Card-point mounting

This technique is used mainly for ants, dryinids and bethylids, but can also be employed for other small aculeates which may be difficult to pin because of their very small size or the coarseness of the integument – for example, some *Diodontus* and *Spilomena* species, female *Methoca ichneumonides*, *Myrmosa atra* and *Smicromyrme rufipes*. With winged aculeates, avoid gluing the wings down.

The card point is triangular, about 10.0 mm long and 3.5 mm wide at the base. A slightly blunt tip increases adhesion when the specimen is glued to the point. A water-soluble glue should be used (eg 'Seccotine') in case the specimen needs to be removed from the point at a later date. Actual gluing is much easier – and less messy – if done under the low power of a binocular microscope. Tease out the insect's legs and antennae first, apply a **small** drop of glue to the point and place the specimen across the apex, ensuring that the attachment is concentrated between the second and third pair of coxae (base of legs). Ensure that the specimens face the same direction (traditionally to the right). Several pointed ants can be carried on the same pin (one ant per point), provided they are from the same nest. This saves space in the collection and is more economical of pins.

Labelling material

All specimens should carry full, **permanent** data labels. Each of these should include (in the following order from the top of the label): country, county, nearest town, locality name (with distance and direction from the nearest town expressed in kilometres), date of capture and name of collector. A six-figure grid reference and a very brief description of the habitat should be supplied on a second label (**not** on the back of the first). Bionomic information should similarly be given; for example, scientific name of the flower being visited, etc. Labels should not be too large. It is better to use two or three separate labels, rather than one as large as a commemorative postage stamp. Finally, an identification label for the specimen can be added to the pin. Ensure that each specimen is separately labelled: do not provide a single covering label for an entire series. At all costs **avoid** labelling the specimen with only a number, with the full data being cross-referenced in a notebook. Notebooks are frequently lost, resulting in the complete devaluation of the specimens. I have occasionally encountered in such a book the data for an insect other than that which carries the number (even of an insect in a different order!).

Preservation of the collection

Once a specimen has been collected, one might be forgiven for thinking that its long-term future is reasonably assured. Unfortunately this is not the case, unless measures are taken to protect the collection from a host of pests. From the moment a specimen is mounted it will be prey to other insects (and even small vertebrates). If mounted specimens are left to dry, place them in an enclosed box and add a few crystals of paradichlorbenzene (or naphthalene). A collection should be housed in a tight-fitting store box (the extra-deep variety to accommodate the long Continental pins on each side of the box), or in a cabinet drawer. These will also protect the contents against dust and exposure to sunlight.

One of the most common pests in insect collections is the dermestid beetle *Anthrenus verbasci*. Its larva, which is colloquially known as a 'woolly bear', has posterior hair tufts which are erected when it is disturbed. The larvae are often noticed crawling around on indoor house walls, as is the almost spherical adult. The larva's diet includes all kinds of detritus of animal origin. Larvae seem capable of entering the tightest fitting boxes or drawers and having done so wreak havoc amongst the collection, rapidly reducing specimens to heaps of frass and fragments, leaving only the pins and labels. There can be nothing more distressing than finding some rare and treasured specimen ruined in this manner. Attacks are more prevalent from spring to autumn, the winter usually being passed in larval diapause. Several other *Anthrenus* species present a danger, including *A.sarnicus*. Another dermestid which can cause similar damage is *Reesa vespulae*. This species potentially presents a greater threat as it reproduces parthenogenetically, the population rapidly increasing inside a box or drawer. The anobiid beetle *Stegobium paniceum* and the ptinid *Ptinus tectus* are also known to attack dead insects in collections. Certain Hymenoptera nests brought indoors may contain beetles which, given the opportunity, will also invade and destroy collections. Examples are *Ptinus sexpunctatus* and the dermestids *Anthrenus fuscus* and *Attagenus pellio*.

The best protection for a collection against all these pests is the liberal addition of paradichlorbenzene crystals. Unfortunately these evaporate quite quickly, so a fairly regular topping-up exercise is required, generally every two to three months. **Avoid inhaling the fumes**. Paradichlorbenzene has the added advantage that it kills beetle pests as well as deterring their entry into a box. Naphthalene is an alternative: it is not as effective, but it lasts for months, sometimes years, before replacement becomes necessary. In an emergency a dose of ethyl acetate will quickly decontaminate an affected box, as will exposure of the box to the very low temperatures in a freezer.

A damp environment will cause mould to grow in a collection (and will also relax specimens). Dampness also encourages Psocoptera: these small insects have been observed feeding on the pollen loads still adhering to the legs of female bees in collections (but not the specimens themselves, though this may occur). A suitable deterrent for mould is thymol crystals. Silica gel will help control moisture.

D. COLLECTING AND PRESERVING DRYINIDAE, BETHYLIDAE AND EMBOLEMIDAE

John Burn

Introduction

The following account is based on my own experience of collecting and studying these groups; I hope that some of the ideas and innovations described here will be useful.

Firstly, I would strongly advise anyone wanting to learn more about these insects to obtain a copy of the R.E.S. Handbook on the Bethyloidea by Perkins (1976). It must be pointed out, however, that, with the publication of Prof. Masimo Olmi's *Revision of the Dryinidae* (1984), nomenclatural changes have been made.

Collecting

I usually start looking for these insects from the first week in May, and continue until the end of September. My equipment is as follows.

Sweep net. Very few aculeate hymenopterists use one of these. As well as an ordinary net, I carry a sweep net made from net curtain which I find essential for dryinids and bethylids, as well as being generally useful around brambles.

Having made a few sweeps of the vegetation, it is best to make a few more in the air, to drive the insects toward the bottom of the bag, before it is opened. Dryinids and bethylids, besides being rather small, are not over-active, so the problem of them flying away on opening the bag does not arise. The vast majority always walk upwards on the net material until they get near the rim; some bethylids will try to hide among the debris at the bottom.

Garden sieve and bowl. These are useful for separating ground-dwelling insects from grass tussocks, nest debris and the like. I find it best to have a bowl which fits inside the sieve as they can then be carried with one hand.

Trapping. Several trapping methods can produce good numbers of dryinids and bethylids in suitable sites. Malaise traps and water traps were described earlier in this Section. Pitfall traps are basically containers which are filled in the same way as water traps and sunk into the ground. I do not leave my Malaise traps unattended for long periods, as I find it better to collect the insects live (no alcohol or water in the collecting jar) and sort them at the end of a day, only retaining those which interest me. This also allows me to attempt to breed any species of interest.

Glass tubes and stoppers. Tubes of about 5 cm by 1.2 cm are ideal for field use. I put self-adhesive labels on the outsides for recording locality and bionomic data.

Pooter. This can be a better way of removing specimens from the net if you find a lot in one sweeping. One collector found 39 male and 9 female *Aphelopus melaleucus* within seconds of beating a hornbeam *(Carpinus betulus)* tree! (Jervis 1979b).

Killing tubes. Mine are made from 7.6 cm by 1.7 cm glass tubes with cork stoppers (Figure 1). The centre of the cork is hollowed out and filled with cotton wool, which can be soaked in ethyl acetate as required. A paper lining is fitted inside the tube, to absorb any excess moisture, and the bottom is loosely filled with tissue paper.

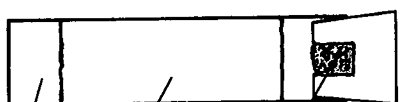


Figure 1. Killing tube

Where to look

Dryinids can be found wherever their hosts (Homoptera, Auchenorrhyncha) are to be found. Sweeping low vegetation such as that at woodland edges, behind coastal sand dunes and around scrub will often produce *Anteon pubicorne, A. gaullie, A. fulviventre* and *Lonchodryinus ruficornis.* Sweeping the foliage of trees will often produce *Anteon infectum* and *A. scapulare* from oak *(Quercus)*; *A. jurineanum* and *A. brachycerum* on birch; *A. arcuatum* and *A. flavicorne* on willow; *Aphelopus* species may be found on any of these trees. None of these species are restricted to these situations, but they are most commonly encountered there.

Other species, such as the wingless females of Gonatopodinae and *Embolemus*, and the almost wingless females of *Lonchodryinus subapterus*, together with both sexes of *Mystrophorus formicaeformis,* are best found by searching short, sparse vegetation and bare patches of soil. Sieving grass tussocks taken from such situations can be profitable, or, better still, pitfall or water traps buried level with the soil can be used.

The more common bethylids can be encountered by sweeping grasses and low mixed vegetation. *Cephalonomia* spp. are best found on fungi on birch and oak where they prey on the larvae of *Cis* (Coleoptera). The way to extract them is to tap the fungi on a bowl or plate so that the bethylids fall out. Fungi can be carried home in polythene bags (those with a sealing strip are best) and the bethylids extracted later.

Rearing dryinids

When a dryinid has successfully attacked its prey (nymph or hopper), a sac containing the developing parasite eventually forms on the thorax or abdomen of the host. This sac is usually dark in colour. Any hopper found with such a sac should be kept in a separate tube and the adult dryinid reared. The following method was suggested to me by Mark Jervis. (See also *The dipterist's handbook*, AES – address in Section 6.)

A rearing container consists of a glass tube about 7.5 cm by 2.5 cm. One third of the tube is filled with plaster of Paris and a small amount of Vermiculite is placed on top. A few drops of water are added for humidity, followed by the hopper and some foodplant. The tube is capped by a perforated plastic stopper. Mark suggests placing it in an outdoor shed, but I usually keep mine indoors for a while, out of direct sunlight, so that I can observe the hopper easily. The foodplant should be changed frequently. When the parasite emerges to pupate in the Vermiculite, I remove the foodplant and the dead hopper, which is then mounted, together with the information needed to associate it with the wasp when it emerges. The tube is then placed in an outdoor shed. Alternatively, acid-washed sand is recommended instead of plaster of Paris and Vermiculite. Chambers (1955) records successfully rearing *Anteon flavicorne* and *A. brachycerum* using damp sand in an 8 oz capped bottle.

Female dryinids can be induced to lay eggs on captive hoppers. On finding a female dryinid, tube it separately and make some further sweeps in the same area, retaining any hoppers found. These can be kept, three to four in a tube, until you are able to introduce the female dryinid to them. This I do as follows:

- invert the tube of hoppers until they walk upwards;
- remove the stopper, place inverted tube on flat surface;
- do the same with the wasp tube;
- when the insects in both tubes are in a suitable position, take a tube in each hand and put the open ends together, keeping the hopper tube higher;
- when the wasp enters the hopper tube replace its stopper;
- now watch until the wasp attacks a hopper. It does this by seizing hold of its prey's legs with its chelate forelegs (*Aphelopus* females lack chela and seize their prey with their fore and middle legs); the hopper will become temporarily paralysed after being stung;

- as soon as the wasp lets go of its victim, retrieve it by inverting the tube, whereupon the hopper will fall into the stopper. This can be removed and the victim transferred to a rearing tube (see above). Replace the stopper and repeat the process to see if further hoppers are attacked.

If the dryinid lays on any hoppers in a tube, the others, which it has refused, should be killed and mounted, together with the relevant data. This negative information helps determine the host range and preferences. If the dryinid refuses all the hoppers then it may be that she was not ready to lay; consequently there is then no need to retain any.

On one occasion I observed a female *Anteon scapulare* attack two late instar hoppers of *Iassus lanio* and apparently lay on both. They were duly placed in separate rearing tubes, but on this occasion only one dryinid larva developed. It would be interesting to know whether females can lay several eggs in rapid succession. Reared females can be used to find out which species are capable of reproducing parthenogenetically.

There is always the possibility that a hopper has already been parasitised; if this was only shortly before you collected them, then there will be no obvious sac. Sometimes hoppers are found with two sacs attached to their bodies; these may be of equal size.

Mounting

I must stress that this is the way I personally mount these groups; it is not necessarily the way you must work. One thing you should do when mounting male dryinids is to expose the genitalia as these have important characters used in separating the species. This is best accomplished shortly after death and involves turning the insect on to its back, under a microscope; holding it steady with a pin across the petiole area; then, taking another pin, pressing lightly down on the sterna in front of the genitalia; sliding the pin toward the end of the abdomen will force the genitalia out. If you do not want to mount the genitalia separately, then leave them exposed at the end of the abdomen.

In addition to the male genitalia of all species, the palpi of both sexes of Gonatopodini should always be mounted: they can be extracted using a hooked pin. I make my slide mounts in the following way:

- make a card rectangle 1.6 cm by 1.0 cm (use Bristol board);
- with an office paper hole-punch make a hole at one end;
- glue a cover slip on to the card, under the hole, so that the hole is in the centre of the cover slip;

- after setting the specimen within the hole on top of the lower cover slip, in whatever mountant you use, place another cover slip above it to close the mount (Figure 2).

Card point mounting is covered under 'Preserving aculeates', above. However, whilst mounting as in Figure 3 may not leave a specimen quite as visible as on a point, it does offer some protection to the specimen. A celluloid mount and gum tragacanth go some way toward overcoming the visibility problem. Most of the characters used in separating the British species can be viewed dorsally. Mounting is best accomplished soon after death, and in the following way:

- use only a thin line of gum where the thorax and abdomen will lie;
- move legs into position, being careful to avoid clogging the teeth on the fifth tarsal segment with gum. Try and keep the chela open by gumming the tip of the enlarged claw. Move the antennae into position using a fine brush;
- avoid using gum to hold down the wings; these may fold as you are setting them, or they may have to be moved sometime. I use a small amount of saliva (or water) which is placed on the mount where the apex of the wings will rest, then move the wings into position, using a fine pin or fine brush. Repeat the process if the wings move when the liquid dries;
- if the specimen should start to revive at this stage due to its not being kept in the killing bottle long enough, place the complete mount in a killing tube, placed horizontally. The tube can be stopped from rolling by being wedged at either side (Blu-Tac is ideal). If using celluloid mounts, the killing fluid must be carbon tetrachloride, as ethyl acetate reacts with the celluloid.

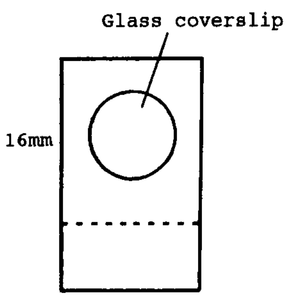


Figure 2. Slide mount for genitalia and palpi

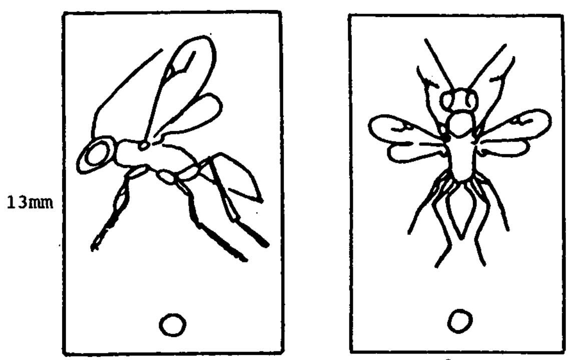


Figure 3. Card mounting complete insects
Note that a width of 6 mm is large enough for smaller species.

Labelling

In addition to the information suggested in 'Labelling material' above, the following dates should be given for reared material:

- when host was attacked;
- when the larva emerged (and its colour – this does vary in dryinids);
- when the larva spun its cocoon;
- when the adult wasp emerged.

Identification

These are small insects and many of the diagnostic features are extremely small, hence a higher-powered microscope is essential. I use a binocular with a magnification range of x25 to x100.

Further reading

The following references to Bethylids, etc, will be found in the Bibliography, Section 7: Chambers, V.H. 1955; Chambers, V.H. 1966; Jervis, M.A. 1977; Jervis, M.A. 1979a; Jervis, M.A. 1979b; Olmi, N. 1984; Perkins, J.F. 1976; Richards, O.W. 1939; Richards, O.W. 1948; Waloff, N. 1974; Waloff, N. 1975.

SECTION 3

IDENTIFICATION OF ACULEATES

Michael E. Archer & Robin Edwards

Introduction

The identification of specimens may be made before or after the preservation methods described above. A few common species can be determined by reference to a popular insect book, but it is much more likely that one or more specialist keys to the group involved will be required. These may be found in books or in papers in scientific journals. It has to said, however, that *positive* identification can be difficult or even impossible for the beginner (and is often quite difficult for the expert!).

Firstly, there are no 'user-friendly' identification keys: in order to obtain *accurate* results, keys have to be very precise and so tend to contain much technical jargon (see example below). Secondly, there is the problem of size: many aculeates are small or very small (some Sphecidae, for instance, are only 3 mm or 4 mm in length), or the features described in keys may be very small. Thus, a good binocular microscope is essential for any serious study (see Section 6 for a supplier).

There is no way round these problems, but the beginner could start by using a ×10 hand lens and a general insect book, such as one of Chinery's *Field guides*, where the illustrations of whole insects should get the reader close to the actual species. From then on, it may be necessary to obtain help from an expert BWARS member (see list in the Newsletters).

Keys to the families and subfamilies of the Aculeata can be found in Gauld and Bolton 1988; Chinery 1986; 1993; Richards 1977; Richards & Davies 1977; Yarrow 1945. Most of these books are now out of print, but a local library or museum may have copies you can refer to. Unfortunately, older keys will not give all species currently found in Britain, and the names of many species will have changed: nevertheless, they are still useful for separating the families and subfamilies. Yarrow's key, although a war-time publication, probably gives the easiest key for a beginner to use (but note that a later edition is not so good).

As an example of a key, we include below one to the social wasps: these insects are large and easily collected (**but do not risk being stung**). As is usual with keys, the text is split into 'couplets', where one has to decide which of a pair of characters best fits the specimen. The name of the species may be given at the end of the description, or there may be the number of the next couplet to be considered. It is always advisable to check **all** the characters described in a couplet, although the best or most important character is generally given first. Basing an identification on just one character may lead to an erroneous determination.

Key to British social wasps by Michael Archer

This key is based on worker specimens which may be collected by net as they leave or enter the nest. Because of the difficulty in separating workers of *Vespula vulgaris* from *V. germanica* I have included a character which relates to the colour of the paper envelope that surrounds the combs of the nest. The envelope will be found when trying to measure the depth of the nest at the end of the season. Except for body length measurements, the key will work for the queens but not for the males. The males have distinctly longer antennae of 13 segments, seven visible gastral terga, and no sting, while the queens and workers have shorter antennae of 12 segments, six visible gastral terga and a sting.

1 Head with an extended vertex; posterior ocelli more than twice as far from the back of the head as from each other (Figure 2A). Body predominantly yellow and brown. Larger, body length 17–24 mm .... *Vespa crabro* Linn. (Hornet)

– Head with short vertex; the posterior ocelli almost as far from each other as from the back of the head (Figure 2B). Body predominantly yellow and black. Smaller, body length 9–15 mm 2

2 Malar space short; as short as or shorter than the maximum diameter of the scape (Figures 1, 3A). Pronotal carina absent or faintly marked vertically (Figure 1) .... (*Vespula*) 3

– Malar space long; considerably longer than the maximum diameter of the scape (Figures 1, 3B, 3C). Pronotal carina well developed (Figure 1) .... (*Dolichovespula*) 5

3 Long hairs on the first gastral tergum black (Figure 1). Occipital carina does not extend to the base of the mandible (Figure 1) .... *Vespula rufa* (Linn.) (Red wasp)

Note: some nests of *V. rufa* may contain queens and males of the cleptoparasite (cuckoo wasp) *Vespula austriaca* (Panzer). This species has no worker caste, and queens and males can be distinguished by the presence of long black hairs on the hind tibiae (Figure 4), which are absent in *Vespula rufa*.

– Long hairs on the first gastral tergum pale (Figure 1). Occipital carina extends to the base of the mandible 4

4 Margin behind the third mandibular tooth straight or at most slightly concave (Figure 5A). Yellow genal band (Figure 1) interrupted by a black bar which may be reduced to a black spot, rarely genal band entirely yellow. Envelope of nest light brown .... *Vespula vulgaris* (Linn.) (Common wasp)

– Margin behind the third mandibular tooth distinctly concave (Figure 5B). Genal band (Figure 1) entirely yellow, rarely interrupted by black markings. Envelope of nest grey .... *Vespula germanica* (Fabricius) (German wasp)

5 Lateral surface of the pronotum covered with ridges (Figure 6). The ocular sinus (Figure 3C) is entirely or mainly yellow .... *Dolichovespula media* (Retzius) (Median wasp)

– Lateral surface of the pronotum not covered with ridges. The ocular sinus is mainly black 6

6 Large punctures more numerous on the clypeus so that generally puncture diameter is equal to or more than inter-puncture distance. Clypeus entirely yellow or with a central black spot (Figure 3B).. .... *Dolichovespula sylvestris* (Scopoli) (Tree wasp)

– Large punctures less numerous on the clypeus so that generally puncture diameter is less than inter-puncture distance. Clypeus with a broad black line usually widened at the centre 7

7 Head long, POL divided by PBHL (Figure 7) equal to or less than one. Hairs on the side of the thorax of light brownish colouration. No orange marks on the first and second gastral terga .....*Dolichovespula saxonica* (Fabricius) (Saxon wasp)

– Head short, POL / PBHL more than one. Hairs on the side of the thorax black or at least with some black hairs. Orange marks often present on the first and second gastral terga .....*Dolichovespula norwegica* (Fabricius) (Norwegian wasp)

Note: two further *Dolichovespula* species, both cleptoparasites (cuckoo wasps), are found on mainland Europe but are not yet recorded from Britain and Ireland. They are *D. adulterina* (du Buysson) (on *D. norwegica* and *D. saxonica*) and *D. omissa* (Bischoff) (on *D. sylvestris*). Females of both species can be separated from other species of *Dolichovespula* by having the anterolateral angles of the clypeus greatly projecting and pointed. In other species of *Dolichovespula* these angles are much less projecting and rounded. The puncture density on the clypeus of *D. omissa* is similar to that of *D. sylvestris*, and on *D. adulterina* similar to that of *D. saxonica* and *D. norwegica*. The clypeus of *D. omissa* is covered by long pale hairs and of *D. adulterina* by long black hairs.

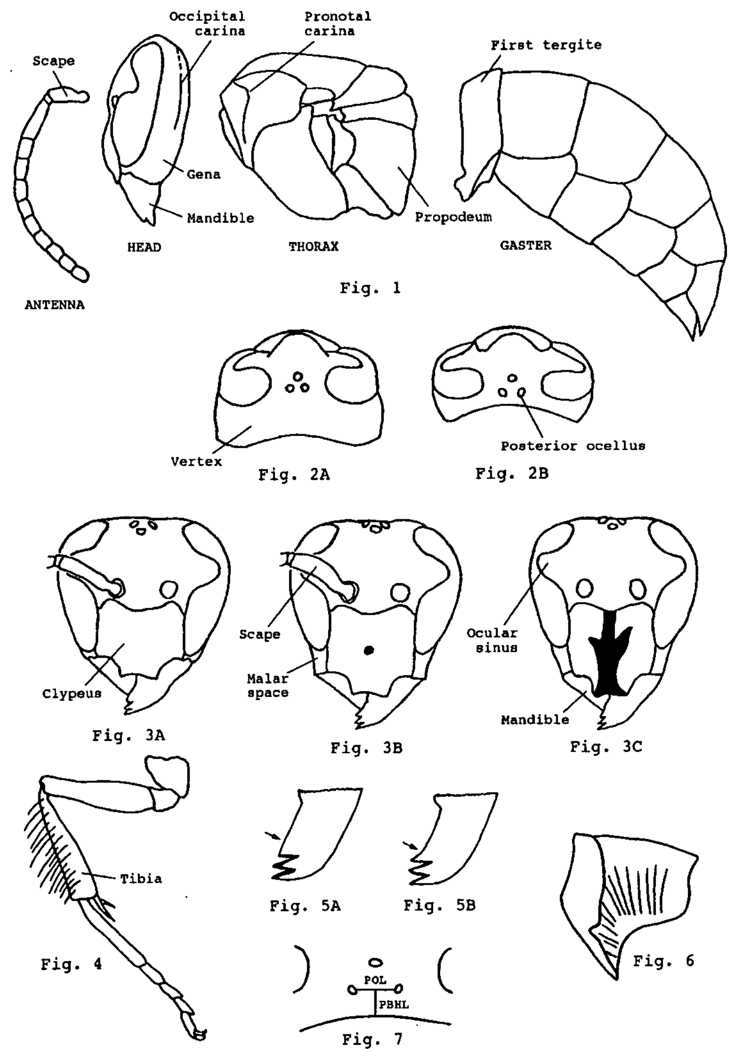


Figure 1. Lateral view of a worker
Figure 2. Dorsal view of head: A) Hornet, *Vespa crabro*; B) Common wasp, *Vespula vulgaris*
Figure 3. Front view of head: A) Common wasp, *Vespula vulgaris*; B) Tree wasp, *Dolichovespula sylvestris*; C) Norwegian wasp, *D. norwegica*
Figure 4. Hind leg of the cuckoo wasp, *Vespula austriaca*
Figure 5. Female mandible: A) Common wasp, *Vespula vulgaris*; B) German wasp, *V. germanica*
Figure 6. Lateral view of the pronotum of the median wasp, *Dolichovespula media*
Figure 7. Dorsal view of the head between the eyes of *D. norwegica*. POL = Postocellar line, PBHL = Postocellar to back-of-head line

SECTION 4

RECORDING

Mike Edwards

Making a record

It is envisaged that most contributors will be submitting records based on their own collecting. Records for mapping will be requested on a 'target species' basis: these target species are announced yearly in the Newsletter. However, any other records may be sent to the relevant recorders at any time (names and addresses in the latest Newsletter). In all cases each record should have as a base a voucher specimen which should be available for inspection in the future, should the need arise.

Some contributors may wish to undertake searches of museum collections or of published papers and articles. It will save unnecessary duplication of effort, and of records, if people embarking on such ventures would first inform the Recording Co-ordinator, who keeps a register of searches being undertaken (name and address in the latest Newsletter).

Recording formats

Records may be submitted in either a computer-readable format or hand-written on one of the standard BRC Record Cards. Both systems are explained in this section. If at all possible, records should be submitted as one of the computer-readable formats, as this saves a lot of time.

What is a record?

There are certain basic requirements which must be met when a record is submitted to the scheme. These requirements are outlined below.

Species name

The names under which insects are recorded have changed over the years. When submitting records, please try to use the most up-to-date name. (Paradoxically, this should usually be the earliest published name!)

At the moment there is considerable flux regarding the correctness of some insect names. Where the name has been changed recently it would help to include the author, or person who first described the species. This is given after the two parts of the scientific name; it may or may not be within brackets, eg *Bombus pratorum*

(Linnaeus), *Psithyrus sylvestris* Lepeletier. If a target species is one where the correctness of the name is important, this fact will be clearly indicated at the time of calling for records. The 'Kloet and Hincks' Checklist and the relevant *Handbooks for the identification of British insects* produced by the Royal Entomological Society will give help here. BWARS will be producing a revised checklist.

BRC species numbers

These are found on the two recording cards, RA 43 and RA 44 (available free of charge from the Biological Records Centre, Monks Wood, Abbots Ripton, HUNTINGDON, Cambs PE17 2LS). Species numbers will also be given in the Newsletter together with the list of target species. An updated list of species and numbers is being prepared by BWARS. Please note that some species do not appear on the original cards.

Locality name

How you refer to the site. If using a local name please also give the nearest place name (town or village is best) which appears on the current 1:50 000 Ordnance Survey map for the area. It can be helpful to give the direction and distance (in km) of the site from the named place.

Locality grid reference

This is based on an eight-figure grid reference, eg SU873210. Only use this level of precision when it is appropriate; remember it refers to a square 100 metres by 100 metres. It is permissible to use such a reference as the centre of a rather larger reasonably homogeneous site. If the exact location is unsure, it is best to use a lower-precision grid reference, eg SU8721 or SU82. Irish and Channel Islands grids are different from those in Britain, and should be used where relevant.

Watsonian vice-county number

Local government re-organisations complicate the issue of county names. The Watsonian vice-counties, on the other hand, have the advantage of being fixed in time. Each Watsonian vice-county has a number. You will need the map showing these (available from BRC).

Collector/s name

Given in full, if known, eg G.M. Spooner.

Determiner/s name

Given in full, if known, eg G.R. Else.

Where voucher specimen is to be found

Given in full. These three examples may help.

- The voucher is in the collections of a recognised institution: record as (eg) 'Dorset County Museum'.
- The voucher is in the collection of a private individual: record as (eg) 'S.P.M. Roberts coll.'.
- A field record, without voucher: record as 'Field record'.

Date collected

Please submit in the form DD/MM/YYYY, eg 24/03/1995. If no day of month is known, then 00/03/1995; if no month 00/00/1995.

Any further information

There may well be data that could make a basic record more valuable. The aim is to collect information which will allow us to add to our knowledge of Aculeates beyond just producing dot-maps of distributions.

Sometimes this kind of information will be written up by the observer for publication in one of the entomological journals. However, it can also be true that the importance of individual observations only becomes apparent when they are combined with many other similar ones. The following should give you some idea of the kind of information that might fall into this category: plant association records; prey taken with specimen; observations on nesting; other behavioural observations; number of specimens and their sex.

If submitting computer-readable data all that is required in the first instance is an indication that further information is available. It is hoped eventually to collect all such data into a second database and a suggested format is given in the section on machine-readable data.

If submitting hand-written data, it would be useful for such observations to be written on the cards alongside the records to which they refer.

Submitting records in computer-readable format

There are two main methods of storing information on computer:

- as a database (preferred), and
- as text produced with a word-processor.

(Please note that a spreadsheet is only a particular way of viewing a database, and is best not used.) The implications of using either storage system are explained below.

Using a database

Increasingly, databases are able to pass information between each other with minimal input from the operator. However, setting up and operating a database require some understanding of quite a complex piece of software.

It is essential that all recorders adhere to the same file structure if data are to be shared easily between databases. It is possible to transfer data directly between many databases, especially those using a DBF format. Failing this, if the data file can be written to disc as a text file (usually in what is known as a comma- or tab-delineated file), then it can be transferred.

The following file structure will allow all the essential information for a record to be passed between databases. (For an explanation of the content of fields see 'What is a record', above). Any additional information which the collector wishes to keep should be added as additional fields **after** the basic information.

Field	Character length
Species name, including author	40
BRC species number	6
Locality name	40
Locality grid reference	8
Watsonian vice-county number	3
Collector/s name	20
Determiner/s name	20
Where voucher specimen(s) is/are to be found (source)	40
Date collected	10
Any further information for record (T/F)	1

(Type T or F here, standing for True or False. 'True' indicates that reference to the original source of the record can give details such as number and sex of individuals, information on habits, flowers visited, parasites reared from host, etc).

Using a word-processor

This section explains how to submit computer-readable data without the use of a database.

Transfer of data between computers has been possible at a very basic level for a long time. A standard, known as ASCII, has been established whereby all computers can recognise text produced by any other machine. Computer files produced in this format are known as TEXT or ASCII files. It is possible to save information from a word-processing program as a text file. This file can then be read into a main database without change, provided the agreed set of rules, or format, are adhered to.

Files must be saved as TEXT (ASCII) files on a floppy disc for sending to the relevant person. Both 3.5 inch and 5 inch double-density discs can be read, although the smaller disc is preferred. Please note that, whilst both IBM compatible and Apple Macintosh discs can be read, some older computers, notably Apricot and 3 inch Amstrad, do not use either format and may not be directly readable. If in doubt, please send a test disc to the Recording Co-ordinator before entering a large amount of data.

Format for submission of records as a text file

The basic rule is that individual pieces of information, which are known as 'fields' in a database (see above), are separated by commas and the maximum number of characters is fixed. This is known as a comma-delineated file.

- No spaces are left between commas and the text, although spaces may be left within the text.
- Type all information as plain text; do not italicise Latin names or use accents; use capital letters at the start of proper nouns only.
- Each letter is worth one character; spaces and other punctuation are also worth a character each.
- Each field has a maximum character length. However, it is not necessary to use all the characters available.
- Each piece of information must be entered strictly in the given order.
- Commas must not be used within a field.
- A carriage return (press 'enter' or 'return' key) marks the end of a record.

Using record cards

Single species (Gen. 13 or 7)

A very useful card when extracting records from collections or other situations where several records of one species from different sites are being recorded. It has space for noting habitat, associations, etc.

Single site (RA 43, RA 44)

One of these lists the wasps (Dryinidae to Sphecidae), the other the bees (Colletidae to Apidae). These are meant for use when recording one visit to a specific site. They may not be ideal if you save all your identifying until winter and process all similar insects together.

Single record (Gen. 8)

Records of exceptional interest (eg new species for the country; species which are thought to be extremely unusual; direct observations of behaviour) should go on to a single record card complete with all the additional data. See above for more information about the kind of data that might be considered for this treatment.

Further information and enquiries on recording should be sent to the Recording Co-ordinator whose address will be found in the most recent BWARS Newsletter.

SECTION 5

A. ENCOURAGING BEES IN THE GARDEN

Chris O'Toole

Introduction

The familiar honey bees (*Apis mellifera*) and bumble bees (*Bombus*) are social insects, living in colonies comprising (generally) a single egg-laying female and many of her sterile female offspring – the workers. Workers co-operate to gather food, build the nest and rear offspring; males and females are only produced at the end of the season. However, the great majority of bees are solitary (non-social) with no worker caste. Instead, each female constructs her own nest and lays an egg in each cell after it has been provisioned with pollen and nectar. Both social and solitary bees can be encouraged to visit and nest in gardens, but solitary bees are the easiest to encourage with artificial nests.

The need for conservation

Modern, intensive agriculture has brought about a decline in the number and diversity of wild flowers and suitable nest sites for wild bees. Bees depend entirely on flowers to feed themselves and their offspring. They collect both nectar (which most bees convert into honey) and pollen. Wild bees like to nest in old logs and in areas of marginal land in hedgerows and the corners of fields, and these habitats are fast disappearing in parts of Britain.

The richest bee faunas are often to be found in town and village gardens, where there is a high diversity of flowering plants and suitable nest sites like old fence posts and walls; others nest in burrows which they excavate in the soil. Nevertheless, there is much we can do to make our gardens even more attractive to bees, and we can benefit by increased pollination of apple, pear and other fruit trees.

What we can do

The easiest way to attract bees is to grow flowers that the bees like to visit. Most seed merchants produce a list of plants which are valuable for bees and sell special seed packs for bee forage. Goodson (1986) and Martin (1986) give more detailed accounts of plants attractive to bees (and other Hymenoptera); the former also has some useful advice on selection and cultivation. Early flowering heather and flowering currant *(Ribes sanguineum)* are attractive to early spring bees, as are all the fruit trees. Later in the year, catmint *(Nepeta cataria)*, lavender *(Lavendula vera)*, sage *(Salvia officinalis)*, stonecrops *(Sedum)* and toadflax *(Linaria)* are all useful bee plants. Flowers such as daisies *(Bellis perennis)*, deadnettles *(Lamium)*, and dandelions *(Taraxacum)* are all attractive, and perhaps a little marginal area with these 'weeds' can be left undisturbed.

Many solitary bees nest in the ground and like well-drained sunny banks with short turf. The small mounds of soil thrown up by females as they burrow into the ground are often conspicuous. It is difficult to encourage mining bees to nest and the best one can do is to keep the lawn well mown!

There are, however, several species of wild bee that will readily nest in artificial nests placed in the garden. They differ from mining bees in a number of ways:

- they nest in existing cavities – beetle borings in dead wood, the hollowed-out pith cavities of bramble, dock, elder or hogweed stems;
- they have dense fringes of stiff hairs for pollen transport on the underside of the abdomen, instead of the legs;
- they collect, according to species, either mud, pieces of fresh leaf or downy plant hairs, which they use to make the cells in which the food is stored and eggs laid.

Artificial nests

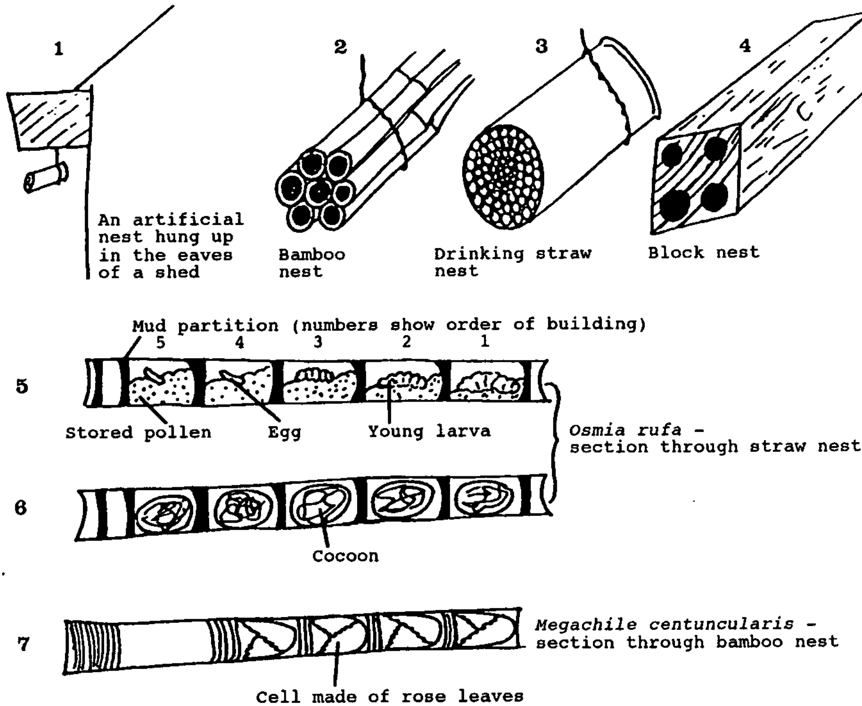
There are several simple designs (see Figures; also Betts 1986). Whichever you choose, they should all be hung at a slight angle so the entrance holes are slightly lower than the back of the nest, thus preventing rain from entering the nest.

The eaves of low outbuildings are ideal places and it is a good idea to hang the nests at eye-level for observation (Figure 1). It is possible to direct a beam of light into the nest using a mirror which does not seem to bother the bees unduly.

When a nest is completed, the female plugs the entrance with mud, discs of cut leaf, or downy plant hairs, according to species. The female dies after making two to three nests. Her offspring develop inside the nest and pass the winter as full-grown larvae or adults. The cycle starts again the following spring. The artificial nests can be used repeatedly and female bees will clear away debris from the previous year.

1. The bamboo nest (Figure 2)

Tie 15 cm lengths of bamboo (garden cane) with a tube diameter of 7–10 mm into bundles of 10–15. Alternatively pack the lengths of bamboo into a tin can.



2. The drinking straw nest (Figure 3)

Carefully clean out an old food can. Place some flakes of paraffin wax or sealing wax in the bottom and heat can gently until wax melts. Allow wax to cool and form a continuous layer over bottom of can – about 8 mm is adequate. Tightly pack some 7 mm diameter drinking straws into the can (Artstraws – from craft shops). Now reheat the bottom of the can until the wax is soft enough to press the straws into it. Allow wax to cool: straws are now firmly anchored in the wax and the nest can be sited in the garden. This nest is suitable for use by schools because at the end of the season some straws can be opened lengthwise with fine scissors and the contents examined.

3. The wooden block nest (Figure 4)

Bore holes 7 mm and 10 mm in diameter and about 15 cm deep into softwood blocks of 50 mm by 50 mm, obtainable from timber merchants. Fit about four holes into each block and put the blocks out singly or tied together in bundles of two or four.

Bees to expect

1. *Osmia rufa* (common or red mason bee) (Figures 5 & 6)

Females of this species will nest in any of the three types of artificial nest described above, and have been recorded nesting in a variety of strange places: a fife, keyholes and spouts of kettles and teapots. Females are about 1 cm long, reddish-brown and can be seen searching for nest sites in late April and May. They seal their cells with mud which they mould into discs and tamp into place using a pair of horns (**not** the antennae) on the front of the head. Pollen is collected mainly from oak and horse chestnut *(Aesculus hippocastanum)*, although daisies, dandelions and deadnettle will also be visited. The larva spins a tough, brown, bullet-shaped cocoon before pupation (Figure 6). This species passes the winter as an adult in its sealed cocoon.

2. *Megachile centuncularis* (rose leaf-cutter bee) (Figure 7)

Females of this species will nest in the bamboo and block nests and only rarely in the drinking straw nest. Their life history is similar to the previous species, except that the cells are made out of pieces of cut leaf, rose leaves being a particular favourite. Rose leaves with characteristically shaped pieces missing are a common sight. This bee flies in late May, June and July. The females are about 1 cm long, black with conspicuous orange red fringes of hair on the underside of the abdomen which is the pollen transport apparatus.

3. *Anthidium manicatum* (solitary or wool carder bee)

Instead of using mud or leaves for cell construction, the females of this species collect balls of plant hairs which they scrape off the leaves with their powerful jaws. They will nest in either the bamboo or block nests and fly in June and July. The females are about 1 cm long, dark brown in colour, with a row of yellow spots on each side of the abdomen. The males are much larger and, in addition to the yellow spots on the abdomen, they also have pale facial markings. It is unusual for male bees to be larger than the females.

4. *Anthophora quadrimaculata*

A good way of attracting this bee to the garden is to plant beds of catmint and lavender. This local bee is a garden species, rarely found in other habitats. It flies from late June to August. Nest burrows are made in the soil, in old walls and sometimes in chimneys where the pointing has deteriorated.

Male bees

The males of *Osmia rufa* and *Megachile centuncularis* emerge before the females and can be seen flying about and sunning themselves in the vicinity of the nest site. The males seek females in the nesting area, but those of *Anthidium manicatum* set up territories in the females' foraging area. They defend the territories against other males and will even fight off bumble bees. Both the male and female of *A. manicatum* have a characteristic rapid darting flight and they can hover and fly backwards.

Some questions to answer

1. How many trips does an *Osmia* female make before she has enough mud to seal a cell, or a *Megachile* enough pieces of leaf?

2. How many foraging trips before the egg is laid and the cell sealed?

Identification

Some care is required when reading the above: there are about ten species of solitary bees that may utilise artificial nests, depending on the diameter of the holes.

B. PHOTOGRAPHING ACULEATES

David F. Lloyd

Introduction

The photography of small subjects such as aculeates is a speciality known as macro-photography, and requires a modicum of specialised photographic equipment.

Though it is possible to produce close-up photographs of insects with black-and-white or colour negative film (allowing prints to be made), most macro work is done using transparency films. In order to produce sharp and grain-free slides, a slow film is necessary, of the order of 25, 50 or 64 ASA. This means that flash equipment will be required to illuminate the subject adequately. The use of flash has the added advantage that it is possible to use small apertures, which give a greater depth of field. Flash also freezes movement and generally gives a sharper result.

It is important to realise that all close-up photographers have their own technique and what follows is the author's interpretation!

The camera

The camera is a body which transports film past the lens. As such, the less complicated the camera the better. Automatic systems have advantages for general photography, but, for macro work, manual facilities are simpler to use. Most modern single-lens reflex cameras have a manual setting, and there are inexpensive makes which are primarily manual. The latter may be advantageous if you frequent sand dunes or other gritty places!

The lens

The lens takes the picture! Therefore it makes sense to spend as much as you can afford on the lens. Dedicated macro lenses are available which will give a 1:1 ratio (ie the insect is reproduced life-size on the slide). However, excellent results can be obtained using a standard 50 mm lens and extension tubes. These tubes fit between the camera body and the lens, preferably maintaining lens shutter operation. They come in several lengths and you must select and fit the correct one for the subject size before taking the shot! In this respect, dedicated macro lenses have an advantage, but in many cases even macro lenses require extension tubes for ratios in excess of 1:1. A fast 50 mm lens means less light is needed and a smaller flash gun will be satisfactory. The lens must stop down to at least f 16 and preferably f 22 to give adequate depth of field at a 1:1 ratio.

The flash

Flash must be synchronised with the lens shutter operation and most cameras work at 1/125th of a second. This will effectively freeze most action, with the exception of wing-beats: also, a limited amount of hand-shake will be eliminated. Most cameras have a 'hot shoe' arrangement by which an attached flash gun can be synchronised to the shutter operation. However, it is recommended that you do not mount the flash gun on the top of the camera: instead, have it to one side. You may find that the most convenient way to do this is to use a bracket attached to the camera tripod socket.

The flash gun must have enough power to expose the film adequately (say, 50 ASA) at 1/125th second using an aperture of f16 when reflecting off the subject. A fast recharge time is also valuable.

The precise arrangement of flashgun on the bracket, distance to the subject, lens aperture and film speed is subject to trial and error and may require the expenditure of several rolls of 24 or 36 exposures to calibrate. Once satisfactory results are obtained, lock all positions with a contact adhesive and get used to shooting with one group of camera settings. This allows you to concentrate on composition of the frame and capturing the action!

Photographing aculeates

Whilst a tripod may be useful for static objects such as pinned specimens or a wasps' nest, to shoot live insects in the field successfully the camera and bracket should be hand-held. Trust the shutter synchronisation speed and flash duration (*ca* 1/10,000th sec) to freeze subject motion.

The generally held view is that it is not possible positively to identify many subjects from a slide. However, a good portrait with the axis of the subject parallel to the film plane, sharp and well exposed may enable a specialist to make an identification to genus, if not to species. Much scientifically useful information concerning the biological activity of the subject can be captured on a good slide. Consequently, every effort should be made to identify the subject, and all relevant data should be inscribed on the slide mount, including date, locality, identification and determiner, identification of plant if captured foraging, and a grid reference if possible. Use a notebook in the field!

I feel that three slides make up a biological record. The first slide should be a habitat shot taken with the 50 mm lens and should include the subject, even if not obviously visible. The second shot should be 1/5th life size and include any interesting activity, eg on a flower, or at the burrow. The last shot should be a 1:1 portrait or study good enough to make a tentative identification.

Bear in mind when shooting insects in close-up that the lens will be only two inches (*ca* 5 cm) or less from the subject for a 1:1 ratio. The subject often will not sit still under these conditions, particularly when the flash fires. You must be prepared for a large number of failures!

With practice, you will find that some individuals do 'pose' for the camera and will sit still long enough for the flash to recharge and take a second or more shots.

Finally, do test the whole set-up without film in the camera before every expedition. There is nothing so frustrating as carefully stalking a subject only to find that when the shutter is pressed, the flash does not fire or some other failure occurs, and, of course, the specimen disappears!

SECTION 6

DEALERS IN ENTOMOLOGICAL SUPPLIES

Amateur Entomologists' Society Publications, The Hawthorns, Frating Road, Great Bromley, COLCHESTER CO7 7JN. Tel: 01 206 251 600. For *The hymenopterist's handbook* (1986) and Supplement (1988). The latter is essential, but must be purchased separately (total price in 1995 - £10.50).
Also, *The dipterist's handbook* (1978) (£9.50).

BCB International Ltd, Freepost CF4037, CARDIFF CF1 1YS, Wales. Market a 5 by 2.5cm thermometer with a standard key ring fitting. Calibrated in Centigrade and Fahrenheit. The instrument is called 'Zip-o-Gage', and is ideal for field naturalists.

Davcaps, Medical & Scientific Supplies, PO Box 48, HITCHIN, Hertfordshire SG4 9BT. Tel: 01 462 433 210. For gelatine capsules. These are sold by the hundred or thousand (prices upon request). The sizes useful for the immature stages of aculeates fall in the range 000 (the largest), 00, 1, 2, 3, 4 (the smallest).

Hampshire Micro, The Microscope Shop, Oxford Road, SUTTON SCOTNEY, Hampshire SO21 3JG. Tel: 01 962 760 228. For microscopes and hand lenses.

Henshaw, D.J. & D., 34 Rounton Road, WALTHAM ABBEY, Essex EN9 3AR. Tel: 01 992 717 663. Entomological supplies, including containers, tubes, chemicals and pins. Will print specimen labels to your exact requirements – right down to 4pt (this is 4pt!). Send for samples.

Marris House Nets, 54 Richmond Park Avenue, Queen's Park, BOURNEMOUTH, Dorset BH8 9DR. Tel: 01 202 515 238. For Malaise traps, moth traps, nets and netting.

Watkins and Doncaster, Four Throws, HAWKHURST, Kent TN18 5ED. Tel: 01 580 753 133. This long-established and renowned firm stocks most collecting and mounting equipment, including celluloid. Illustrated catalogue available.

Worldwide Butterflies, Compton House, SHERBORNE, Dorset DT9 4QN. Tel: 01 935 746 08. Market the best kite net (frame and net bag) currently available: it has even won a design award!

SECTION 7

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[See Section 6 for the address of the Amateur Entomologists' Society.]

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