The British Arachnological Society

Members' Handbook

First edition

Edited for the British Arachnological Society by
Tony Russell-Smith

www.britishspiders.org.uk

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Cover picture: The fen raft spider *Dolomedes plantarius* ♀ and ♂; in drainage ditch at Pevensey Levels, Sussex. *Dolomedes plantarius* is one of our rarest spiders, found at only three sites in Britain and fully protected under both British and European laws. Photo © O. Jones.

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Copy-edited and typeset by P. A. Selden (paulselden@mac.com)
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The British Arachnological Society

The Society grew out of the Flatford Mill Spider Group formed in 1958 by a small band of enthusiasts led by D. W. Mackie. By 1963, a regular news bulletin was being produced and it was renamed the British Spider Study Group. At its tenth annual meeting in 1968, it was agreed that the Group would change its status and become the British Arachnological Society. Currently the Society has just under 300 British members and 220 overseas members.

The prime objective of the Society is to further the education of the public by promoting the study of the Arachnida, particularly spiders, opiliones and pseudoscorpions. Membership of the British Arachnological Society is open to any individual interested in the study of spiders and other arachnids. Advantages of membership include:

- The Society's *Bulletin* and *Newsletter* published three times a year and free to members and subscribers. The Bulletin publishes peer-reviewed scientific papers on all aspects of arachnology world-wide. The *Newsletter* contains short articles and notes, principally of interest to UK members, and now incorporates the Spider Recording Scheme newsletter.
- Access to the Society's extensive library of arachnological papers. The society also holds an important collection of photographic slides of spiders and other arachnids which are available for loan to members.
- Access to the members' section of the website which is gradually developing into an online resource for arachnologists
- Access to an email discussion group – the ideal place for advice, help or just a chat!

Other activities organised or supported by the Society are:

- Running the Spider Recording Scheme to develop our knowledge of the distribution, habitat preferences and phenology of the British spider fauna. Full details of the scheme are provided in Chapter 7.
- Holding and encouraging the holding of courses, meetings and other gatherings to foster the study of arachnids. These include short one day or weekend meetings and courses as well as occasionally longer one week courses, often in association with the Field Studies Council.
- Producing Occasional Publications dealing with different aspects of arachnid systematics and biology.
- Providing impartial scientific advice and guidance to individuals, NGOs and government bodies on the conservation of the British arachnid fauna.
- Those interested in joining should visit the website at: http://www.britishspiders.org.uk or contact the Membership Secretary at: membership@britishspiders.org.uk.

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In addition to the authors of each chapter, many members of the BAS have contributed to the production of this handbook. I am particularly grateful to the following for taking the time to peer-review the first drafts of chapters; Martin Askins, Lawrence Bee, Ian Dawson, Peter Harvey, Paul Lee, Peter Merrett, David Nellist and Chris Spilling. For permission to use their excellent photographs, without which the book would have been considerably less interesting, I thank John Murphy (for allowing us to use the splendid photos taken by the late Frances Murphy), Martin Askins and Peter Harvey. I am also grateful to Peter Harvey for scanning many of the original 35 mm transparencies. Paul Selden kindly prepared the final PDF file for the printers and provided helpful advice on other editorial aspects. Finally, Rod Allison provided constant support and advice throughout the project as well as liaising with the printers.

Tony Russell-Smith
October 2007

[NOTE: The first paragraph of these acknowledgements was inadvertently omitted from the printed version of the handbook. We are extremely sorry for this and apologise to Stan for the error.]

Contents

1. WHY STUDY SPIDERS?

A. Russell-Smith

1.1 Introduction

Spiders are found in virtually every environment on earth, from the driest deserts to near the summit of Mt Everest and on all continents other than Antarctica. In Britain, they are abundant in almost any habitat type including regularly inundated areas such as salt marshes, the highest mountain tops and even man-made habitats such as buildings and motorway verges. In terms of their diversity, spiders, with 40,000 species worldwide, rank second only to insects among terrestrial animals. However, in Britain we have a fairly restricted spider fauna (about 650 species) compared to that of other countries of north-west Europe. This is thought to have come about as a result of the relatively short period in post-glacial times when Britain still had a land connection to Europe. Once the English channel opened up about 8000 years ago, immigration into the British Isles would have become significantly more difficult, although not impossible, for spiders. The British spider fauna, however, is not static, with new species being added almost every year and the distribution of many established species changing constantly, partly in response to climate change.

1.2 The Uniqueness of Spiders

A common misperception of spiders, which is sometimes found even among those who should know better, is that they are merely slightly odd insects which have an additional pair of legs and lack wings. Nothing, in fact, could be further from the truth. Although they share an external jointed skeleton with other arthropods such as insects, crustaceans and myriapods (centipedes and millipedes), they have evolved independently from any of these other groups for hundreds of millions of years. In so doing, they have developed a series of adaptations to survival on land which are quite unique among animals. Although this is not the place to go into the details of anatomy and physiology of spiders, they differ completely from insects in their locomotion, prey capture and feeding, respiration and their reproductive systems and behaviour. But, above all, they differ from the insects in their production and use of silk in a whole range of different aspects of their biology and in injecting venom through their fangs to immobilise their prey. Some insect groups do indeed produce silk, for example to provide a protective cocoon for the pupae in butterflies and moths or for prey capture amongst some aquatic caddis-fly larvae, but these are exceptions rather than the rule.

1.3 Silk and its Uses

Spiders use silk in almost all aspects of their life. Most obvious to the casual observer are the various types of web that many use in prey capture. But silk is also used to build retreats and line burrows, to protect the eggs from parasites and desiccation and as a safety device when moving around. It is not widely realised that all spiders lay down a silken dragline wherever they go that can act as a climbing rope to retreat to safety should they lose their footing or be disturbed. Silk is also used for dispersal of spiders by so-called ballooning. If climatic conditions are suitable, spiders will climb to the top of the vegetation and then release a long strand of silk into the breeze. Eventually the spider takes off on the end of the silken strand and can be carried to considerable heights by thermals. Drifting in the wind, they eventually come down to earth again, where, if the habitat is suitable, they can establish new populations. Spiders can travel considerable distances in this way and it seems more than likely that some species that have been newly found in our southern counties have arrived from the near continent by ballooning. The strangest use of silk is perhaps in making of the sperm-web which is spun by male spiders prior to mating. This is a minute sheet of silk onto which the male deposits a small drop of sperm which is then taken up into the pedipalp (functionally equivalent in spiders to the penis in mammals). The pedipalp is then used to introduce the sperm into the female sexual orifice, the epigyne, during mating.

Uniquely among land animals, spiders produce a different type of silk for each of the various purposes mentioned above. In orb-web spiders (Araneidae) there are no less than six separate types of silk-producing glands in the abdomen, each supplying different pairs of spigots in the spinnerets, through which the silk is secreted to the exterior. Three of these are used in the production of the complex orb-web, one for the production of dragline silk, one is used for both swathing prey items in silk and producing the sperm web and the last is used to produce the egg cocoon. The silk from each type of gland differs somewhat in its chemical and physical properties, ensuring that it is optimally fitted to its function. Compared to this diversity, the simple silk used by insects seems positively rudimentary. The silks produced by spiders are liquid proteins which solidify on contact with the air during extrusion. Interestingly, this actually happens just inside the spigot so that by the time it reaches the exterior it is fully functional.

1.4 Prey Capture

Just as spiders have developed a range of uses for silk, their prey capture techniques are equally diverse. For convenience sake, spiders are normally divided into two groups, hunting spiders that do not use a web to catch prey and web-builders that always catch their prey in a silk snare. In reality, this division into active hunters and passive trappers is not quite as hard and fast as it might seem. In the most primitive living spiders (members of the sub-order Mesothelae, found only in south-east Asia and Japan), the spiders live in a silk-lined burrow in soil, from the mouth of which strands of silk radiate on the soil surface, like the spokes of a wheel. When a prey item disturbs one or more of these strands, the vibrations are transmitted to the spider in the burrow which then rushes out and attacks and kills its prey. Thus, although silk is used in detecting prey, it is not used to make a snare. It seems likely therefore that the potential for becoming either free living hunters or sedentary web-builders already existed in the earliest spiders and that most modern groups of spiders have evolved to use one or other of these strategies. However, some families, such as wolf spiders (Lycosidae) and nursery-web builders (Pisauridae), include some members that are web-builders and others that are not.

Among families of spiders that are principally free-living hunters, we can see a further sub-division into those with poorly developed eyes that hunt largely by detecting the vibrations caused by their prey and those with well-developed eyes that hunt largely by sight. Good examples of the first group in the British fauna include the sac spiders (Clubionidae) and the ground spiders (Gnaphosidae). Species of both these groups have two rows of four small eyes on the carapace which are probably only able to detect light and dark. Both families are largely nocturnal and sight probably plays a relatively small part in prey detection. They rely more on special sensory hairs on the legs (called trichobothria), which are highly sensitive to small movements of air, to detect the movement of prey.

Examples of spider families that hunt visually include the wolf spiders (Lycosidae) and, particularly, the jumping spiders (Salticidae). In both these families, the eight eyes are arranged in three rows and at least one pair of eyes is greatly enlarged and is capable of forming a proper image of objects. They are most highly developed in the jumping spiders which have the anterior median eyes enormously enlarged and pointing forward on the carapace. These eyes are capable of adaptation, the ability to focus on objects at different distances, which allows the spider to accurately estimate the distance of a prey item it has stalked and finally leap onto and kill it. With the notable exception of the cephalopods (octopuses, squids and their relatives), the eyes of jumping spiders are probably the most highly evolved amongst those of all invertebrates. It is perhaps in part because of their exceptional vision that the salticid spiders are the most species-rich spider family, with over 4000 species worldwide, although in Britain we have a rather impoverished fauna of some 38 species.

In web-building spiders, a considerable range of different prey capture devices have evolved, each of which is associated with a different range of prey capture behaviours. Some webs are specifically designed to catch flying insects while others trap insects and other invertebrates crawling on the ground or the trunks of trees. An example in the latter category is the sheet web of *Agelena labyrinthica*, common in the southern half of Britain in hedgerows, bushes and grassy areas in autumn. Insects that leap onto or crawl across the dense sheet of silk are ambushed by the spider emerging from a tubular retreat in one corner of the web. Some types of sheet webs have added refinements and are designed to capture small flying insects. In the money spiders (Linyphiidae), the thin sheet of silk has a dense scaffolding of criss-cross threads above it. Insects that fly into the scaffold are knocked down onto the sheet below. The spider hangs upside down on the underside of the sheet and, when an insect falls on it, attacks and bites it through the sheet. As with all spiders, the bite rapidly paralyses and finally kills the insect which is then fed on at leisure. Although this type of web may look untidy and much less organised than, for example, an orb-web, its functional efficiency is attested to by the fact that over 40% of our species belong to this family which ranks second after the jumping spiders in numbers of species worldwide.

However, it is the orb-web weavers, belonging to the families Araneidae and Tetragnathidae, that perhaps attract most attention and admiration. To the casual observer, the organised, geometrically regular webs they produce might appear to be the product of conscious design. While we know that this is not the case, but rather that they are the products of a series of highly stereotyped behavioural steps over which the animal has relatively little conscious control, they are nevertheless fascinating examples of how complex structures can result from simple behavioural steps. Although to the casual eye all orb-webs look very similar, careful examination reveals differences in construction and use between the webs of different genera. In Britain, an obvious example is the web of *Zygiella*, a genus often found in and on houses, which has one sector of the spiral threads missing, leaving one of the radial threads free. During the day, the spider sits in a retreat constructed at the end of this radial thread which is used as a vibration detector. As soon as an insect contacts the web, the spider rushes from the retreat along the free radial thread to the hub of the web, from where she attacks and kills the prey.

A number of spider species have evolved specialised webs, designed to capture particular prey types. In the comb-footed spiders (Theridiidae) several genera specialise on feeding on ants. This is unusual among invertebrates in general since ants have formidable defences and live in colonies, thus making them risky prey. They may have evolved to feed on ants in part because theridiids possess extremely potent venom which paralyses and kills their prey very rapidly. In *Achaearanea riparia* (Fig. 5), the web is an untidy tent-like structure, built just above the ground or other flat surface over which ants crawl. Hanging down from the web are a number of straight vertical threads attached to the ground, the bottom part of which have drops of gum along them. The threads are stretched extremely taught and, when an ant brushes against one, it sticks to the gum, breaks the thread and is lifted off the ground by its contraction. The spider then immediately starts hauling up the thread using her first pair of legs until the ant is just below her. She then turns round so her spinnerets point towards the ant and flings more gummy silk over it, using her fourth pair of legs. Once the ant is subdued, the spider bites it and subsequently feeds on it. In the genus *Episinus* (Fig. 8), the web is highly reduced and consists of an H-shaped framework within which the spider hangs head downwards by her hind pair of legs, just above the ground surface. These spiders are often found in heathland where they construct the web beneath a heather bush or similar small shrub. Her front legs are stretched out at an angle to hold the two vertical arms of the H-shaped web which are attached to the ground and have their bottom portion coated in drops of gum, as in the web of *Achaearanea*. As soon as an ant (or other crawling insect) touches the base of the vertical thread, it is attacked, swathed in silk and bitten.

For anyone who is interested in learning more of the webs and prey capture behaviour of British spiders, an excellent starting point is Bristowe's book *The world of spiders* (see Recommended Reading). His vivid and well illustrated accounts have been the stimulus for many, myself included, to take an interest in this fascinating group. A more scientific account, which covers spiders worldwide and deals with the evolution of web-building and the evidence webs provide for the relationships of different spider families, can be found in the compilation edited by Shear entitled *Spiders. Webs, behavior, and evolution*. This is written by a series of authors and the chapters vary in their readability but it does pull together all that is known on the topic of spider webs and their use in prey capture.

1.5 Courtship Behaviour

The majority of spiders are equipped with very simple eyes and have relatively poor vision, in some cases possibly only being able to distinguish light and dark. This is particularly true of web-building spiders, the vast majority of which probably only use vision at very close range. This presents a problem for mating, as both the male and the female spider need to know whether the opposite sex represents a predator, a prey or a potential mate before the other has come too close. The need is particularly great for the male as, in some species at least, the larger female is quite capable of attacking and killing him. All spiders are extremely sensitive to vibrational stimuli, whether these are transmitted through the air or through the substrate on which they sit. The legs carry special sensory hairs called *trichobothria* which detect the slightest vibration in the air, and the cuticle is furnished with slit sense organs, some of which are designed to detect vibrations transmitted through the substrate. In many web building spiders, the males court the female in the web by either plucking the edge of the web with a leg or drumming on it with the abdomen, legs or palps. In the genus *Amaurobius*, for example, it has been shown that the common European species each has a specific pattern of vibration of the web which allows the female to know that it is a male ready to mate and not a potential prey item or predator. A similar courtship behaviour is found in orb-web spiders, which either pluck the edge of the female web or, in some *Argiope* species, dangle on a single silk thread near the hub of the web and then pluck this thread to signal their presence to the female.

Some families of hunting spiders, such as the wolf spiders, lynx spiders (Oxyopidae) and jumping spiders, have well developed eyes and excellent vision. Many of these use visual displays in courtship, allowing the male to signal to the female that he is ready to mate. Among wolf spiders, males of the genus *Pardosa* usually have palps covered in dense black hairs. During courtship, the palps are waved up and down or side to side, the first pair of legs may be waved or vibrated and the abdomen raised and lowered or vibrated against the substrate as the male faces and slowly approaches the female (Fig. 10). Each species has a different and specific pattern of palpal and leg movements which signals to the female that it is a male of her own species ready to mate which is approaching and not a prey item or predator. The displays have an uncanny resemblance to the semaphore system with flags which was used by the Royal Navy in the past to signal from ship to ship. Such visual courtship is also used by other genera among lycosids, including *Trochosa*, *Alopecosa* and *Aulonia*.

A fascinating study of the courtship behaviour of a group of European *Pardosa* species related to *P. lugubris* demonstrated that there were in fact no fewer than six closely related cryptic species present in Europe that differed in the details of male courtship displays (Töpfer-Hofmann *et al.*, 2000). It was only after these species had been separated on the basis of male courtship that it was realised that there were small but significant differences in the structure and colouration of the male palps which allowed them to be separated on morphological characteristics. An interesting secondary consequence of the study was the discovery that the common species in Britain is *Pardosa saltans* (Fig. 14), one of the newly described species, and not *P. lugubris*, as previously thought. In fact, the true *Pardosa lugubris* has since been discovered in Scotland and northern England but appears to be a rare species in Britain.

As was mentioned earlier, the salticids have the best developed eyes and most acute vision of all spiders. While not proven, all the evidence suggests that salticids possess colour vision. Males of jumping spiders are normally very brightly coloured and in particular have strongly coloured hairs on the palps and the front of the cephalothorax. The facial region of the cephalothorax is often decorated with bands or patches of hairs of markedly contrasting colour (Fig. 9), sometimes resembling nothing so much as a native American in full war paint! As with lycosids, the male performs an elaborate courtship dance which involves palpal movements, leg movements and a specific pattern of steps as he slowly approaches the female. Here again, the purpose of the courtship display is to establish that he is a male of her own species and consequently each species has a different sequence of palpal and leg movements as well as a specific pattern of steps. It is thought likely that the precise pattern of coloured hairs on the facial region also plays a role in recognition of the male by the female as a mate of her own species.

1.6 The Diversity of Spiders in Britain

The British spider fauna, in common with that of most countries of north-west Europe, is dominated by money spiders, members of the family Linyphiidae, which account for over 40% of our spider species. This is a considerable contrast to the situation in warmer areas of the world, such as the Mediterranean, where linyphiids typically form a small proportion of all spiders. For example, they constitute only 7% of the Greek spider fauna. While there are a few larger linyphiids, such as the ubiquitous *Linyphia triangularis* found on shrubs and tall grass in autumn, the majority of species are small (less than 3.0 mm in total length) with black or grey abdomens which lack a dorsal pattern. While they are found in virtually all possible habitats, including on trees and shrubs, the majority of species are found either on the ground surface, in leaf litter or even in the soil. Here they produce minute sheet webs which catch small insects such as springtails and midges. It seems that the small size of money spiders and their habit of living at or even below the ground surface are likely adaptations to the relatively cold and wet climates of northern latitudes. Many species, particularly among those living in the shelter of woodland litter, are adult and fully active throughout the winter, at least in southern Britain.

Linyphiids include many of our commonest spiders which are found throughout the country and in a wide variety of habitats. These include several species, such as *Tenuiphantes tenuis*, *Bathyphantes gracilis* and *Oedothorax fuscus*, which are well adapted to surviving in intensively managed arable fields, where they can occur in high densities. However, as might be expected in such a diverse family, there are many which are rare and known from very few localities. In fact, 30% of our Red Data Book listed species and 39% of Nationally Notable species are money spiders. As with many other families, there are several different reasons for their rarity. In some cases this may be a result of very specialised habitat or micro-habitat requirements that are only found in a few places. Examples include *Diplocephalus connatus*, confined to small cavities under large stones and boulders along two major rivers in Northumberland and *Midia midas*, only found on ancient trees in a few forests and parklands scattered from Nottingham southwards. In other cases, the species concerned seem to be restricted to the extreme south of Britain and may well be at the north-west edge of their range in this country, with adverse climate restricting their spread northwards. These include such species as *Trichoncus saxicola* (Nb), *Meioneta simplicitarsis* (Na) and *Neriene furtiva* (Nb). Finally, there is an unfortunately large group of species which are apparently rare but, because we know so little of their detailed habitat requirements, our real knowledge of their distribution and abundance is simply inadequate. A good example is *Pseudomaro aenigmaticus*, a tiny, pale species with reduced eyes found in four or five localities in southern England. It has been suggested that it may live in fissures in stony soils and have been overlooked in studies using normal sampling techniques.

Among web-building spiders, two other families are numerically important in the British fauna, the comb-footed spiders (Theridiidae) which include 8.5% of our species and the orb-web weavers (Araneidae) which include 5%. Unlike linyphiids, the theridiids are characteristically found in the field and shrub layers of most habitats. Only a few genera such as *Enoplognatha*, *Robertus* and *Pholcomma*, are characteristically found in the ground layer. Again, many species, such as *Theridion sisyphium* (Fig. 17), *Enoplognatha ovata* and *Paidiscura pallens* are very widespread and common in Britain. However, unlike linyphiids, only a small proportion of British theridiids are uncommon, with only 14% of the 56 species included in the British RDB or Notable species lists. However, among them are a number of interesting and rather poorly known spiders. Members of the genus *Dipoena* are all, so far as we know, specialist feeders on ants and predominantly occur in dry habitats in the South. Among the seven species recorded in Britain, no less than four are RDB-listed and the other three species are on the Notable list. *Robertus insignis* (RDB1), a fenland species from East Anglia, was rediscovered near Norwich in 1988 after an 80 year gap. Not only is it extremely rare in Britain, it is scarce throughout Europe, being known from only a few individuals from Sweden, Estonia and Germany.

The British orb-web spiders are again found characteristically in the field and shrub layers but also extend into the higher tree layer. Among our 33 species, only three are RDB listed with a further eight (24%) being listed as Nationally Notable. Almost everyone will be familiar with the common garden orb-web spider, *Araneus diadematus* (Fig. 6) which, together with another eight species in this family, is widespread and abundant throughout Britain. Several other species, while still relatively common, are confined to more southern areas of Britain where they tend to be found in warmer and drier habitats. They include *Agalenatea redii*, *Zilla diodia*, *Hypsosinga pygmaea* and *Mangora acalypha*. A particularly interesting and beautiful species, the so-called wasp spider (*Argiope bruennichi*), is a relatively recent addition to the British fauna. It was first recorded near Rye in East Sussex in 1922 and for many years was only recorded from a few places along the south coast, from Kent to Dorset. However, from the 1970s it had started to spread northwards and is now common throughout the South-East with a few isolated records as far north as Derbyshire and several from Devon and Cornwall. It is to be found in tall grass where the female weaves a characteristic web with a cross-shaped structure of iridescent silk in the centre and is mature in late summer and autumn.

The hunting spiders can be divided, for the sake of convenience, into those with poor eyesight that hunt largely by feel and those with well developed eyes that hunt by sight. Among the former group, the ground spiders (Gnaphosidae) include 33 species, representing 5% of the British fauna. The majority of gnaphosids are medium-sized, fairly dark spiders with an elongated body form which, as the name suggests, live almost exclusively on the ground surface. They are very largely nocturnal hunters, spending the day hidden beneath stones or litter, but the ant-mimicking *Micaria* species run in sunshine. Although about a quarter of the species are widespread in Britain, a majority have a markedly southern distribution with over half (17 species) being limited to an area south of a line between the rivers Humber and Tees and all but four of these more or less confined to an area south of the Thames–Severn line. Worldwide, ground spiders are typical of hot and dry climates and it is therefore perhaps not surprising that we have a relatively small fauna in this country or that most are confined to the warmer and drier areas of Britain. It is perhaps because many of the species reach the edge of their natural range in Britain that they are rare or uncommon. More than half (18 species) are either included in the current Red Data Book list (six species) or the Nationally Notable list (12 species).

The other common night-hunting family of spiders in Britain is the sac-spiders (Clubionidae). Although they resemble the gnaphosids in body form, they are distinguished by the conical (rather than cylindrical) anterior spinnerets and their pale colouration. Unlike the ground spiders, the majority of species are found in the field or shrub layer with a few species also occurring in the tree layer. Among the 24 species, 60% are widespread in Britain and only five species have a predominantly southern distribution. Indeed, unlike gnaphosids, the family in general is much more diverse in temperate climates than in hot or dry areas. Six species are listed in the current British Red Data Book, two from coastal habitats (*Clubiona frisia* and *C. genevensis*), two from fens (*C. juvenis* and *C. rosserae*) one from Caledonian pine forests (*C. subsultans*) and one from southern heathland (*Cheiracanthium pennyi*).

The wolf spiders (Lycosidae) are characteristically diurnal hunters with well developed eyes and excellent eyesight. Our 38 species (5% of the total) include many that are familiar inhabitants not only of grasslands, heathlands and other open habitats but domestic gardens and urban wasteland as well. Females may be immediately recognised in the breeding season by their habit of carrying their egg-sac attached to their spinnerets, a behavioural trait almost unique to this family. They are almost exclusively ground-active, with only *Pardosa nigriceps* regularly found in the field layer of grasslands and heaths. The largest genus is *Pardosa* (15 species) which includes some of our commonest spiders. *Pardosa amentata*, *P. palustris* and *P. pullata* are found throughout Britain and are often very abundant. In addition to the open habitats already mentioned, wolf spiders are also characteristic inhabitants of wetlands. Typical species of marshes, bogs and fens include the six species of *Pirata*, of which *P. piraticus* is the commonest and most widespread, *Pardosa purbeckensis* (salt marshes), *Arctosa leopardus* and the rare *Hygrolycosa rubrofasciata*, found in fenlands mainly in East Anglia. Our wolf spiders include five Red Data Book species and six Nationally Notable species. The RDB listed species include *Pardosa paludicola* (RDB3) from half a dozen sites in southern Britain where it usually occurs in long grass near water, *Alopecosa fabrilis* (RDB1) known from two heathland sites in Dorset and Surrey, *Arctosa fulvolineata* (RDB3) from a few saltmarsh sites in southern and eastern England, *Arctosa alpigena* from some 10 sites on mountains above 1000 m in northern Scotland and *Aulonia albimana*, known from only one grassland site on the Isle of Wight. In addition, four of the six Nationally Notable species are restricted to the southern half of Britain.

The jumping spiders (Salticidae) are arguably our most distinctive and beautifully marked family of spiders. As already noted, their excellent eyes allow them to stalk and capture their prey visually and they are entirely diurnal hunters. This is predominantly a family of warm climates and in the tropics and Mediterranean they dominate the spider faunas. For example they form 10.3% of the spiders of Spain and 9.6% of those of Italy but account for only 5.8% of the British fauna. As with the gnaphosids, a large proportion of our 38 species have a southern distribution, 18 (50%) being more or less confined to an area south of a line between the Wash and the Severn estuary. Even some of our commonest species in the southern part of Britain, such as *Salticus scenicus*, *Heliophanus flavipes* and *Euophrys frontalis*, are either very much less widely distributed in or, as in the last species almost absent from, most of Scotland. Like the gnaphosids, many of our jumping spiders are probably near the north-west limit of their distribution in southern England. Interestingly, there is a group of six species that are confined in Britain either to shingle or sand dunes in the south but in continental Europe are found in a much wider range of habitats. They include *Heliophanus auratus*, *Pseudeuophrys obsoleta*, *Sitticus inexpectus* and *Phlegra fasciata*. It seems probable at least that the warm, dry and open habitats provided by shingle or sand dunes allow them to maintain a foothold in southern Britain by providing micro-climatic conditions similar to those further south in Europe.

This is another family which has an unusually large number of rare species with seven included in the British Red Data Book and 14 Nationally Notable, accounting for 57% of all our jumping spiders. The seven RDB listed species include *Heliophanus auratus* (RDB2), *Pseudeuophrys obsoleta* (RDB3), *Sitticus inexpectus* (RDB3) and *Phlegra fasciata* (RDB3) mentioned above, *Neon valentulus* (RDB 2), known from only six fenland sites in East Anglia, *Pellenes tripunctatus* (RDB1) known from only two shingle sites on the South coast, *Heliophanus dampfi* (RDBK), a species found in bogs in one site in Wales and two in Scotland and finally *Sitticus floricola* (RDB3) from some half dozen bogs in Cheshire and southern Scotland. Among the 14 Nationally Notable species, only two extend into northern England or Scotland and 11 are essentially confined to the area south of the Wash–Severn estuary line.

Given their distinctive appearance and small numbers, it might be thought unlikely that many new species of jumping spiders would be added to the British list. However, in the past 10 years three new species have been found in this country and consequently do not yet have an official conservation status. The first was the small and rather inconspicuous species *Neon pictus*, first discovered on shingle at Rye Harbour (Sussex) in 1998 and then a little way along the coast at Dungeness (Kent) in 1999. To date, these remain the only localities for this species. *Macaroeris nidicolens* is a larger and more conspicuous species, widespread on shrubs and trees in continental Europe. It was first taken in Britain from small pine trees in a park in East London and thought at the time to have been imported with the trees. However, it has subsequently been collected from young pine trees in Surrey, and also in Essex, and it is now thought it may have colonised the country without human assistance. Finally, a small species of *Sitticus*, *S. distinguendus* was first reported from a brownfield site in south Essex in 2003 and then from a somewhat similar site in north Kent in 2004. Again, these are currently the only known sites in the UK for this species and since both are scheduled for development in the near future, this species must be regarded as highly threatened. In northern France and Belgium the species is associated with stable "grey" dunes and it is possible that it might occur in a similar habitat on the south coast of England.

The crab spiders represent a totally different type of diurnal hunting spider. I deliberately include the families Thomisidae and Philodromidae here because, although distinct, they are closely related and were formerly considered a single family. The two families combined include 41 species (6.3% of our species). The true crab spiders (Thomisidae) are flattened with the legs held out sideways and the first two pairs of legs much stouter and longer than the last two. The so-called running crab spiders (Philodromidae) resemble thomisids in body form but have much longer legs which are all more or less equal in length. True crab spiders differ from other daytime hunters in that they are lie-in-wait predators which rarely pursue their prey but ambush them when they come close enough to be seized in the powerful front legs and rapidly subdued with a bite that injects fast-acting venom. The mammalian equivalent would perhaps be the leopard which, among the big cats, rarely pursues prey any distance but ambushes it from a tree or other suitable cover. Running crab spiders, as the name suggests, are much more active, most members chasing their prey on the foliage of shrubs and trees.

Two different ecological groups can be distinguished among the true crab spiders. The first, which includes the genera *Xysticus* (Figs. 3, 11) and *Ozyptila*, is predominantly ground-active (although also found in the field layer of grasslands) and its members are cryptically coloured in subdued mottling of brown, black and fawn. The second group, which lie in wait in the field and shrub layers are much more brightly coloured to blend in with the foliage or flowers they haunt. It includes genera such as *Thomisus*, *Misumena* and *Diaea*. Among our 26 species of Thomisidae, only seven extend as far north as Scotland and nine (35%) are confined to the area south of the Wash–Severn estuary line. The family includes only two Red Data Book species, *Pistius truncatus* and *Xysticus luctator* but seven other species are listed as Nationally Notable. Among the latter is *Thomisus onustus* (Nb) (Fig. 4), the species which gives its name to the family. It is found on heathland in Dorset, Hampshire and Surrey where the females are to be found on heather and other flowers. They can vary in colour from pink through yellow to white to match the background colour of the flower they sit on. Another much commoner species, *Misumena vatia*, can also change its colour to match that of the flowers it selects to wait for prey on. Both these species can catch and subdue insects many times their own size. It is not uncommon to see a butterfly or hoverfly apparently feeding at a flower but which does not fly off when approached. Closer examination reveals that the prey is held in the jaws of a female *Misumena* while she sucks the body fluids out if it.

The 15 species of Philodromidae again include two different ecological groups. The first, represented by the genera *Philodromus* and *Thanatus*, have body forms similar to our thomisid species as described above. The second, represented by the genus *Tibellus*, includes just two species found in grasslands. They have elongated bodies and are to be found lying along the stems of grasses with the first two pairs of legs held forward and the second two backward so that they are superbly camouflaged, particularly when viewed from above. Whether this cryptic form and behaviour is an adaptation to predator avoidance or to prey capture is not really clear at present. Like the thomisids, a large proportion of philodromids have a southern distribution in Britain with nine of the 15 species found only in the southern half of the country. The family includes one RDB listed and six Nationally Notable species.

While this brief outline includes most of the larger families of spiders, representing about 83% of all our species, there are many smaller families which there is no space to mention here and which include a diversity of body forms, prey capture strategies and ecological adaptations. For those who wish to read more about our fauna, an ideal starting point is Bristowe's (1958) *The world of spiders* in the Collins New Naturalist series. Excellent colour figures of all our larger species can be found in the *Collins field guide: spiders of Britain and northern Europe* by Roberts (1995). Finally, comprehensive information on distribution and individual species accounts can be found in *The provisional atlas of British spiders* edited by P. Harvey *et al.* (2002).

1.7 Human Attitudes to Spiders

Spiders seem to inspire a combination of fascination and fear amongst the public in general. I believe that the fascination is entirely justified by their beauty, complex behaviour, varied lifestyles and extraordinary diversity. On the other hand, fear of spiders, which can range from mild aversion to a fully fledged clinical condition known as arachnophobia, is less easy to explain. It seems unlikely that an innate fear of poisonous bites can really account for such apprehension. Among the 40,000 or so described species of spiders world-wide, only some 20 or 30 are reliably reported to cause seriously dangerous bites to humans and none of these are found in Britain. Unless you happen to have the misfortune to be highly allergic, the most unpleasant symptom you are likely to suffer from a spider bite in this country is a relatively mild and short-lived local reaction, equivalent to a bee or wasp sting. Other reasons that are sometimes put forward for this fear are that they move incredibly fast or are hairy, both of which equally well describe many other animals (such as domestic cats) which rarely inspire similar anxieties. Whatever the causes, innate or learned, most people can overcome their fear of spiders once they become more familiar with them. I hope that this book will help those who have decided to learn more of these fascinating animals to understand them better.

1.8 So, What Next?

If, having read this far, you have decided that spiders are still uninteresting, clearly they may not be for you! By contrast, if this short account has stimulated you to try and find out more about these fascinating and beautiful creatures, you should read on. Although spiders in Britain are probably better known than those of almost any other part of the world, we are surprisingly ignorant about many aspects of the distribution, habitats and biology of our fauna. For example, although the recent *Provisional atlas of British spiders* (Harvey *et al*. 2002) and updated maps to the end of 2005 on the BAS website (see also the NBN Gateway at http://www.searchnbn.net) have added enormously to our knowledge of the overall distribution of all our spider species, there are clearly still considerable gaps. A glance at Figure 5 in this atlas, which shows the distribution of all spider records by 10 km square, shows that there are many areas for which there are no records at all. These include, for example, parts of the Hampshire/Wiltshire borders, much of Exmoor and the inland margin of the Wash in Lincolnshire. The situation is even poorer in many areas of Scotland which is clearly still greatly under-recorded. There is much work for the keen arachnologist to do in both filling these gaps and in recording at a more detailed level (tetrad or even 1 km square), particularly for species that are rare or apparently declining.

As with distribution, our knowledge of the detailed habitat requirements of all but a very few of our spider species is quite limited. Again, a glance at the individual species accounts in the *Provisional Atlas* shows how often habitats are only recorded in the most general of terms. Phrases such as "Species X may be found by beating trees and bushes and by sweeping herbaceous vegetation" or "The spider occurs in a variety of situations, both damp and dry" abound. In Phase 2 of the Spider Recording Scheme, recorders are being asked to note a great deal more about the habitat and micro-habitat in which specimens are collected and it is hoped that this will help to provide a more precise idea of the requirements of our species. This is one way that keen naturalists can help to improve our understanding but beyond that there is a need for detailed studies of individual species, particularly those that are endangered. Once again there is an important role for keen field naturalists in such detailed studies.

Finally, our understanding of the biology of many of our species, whether it be life cycles, web construction and use, feeding habits or courtship and mating behaviour, is woefully inadequate. For those who have the time and patience to observe live spiders, either in the field or captivity, there are important discoveries to be made in all these fields which in many cases can help us understand their conservation needs. For example, despite the fact that the linyphiids include over 40% of our species, very little indeed is known about web construction and use or the range of prey taken in all but a very few of our largest species. Detailed observation of web production and prey taken in captivity could add substantially to our knowledge of this important family. Likewise, among our 38 salticid species, courtship behaviour has only been properly studied in about one third and much could be learned about their relationships from a better understanding of this aspect of their biology. The keen amateur naturalist could potentially make a significant contribution to all of these aspects of spider biology and in doing so help safeguard their populations for the future .

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2. THE BRITISH ARACHNID FAUNA

2.1 Notes on the Commoner Families of Spiders

I. Dawson

These notes are primarily intended for those who are beginning the study of spiders. It is helpful to read them in conjunction with Michael Roberts's (1995) book *Collins field guide: spiders of Britain and northern Europe* which provides both formal keys to the families and coloured plates of all the species mentioned below.

Pholcidae

The daddy-long-legs spider *Pholcus phalangioides* is an indoor spider in the southern half of Britain, commonly seen lurking in a tangle of thin threads in the angle of wall and ceiling (Fig. 13). The very long legs and tubular abdomen are unmistakable. When disturbed, the spider gyrates wildly in its web, becoming a blur. Although looking fragile and clumsy, it is an efficient predator on other spiders, including even the large house spiders (*Tegenaria* spp.) which share our homes.

Segestriidae

The six-eyed *Segestria senoculata* is very common in stone walls in the north and west of Britain; elsewhere the web may be found on walls and tree trunks with careful searching. This is a silken collar at the rim of a hole with a number of single strands of silk radiating, star-like, from it. These are trip lines which alert the spider living in the hole to possible prey. It may be enticed out briefly by tweaking a trip line, but moves very rapidly and realising the deception darts back to safety. *Segestria* has a beautifully adapted tubular body with the first three pairs of legs directed forwards, and a clear zigzag adder pattern on the abdomen.

Dysderidae

The spectacular and rather menacing-looking pink or red *Dysdera crocata* (Fig. 2) is widespread in gardens, though not often seen. It is a specialist predator on woodlice which are rejected by most other spiders, using its fearsome jaws to penetrate the hard cuticle. Its smaller relative *Harpactea hombergi* is frequent in dry litter and under loose bark. In these relatively primitive spiders the female lacks an external epigyne. *Dysdera* has a characteristic slow knock-kneed gait, while the slim, tubular *Harpactea* has a fair turn of speed. Like the next family, these spiders have only six eyes, unlike the usual eight of most spiders.

Oonopidae

These minute, six-eyed spiders are easy to overlook. In Britain there are only two species of *Oonops* and the most likely to be encountered first is *Oonops domesticus* which is found on the internal walls of buildings. Apart from its minute size and pale colouration, it can be recognised by the peculiar jerky gait, with short dashes interspersed with longer periods of very slow walking.

Mimetidae

Two common species of Pirate spiders, *Ero*, turn up occasionally when beating or shaking dry grass. They live solitary lives and move slowly and deliberately, preying on other spiders, usually theridiids. Although quite small, they are recognisable with a lens, having a rather globular abdomen with a pair of humps, long curved spines on the legs, and a well marked pattern on the carapace. The distinctive long-stalked egg sacs are seen as frequently as the spiders.

Uloboridae

The two native British species are both rarities. *Uloborus walckenaerius* is known from a few heathland sites in southern England, while *Hyptiotes paradoxus* (Fig. 7) occurs on yew trees in scattered sites across

England from Dorset as far north as Cumbria. A third species, *Uloborus plumipes*, is a recent colonist and hence is not to be found in any of the standard spider identification books. First found in Britain as recently as the early 1990s, it is now very common in garden centres, and is thought to be native to southern Europe and Africa. This family is unique among the world's spiders in lacking poison glands. *U. plumipes* is a small cryptically coloured spider which weaves a horizontal orb-web, though once you know what to look for it is surprisingly easy to find. The egg-sac, which resembles a small, beige, holly leaf suspended in the web, is often the first indication of its presence. In all British uloborid species the abdomen is triangular in side profile.

Theridiidae

These are the so-called comb-footed spiders, although the comb is actually on the tarsus of the fourth pair of legs and, in any case, is not easy to see. However, the general appearance of most species allows easy recognition to family. Most of the species are small to medium-sized globular spiders with relatively short legs with few or no spines. Many are also colourful, with species-specific abdominal and carapace patterns. Most species build a scaffold web of tangled threads. The infamous Black Widow genus *Latrodectus* belongs to this family, though no members occur in Britain. Indeed, we have no spiders dangerous to man in Britain.

You will soon come across several members of this family. Some of the more obvious include *Paidiscura* (formerly *Theridion*) *pallens* which, despite its tiny size, is instantly recognisable, guarding the bizarre, sputnik-shaped egg-sac several times larger than herself; in June and early July you can almost guarantee finding this species under an oak leaf within a minute of starting to search. Various other *Theridion* species are frequent in a variety of habitats. *Anelosimus vittatus* is a lovely golden-brown spider with dark central band, commonly beaten from oak. In and around houses, *Steatoda bipunctata*, a shining nut-brown globular or slightly flattened spider sometimes with a white or cream band across the front of the abdomen, is common. *Episinus angulatus*, not uncommon in low vegetation, has an abdomen the shape of an isosceles triangle.

The candy-stripe spider *Enoplognatha ovata* comes in three main colour forms: plain cream, with two pink stripes down the sides of the abdomen, or with a single broad carmine central stripe, and is very common in low vegetation in mid to late summer, females guarding their curiously coloured turquoise-blue egg sacs.

Linyphiidae

This is the largest family of spiders in the British fauna, accounting for over 40% of all our species. Most are very small and only a minority can be identified in the field. The money spiders fall very roughly into two groups. Tiny spiders less than 3 mm long, usually dark and lacking any pattern (the archetypal money spiders) and generally larger spiders with a pattern on the abdomen and longer and more spiny legs. Males of the former group often show bizarre modifications of the head which are fascinating when seen under the microscope. Two ubiquitous *Erigone* species can be recognised to genus with a lens by their spike-fringed carapace.

A very common late summer spider is *Linyphia triangularis*, one of our largest 'linys', which can be seen hanging upside down below its sheet web in almost every bush. Apart from the clear black and white pattern on the abdomen, a dark tuning fork mark on the yellowish carapace is distinctive. Other patterned species which are all locally common and can often be identified in the field include *L. hortensis*, *Neriene clathrata*, *N. peltata*, *N. montana* and *Microlinyphia pusilla*.

The sheer numbers of money spiders present is often obvious only in autumn when sheets of spider silk (gossamer) may be seen carpeting the ground in certain weather conditions. Perhaps our commonest and most abundant British spider is *Lepthyphantes tenuis* which can be tentatively identified in the field with its weak abdominal pattern, and relatively long body and legs which bear quite long spines, but there are several similar species, so field identifications always need checking under the microscope. For those who progress to looking at spiders under the microscope, *L. tenuis* is the first spider worth learning, as recognising it quickly will save a great deal of time and anguish!

Tetragnathidae

There are three main genera in this family, all quite recognisable and distinctly different. *Tetragnatha* are the long, thin-bodied spiders with long legs and enormous jaws which are often found hanging in their orb webs near water. Two species, *Tetragnatha extensa* and *T. montana* are common. The three species of *Metellina* also build orb webs and indeed look very like members of the next family, though their webs (and those of *Tetragnatha*) have an open hub (closed in araneid webs). The third genus, *Pachygnatha* has two common species, one small and quite likely to be overlooked, which have abandoned web-making and are ground hunters. Like *Tetragnatha* they have disproportionately large jaws.

Araneidae

The classic orb-web weavers, including some of our largest and most familiar spiders. Though the colours are often very variable within a single species, the abdominal patterns enable most araneids to be identified in the field.

The garden spider *Araneus diadematus* is widespread in gardens and low vegetation across the whole of Britain. When they hatch from the egg, spiderlings are bright yellow with a black patch on the rear of the abdomen, quite unlike the familiar adult with its white abdominal cross which is mature in late summer. *Larinioides cornutus* is a white and greenish patterned spider which can be found in tall grass and herbage by all sorts of wet and damp habitats: opening silken retreats in dead flower heads or in reedheads often reveals this spider. Its darker and larger relative *L. sclopetarius* is common in some areas on bridges and buildings adjacent to water. The impressive black flattened *Nuctenea umbratica* probably lives on every stretch of lap fencing and shed in the country but comes out only at night, when it is easy to find by torchlight. Its natural habitat is under bark. The web of *Nuctenea* has relatively few, widely spaces spirals.

Araniella species are apple green and weave a miniature orb web across a single leaf. Our two common species are closely similar and need microscopic examination to distinguish them. Indeed, they hid under a single name for many years before being recognised as different species. The ubiquitous *Zygiella x-notata* weaves her characteristic web with a missing sector on window frames. The missing slice allows the spider to have her retreat close to the plane of the web. The dark head region contrasting with paler thoracic area, and the yellow centred sternum allow for easy identification, even when the silvery leaf pattern (folium) on the abdomen is indistinct.

Several other species are widespread, including *Araneus quadratus* which holds the title of heaviest British spider. *Zilla diodia*, with its death-mask pattern, sits in the middle of her web with tightly packed spirals, spun often low down in shady places in the south and east. One to look out for is the yellow-and-black wasp spider *Argiope bruennichi* which has been spreading recently from its stronghold on the Hampshire and Dorset coast. It is now known from a good many sites north of the Thames and has recently been reported in Nottinghamshire. It weaves its web low down in rough grassland in late summer. Most araneids sit in a hidden retreat with a silk line leading to the web, but *Argiope* sits out in the middle of her web.

Lycosidae

The wolf spiders, like the jumping spiders, are mostly daytime hunters and also appear to have good vision, though in these it is the posterior median eyes which are the largest. There are several common *Pardosa* species which are to be seen running on the ground in spring. The males only have a short season and die soon after mating. In early summer the females can be seen with their globular egg-sac attached to their spinnerets. When the young spiderlings hatch they are carried around on the female's back for a few days before dispersing.

Several other genera are also common. These include the water-loving *Pirata* species which are a beautiful velvety brown with paired blue or white spots on the abdomen and a darker fork mark on the carapace. *Trochosa* species are larger and relatively shorter-legged nocturnal hunters, recognisable by two short, dark, parallel lines within the pale central carapace band. *Alopecosa pulverulenta* is common in grassland and open habitats, with a distinctive pattern on the carapace and abdomen.

Pisauridae

The Nursery-web Spider *Pisaura mirabilis* is widespread and common in rough grassland. Though superficially recalling a wolf spider, the eyes are smaller and the pattern on the carapace and abdomen is unique to this species, enabling even the tiniest spiderlings to be identified (Fig. 12). The female carries her large egg-sac in her jaws (unlike the wolf spiders which attach their egg-sac to their spinnerets) until the eggs are about to hatch (Fig. 15). She then hangs it near the top of some low vegetation and weaves a conspicuous silken tent (the nursery web) around it to provide protection to the young spiderlings when they hatch (Fig. 16). The two members of the genus *Dolomedes*, the so-called raft spiders are rare, semi-aquatic spiders found in bogs and fens. They are both very large, chocolate-brown spiders with two yellow bands along the abdomen and are capable of catching prey, including small fish, under water.

Agelenidae

Agelena labyrinthica builds a large and spectacular sheet web leading to a funnel retreat low down in rough grass, bramble etc. The spider which has a most attractive herringbone pattern down its abdomen can be seen lurking in the entrance to the funnel. The web is designed to catch grasshoppers. Most of the other members of this family are species of *Tegenaria*, the familiar large hairy house spiders which turn up in the bath. Although several species are common, the vast majority are *Tegenaria gigantea* or, in the West, *T. saeva*.

Hahniidae

These small web-building spiders can easily be mistaken for money spiders (Linyphiidae) but, with the aid of a hand lens, they are readily distinguished by the arrangement of the spinnerets which are placed in a transverse row across the tip of the abdomen. *Hahnia montana* is probably the commonest of our six species and occurs in litter and moss, primarily in woodlands. *Antistea elegans* is found in wet marshy places and is distinguished by the overall reddish colouration of the body.

Dictynidae

Small web-building spiders, the only species likely to be found initially are *Dictyna uncinata*, *D. arundinacea*, and *Lathys humilis*. A clear pattern on the abdomen and carapace formed by dark and pale hairs characterises *Dictyna*, while *Lathys* has a unique abdominal pattern, rather like a tiny *Amaurobius*, and strongly annulated (dark-ringed) legs. *Dictyna* occur typically on bushes, heather and in dead flower heads, *Lathys* on bushes and trees in woodland and hedgerows.

Amaurobiidae

The abdominal pattern and rather large, slightly raised, parallel-sided head region are unmistakeable. However, the key feature alerting us to their presence is the web: a closely woven lacy mesh of bluish silk surrounding a collar of silk lining holes and crevices. Two closely similar common species are *Amaurobius similis* and *A. fenestralis*. The former typically occurs on walls and houses, the latter in more natural habitats such as tree trunks. The spiders are nocturnal and can easily be found by torchlight. By day they can often be enticed out of their retreats by using a tuning fork to simulate a fly in the web.

Anyphaenidae

Superficially very similar to the clubionids, *Anyphaena accentuata* is immediately identifiable at any age by two dark marks resembling circumflex accents on the back of the abdomen. It can be found commonly on the foliage of deciduous trees, where the males drum out a message to the female on a leaf, giving it its popular name of the buzzing spider.

Liocranidae

These mostly have a pattern of different shades of brown on both abdomen and carapace, together with a narrow eye region relative to the width of the carapace. Members of the genus *Agroeca* are found in a range of mainly dry habitats (including heathland and sand dunes) and mature in autumn. They bear a superficial resemblance to lycosids with which they are sometimes confused. The rather atypical-looking ant-mimic *Phrurolithus festivus*, recognisable with a lens by a long bristle projecting forwards from each chelicera, is often loosely associated with colonies of ants. Our other common ant-mimic is the gnaphosid *Micaria pulicaria* which has a beautiful iridescence on the abdomen.

Clubionidae

The genus *Clubiona* comprises some 20 species, many of which are common in a wide range of habitats. Although there is some variation in size and colour, the genus has a very uniform general appearance such that even tiny juveniles are recognisable. However, identifying to species level always requires microscopic examination. They usually occur in a variety of grassy and vegetated habitats (including some on deciduous trees) where they are active nocturnal hunters. Females with egg-sacs can be found in their characteristic leaf folds.

Gnaphosidae

Many gnaphosids have a dense coat of smooth silky hairs, looking almost like fur. The mouse spider *Scotophaeus blackwalli* occurs mainly indoors and may be seen after dark on walls and ceilings, where it is an active hunter. The paler *Drassodes lapidosus* lives in a silken cell under large stones (and sometimes in the gap below opening windows in our homes), emerging at night to hunt. Bristowe found that *Drassodes* was a fierce and skilful predator, able to overcome almost any other spider. Members of the genus *Zelotes* are jet black in colour and are fast moving night-time hunters often found under stones in dry habitats.

This and the next family are rather similar-looking spindle-shaped spiders which can often move rapidly. Although the book keys use the shape and arrangement of the spinnerets as the main separating character, I find the eye arrangement a very useful check. In Gnaphosids the posterior median eyes are close together and in most species are oval or slit-shaped whilst in Clubionids these eyes are widely separated and usually closer to the laterals than each other (naph = near, club = clear) and moreover they are always round.

Zoridae

Zora spinimana is another active hunting spider, rather like a cross between a small well-patterned *Clubiona* and a wolf-spider. The strongly contrasting stripes on the very pointed carapace make for simple recognition. It occurs in dry grasses in a variety of habitats where its yellow and brown colouration makes for good camouflage.

Philodromidae

Formerly included with the Thomisidae, these are the running crab spiders, and are usually beaten from tree foliage. The long legs, held out from the body, are more equal in length and diameter than in the thomisids, and, as both the scientific and English names imply, they can run extremely rapidly! Several species of *Philodromus* are common and widespread but, although simple to assign to genus even as spiderlings, the adults are probably among the trickiest groups of species in Britain to identify confidently. Also in this family are our two *Tibellus* species, elongated straw-coloured spiders which live in lush grassy habitats and which may at first sight recall a *Tetragnatha*.

Thomisidae

Crab spiders are so-called because of their superficial resemblance to the crustacean. The first two pairs of legs are disproportionately long and stout and all the legs are held in a crab-like posture. They are usually sluggish, ambush predators with a powerful toxin capable of immobilising even large insects such as bees almost instantaneously. *Misumena vatia* is able to change colour to match the white or yellow flower head it sits in wait on. *Diaea dorsata* is one of our more colourful spiders with a bright green cephalothorax and legs which, presumably, help to disguise it in its favoured tree foliage habitat. The largest genus in Britain is *Xysticus*, two species of which are often swept from vegetation. *Ozyptila* species are smaller and tend to occur at ground level, often in damp habitats. The shape of the initial letters is a useful mnemonic for separating these two genera. *Xysticus* (X) tend to have a pattern of angled lines and the bristles on the abdomen and carapace are pointed, while *Ozyptila* (O) have a pattern of swirling lines and blunt or club-tipped bristles.

Salticidae

The zebra spider *Salticus scenicus*, with its black and white pattern and habit of living on sunny walls and buildings, is familiar to nearly everyone. Jumping spiders are daytime predators and have been shown to have excellent binocular vision with their huge forward-facing, anterior median eyes. Indeed, by watching one closely it is possible to see the eyes focusing – it is fascinating to watch the spider watching you back. Again, as the name suggests, some species – including the Zebra Spider – are good jumpers, but not every salticid can jump. Most of our jumping spiders have a markedly southern distribution in Britain and many of the species are rare or very local. *Heliophanus flavipes* and *H. cupreus* are small, very dark green (appearing almost black) jumping spiders with paler legs and are both widespread in grassland and other dry habitats. Another common small species, *Euophrys frontalis*, has a mottled abdomen and bright orange hairs around the eyes. It occurs in a wide range of habitats.

2.2 Checklist of British Spiders

This list is based on Merrett & Murphy (2000), but has been updated to include additional species and some taxonomic changes since then.

Scientific name and authority	Earlier name(s) and comments
Atypidae	
Atypus affinis Eichwald, 1830	
Scytodidae	
Scytodes thoracica (Latreille, 1802)	
Pholcidae	
Pholcus phalangioides (Fuesslin, 1775)	
Psilochorus simoni (Berland, 1911)	*Physocyclus simoni*
Segestriidae	
Segestria senoculata (Linnaeus, 1758)	
Segestria bavarica C. L. Koch, 1843	
Segestria florentina (Rossi, 1790)	
Dysderidae	
Dysdera erythrina (Walckenaer, 1802)	
Dysdera crocata C. L. Koch, 1838	
Harpactea hombergi (Scopoli, 1763)	
Harpactea rubicunda (C. L. Koch, 1838)	
Oonopidae	
Oonops pulcher Templeton, 1835	
Oonops domesticus Dalmas, 1916	
Orchestina sp.?	recorded in Essex in 1992
Mimetidae	
Ero cambridgei Kulczyński, 1911	
Ero furcata (Villers, 1789)	
Ero aphana (Walckenaer, 1802)	
Ero tuberculata (De Geer, 1778)	

Eresidae

Eresus sandaliatus (Martini & Goeze, 1778)	*E. cinnaberinus*, *E. niger*

Uloboridae

Uloborus walckenaerius Latreille, 1806
Hyptiotes paradoxus (C. L. Koch, 1834)

Nesticidae

Nesticus cellulanus (Clerck, 1757)

Theridiidae

Episinus angulatus (Blackwall, 1836)	
Episinus truncatus Latreille, 1809	
Episinus maculipes Cavanna, 1876	
Euryopis flavomaculata (C. L. Koch, 1836)	
Dipoena erythropus (Simon, 1881)	
Dipoena prona (Menge, 1868)	
Dipoena inornata (O. P.-Cambridge, 1861)	
Dipoena tristis (Hahn, 1833)	
Dipoena coracina (C. L. Koch, 1837)	
Dipoena melanogaster (C. L. Koch, 1837)	
Dipoena torva (Thorell, 1875)	
Crustulina guttata (Wider, 1834)	
Crustulina sticta (O. P.-Cambridge, 1861)	
Steatoda phalerata (Panzer, 1801)	*Asagena phalerata*
Steatoda albomaculata (De Geer, 1778)	*Lithyphantes albomaculatus*
Steatoda bipunctata (Linnaeus, 1758)	
Steatoda grossa (C. L. Koch, 1838)	*Teutana grossa*
Steatoda nobilis (Thorell, 1875)	
Steatoda triangulosa (Walckenaer, 1802)	found in Leicester in 1996
Anelosimus vittatus (C. L. Koch, 1836)	*Theridion vittatum*
Anelosimus aulicus (C. L. Koch, 1838)	*Theridion aulicum*
Achaearanea lunata (Clerck, 1757)	*Theridion lunatum*
Achaearanea riparia (Blackwall, 1834)	*Theridion saxatile*
Achaearanea tepidariorum (C. L. Koch, 1841)	*Theridion tepidariorum*
Achaearanea simulans (Thorell, 1875)	*Theridion tepidariorum simulans*
Achaearanea veruculata (Urquhart, 1885)	
Theridion sisyphium (Clerck, 1757)	
Theridion impressum L. Koch, 1881	
Theridion pictum (Walckenaer, 1802)	
Theridion hemerobium (Walckenaer, 1802)	described from Britain in 1994
Theridion varians Hahn, 1833	
Theridion pinastri L. Koch, 1872	
Theridion familiare O. P.-Cambridge, 1871	
Theridion melanurum Hahn, 1831	*T. denticulatum*
Theridion mystaceum L. Koch, 1870	
Theridion blackwalli O. P.-Cambridge, 1871	
Theridion tinctum (Walckenaer, 1802)	
Simitidion simile (C. L. Koch, 1836)	*Theridion simile*
Neottiura bimaculata (Linnaeus, 1767)	*Theridion bimaculatum*
Paidiscura pallens (Blackwall, 1834)	*Theridion pallens*
Rugathodes instabilis (O. P.-Cambridge, 1871)	*Theridion instabile*

Rugathodes bellicosus (Simon, 1873)	*Theridion bellicosum*
Enoplognatha ovata (Clerck, 1757)	*Theridion ovatum*
Enoplognatha latimana Hippa & Oksala, 1982	
Enoplognatha thoracica (Hahn, 1833)	
Enoplognatha mordax (Thorell, 1875)	*E. schaufussi, E. crucifera*
Enoplognatha tecta (Keyserling, 1884)	*E. caricis* sensu auct. not (Fickert)
Enoplognatha oelandica (Thorell, 1875)	*E. mandibularis*
Robertus lividus (Blackwall, 1836)	
Robertus arundineti (O. P.-Cambridge, 1871)	
Robertus neglectus (O. P.-Cambridge, 1871)	
Robertus scoticus Jackson, 1914	
Robertus insignis O. P.-Cambridge, 1907	
Pholcomma gibbum (Westring, 1851)	
Theonoe minutissima (O. P.-Cambridge, 1879)	

Theridiosomatidae

Theridiosoma gemmosum (L. Koch, 1877)

Linyphiidae

Ceratinella brevipes (Westring, 1851)	
Ceratinella brevis (Wider, 1834)	
Ceratinella scabrosa (O. P.-Cambridge, 1871)	
Walckenaeria acuminata Blackwall, 1833	*Walckenaera acuminata*
Walckenaeria mitrata (Menge, 1868)	*Walckenaera mitrata*
Walckenaeria antica (Wider, 1834)	*Wideria antica*
Walckenaeria alticeps (Denis, 1952)	
Walckenaeria cucullata (C. L. Koch, 1836)	*Wideria cucullata*
Walckenaeria nodosa O. P.-Cambridge, 1873	*Wideria nodosa*
Walckenaeria atrotibialis (O. P.-Cambridge, 1878)	*Wideria melanocephala*
Walckenaeria capito (Westring, 1861)	*Wideria capito*
Walckenaeria incisa (O. P.-Cambridge, 1871)	*Wideria polita*, *Prosopotheca incisa*
Walckenaeria dysderoides (Wider, 1834)	*Wideria fugax*
Walckenaeria stylifrons (O. P.-Cambridge, 1875)	*Wideria stylifrons*
Walckenaeria nudipalpis (Westring, 1851)	*Trachynella nudipalpis*
Walckenaeria obtusa Blackwall, 1836	*Trachynella obtusa*
Walckenaeria monoceros (Wider, 1834)	*Prosopotheca monoceros*
Walckenaeria corniculans (O. P.-Cambridge, 1875)	*Prosopotheca corniculans*
Walckenaeria furcillata (Menge, 1869)	*Tigellinus furcillatus*
Walckenaeria unicornis O. P.-Cambridge, 1861	*Cornicularia unicornis*
Walckenaeria kochi (O. P.-Cambridge, 1872)	*Cornicularia kochi*
Walckenaeria clavicornis (Emerton, 1882)	*Cornicularia karpinskii*
Walckenaeria cuspidata Blackwall, 1833	*Cornicularia cuspidata*
Walckenaeria vigilax (Blackwall, 1853)	*Cornicularia vigilax*
Dicymbium nigrum (Blackwall, 1834)	
Dicymbium brevisetosum Locket, 1962	*D. nigrum* form *brevisetosum*
Dicymbium tibiale (Blackwall, 1836)	
Entelecara acuminata (Wider, 1834)	
Entelecara congenera (O. P.-Cambridge, 1879)	
Entelecara erythropus (Westring, 1851)	
Entelecara flavipes (Blackwall, 1834)	
Entelecara omissa O. P.-Cambridge, 1902	

Entelecara errata O. P.-Cambridge, 1913	
Moebelia penicillata (Westring, 1851)	
Hylyphantes graminicola (Sundevall, 1830)	*Erigonidium graminicola*
Gnathonarium dentatum (Wider, 1834)	
Trematocephalus cristatus (Wider, 1834)	
Tmeticus affinis (Blackwall, 1855)	
Gongylidium rufipes (Linnaeus, 1758)	
Dismodicus bifrons (Blackwall, 1841)	
Dismodicus elevatus (C. L. Koch, 1838)	
Hypomma bituberculatum (Wider, 1834)	
Hypomma fulvum (Bösenberg, 1902)	
Hypomma cornutum (Blackwall, 1833)	
Metopobactrus prominulus (O. P.-Cambridge, 1872)	
Hybocoptus decollatus (Simon, 1881)	
Baryphyma pratense (Blackwall, 1861)	*B. pratensis*
Baryphyma gowerense (Locket, 1965)	*Acanthophyma gowerensis, Lasiargus gowerensis*
Baryphyma trifrons (O. P.-Cambridge, 1863)	*Minyrioloides trifrons*
Baryphyma maritimum (Crocker & Parker, 1970)	*Minyrioloides maritimus*
Praestigia duffeyi (Millidge, 1954)	*Baryphyma duffeyi*
Gonatium rubens (Blackwall, 1833)	
Gonatium rubellum (Blackwall, 1841)	
Gonatium paradoxum (L. Koch, 1869)	*Gonatium corallipes*
Maso sundevalli (Westring, 1851)	
Maso gallicus Simon, 1894	*Maso gallica*
Minicia marginella (Wider, 1834)	
Peponocranium ludicrum (O. P.-Cambridge, 1861)	
Pocadicnemis pumila (Blackwall, 1841)	
Pocadicnemis juncea Locket & Millidge, 1953	*P. pumila* var. *juncea*
Hypselistes jacksoni (O. P.-Cambridge, 1902)	
Oedothorax gibbosus (Blackwall, 1841)	includes *O. tuberosus*
Oedothorax fuscus (Blackwall, 1834)	
Oedothorax agrestis (Blackwall, 1853)	
Oedothorax retusus (Westring, 1851)	
Oedothorax apicatus (Blackwall, 1850)	
Trichopterna thorelli (Westring, 1861)	
Trichopterna cito (O. P.-Cambridge, 1872)	
Pelecopsis mengei (Simon, 1884)	*Trichopterna mengei*
Pelecopsis parallela (Wider, 1834)	*Lophocarenum parallelum*
Pelecopsis nemoralis (Blackwall, 1841)	*Lophocarenum nemorale*
Pelecopsis nemoralioides (O. P.-Cambridge, 1884)	*Lophocarenum stramineum*, *P. mediocris*, *P. locketi*
Pelecopsis elongata (Wider, 1834)	*Lophocarenum elongatum*
Pelecopsis radicicola (L. Koch, 1872)	*Lophocarenum radicicola*
Silometopus elegans (O. P.-Cambridge, 1872)	
Silometopus ambiguus (O. P.-Cambridge, 1905)	includes *S. curtus*
Silometopus reussi (Thorell, 1871)	*S. interjectus*
Silometopus incurvatus (O. P.-Cambridge, 1873)	
Mecopisthes peusi Wunderlich, 1972	*M. pusillus*
Cnephalocotes obscurus (Blackwall, 1834)	
Acartauchenius scurrilis (O. P.-Cambridge, 1872)	
Trichoncus saxicola (O. P.-Cambridge, 1861)	

Trichoncus hackmani Millidge, 1955	
Trichoncus affinis Kulczyński, 1894	
Ceratinopsis romana (O. P.-Cambridge, 1872)	*Styloctector romanus*
Ceratinopsis stativa (Simon, 1881)	*Anacotyle stativa*
Evansia merens O. P.-Cambridge, 1900	
Tiso vagans (Blackwall, 1834)	
Tiso aestivus (L. Koch, 1872)	
Troxochrus scabriculus (Westring, 1851)	includes *T. cirrifrons*
Minyriolus pusillus (Wider, 1834)	
Tapinocyba praecox (O. P.-Cambridge, 1873)	
Tapinocyba pallens (O. P.-Cambridge, 1872)	
Tapinocyba insecta (L. Koch, 1869)	
Tapinocyba mitis (O. P.-Cambridge, 1882)	
Tapinocyboides pygmaeus (Menge, 1869)	*T. pygmaea*, *T. antepenultima*
Microctenonyx subitaneus (O. P.-Cambridge, 1875)	*Aulacocyba subitanea*
Satilatlas britteni (Jackson, 1913)	*Perimones britteni*
Thyreosthenius parasiticus (Westring, 1851)	
Thyreosthenius biovatus (O. P.-Cambridge, 1875)	
Monocephalus fuscipes (Blackwall, 1836)	
Monocephalus castaneipes (Simon, 1884)	
Lophomma punctatum (Blackwall, 1841)	
Saloca diceros (O. P.-Cambridge, 1871)	
Gongylidiellum vivum (O. P.-Cambridge, 1875)	
Gongylidiellum latebricola (O. P.-Cambridge, 1871)	
Gongylidiellum murcidum Simon, 1884	
Micrargus herbigradus (Blackwall, 1854)	
Micrargus apertus (O. P.-Cambridge, 1871)	*M. herbigradus* in part
Micrargus subaequalis (Westring, 1851)	
Micrargus laudatus (O. P.-Cambridge, 1881)	
Notioscopus sarcinatus (O. P.-Cambridge, 1872)	
Glyphesis cottonae (La Touche, 1945)	
Glyphesis servulus (Simon, 1881)	
Erigonella hiemalis (Blackwall, 1841)	
Erigonella ignobilis (O. P.-Cambridge, 1871)	
Savignia frontata Blackwall, 1833	
Diplocephalus cristatus (Blackwall, 1833)	
Diplocephalus permixtus (O. P.-Cambridge, 1871)	
Diplocephalus latifrons (O. P.-Cambridge, 1863)	
Diplocephalus connatus Bertkau, 1889	*D. jacksoni*, *D. adjacens*
Diplocephalus picinus (Blackwall, 1841)	
Diplocephalus protuberans (O. P.-Cambridge, 1875)	
Araeoncus humilis (Blackwall, 1841)	
Araeoncus crassiceps (Westring, 1861)	
Panamomops sulcifrons (Wider, 1834)	
Lessertia dentichelis (Simon, 1884)	
Scotinotylus evansi (O. P.-Cambridge, 1894)	*Caledonia evansi*
Typhochrestus digitatus (O. P.-Cambridge, 1872)	*Typhocrestus digitatus*
Typhochrestus simoni de Lessert, 1907	
Milleriana inerrans (O. P.-Cambridge, 1885)	*Scotargus inerrans*
Diplocentria bidentata (Emerton, 1882)	

Wabasso replicatus (Holm, 1950)	first recorded from Britain in 1999
Erigone dentipalpis (Wider, 1834)	
Erigone atra Blackwall, 1833	
Erigone promiscua (O. P.-Cambridge, 1872)	
Erigone arctica (White, 1852)	
Erigone longipalpis (Sundevall, 1830)	
Erigone tirolensis L. Koch, 1872	
Erigone capra Simon, 1884	
Erigone welchi Jackson, 1911	
Erigone psychrophila Thorell, 1871	
Erigone aletris Crosby & Bishop	
Prinerigone vagans Audouin, 1826	*Erigone vagans*
Mecynargus morulus (O. P.-Cambridge, 1873)	*Rhaebothorax morulus*
Mecynargus paetulus (O. P.-Cambridge, 1875)	*Rhaebothorax paetulus*
Latithorax faustus (O. P.-Cambridge, 1900)	*Eboria fausta*
Semljicola caliginosus (Falconer, 1910)	*Eboria caliginosa*
Donacochara speciosa (Thorell, 1875)	
Leptorhoptrum robustum (Westring, 1851)	
Drepanotylus uncatus (O. P.-Cambridge, 1873)	
Leptothrix hardyi (Blackwall, 1850)	*Phaulothrix hardyi*
Hilaira excisa (O. P.-Cambridge, 1871)	
Hilaira frigida (Thorell, 1872)	
Hilaira nubigena Hull, 1911	
Hilaira pervicax Hull, 1908	
Halorates reprobus (O. P.-Cambridge, 1879)	
Halorates distinctus (Simon, 1884)	*Collinsia distincta*
Halorates holmgreni (Thorell, 1871)	*Collinsia holmgreni*
Carorita limnaea (Crosby & Bishop, 1927)	
Carorita paludosa Duffey, 1971	
Wiehlea calcarifera (Simon, 1884)	*Gongylidiellum calcariferum*
Mioxena blanda (Simon, 1884)	
Caviphantes saxetorum (Hull, 1916)	*Lessertiella saxetorum*
Asthenargus paganus (Simon, 1884)	
Jacksonella falconeri (Jackson, 1908)	
Pseudomaro aenigmaticus Denis, 1966	
Ostearius melanopygius (O. P.-Cambridge, 1879)	
Aphileta misera (O. P.-Cambridge, 1882)	*Hillhousia misera*
Porrhomma pygmaeum (Blackwall, 1834)	
Porrhomma convexum (Westring, 1851)	
Porrhomma rosenhaueri (L. Koch, 1872)	
Porrhomma pallidum Jackson, 1913	
Porrhomma campbelli F. O. P.-Cambridge, 1894	
Porrhomma microphthalmum (O. P.-Cambridge, 1871)	
Porrhomma errans (Blackwall, 1841)	
Porrhomma egeria Simon, 1884	
Porrhomma oblitum (O. P.-Cambridge, 1871)	
Porrhomma cambridgei Merrett, 1994	as *Porrhomma* sp. in previous list
Porrhomma montanum Jackson, 1913	
Agyneta subtilis (O. P.-Cambridge, 1863)	
Agyneta conigera (O. P.-Cambridge, 1863)	

Agyneta decora (O. P.-Cambridge, 1871)	
Agyneta cauta (O. P.-Cambridge, 1902)	
Agyneta olivacea (Emerton, 1882)	
Agyneta ramosa Jackson, 1912	
Meioneta innotabilis (O. P.-Cambridge, 1863)	*Syedrula innotabilis*
Meioneta rurestris (C. L. Koch, 1836)	
Meioneta mollis (O. P.-Cambridge, 1871)	
Meioneta saxatilis (Blackwall, 1844)	
Meioneta mossica Schikora, 1993	described from Britain in 1993
Meioneta simplicitarsis (Simon, 1884)	
Meioneta beata (O. P.-Cambridge, 1906)	
Meioneta fuscipalpa (C. L. Koch, 1836)	found in Suffolk in 1998
Meioneta gulosa (L. Koch, 1869)	
Meioneta nigripes (Simon, 1884)	
Microneta viaria (Blackwall, 1841)	
Maro minutus O. P.-Cambridge, 1906	
Maro sublestus Falconer, 1915	
Maro lepidus Casemir, 1961	
Syedra gracilis (Menge, 1869)	
Centromerus sylvaticus (Blackwall, 1841)	
Centromerus prudens (O. P.-Cambridge, 1873)	includes *C. parkeri* & *C. subacutus*
Centromerus arcanus (O. P.-Cambridge, 1873)	
Centromerus levitarsis (Simon, 1884)	
Centromerus dilutus (O. P.-Cambridge, 1875)	includes *C. tantulus* & *C. laevitarsis*
Centromerus capucinus (Simon, 1884)	
Centromerus incilium (L. Koch, 1881)	
Centromerus semiater (L. Koch, 1879)	*C. incultus*
Centromerus brevivulvatus Dahl, 1912	*C. aequalis*
Centromerus serratus (O. P.-Cambridge, 1875)	
Centromerus albidus Simon, 1929	
Centromerus cavernarum (L. Koch, 1872)	*C. jacksoni*
Centromerus persimilis (O. P.-Cambridge, 1912)	
Centromerus minutissimus Merrett & Powell, 1993	as *Centromerus* sp. in previous list
Tallusia experta (O. P.-Cambridge, 1871)	*Centromerus expertus*
Centromerita bicolor (Blackwall, 1833)	
Centromerita concinna (Thorell, 1875)	
Sintula corniger (Blackwall, 1856)	*S. cornigera*
Oreonetides vaginatus (Thorell, 1872)	
Saaristoa abnormis (Blackwall, 1841)	*Oreonetides abnormis*
Saaristoa firma (O. P.-Cambridge, 1905)	*Oreonetides firmus*
Macrargus rufus (Wider, 1834)	
Macrargus carpenteri (O. P.-Cambridge, 1894)	*M. rufus carpenteri*
Bathyphantes approximatus (O. P.-Cambridge, 1871)	
Bathyphantes gracilis (Blackwall, 1841)	
Bathyphantes parvulus (Westring, 1851)	
Bathyphantes nigrinus (Westring, 1851)	
Bathyphantes setiger F. O. P.-Cambridge, 1894	
Kaestneria dorsalis (Wider, 1834)	*Bathyphantes dorsalis*
Kaestneria pullata (O. P.-Cambridge, 1863)	*Bathyphantes pullatus*
Diplostyla concolor (Wider, 1834)	*Bathyphantes concolor*

Poeciloneta variegata (Blackwall, 1841)	*Poecilonota globosa*
Drapetisca socialis (Sundevall, 1833)	
Tapinopa longidens (Wider, 1834)	
Floronia bucculenta (Clerck, 1757)	
Taranucnus setosus (O. P.-Cambridge, 1863)	
Labulla thoracica (Wider, 1834)	
Stemonyphantes lineatus (Linnaeus, 1758)	
Bolyphantes luteolus (Blackwall, 1833)	
Bolyphantes alticeps (Sundevall, 1833)	
Nothophantes horridus Merrett & Stevens, 1995	as *Lepthyphantes* sp. in previous list
Megalepthyphantes nebulosus (Sundevall, 1830)	*Lepthyphantes nebulosus*
Megalepthyphantes sp. n.	discovered in Kent in 1999
Lepthyphantes leprosus (Ohlert, 1865)	
Lepthyphantes minutus (Blackwall, 1833)	
Agnyphantes expunctus (O. P.-Cambridge, 1875)	*Lepthyphantes expunctus*
Improphantes complicatus (Emerton, 1882)	*L. umbraticola* & *L. audax*
Mughiphantes whymperi (F. O. P.-Cambridge, 1894(	*Lepthyphantes whymperi*
Obscuriphantes obscurus (Blackwall, 1841)	*Lepthyphantes obscurus*
Oryphantes angulatus (O. P.-Cambridge, 1881)	*Lepthyphantes angulatus*
Palliduphantes ericaeus (Blackwall, 1853)	*Lepthyphantes ericaeus*
Palliduphantes pallidus (O. P.-Cambridge, 1871)	*Lepthyphantes pallidus*
Palliduiphantes insignis (O. P.-Cambridge, 1913)	*Lepthyphantes insignis*
Palliduphantes antroniensis (Schenkel, 1933)	*Lepthyphantes antroniensis*
Piniphantes pinicola (Simon, 1884)	*Lepthyphantes pinicola*
Tenuiphantes alacris (Blackwall, 1853)	*Lepthyphantes alacris*
Tenuiphantes tenuis (Blackwall, 1852)	*Lepthyphantes tenuis*
Tenuiphantes zimmermanni (Bertkau, 1890)	*Lepthyphantes zimmermanni*
Tenuiphantes cristatus (Menge, 1866)	*Lepthyphantes crsitatus*
Tenuiphantes mengei Kulczyński, 1887	*Lepthyphantes mengei*
Tenuiphantes flavipes (Blackwall, 1854)	*Lepthyphantes flavipes*
Tenuiphantes tenebricola (Wider, 1834)	*Lepthyphantes tenebricola*
Midia midas (Simon, 1884)	*Lepthyphantes carri*
Helophora insignis (Blackwall, 1841)	
Pityohyphantes phrygianus (C. L. Koch, 1836)	
Linyphia triangularis (Clerck, 1757)	
Linyphia hortensis Sundevall, 1830	
Neriene montana (Clerck, 1757)	*Linyphia montana*
Neriene clathrata (Sundevall, 1830)	*Linyphia clathrata*
Neriene peltata (Wider, 1834)	*Linyphia peltata*
Neriene emphana (Walckenaer, 1841)	found on the Isle of Wight in 2000
Neriene furtiva (O. P.-Cambridge, 1871)	*Linyphia furtiva*
Neriene radiata (Walckenaer, 1841)	*Linyphia marginata*
Frontinellina frutetorum (C. L. Koch, 1834)	
Microlinyphia pusilla (Sundevall, 1830)	*Linyphia pusilla*
Microlinyphia impigra (O. P.-Cambridge, 1871)	*Linyphia impigra*
Allomengea scopigera (Grube, 1859)	*Mengea scopigera*
Allomengea vidua (L. Koch, 1879)	*Mengea warburtoni*

Tetragnathidae

Tetragnatha extensa (Linnaeus, 1758)
Tetragnatha pinicola L. Koch, 1870

Tetragnatha montana Simon, 1874	
Tetragnatha obtusa C. L. Koch, 1837	
Tetragnatha nigrita Lendl, 1886	
Tetragnatha striata L. Koch, 1862	*Eugnatha striata*
Pachygnatha clercki Sundevall, 1823	
Pachygnatha listeri Sundevall, 1830	
Pachygnatha degeeri Sundevall, 1830	
Metellina segmentata (Clerck, 1757)	*Meta segmentata*
Metellina mengei (Blackwall, 1869)	*Meta mengei*
Metellina merianae (Scopoli, 1763)	*Meta merianae*
Meta menardi (Latreille, 1804)	
Meta bourneti Simon, 1922	

Araneidae

Gibbaranea bituberculata (Walckenaer, 1802)	*Araneus bituberculatus*
Gibbaranea gibbosa (Walckenaer, 1802)	*Araneus gibbosus*
Araneus angulatus Clerck, 1757	
Araneus diadematus Clerck, 1757	
Araneus quadratus Clerck, 1757	
Araneus marmoreus Clerck, 1757	
Araneus alsine (Walckenaer, 1802)	
Araneus sturmi (Hahn, 1831)	*Atea sturmi*
Araneus triguttatus (Fabricius, 1775)	*Atea triguttata*
Larinioides cornutus (Clerck, 1757)	*Araneus cornutus*
Larinioides sclopetarius (Clerck, 1757)	*Araneus sclopetarius*
Larinioides patagiatus (Clerck, 1757)	*Araneus patagiatus*
Nuctenea umbratica (Clerck, 1757)	*Araneus umbraticus*
Agalenatea redii (Scopoli, 1763)	*Araneus redii*
Neoscona adianta (Walckenaer, 1802)	*Araneus adiantus*
Araniella cucurbitina (Clerck, 1757)	*Araneus cucurbitinus*
Araniella opisthographa (Kulczyński, 1905)	*Araneus cucurbitinus opistographus*
Araniella inconspicua (Simon, 1874)	*Araneus inconspicuus*
Araniella alpica (L. Koch, 1869)	*Araneus alpicus*
Araniella displicata (Hentz, 1847)	*Araneus displicatus*
Zilla diodia (Walckenaer, 1802)	
Hypsosinga albovittata (Westring, 1851)	*Singa albovittata*
Hypsosinga pygmaea (Sundevall, 1831)	*Singa pygmaea*
Hypsosinga sanguinea (C. L. Koch, 1844)	*Singa sanguinea*
Hypsosinga heri (Hahn, 1831)	*Singa heri*
Singa hamata (Clerck, 1757)	
Cercidia prominens (Westring, 1851)	
Zygiella x-notata (Clerck, 1757)	
Zygiella atrica (C. L. Koch, 1845)	
Zygiella stroemi (Thorell, 1870)	
Mangora acalypha (Walckenaer, 1802)	
Cyclosa conica (Pallas, 1772)	
Argiope bruennichi (Scopoli, 1772)	

Lycosidae

Pardosa agricola (Thorell, 1856)	includes *P. arenicola*
Pardosa agrestis (Westring, 1861)	*Lycosa agrestis*

Pardosa purbeckensis F. O. P.-Cambridge, 1895	*Lycosa purbeckensis*
Pardosa monticola (Clerck, 1757)	*Lycosa monticola*
Pardosa palustris (Linnaeus, 1758)	*Lycosa tarsalis*
Pardosa pullata (Clerck, 1757)	*Lycosa pullata*
Pardosa prativaga (L. Koch, 1870)	*Lycosa prativaga*
Pardosa amentata (Clerck, 1757)	*Lycosa amentata*
Pardosa nigriceps (Thorell, 1856)	*Lycosa nigriceps*
Pardosa lugubris (Walckenaer, 1802)	described from Britain in 2002
Pardosa saltans Topfer-Hofmann, 2000	*Lycosa lugubris* in part
Pardosa hortensis (Thorell, 1872)	*Lycosa hortensis*
Pardosa proxima (C. L. Koch, 1847)	*Lycosa proxima*
Pardosa traillі (F. O. P.-Cambridge, 1873)	*Lycosa trailli*
Pardosa paludicola (Clerck, 1757)	*Lycosa paludicola*
Hygrolycosa rubrofasciata (Ohlert, 1865)	*Lycosa rubrofasciata*
Xerolycosa nemoralis (Westring, 1861)	
Xerolycosa miniata (C. L. Koch, 1834)	
Alopecosa pulverulenta (Clerck, 1757)	*Tarentula pulverulenta*
Alopecosa cuneata (Clerck, 1757)	*Tarentula cuneata*
Alopecosa barbipes (Sundevall, 1833)	*T. barbipes*, *Alopecosa accentuata*
Alopecosa fabrilis (Clerck, 1757)	*Tarentula fabrilis*
Trochosa ruricola (Degeer, 1778)	
Trochosa robusta (Simon, 1876)	
Trochosa terricola Thorell, 1856	
Trochosa spinipalpis (F. O. P.-Cambridge, 1895)	
Arctosa fulvolineata (Lucas, 1846)	*Trochosa fulvolineata*
Arctosa perita (Latreille, 1799)	
Arctosa leopardus (Sundevall, 1833)	
Arctosa cinerea (Fabricius, 1777)	
Arctosa alpigena (Doleschall, 1852)	*Tricca alpigena*
Pirata piraticus (Clerck, 1757)	
Pirata tenuitarsis Simon, 1876	
Pirata hygrophilus Thorell, 1872	
Pirata uliginosus (Thorell, 1856)	
Pirata latitans (Blackwall, 1841)	
Pirata piscatorius (Clerck, 1757)	
Aulonia albimana (Walckenaer, 1805)	

Pisauridae

Pisaura mirabilis (Clerck, 1757)
Dolomedes fimbriatus (Clerck, 1757)
Dolomedes plantarius (Clerck, 1757)

Oxyopidae

Oxyopes heterophthalmus Latreille, 1804

Agelenidae

Agelena labyrinthica (Clerck, 1757)	
Textrix denticulata (Olivier, 1789)	
Tegenaria gigantea Chamberlin & Ivie, 1935	*T. duellica*, *T. propinqua*, *T. atrica* in part
Tegenaria saeva Blackwall, 1844	*T. atrica* in part
Tegenaria atrica C. L. Koch, 1843	*T. larva*
Tegenaria parietina (Fourcroy, 1785)	

Tegenaria ferruginea (Panzer, 1804)	described from Britain in 1999
Tegenaria agrestis (Walckenaer, 1802)	
Tegenaria domestica (Clerck, 1757)	
Tegenaria silvestris L. Koch, 1872	
Tegenaria picta Simon, 1870	

Cybaeidae

Argyroneta aquatica (Clerck, 1757)

Hahniidae

Antistea elegans (Blackwall, 1841)
Hahnia montana (Blackwall, 1841)
Hahnia candida Simon, 1875
Hahnia microphthalma Snazell & Duffey, 1980
Hahnia nava (Blackwall, 1841)
Hahnia helveola Simon, 1875
Hahnia pusilla C. L. Koch, 1841

Dictynidae

Dictyna arundinacea (Linnaeus, 1758)	
Dictyna pusilla Thorell, 1856	
Dictyna major Menge, 1869	
Dictyna uncinata Thorell, 1856	
Dictyna latens (Fabricius, 1775)	
Nigma puella (Simon, 1870)	*Dictyna puella*
Nigma walckenaeri (Roewer, 1951)	*Dictyna viridissima*
Cicurina cicur (Fabricius, 1793)	
Cryphoeca silvicola (C. L. Koch, 1834)	
Tuberta maerens (O. P.-Cambridge, 1863)	*T. moerens*
Mastigusa arietina (Thorell, 1871)	*Tetrilus arietinus*
Mastigusa macrophthalma (Kulczyński, 1897)	*Tetrilus macrophthalmus*
Lathys humilis (Blackwall, 1855)	
Lathys nielseni (Schenkel, 1932)	
Lathys stigmatisata (Menge, 1869)	
Argenna subnigra (O. P.-Cambridge, 1861)	
Argenna patula (Simon, 1874)	*Protadia patula*
Altella lucida (Simon, 1874)	

Amaurobiidae

Amaurobius fenestralis (Stroem, 1768)	*Ciniflo fenestralis*
Amaurobius similis (Blackwall, 1861)	*Ciniflo similis*
Amaurobius ferox (Walckenaer, 1830)	*Ciniflo ferox*
Coelotes atropos (Walckenaer, 1830)	*Amaurobius atropos*
Coelotes terrestris (Wider, 1834)	*Amaurobius terrestris*

Anyphaenidae

Anyphaena accentuata (Walckenaer, 1802)

Liocranidae

Agroeca brunnea (Blackwall, 1833)
Agroeca proxima (O. P.-Cambridge, 1871)
Agroeca inopina O. P.-Cambridge, 1886
Agroeca lusatica (L. Koch, 1875)
Agroeca cuprea Menge, 1873

Agroeca dentigera Kulczyński, 1913	described from Britain in 1989 as *A. lusatica*; as *A. dentigera* in 2004
Agraecina striata (Kulczyński, 1882)	
Apostenus fuscus Westring, 1851	
Scotina celans (Blackwall, 1841)	
Scotina gracilipes (Blackwall, 1859)	
Scotina palliardii (L. Koch, 1881)	*S. palliardi*
Liocranum rupicola (Walckenaer, 1830)	

Clubionidae

Clubiona corticalis (Walckenaer, 1802)	
Clubiona reclusa O. P.-Cambridge, 1863	
Clubiona subsultans Thorell, 1875	
Clubiona stagnatilis Kulczyński, 1897	
Clubiona rosserae Locket, 1953	
Clubiona norvegica Strand, 1900	
Clubiona caerulescens L. Koch, 1867	*S. coerulescens*
Clubiona pallidula (Clerck, 1757)	
Clubiona phragmitis C. L. Koch, 1843	
Clubiona terrestris Westring, 1851	
Clubiona frutetorum L. Koch, 1866	found in Ireland in 2007
Clubiona neglecta O. P.-Cambridge, 1862	
Clubiona pseudoneglecta Wunderlich, 1994	separated from *C. neglecta* (1994)
Clubiona frisia Wunderlich & Schutt, 1995	*C. similis*
Clubiona lutescens Westring, 1851	
Clubiona comta C. L. Koch, 1839	*C. compta*
Clubiona brevipes Blackwall, 1841	
Clubiona trivialis C. L. Koch, 1843	
Clubiona juvenis Simon, 1878	
Clubiona genevensis L. Koch, 1866	
Clubiona diversa O. P.-Cambridge, 1862	
Clubiona subtilis L. Koch, 1867	
Cheiracanthium erraticum (Walckenaer, 1802)	
Cheiracanthium pennyi O. P.-Cambridge, 1873	
Cheiracanthium virescens (Sundevall, 1833)	

Corinnidae

Phrurolithus festivus (C. L. Koch, 1835)	
Phrurolithus minimus C. L. Koch, 1839	

Zodariidae

Zodarion italicum (Canestrini, 1868)	
Zodarion vicinum Denis, 1935	as *Zodarion* sp. in 1992 list
Zodarion rubidum Simon, 1914	collected in Essex in 1997
Zodarion fuscum (Simon, 1870)	found in Wiltshire in 1999

Gnaphosidae

Drassodes lapidosus (Walckenaer, 1802)	
Drassodes cupreus (Blackwall, 1834)	*D. lapidosus* var. *cupreus, D. macer*
Drassodes pubescens (Thorell, 1856)	
Haplodrassus signifer (C. L. Koch, 1839)	*Drassodes signifer*
Haplodrassus dalmatensis (L. Koch, 1866)	*Drassodes dalmatensis*
Haplodrassus umbratilis (L. Koch, 1866)	

Haplodrassus soerenseni (Strand, 1900)	*Drassodes sorenseni*
Haplodrassus silvestris (Blackwall, 1833)	*Drassodes silvestris*
Haplodrassus minor (O. P.-Cambridge, 1879)	*Drassodes minor*
Scotophaeus blackwalli (Thorell, 1871)	*Herpyllus blackwalli*
Scotophaeus scutulatus (L. Koch, 1866)	
Phaeocedus braccatus (L. Koch, 1866)	
Zelotes electus (C. L. Koch, 1839)	
Zelotes latreillei (Simon, 1878)	
Zelotes apricorum (L. Koch, 1876)	
Zelotes subterraneus (C. L. Koch, 1833)	
Zelotes longipes (L. Koch, 1866)	*Z. serotinus*
Zelotes petrensis (C. L. Koch, 1839)	
Trachyzelotes pedestris (C. L. Koch, 1837)	*Zelotes pedestris*
Urozelotes rusticus (L. Koch, 1872)	*Zelotes rusticus*
Drassyllus lutetianus (L. Koch, 1866)	*Zelotes lutetianus*
Drassyllus pusillus (C. L. Koch, 1833)	*Zelotes pusillus*
Drassyllus praeficus (L. Koch, 1866)	*Zelotes praeficus*
Gnaphosa lugubris (C. L. Koch, 1839)	
Gnaphosa occidentalis Simon, 1878	
Gnaphosa nigerrima L. Koch, 1877	described from Britain in 1997
Gnaphosa leporina (L. Koch, 1866)	
Callilepis nocturna (Linnaeus, 1758)	
Micaria pulicaria (Sundevall, 1831)	
Micaria romana L. Koch, 1866	*Micaria scintillans*
Micaria alpina L. Koch, 1872	
Micaria subopaca Westring, 1861	
Micaria silesiaca L. Koch, 1875	

Zoridae

Zora spinimana (Sundevall, 1833)
Zora armillata Simon, 1878
Zora nemoralis (Blackwall, 1861)
Zora silvestris Kulczyński, 1897

Sparassidae

Micrommata virescens (Clerck, 1757)

Philodromidae

Philodromus dispar Walckenaer, 1826	
Philodromus aureolus (Clerck, 1757)	
Philodromus praedatus O. P.-Cambridge, 1871	*P. aureolus* in part
Philodromus cespitum (Walckenaer, 1802)	*P. aureolus* var. *caespiticolis*
Philodromus longipalpis Simon, 1870	separated from *P. aureolus* in 1992
Philodromus collinus C. L. Koch, 1835	
Philodromus fallax Sundevall, 1833	
Philodromus histrio (Latreille, 1819)	
Philodromus emarginatus (Schrank, 1803)	
Philodromus albidus Kulczyński, 1911	*P. rufus*
Philodromus margaritatus (Clerck, 1757)	
Thanatus striatus C. L. Koch, 1845	
Thanatus formicinus (Clerck, 1757)	
Tibellus maritimus (Menge, 1875)	

Tibellus oblongus (Walckenaer, 1802)	

Thomisidae

Thomisus onustus Walckenaer, 1806	
Diaea dorsata (Fabricius, 1777)	
Misumena vatia (Clerck, 1757)	
Pistius truncatus (Pallas, 1772)	
Xysticus cristatus (Clerck, 1757)	
Xysticus audax (Schrank, 1803)	
Xysticus kochi Thorell, 1872	
Xysticus erraticus (Blackwall, 1834)	
Xysticus lanio C. L. Koch, 1835	
Xysticus ulmi (Hahn, 1831)	
Xysticus bifasciatus C. L. Koch, 1837	
Xysticus luctator L. Koch, 1870	
Xysticus sabulosus (Hahn, 1832)	
Xysticus luctuosus (Blackwall, 1836)	
Xysticus acerbus Thorell, 1872	
Xysticus robustus (Hahn, 1832)	
Ozyptila blackwalli Simon, 1875	*Oxyptila blackwalli*
Ozyptila scabricula (Westring, 1851)	*Oxyptila scabricula*
Ozyptila nigrita (Thorell, 1875)	*Oxyptila nigrita*
Ozyptila pullata (Thorell, 1875)	described from Britain in 1999
Ozyptila sanctuaria (O. P.-Cambridge, 1871)	*Oxyptila sanctuaria*
Ozyptila praticola (C. L. Koch, 1837)	*Oxyptila praticola*
Ozyptila trux (Blackwall, 1846)	*Oxyptila trux*
Ozyptila simplex (O. P.-Cambridge, 1862)	*Oxyptila simplex*
Ozyptila atomaria (Panzer, 1801)	*Oxyptila atomaria*
Ozyptila brevipes (Hahn, 1826)	*Oxyptila brevipes*

Salticidae

Salticus scenicus (Clerck, 1757)	
Salticus cingulatus (Panzer, 1797)	
Salticus zebraneus (C. L. Koch, 1837)	
Heliophanus cupreus (Walckenaer, 1802)	
Heliophanus flavipes (Hahn, 1832)	
Heliophanus auratus C. L. Koch, 1835	
Heliophanus dampfi Schenkel, 1923	
Marpissa muscosa (Clerck, 1757)	
Marpissa radiata (Grube, 1859)	*Marpissa pomatia* sensu auct. Brit.
Marpissa nivoyi (Lucas, 1846)	*Hyctia nivoyi*
Sibianor aurocinctus (Ohlert, 1865)	*Bianor aurocinctus*, *B. aenescens*
Ballus chalybeius (Walkenaer, 1802)	*B. depressus*
Neon reticulatus (Blackwall, 1853)	
Neon robustus Lohmander, 1945	described from Britain in 1999
Neon valentulus Falconer, 1912	
Neon pictus Kulczyński, 1891	described from Britain in 1999
Euophrys frontalis (Walckenaer, 1802)	
Euophrys herbigrada (Simon, 1871)	*E. molesta*
Pseudeuophrys erratica (Walckenaer, 1826)	*Euophrys erratica*
Pseudeuophrys lanigera (Simon, 1871)	*Euophrys lanigera*

Pseudeuophrys obsoleta (Simon, 1871)	*Euophrys browningi*
Talavera petrensis (C. L. Koch, 1837)	*Euophrys petrensis*
Talavera aequipes (O. P.-Cambridge, 1871)	*Euophrys aequipes*
Talavera thorelli (Kulczyński, 1891)	*Euophrys thorelli*
Sitticus pubescens (Fabricius, 1775)	
Sitticus caricis (Westring, 1861)	
Sitticus floricola (C. L. Koch, 1837)	
Sitticus inexpectus Logunov & Kronestadt, 1997	*Sitticus rupicola*
Sitticus saltator (O. P.-Cambridge, 1868)	*Attulus saltator*
Sitticus distinguendus (Simon, 1868)	described from Britain in 2003
Evarcha falcata (Clerck, 1757)	
Evarcha arcuata (Clerck, 1757)	
Aelurillus v-insignitus (Clerck, 1757)	
Phlegra fasciata (Hahn, 1826)	
Synageles venator (Lucas, 1836)	
Myrmarachne formicaria (Degeer, 1778)	
Pellenes tripunctatus (Walckenaer, 1802)	
Macaroeris nidicolens (Walckenaer, 1802)	described from Britain in 2002

Additional species not yet accepted onto the British list

Scientific name and authority	Comments
Uloboridae	
Uloborus plumipes Lucas, 1846	found in glasshouses
Theridiidae	
Coleosoma floridanum Banks, 1900	found in glasshouses
Philodromidae	
Thanatus vulgaris Simon, 1870	imported, no established population

Spiders of the Channel Islands

The following additional species have been confirmed as having been recorded from the Channel Islands, but not from the British mainland.

Scientific name and authority

Linyphiidae
Sintula retroversus (O. P.-Cambridge, 1875)

Lycosidae
Alopecosa albofasciata (Brullé, 1832)

Dictynidae
Archaeodictyna ammophila (Menge, 1871)

Gnaphosidae
Zelotes civicus (Simon, 1878)
Micaria albimana O. P.-Cambridge, 1872

Philodromidae
Philodromus pulchellus Lucas, 1846

Thomisidae
Heriaeus melloteei Simon, 1886

Salticidae
Heliophanus aeneus (Hahn, 1831)
Heliophanus tribulosus Simon, 1868
Dendryphantes rudis (Sundevall, 1833)
Pellenes nigrociliatus (Simon, 1875)

2.3 Checklist of British Pseudoscorpions

This checklist is taken from Legg & Jones (1988).

Scientific name and authority

Chthoniidae
Kewochthonius halberti (Kew, 1916)
Chthonius tetrachelatus (Preyssler, 1790)
Chthonius kewi Gabbutt, 1966
Chthonius ischnocheles (Hermann, 1804)
Chthonius tenuis L. Koch, 1873
Chthonius orthodactylus (Leach, 1817) *sensu stricto*

Neobisiidae
Neobisium maritimum (Leach, 1817)
Neobisium carpenteri (Kew, 1910)
Neobisium carcinoides (Herman, 1804)
Roncus lubricus L. Koch, 1873
Roncocreagris cambridgei (L. Koch, 1873)
Microbisium brevifemoratum (Ellingsen, 1903)

Cheiridiidae
Cheiridium museorum (Leach, 1817)

Chernetidae
Lamprochernes savignyi (Simon, 1881)
Lamprochernes nodosus (Schrank, 1803)
Lamprochernes chyzeri (Tömösváry, 1882)
Pselaphochernes scorpioides (Hermann, 1804)
Pselaphochernes dubius (O. P.-Cambridge, 1892)
Allochernes powelli (Kew, 1916)
Allochernes wideri (C. L. Koch, 1843)
Dinocheirus panzeri (C. L. Koch, 1837)
Chernes cimicoides Menge, 1855
Dendrochernes cyrneus (L. Koch, 1873)

Cheliferidae
Withius piger (Simon, 1878)
Chelifer cancroides (Linnaeus, 1758)
Dactylochelifer latreillei (Leach, 1817)

Garypidae
Larca lata Hansen, 1884

2.4 Checklist of British Opiliones

This checklist is taken from Hillyard (2005).

Scientific name and authority	**Comments**
Nemastomatidae	
Nemastoma bimaculatum (Fabricius, 1775)	
Mitosloma chrysomelas (Hermann, 1804)	
Centetostoma bacilliferum Simon, 1879	recent introduction
Trogulidae	
Trogulus tricarinatus (Linnaeus, 1767)	
Anelasmocephalus Cambridgei (Westwood, 1874)	
Sabaconidae	
Sabacon viscayanum ramblaianum Simon, 1881	recent introduction
Sclerosomatidae	
Homalenotus quadridentatus (Cuvier, 1795)	
Phalangiidae	
Oligolophus tridens (C. L. Koch, 1836)	
Oligolophus hanseni (Kraepelin, 1896)	
Paroligolophus agrestis (Meade, 1855)	
Paroligolophus meadii (O. P.-Cambridge, 1890)	
Lacinius ephippiatus (C. L. Koch, 1835)	
Odiellus spinosus (Bosc, 1792)	established introduction
Mitopus morio (Fabricius, 1799)	
Phalangium opilio Linnaeus, 1758	
Opilio parietinus (De Geer, 1778)	
Opilio canestrinii (Thorell, 1876)	recent introduction
Opilio saxatilis C. L. Koch, 1839	
Megabunus diadema (Fabricius, 1779)	
Platybunus triangularis (Herbst, 1799)	
Lophopilio palpinalis (Herbst, 1799)	
Dicranopalpus ramosus (Simon, 1909)	established introduction
Leiobunidae	
Leiobunum rotundum (Latreille, 1798)	
Leiobunum blackwalli Meade, 1861	
Nelima gothica Lohmander, 1945	

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3. COLLECTING ARACHNIDS

3.1 Techniques

Lawrence Bee & Peter Smithers

This section applies to the three main British arachnid groups, namely spiders, harvestmen and pseudoscorpions; however, since spiders are more diverse in their habits and habitat, most of the notes given below refer to them. Harvestmen, in general, are easier to see and capture, although more care must be taken in handling them and putting them in tubes because of the long legs of the majority. Pseudoscorpions are generally found in the ground layer in similar habitats to spiders but, being extremely small, often require specialised collection techniques.

3.1.1 Ground Sampling

Grubbing about

Grubbing about in leaf litter or ground vegetation can be a very productive method of collecting spiders. Half an hour or so spent at any one place is often far more rewarding than moving about investigating a large number of locations and spending only a few minutes at each. Collectors should get down as close to their prey as possible, so kneeling or even lying on the ground will probably yield far more individual spiders than simply looking at the ground from a distance of more than half a metre. Also, give yourself time to get your eye in; it may be five minutes or so before you start to pick out individual spiders from the background of leaves or grass.

Sieving ground vegetation

Dead leaves, grass tussocks, dry vegetation and moss can all be shaken in a sieve, encouraging any creatures present to fall through the mesh. A sheet (see Section 3.2) placed underneath the sieve will catch any animals that drop through and enable you to see them quite easily. Any material sieved should be returned to the site from which it was collected. When grass tussocks are sieved, please be aware of the destructive nature of this activity. Only tussocks of dead grass should be pulled up and sieved; tussocks of living grass can be investigated by grubbing about.

Checking under stones and other objects on the ground

Many spiders live in dark, damp conditions away from sunlight, underneath fallen logs and bark, stones and even man-made materials lying on the ground for some time. It is always worth investigating these sites, but always restore whatever has been disturbed to its original position. It is worth bearing in mind that many small spiders cling upside-down under these objects and are lifted up with them. Get into the habit of lifting the object and immediately holding it over a sheet or sweep-net in case anything drops off. Any resulting finds can then be dealt with at leisure after the ground below has been examined.

A similar technique is particularly useful on dry-stone walls, where the handle of the sweep-net can be inserted into a crevice in the wall. Where stones are lifted off immediately above the net. This is very effective for catching such fast-moving spiders as *Segestria* and *Textrix*. Once again, please ensure that stones are replaced on the wall exactly as they were previously. Pieces of wood, tiles, corrugated roofing, etc. may be deliberately placed in areas of short grassland or heathland which can otherwise be difficult to survey and examined later, as with natural objects. Egg-boxes, pegged down in grassy areas, have been found useful for attracting harvestmen. All such techniques are best reserved for areas with little or no public access, otherwise the traps are likely to be disturbed.

Pitfall trapping

For ground-dwelling spiders, particularly those which move around at night, one of the most productive collecting methods is to set pitfall traps. They are cheap and easy to set up, sample over extended periods and collect a wide range of ground living invertebrates. At their simplest, they can be plastic coffee cups sunk in the ground with their rims flush with the soil surface. Animals running over the ground then fall into the traps and are unable to escape.

Traps can be made from plastic coffee cups or yoghurt pots which are cheap and easily obtained but any convenient non-breakable container can be used. Translucent cups are best as they become invisible when set in the soil and do not attract the attention of vertebrates as readily as white cups. It is best not to use glass jars as these break easily and can cause injury to wildlife and humans alike if trodden on. The size of the container should reflect the size of the spiders being investigated. In Britain, small containers such as plastic cups are perfectly adequate but in those parts of the world where very much larger species are found they should be both deeper and have a wider mouth. Trowels can be used to dig a hole but if the soil is light and you have a number of traps to dig in, hand bulb planters can speed up the process immensely. Care should be taken not to leave small gaps between the rim of the trap and the surrounding soil and to keep the soil surface flush with the cup rim.

Disturbance is another important factor, the less the surrounding area is disturbed the more representative the catch. So dig the smallest hole possible for your cup and pile displaced soil at least an arms reach away. Freshly disturbed soil has been shown to temporarily increase the activity of invertebrates in the vicinity of the trap. It is thought that this is due to an increase in soil respiration, which elevates local CO_2 levels. If traps are to be used over a period of weeks or more, disturbance can be reduced by having two plastic coffee cups, one nested inside the other. When the traps are being emptied, the inner trap, containing the catch, can be lifted from the outer one without the risk of disturbing the surrounding soil or the need to re-dig a hole.

Always mark your trap line clearly. Coloured string or PVC tape works well either tied to nearby vegetation or as a flag. Mark the ends of each row and draw a sketch map noting the positions of the traps plus features in the micro-landscape, as it is amazing how difficult the traps can be to relocate one or two weeks later.

Where traps are operated over longer periods, a preservation fluid is essential. Ethylene glycol (antifreeze) diluted 50% with water is easily obtained and keeps the catch in good condition. Unfortunately, it is extremely toxic to mammals which find it very attractive. So, in order to avoid killing them, the bactericide propylene phenoxitol is recommended. This is used as a 1% solution and is available from Blades Biological at £14.00/100ml (Blades-bio.co.uk). If the animals are required alive, vegetation can be added to the cup which minimises encounters and provides retreats where smaller invertebrates can hide. The traps should then be emptied at least daily. If the animals are not required alive, filling the cup one third full with water and adding a drop of detergent to break the surface tension ensures that any invertebrates that fall in will be retained. Again, the traps should be emptied daily, especially in warm weather, to avoid decomposition of the catch.

When traps are to be left out for any length of time, rain covers are essential to prevent flooding and loss of the catch. They can be coffee-cup lids, squares of plastic or plywood or simply a stone held above the cup on wire supports, sticks or smaller stones. Another precaution is to make a series of small holes just below the rim of the container so that any water that does enter will drain away before washing the catch out of the cup. If the traps are to be left out for long periods of time where there are vertebrates, it is often worth placing a coarse wire mesh over the traps to prevent trap robbery.

The number of spiders caught will depend, in part, on the number whose paths intersect the rim of the trap. To increase the catch efficiency and obtain a better sample of the spiders present, one can either increase the size of the container or increase the number of containers. If traps become too large they begin to catch small mammals and reptiles which is undesirable for both practical and conservation reasons and a larger number of smaller traps is usually the best solution. However, if a trapping a regime is run for a long period of time (e.g. to assess seasonal changes in invertebrate species assemblages), care should be taken not to reduce the local population by over-trapping. A series of short trapping periods at regular intervals is recommended. When traps are not in use they should be covered or removed to prevent the unnecessary collection of material. Barriers have also been used to increase the effective trapping area by placing pitfall traps at each end of a linear barrier. Spiders and other invertebrates which intercept the barrier are diverted either left or right and caught in the traps at the ends. While useful in studies of directional movement of surface-active spiders, this is probably not worth the additional effort when undertaking qualitative surveys of particular habitats.

A range of different specialised pitfall traps have been devised for particular micro-habitats. These include subterranean pitfalls for use in shingle and similar loosely packed substrata, platform traps for use in scree slopes, boulder fields and caves where there is no level surface in which to bury conventional traps and even floating pitfall traps for use on the surface of ponds and lakes. While all of these have their uses for specific studies of such micro-habitats, they are not in everyday use in general survey work and readers should refer to the specialist literature if they wish to try them.

Care should be taken when interpreting the results from pitfall surveys. The relative abundance of organisms in pitfall traps rarely accurately reflects their abundance in the trapping area. The activity of the individual species plays an important role in determining the numbers of individuals caught in any particular trap as does the type of vegetation in the area surrounding it. Species that are very active will be more abundant in pitfall samples than more sedentary ones even though there population densities may be similar. Traps set in areas of dense vegetation catch far fewer spiders than those in more open ground, even though the real population densities may be similar in the two areas.

Aeronaut traps

Water traps in trays as large as possible or practicable may be mounted above the ground and will selectively trap "ballooning" spiders. They must not be covered, of course, and must be checked regularly because of evaporation. While these will collect spiders already airborne, a device recently described by Woolley *et al.* (2007) will sample spiders ballooning from a particular spot and intercept them before they take flight. It consists of a two-litre plastic drink bottle with the top cut off and inverted back inside the bottle. This is placed upside down on a bamboo cane that is stuck in the ground. A cone of string netting is attached to the cane below the bottle and fixed to the ground with tent pegs to form a cone shape. Spiders that attempt to balloon will climb the netting and enter the plastic bottle but be unable to leave. The trap should be examined and emptied every day.

3.1.2 Vegetation Sampling

Beating or shaking foliage

The lower branches of a tree can be shaken over a sheet or upturned umbrella to dislodge spiders living amongst the leaves. A more effective way of collecting spiders in trees is to tap the branch sharply with a stick several times – take care not to thrash about and cause damage to leaves or branches. Once the spiders have fallen out, they can be easily collected from the underlying sheet, upturned umbrella or beating tray (see Section 3.2). Some spiders and pseudoscorpions may remain stationary for a time, curled up and playing dead; it is always worth waiting a minute or two when using this method before discarding the results of branch beating to ensure that no spiders have escaped your attention.

Sweeping vegetation

Spiders are often found in the upper levels of grass and other ground vegetation. By briskly sweeping the top of such vegetation using a sweep-net (see Section 3.2) with a steady side-to-side swinging motion, spiders can be dislodged and collected in the sweep-net. A slow walk through an area of long grass, for example, swinging the net as you go, can yield a large number of different invertebrates. After a few paces, check your net, collect any spiders and shake any other creatures back into the grass.

Bark sweeping

On trees where deep fissures in the bark provide a suitable retreat for some spiders, a standard soft household brush or a stiff paintbrush is useful to sweep the bark. Again, a sheet or beating tray or inverted umbrella to catch the sweepings can be usefully employed.

Bark traps

Spiders often travel up and down the bark of trees, particularly during the hours of darkness. If an artificial retreat, in the form of a strip of double corrugated cardboard or polythene bubble wrap (20–30 cm wide) is placed around the trunk of the tree and left in position for some time, it may well provide some spiders with a sufficiently enclosed and compact environment for them to remain within the cardboard corrugations or between the polythene bubbles. If using bubble sheeting, an extra layer of dark material needs to be placed over the trap to prevent light from entering. A period of around one month is probably the minimum time required for a bark trap to remain *in situ*. Traps should therefore only be used on sites where they are not likely to be vandalised.

Artificial birds' nests

Old birds' nests in trees are known to attract some spiders, but it is difficult to examine them without the whole thing falling apart. Artificial birds' nests can be extracted quickly and easily and immediately placed in a plastic bag. They can then be examined later at home where any potential escapees can be caught before they disappear. These can be created by packing thin twigs, wood-wool, small wood shavings, or dry grass into a plastic mesh bag (similar to those used by wholesale greengrocers to pack carrots or brussels sprouts). The filled bag should be placed in hollow tree trunks, holes in branches or cracks in the main trunk and left for some weeks.

Artificial litter heaps

Net bags (1 cm mesh) filled with sterile litter may be left out for weeks or months to allow colonization. (This is useful when natural litter is scarce). Alternatively, leaves heaped up on polythene sheets can convert a thin layer into large piles and can be taken away eventually by lifting and tying the corners of the sheet.

3.1.3 Other Collecting Techniques

Checking webs and retreats

It is always worth taking a close look at any webs which are encountered whilst collecting. Orb webs, sheet webs and other less structured webs may all contain spiders, either in the main web or sheltering in a retreat close by. A convenient method of highlighting webs is to spray them with water from a plant mister, the total extent of the web can then be easily seen. Spiders which do not weave webs may construct some type of silken retreat. These are most commonly found in grassy habitats, in rolled-up leaves, and under bark. These are always worth investigating as the spider may, in fact, be sheltering in the retreat. On walls, rocks, tree-trunks, etc., threads left by wandering spiders often show their presence when viewed so that sunlight is reflected off them.

Leaf litter sorting

It may be more convenient to collect plastic bags full of leaf litter and sort through them at home, collecting any spiders discovered. Empty your bags of litter, a few handfuls at a time, on to a large white tray and carefully sort through it. A lamp shining over the tray will increase illumination and temperature which may well stimulate some spiders to move and seek a cooler, more shaded situation. Try, if at all possible, to return the litter to the area from which it was collected.

Tullgren (or similar) funnels

An alternative to hand-sorting is to use Tullgren funnels. This consists of a sieve containing the litter sample over which is placed a desk lamp. The lamp sets up a gradient of heat, light and humidity within the litter that drives the organisms down to the base of the sieve. Beneath the sieve is a funnel which leads to a container of alcohol or isopropanol to preserve the extracted animals, or damp screwed-up kitchen roll if the fauna is to be kept alive.

Winkler bags

An alternative to Tullgren funnels if electricity is not available is the Winkler bag. This consists of a small mesh bag that contains the litter which is hung inside a canvas funnel, held open with a wire hoop. The top can be tied closed to prevent organisms escaping and the bottom of the funnel leads to a container that contains alcohol to preserve the catch or damp tissue or vegetation if the catch is to be collected alive. The process relies on ambient temperatures and the activity of the litter organisms. They remain active in the litter and eventually approach the outer surface of the mesh bag were they fall out into the funnel and the container below. Tullgren funnels and Winkler bags can be very effective for extracting very small spider species that are easily overlooked when hand collecting.

Simulating insect vibrations in webs

Some spiders, e.g. *Segestria*, *Amaurobius*, *Tegenaria* and *Zygiella* spp. respond to a vibrating tuning-fork being placed on the web. *Segestria florentina*, for example, can be encouraged to come to the entrance of her tubular retreat by placing the tip of the vibrating tuning-fork on one of the lines of silk radiating from the entrance to the tube. With luck, the spider can then be persuaded out of the tube into a sweep net placed immediately underneath.

Night collecting

Many species are active mainly or only at night and it can be really worthwhile going out with a torch. As well as a hand-held torch, a head-light (obtainable from outdoor pursuits shops) can be very useful. Particular care should be taken when collecting at night and someone should always be told where you are going and when you expect to return (see Chapter 9).

Vacuum-sampling

This is the use of a portable vacuum cleaner to suck invertebrates off short vegetation and the surface of the ground. The apparatus (known as the D-vac) used to be bulky and expensive which meant it was a technique only available to professionals. The recent availability of petrol driven garden leaf blowers has made suction samplers available to many more arachnologists and entomologists. A conventional garden leaf blower can be easily modified by inserting a fine net bag in the intake of the machine. Net bags can be hand made from net curtain material or brewers wort bags, which just happen to be the correct size, are a ready made alternative. Such modified machines are now known as G-vacs. For the best results the intake is then held against the surface of the ground and swept from side to side to cover a known area delineated by a quadrat (e.g. 1 m^2). Suction samplers are very efficient at sampling the smaller organisms associated with short vegetation in habitats such as grasslands or heaths. Once an area has been sampled the contents of the bag can be emptied onto a high sided white tray where the invertebrates can sorted and preserved. Alternatively, the contents of the bag can be placed in containers and preserved, to be sorted later.

The G-vac is not without its limitations because, like a sweep net, wet vegetation reduces their efficiency. Care should also be taken when interpreting catches as measures of population density per unit area, as the high speed air flows at the edges of the intake tube tend to drag in items from outside of the area sampled. This effect decreases as the diameter of the intake tube is increased (Bell *et al.* 2002). While they may not generate absolute data they are excellent for generating comparative data.

3.2 Equipment

Lawrence Bee

Some equipment is essential when collecting in the field; however, carrying too much can become something of an encumbrance. It is therefore useful to have adequate storage space on your person, either in the form of a strong and spacious shoulder bag with plenty of separate compartments and pockets, or a jacket with plenty of pockets for collecting tubes, etc., or both.

3.2.1 Essential items of equipment

Notebook and pencil

Record all collections you make in a field notebook, relating entries in the notebook with labelled collecting tubes. Make sure that you have plenty of labels available for immediate use. Information noted should include date, habitat, grid reference number, locality and collector's name. Carry spin pencils and/or a pencil-sharpener/knife.

Collecting tubes

Can be of glass or plastic, preferably with plastic stoppers. Some tubes should contain a killing agent/ preservative (e.g. 70% alcohol) whilst others should be empty for the collection and/or examination of live specimens. Glass tubes are relatively fragile, but do not scratch, which is a drawback with plastic tubes. Corks are preferable to plastic stoppers for tubes for examining live specimens in the field, since the specimen cannot run into the depression in the latter.

Hand lens

A lens with 10× magnification is the most useful in the field because it provides a sharp image when examining a spider in the glass chamber of a pooter (see below) or a glass collecting tube. The hand lens can be in virtually constant use so it is advisable to have it near at hand, and wearing it on a neck strap is strongly recommended.

Pooter

There are a number of pooter designs. One of the best for spider collecting is the so-called "Cooke pooter" which consists of a length of rubber tubing with different sizes of glass tubing attached to one end (see Figure 1). The spider is sucked into the glass or plastic chamber at one end of the pooter and can then be blown into a collecting tube with no damage being caused to the spider's body. A small piece of fine gauze material (e.g. nylon stocking) creates a barrier to the spider being sucked beyond the glass chamber. Again, it is sometimes useful to attach your pooter to a neck strap to have it immediately available.

Polythene Sheet

As well as the uses described in Section 3.1 for sieving and bark-sweeping etc., a polythene sheet can make life a lot more comfortable when kneeling or lying whilst collecting in damp grassland or leaf litter. Another advantage of a plastic sheet is that it can be cleaned in a stream or on wet grass when it becomes dirty. Pale coloured sheeting is preferable so that spiders can be spotted easily against the background. Old fertilizer sacks or polythene obtainable as cheap garden pool liner from garden shops is ideal. White sheeting can be too dazzling in bright sunshine. The size of the sheet is a matter of personal preference: too small and it does not serve the purpose, too large and it becomes unwieldy. The optimum is something like a metre square.

Sweep net

Used to collect in areas of long grass, heather etc. Designs can vary considerably (as does the price) but at its most basic, the sweep-net consists of a bag of finely woven material (preferably white), attached to a stout handle. The bag itself needs to be made of canvas or other strong material to withstand the buffeting

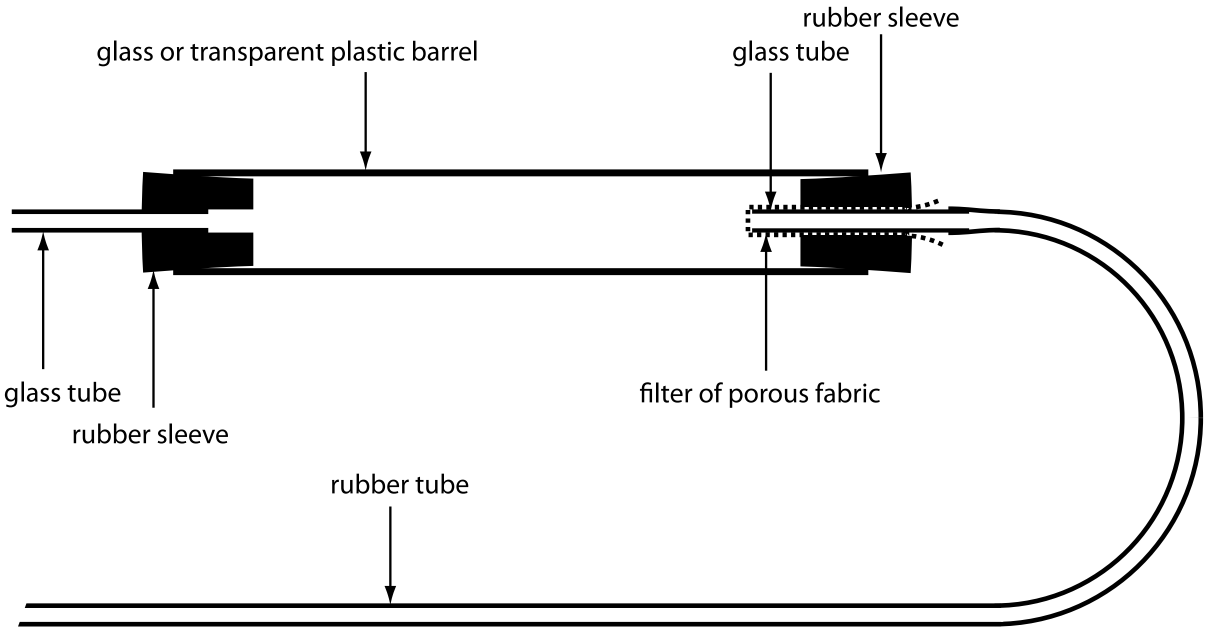

Figure 1. Diagram of a Cooke pooter shown in cross-section. The rubber tubing has been truncated in this figure and would normally be three to four times the length of the barrel.

it will receive in sweeping varying types of vegetation. Butterfly nets, of light flimsy material, are unsuitable as sweep-nets as they are simply not robust enough. Avoid using a long handle – it is much easier to use a sweep-net with a short handle as an extension of the arm. Shallow nets are more manageable, but a deeper net can be used to envelop tree branches (shaking the net with branch enclosed to dislodge spiders), serve as an impromptu beating tray or sweep lower branches of trees and shrubs. If possible, acquire a net with strengthening on the rim because the side away from the handle will get a lot of wear. Collapsible nets are an advantage for portability.

3.2.2 Other useful items of equipment

Beating tray

For collecting from foliage of shrub or trees. It consists of a collapsible framework of cane struts supporting a piece of closely woven white material. When in use, the canes (inserted in pockets at the four corners of the square of material) create a tension which stretches the material into a dish-like shape. Invertebrates falling into the tray can therefore be picked up with ease. The disadvantages are the difficulty of handling in windy weather, and the slit where the two edges meet, which sometimes seems to act as a magnet for fast-running spiders. Non-collapsible beating-trays are available, or can easily be made, but they suffer from the obvious drawback of portability. An old umbrella may be used in a similar fashion and some of these have telescopic handles which enable the umbrella to be collapsed to a very small size.

Garden sieve

This can be used for shaking out spiders from leaf litter etc. Collapsible sieves can be obtained, otherwise a garden riddle, preferably plastic, with a mesh size of 5 to 10 mm. is very suitable.

Trowel and/or strong knife

A small garden trowel or knife can be used for lifting turf or bark from dead or fallen timber. However, such aids must be used with discretion since much damage to the environment can be caused by their indiscriminate use.

Brush for bark sweeping

This can be a paintbrush, not too soft, or a domestic hand-brush. If the brush is too unwieldy, the handle, usually made of wood or plastic, can be cut down. One difficulty with bark brushing is that of getting a net underneath. It is not difficult to make a suitable net using a wire coat-hanger as the rim which can then be bent into any convenient shape. Alternatively, an inverted umbrella may be used and fits snugly against the bark.

Tuning fork

Placing a vibrating tuning fork against the edge of a web can bring some spiders out of hiding (see Section 3.1.3). Middle C is reputed to be the best all-rounder, but precise information is lacking!

Torch

A robust torch is a must for caves, and for night collecting but can also be useful for dark corners such as hollow trees. It is also a sensible thing to have whenever in remote or difficult countryside in case you are inadvertently night bound (see Chapter 9).

Small towel

This does not take up much room, but is invaluable in showery weather or when working in a boggy environment. Once the hands are wet, it is difficult to keep essential equipment dry.

Insect repellent

By the nature of many of the habitats concerned, it is inevitable that arachnologists will encounter biting insects, sometimes in great numbers. We are advised by an expert arachnologist and enthusiastic field-worker who is also a medical practitioner, that the best is "Repel 100" obtainable from Survival Aids Ltd, Morland, Penrith, Cumbria CA1O 3AZ. Although there are reports of toxicity with sustained use, occasional use on collecting trips should present no risk and the benefits are considerable. A fine-mesh head net, worn over a brimmed hat and tied at the neck is also very useful particularly if soaked in "Repel 100".

3.3 Code of Conduct for Collecting

Paul Lee

Preamble

Some people prefer to identify arachnids alive and then to return them to their place of collection. Whilst this will satisfy the unease that many feel about having to kill specimens in order to study them, field identification is normally only possible with certainty for some of the larger species. Often, closely related species within a genus are only separated by small morphological differences, and careful examination of preserved specimens, using a microscope, is then essential for an identification to be made with any confidence. For any serious taxonomic, ecological, recording or distribution studies of arachnids, it is necessary to collect and preserve the specimens. They are then available should it be necessary to obtain confirmation of the identity of new, rare or 'difficult' species by an expert, or for subsequent examination when new techniques become available or new characters are discovered.

Whilst it is unlikely that collecting alone has caused the extinction of any species in the British Isles, the increasing loss of habitats resulting from forestry, agriculture, and industrial, urban and recreational development means that a Code for Arachnid Collecting is required in the interests of arachnid conservation. The BAS subscribes to A Code of Conduct for Collecting Insects and Other Invertebrates issued by Invertebrate Link (formerly Joint Committee for the Conservation of British Invertebrates) on which the following is based.

Permission and conditions for collecting

Permission from a landowner, occupier, warden or other authority should always be sought before collecting on private land. Collecting on a Site of Special Scientific Interest requires permission both from the owner and from the local office of the appropriate national conservation agency. Any conditions which might be imposed should be strictly followed.

It is illegal to collect species listed in Schedule 5 of the Wildlife and Countryside Act except under licence from DEFRA. The two arachnids currently listed in Schedule 5 are the fen raft spider *Dolomedes plantarius*, and the ladybird spider *Eresus sandaliatus*.

After collecting on private land, nature reserves, SSSIs or other sites of known conservation interest, a list of species, annotated with habitat data, should be submitted to the appropriate authority.

Protecting the environment

Whilst collecting, damage to the local environment should be minimised. For example, nesting birds should not be disturbed; if such disturbance occurs unwittingly, the area should be left immediately.

- Excessive trampling of vegetation should be avoided, particularly if rare plants are known to occur on the site. When beating for arachnids, shrubs and trees should not be damaged by the use of excessive force.
- Vegetation, leaf litter, vertebrate nests or other material should not be removed from a site in excessive amounts, and then only if permission has been granted and in compliance with the laws applying to the species concerned.
- Any form of vegetation, such as moss, that is likely to recover, should be replaced in its appropriate habitat once it has been worked for specimens. Logs and stones should be returned to their original positions after searching beneath them.
- Only small areas of bark should be stripped from dead wood and, whenever possible, it should be replaced in position. Piles of litter should be replaced and not left scattered about after sorting.

Trapping

- If a trap is found to be catching large numbers of local or rare species, it should be re-sited if possible.
- Take precautions to ensure that larger creatures such as frogs and shrews cannot fall into pitfall traps.
- Bear in mind that pitfall trapping is indiscriminate. Keep trapping to a minimum commensurate with the studies being undertaken and do not leave traps in position when they are not required.
- Trapping will catch many other creatures other than arachnids. Every effort should be made to contact experts on other groups so that this by-catch material will not be wasted.

3.4 Selection and Use of Microscopes

David Nellist

3.4.1 Introduction

Very few arachnids on the British list can be identified with confidence using only the eye or a hand lens, the majority being of such a size that a microscope is needed to examine the morphological features which determine the species. However, microscopes, which range widely in features and price, are probably the most important financial investment any amateur arachnologist is likely to make. Some understanding of their operation and of the factors which affect their performance are therefore needed in order to make a wise investment and to use the instrument to its full potential. These brief notes have been put together to help those with little experience of using a microscope, perhaps considering the purchase of a microscope for the first time, to make a more informed choice and then, subsequently, to extract the best performance from it.

The components of the microscope

There are basically two types of microscope. The *compound* microscope, with the potential to produce images with magnifications up to 1500×, is generally only used for the examination of extremely thin sections of materials, mounted on glass slides, and illuminated from below using transmitted light. Such microscopes are not suitable for the examination of solid objects and are not recommended for the routine identification of spiders. However they do have one application. From time to time, in order to be confident of a correct identification, when two morphologically similar species are known, it is necessary to examine a detached and cleared epigyne. When mounted on a slide and illuminated from below magnifications of 200× or more can then be used to determine the details of the internal structure. Arachnologists invariably use *stereo* microscopes for the study and identification of preserved spiders and other small arachnids.

Specimens are examined whilst lying in spirit and illuminated with a high-intensity top light. Stereo instruments have a number of important advantages. First, they have a considerable depth of field so that more of the depth of a solid object is in focus at the same time. Second, they present a three-dimensional image to the eyes allowing the spatial relationships of complex structures, such as the palps of mature male spiders, to be more easily understood. Third, the image is upright, in contrast to the two-dimensional image provided by a compound microscope where the bottom of the specimen is seen at the top of the image, and vice versa. Finally, the large depth of field, coupled with the long working distance between the specimen and the bottom objective lens, means that maneuvering a specimen into the desired position for viewing is relatively simple. However, the price range is very wide. Simple instruments with a limited magnification range, often not exceeding 40×, can be purchased for just a hundred pounds or so. At the other end of the scale, high-performance microscopes with first-class, apochromatic optics and sophisticated photographic capability can cost many thousands of pounds.

For all stereo microscopes the stereo image is the result of viewing the specimen using two separate optical paths, one for each eye, and slightly inclined to each other. When separate objective lenses are used the microscope is then referred to as being a *Greenough* type. But the same effect can be achieved by bringing the light from the specimen to the separate eyepieces using the two sides of a single objective lens. This is known as the *common optic* design. It has more flexibility than the Greenough type and is now the usual form for higher-performance instruments. For example, with microscopes of the Greenough type changing the magnification has to be achieved by replacing the two eyepieces, or the two lower objective lenses, or both, with those of higher or lower magnification. This is time-consuming and inconvenient especially when spending several hours at the microscope and requiring frequent changes in magnification. However, with the common optic design the magnification can be changed up or down simply by screwing a single supplementary lens into the existing objective lens, or by rotating a knob or a ring on the lens housing which brings different lenses into the light beam, or changes the lens spacing. This is certainly much more convenient and most stereo microscopes now incorporate such a zoom facility which allows a continuous change in magnification. Anyone using one of these stereo zoom microscopes, as they are known, is unlikely to want to return to a Greenough type especially if the microscope is in use for long periods. The magnifications required for the study of spiders range between 15× and about 100×. It becomes increasingly difficult to maintain image sharpness and a flat field of view as the magnification exceeds 100×, especially with instruments towards the lower end of the price range, but some high quality instruments are advertised as being able to achieve meaningful magnifications somewhat in excess of 200×.

An important point to be borne in mind when purchasing a microscope is the angle of the eye-tubes to the horizontal. Low priced instruments have vertical eye-tubes so that the viewer has to be above the microscope looking directly down to see the images in the eyepieces. To reduce neck strain, especially if working for long periods, it is recommended that an instrument be purchased with inclined eye-tubes. Angles vary between manufacturers, and within the range of instruments offered by a single manufacturer. For example, in the current range of Leica stereo zooms it is possible to purchase instruments with angles of 38° or 60°. The angle on my own instrument is 50° which I find ideal and comfortable even for long periods of use. For the RZ series of high performance stereo zoom microscopes supplied by the Meiji Techno company, an ergonomic head can be supplied in which the angle can be adjusted between 10° and 50° – but this is an expensive option! It is worth sitting down at the microscope and checking that the angle is comfortable before making a purchase. It is also worth checking that the microscope being considered allows the interocular distance to be changed i.e. the eye pieces will swivel slightly to cater for variations in the distance between the eyes of different observers. This will only be a problem with older, second-hand microscopes

Generally, microscopes are supplied with the column supporting the lens housing at the back of the microscope i.e. on the side away from the observer. This has the advantage of allowing a clear view of, and unrestricted access to, the specimen on the stage. However, when one or two high-intensity lights also have to be sited close to the specimen, and frequently moved to adjust the position of the beams and the contrast of the illumination, this arrangement can be inconvenient. It is therefore recommended that a microscope be purchased which allows the eye-tubes to be rotated through 180°. The column supporting the lens housing can then be on the same side as the observer freeing-up space on the far side to accommodate two lights, in any desired position, and with different angles of incidence, without significantly restricting access to the specimen. Generally, a simple flat stage is all that is required for the examination of specimens using a top light. However, stages can be obtained which incorporate a tungsten light source to allow for the examination of thin specimens with transmitted light (e.g. detached and cleared epigynes as described above). From time to time this might be a useful facility, and certainly cheaper than buying a separate compound microscope for this purpose, but the magnification range is then limited and the use of a higher-power, compound microscope would be the preferred option.

It is very often necessary to be able to make measurements using the microscope, for example, to measure the total length of specimens, positions of trichobothria, width of eyes etc. and an eyepiece micrometer is normally used to make such linear measurements. This is a glass disc having a linear scale printed on it, usually 10 mm, divided into 100 parts and inserted into an eyepiece. The length or width of a particular feature of the specimen is then compared directly with the scale, taking into account the magnification being used. The disc is held in the image plane of the eyepiece and can be left in place during normal viewing, but the slight obscuring of the field of view may then be an irritation and it is useful to keep a separate eyepiece for the purpose and simply insert it into the eye-tube when a measurement is necessary. Note that with some lower cost instruments it is not possible to fit a micrometer and this will need to be checked with the supplier.

3.4.3 Using the Microscope

Sitting position

To be able to sit and use the microscope in a comfortable, relaxed position is extremely important. However, microscopes do not always sit comfortably on desks or tables. Generally the whole stand is too short, and the eyepieces then too low, so that a crouching position has to be adopted to see the image. The microscope should therefore be raised using vibration-free, stable, heavy blocks until, when sitting in a comfortable position, the eyes are slightly higher than the eyepieces. Rocking forward, without stretching or crouching, should then allow the eyes to rest comfortably in the eye cups for stress-free viewing. The interocular distance should then be adjusted until the images from each eyepiece fuse together and a clear view of the entire circular field is visible.

Focusing

To obtain the sharpest, high-contrast images possible from the instrument several other adjustments are necessary. First, most microscopes have, in addition to the normal focusing knob, the ability to focus one or both, eyepieces individually. This is to compensate for differences between the eyes and should also allow the image to remain in focus when using the zoom-ring to change the magnification. In fact, invariably some slight refocusing is necessary as one moves through the magnification range. Also one's eyes vary slightly from one viewing session to the next and so adjusting the eyepiece settings is a first priority when beginning a new session. Use the procedure recommended by the manufacturer to do this. Second, before beginning a viewing session GENTLY clean the lenses of the eyepieces using a special lens-cleaning cloth (obtainable from camera shops such as Jessops.) This removes the small quantities of oils and waxes which evaporate from the eye and condense on the lenses during use, especially if the eyes are enclosed in eye cups during viewing. This oily layer, although perhaps barely perceptible, scatters the light leaving the eyepiece degrading the image slightly and thus the ability to resolve very fine detail. If there is any possibility that hard particles have settled on the lenses (*although every care should be taken to prevent this*) then a VERY soft lens brush should be used to remove these before using the lens cloth.

The top light

It seems to be standard practice now to examine specimens whilst they are lying on fine glass beads immersed in spirit and illuminated by one or two high intensity top lights. Top lights are available in two forms. Those using tungsten bulbs as the light source and those using a "cold" halogen light source, generally in combination with fibre optic light guides. The latter are much more expensive than the former, several hundred pounds versus less than one hundred, but both have their devotees. One disadvantage of tungsten sources is that the spirit is warmed by the heat of the bulb and the alcohol gradually evaporates. As the specific gravity of the liquid then rises specimens tend to float and move about in the dish. This can be controlled by inserting a heat-absorbing filter in the light beam but this is not always a convenient option in the limited space available. One advantage of a halogen source is that the lamp is separated from the specimen by about 12 inches of light guide and evaporation is much reduced or even eliminated. Devotees also claim that the light can be directed just where it is needed by manipulating the swan neck light guides, but in practice this is not always easy to accomplish. However, halogen sources do have one distinct disadvantage. The light has a higher blue component than that from tungsten bulbs and is thus scattered to a far greater degree by colloidal material in the spirit (e.g. particles having a diameter less than 0.002 mm). It is extremely difficult to keep spirit free of colloidal material and it will thus appear slightly more opalescent when illuminated with a halogen bulb rather than with a tungsten bulb, and this again does lead to a small degradation of the image as it appears in the eyepiece; maybe not sufficient to compromise one's ability to identify a specimen, but sufficient perhaps to make the resolution of fine detail just a little more uncertain and it can be an irritating nuisance.

Spirit

In the light of what has been said above it should be obvious that every precaution should be taken to keep spirit as clean as possible. Industrial Methylated Spirit as purchased (98%) should be diluted to about 80% with distilled water, allowed to stand for a few weeks and then filtered through a fine filter paper. Dirty and discoloured spirit can be partially reclaimed (see the note in British Arachnological Society *Secretary's Newsletter*, No. 3, March 1972) but the problem of the colloidal material had not been recognised at the time the note was written.

Glass beads

Specimens lying in spirit are generally supported on 80 mesh glass beads in an excavated glass block. The specimen can then be pushed into the beads and supported in any desired position to allow the morphological features to be examined. The glass beads should be kept very clean. Large particles of detritus can easily be removed by using a gentle flow of water, but periodically adding a small amount of household bleach to the beads and allowing them to stand overnight before, again, flushing with a gentle flow of water will dissolve small particles of organic matter and remove any grease adhering to the bead surface so that they slide easily. Of course, when using clear glass beads the specimen is being viewed against a light background. On some occasions, for example trying to detect fine trichobothria or hairs it helps to view the specimen against a dark background. For this purpose a small dish containing a layer of black candle wax can be used. Small depressions of different sizes can be pushed into the wax to hold the specimen in the desired position and the specimen then illuminated with low-angle light.

Photography through the microscope

Photographs may be taken of the image provided by the microscope by mounting a single lens reflex camera onto one of the eye-tubes. The camera lens has to be removed but the microscope eyepiece remains in place. This requires a special adaptor the design of which will depend on the make of camera being used. Obviously, the microscope eye-tube has to be sufficiently sturdy to carry the weight of the camera without distorting. Alternatively one can purchase a microscope with a trinocular head i.e. with a third tube rising vertically from the lens housing between the eye-tubes. The camera is then mounted on the top of this tube, again using a special adaptor. Photography using a trinocular head is most convenient because the camera may be left in place during normal viewing. The image can be positioned and focused using both eyepieces and the light then sent from one of the eyepieces, generally the left, to the camera simply by moving a lever. This is not the place to go into the detail of photomicrography and reference to a specialised text book should be made. However, one cardinal rule is to match the spectral sensitivity of the film being used to the colour temperature of the light source. Using a film balanced for daylight to photograph a specimen illuminated with a tungsten light source will produce a picture with false colours. Video and digital cameras can be substituted for the film camera but, again, reference to specialist literature should be made.

Drawing images

Clearly, it is possible to simply transfer the details of a microscope image as seen in the eyepieces to a sheet of paper using a pencil and freehand, but a high degree of skill is needed. A simple aid is to use an eyepiece containing a squared graticule i.e. simply a large square divided into, say, 16 smaller squares. When the graticule is placed in the focal plane of the eyepiece the grid is in focus and details of the focussed image can be easily transferred to a sheet of paper marked-up in pencil with a similar grid. A further refinement is to use a camera lucida device whereby a prismatic mirror system is placed over the eyepiece which allows the viewer to see an image of the specimen superimposed onto a sheet of paper lying flat at the base of the microscope.

3.4.4 Suppliers

The companies listed in chapter 10 supply microscopes and high-intensity lights of all types. They are listed simply as a guide without any recommendation for one or the other. To the best of my knowledge those marked with an asterisk supply second-hand instruments only. Companies such as Nikon, Olympus, Leica (now incorporating Wild), Kyowa, etc., manufacture high quality instruments but appear to be more oriented towards supplying the professional market. However, they may have second-hand or ex-demonstration models available from time to time. Brunel Microscopes Ltd generally exhibit at the Amateur Entomologists' Society's Exhibition held annually at Kempton Park Racecourse in October, and this provides an excellent opportunity for their various models to be examined. From my limited experience of their instruments, the optical quality appears to be excellent. Meiji Techno UK Ltd, which frequently advertises in the *British Wildlife* magazine, also supplies a wide range of high quality instruments.

Note that, if considering the purchase of a second-hand instrument, or indeed a new model, it is essential to know exactly what type of instrument is required, and to be able to assess the quality and performance of any instrument that is offered before committing oneself to a purchase. Look for smooth movements of all the controls; a wide and bright field of view without shadows due to dirt on internal glass surfaces;

a flat field i.e. an object in focus at the centre of the field should also be in focus at the edge of the field, There may be some slight degradation at the edge but the field should not appear to be dished Finally, check the quality of the optics by ensuring that the tips of hairs etc. are sharp and crisp even at the higher magnifications. If considering using the instrument away from home then purchase of the appropriate case is strongly recommended in order to secure the microscope and prevent damage by vibration or knocks.

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4. STARTING AND USING A COLLECTION

Paul Lee

4.1 Identification

Introduction

There are few species of spider that can be identified in the field by the novice and even experts will only attempt identification of a limited number of species this way. Identification of the majority of spiders requires that they are collected and killed prior to microscopic examination of the male palps or the female epigyne. When specimens are killed in alcohol there is a tendency for the legs to contract obscuring characteristic features of the legs themselves and of the underside of the animal, particularly the epigyne. In such cases, removal of individual limbs may be necessary. Suggestions for avoiding this are made in the section on preservatives but many arachnologists consider such problems a minor drawback compared to the extra steps required to avoid them. Even when the legs do not contract it is usually essential to orientate the spider carefully (and arrange the microscope lighting) to make characteristic features such as trichobothria clearly visible. Fortunately, most of the important features are external and only very rarely is it necessary to dissect a specimen.

Equipment

The basic equipment required for the identification of spiders comprises:

A **stereo microscope** capable of producing good quality images at magnifications of at least 30× and ideally up to 100×. An eyepiece micrometer for taking accurate measurements is a useful addition to the microscope. Further details of microscope selection are given in Section 4.5.

An overhead **light source** of sufficient intensity to provide clear images at the highest magnification of the microscope. A separate light source is far more flexible than one built into the microscope. Ideally it should be possible to focus the light and its intensity should be variable;

An **excavated glass block** for holding specimens in alcohol whilst viewing them under the microscope. A clear glass block is normally used but a black block (or a black card beneath a clear block) can be useful sometimes when pale hairs and trichobothria are proving difficult to see;

A layer of fine **glass beads** can be placed in the excavated glass block allowing the specimen to be gently pushed into the beads and held in position at whatever angle is required;

A pair of **soft forceps** for holding and manipulating specimens without damaging them;

A very fine **mounted needle** (a headless entomological pin in a pin vice is ideal) for the manipulation and dissection of specimens;

A **wash bottle** containing preservative, either **industrial denatured alcohol** (IDA) (this used to be known as industrial methylated spirits – IMS) diluted with distilled water to 70% or **propan-2-ol**, diluted with distilled water to 60%. The specimen must be completely submerged in liquid if surface distortions are not to interfere with the visibility of fine features. This means the alcohol level in the excavated glass block will need to be kept topped up. The alcohol will also need changing at regular intervals as suspended debris builds up and increasingly scatters the light.

Use of keys and other literature

A range of specialist literature is available to assist with the identification of spiders. Details of this literature are given in Section 10.1. As with all specialist keys, one problem the novice has to overcome is that of the terminology used. There is no easy answer to this problem but it can be helpful to have guidance from an experienced arachnologist when interpreting keys initially. This sort of help is available on a range of short courses arranged in different locations around the country by BAS, the Field Studies Council and various other organisations. It is important to be patient when attempting to identify specimens. If you get to an identification that is clearly incorrect, by all means go back to the start of the key and start again reading the couplets carefully. However, you should not allow yourself to become frustrated. If this seems to be happening, stop and move on to another specimen. It is surprising how often the difficulties disappear when you come back to a problem specimen at a later time. The importance of the detailed structure of male palps and female epigynes has already been mentioned. Diagrams of these organs are given for most species in the standard identification literature. If you are experiencing difficulties with keys that do not seem to be working e.g. with the family Linyphiidae, you should not consider it a failure to revert to comparing your specimen to these diagrams. In such cases it is best to approach the task firstly by flicking through the diagrams to find those that appear similar in overall appearance to your specimen. This will usually narrow down the identity to a small number of related species. These can then be separated by looking for more subtle distinctions in the same way as you might in a spot-the-difference competition. Remember, if you are struggling to identify a particular specimen using one key it is always worth looking at a second key or reference work.

Reference collections

The value of a reference collection lies in the opportunity to compare problematic specimens with specimens that have been reliably named. In building their own reference collection, most arachnologists will probably aim to have a typical example of both sexes of each species along with examples of distinct, and potentially confusing, varieties they encounter. In order to be most useful the reference collection needs to be readily accessible. This will usually mean it is kept separately from the general collection. It is important to number each tube (whether in the reference or general collection) so that it can be cross-referenced in the computer database or card index.

The majority of arachnologists, and certainly the beginner, will soon find they need to look at specimens of species not present in their own reference collection. The personal collections of fellow arachnologists may provide access to missing species. Alternatively, one of the museum collections listed in Section 5.5 can be consulted.

Although not necessarily a part of the reference collection, voucher specimens are important in spider identification. At the very least, voucher specimens should be retained to support records of rare and difficult to identify species, or species outside their expected range, in unusual habitats or at unexpected times of year. A voucher specimen may be requested by the Verification Panel of the British Arachnological Society before a record is accepted.

Expert referees

All arachnologists encounter specimens that they find they cannot identify with total certainty. It may simply be due to lack of experience but some specimens will cause difficulties no matter how experienced the arachnologist may be. Possible examples include palps distorted by pitfall trap preservatives, physically damaged specimens or specimens with developmental abnormalities. There will also be occasions when uncertainty is caused by the rarity of a supposed species or by its discovery in an unexpected location. Whatever the reason, it is important that a second opinion is sought. For new BAS members their mentor may be able to help. Area Organisers will also be willing to provide assistance in such circumstances. Ultimately the specimen may be referred to the Verification Panel of the British Arachnological Society. This panel currently comprises:

- Ian Dawson, 100 Hayling Avenue, Little Paxton, St Neots, Cambs. PE19 6HQ
- Peter Harvey, 32 Lodge Lane, Grays, Essex RM16 2YP.
- Peter Merrett, 6 Hillcrest Drive, Durlston Road, Swanage, Dorset BH19 2HS
- Tony Russell-Smith, 1 Bailiffs Cottages, Doddington, Sittingbourne, Kent ME9 0TU
- Rowley Snazell, 10 Bon Accord Road, Swanage, Dorset BH19 2DS

Specimens to be sent to a verifier by post should be well packaged to prevent damage. Glass tubes should be completely filled with preservative or packed with a twist of tissue (not cotton wool) to prevent damage by air bubbles as the package is thrown around in the post. Each tube should be well protected e.g. by wrapping in bubble wrap and the protected tubes then placed in a re-sealable plastic bag. The bag should then be put in a rigid box or tin padded with tissue or further bubble wrap. Much of this is unnecessary with polypropylene tubes, when only some bubble wrap and padded bag are necessary. The box can then be posted in a padded envelope along with a covering letter and return postage. Tubes should not be sent in an ordinary envelope – these split or burst if any bulky items are contained.

4.2 Preservation and Display of Specimens

4.2.1 Types of Preservative

Alcohol

Alcohol, diluted with de-ionised water to about 70%, remains the best and most reliable medium for killing and preserving arachnids. Most collectors use the form of alcohol previously known as industrial methylated spirits (IMS) and now referred to as industrial denatured alcohol (IDA). This consists of ethanol (95% by volume) and methanol (5% by volume). However, the receipt and use of IDA requires authorisation from the National Registration Unit of HM Revenue & Customs. Obtaining such authorisation needs only to be done once and is not difficult. It certainly should not be seen as a problem for those commencing serious study of spiders. Propan-2-ol (iso-propyl alcohol) is a good alternative; it may be diluted with de-ionised water to 50% and, being more viscous than IDA, it evaporates more slowly. It can be bought and used without a licence. When mixed with IDA there is the minor problem of refractive index disturbance.

The main problems with alcohol are that it is flammable, volatile and evaporates rapidly. If specimens are allowed to dry out, they will be virtually ruined by distortions and will float when re-supplied with alcohol. The addition of 5% glycerine to the alcohol as an insurance against complete dehydration is not recommended. It causes glassware and specimens to become somewhat sticky and disturbances of the refractive index result when specimens are transferred to glycerine-free alcohol for examination. Should the alcohol evaporate completely, the glycerine concentration will increase to the point of clearing the specimen and may act as a culture medium for moulds, though this can be prevented by the addition of a small quantity of phenol or similar fungicide. The hygroscopic action of the glycerine may cause damage to a microscope if the preservative is spilt.

A secondary problem is that when specimens are killed in alcohol, there is a tendency for the legs to contract and obscure the underside. This can be avoided by using the entomologist's technique of killing with ethyl acetate vapour prior to preservation in alcohol. This should be done in a glass container as ethyl acetate will dissolve many plastics (but not polythene, polypropylene or nylon). Alternatively, the addition of 5% glacial ethanoic acid to the alcohol will extend the limbs of specimens as it acts to swell the material and balance shrinkage caused by the alcohol. Expansion of the palps of adult male spiders should not be a problem so long as the concentration of ethanoic acid does not exceed 5%. If specimens are required for DNA studies, they should always be preserved in 100% alcohol. The methanol in IDA causes the DNA (or RNA) strands to break, reducing the chances of obtaining usable base sequences.

Propylene phenoxetol

A 1% solution of propylene phenoxetol in distilled water has been used successfully as a preservative for arachnids. It has the advantages of being non-flammable, non-volatile and evaporates very slowly. It keeps specimens supple and helps to preserve specimen colouration including the red pigments that are usually lost in alcohol. However, this substance is a bactericide and a fungicide and **not** a fixative. Propylene phenoxetol cannot, by itself, arrest the autolysis of cells (i.e. self-digestion by enzymes), thus it can only be used after adequate fixation, in alcohol for example. It has been suggested that specimens should be killed in a solution of propylene phenoxetol because in this way they die with limbs extended and much more rapidly than in alcohol. The material should then be transferred to alcohol and fixed for 24 hours (more if specimens are large) before being returned to propylene phenoxetol for storage.

The problem with using propylene phenoxetol is that it is difficult to dissolve in water; it should be diluted to 1% with hot water and stirred very vigorously. Fortunately, the addition of propylene glycol to the propylene phenoxetol will greatly assist the preparation of the solution. This is known as Steedman's post-fixation preservative, and is composed of:

Propylene glycol 10 ml   Propylene phenoxetol 1 ml   Distilled water 89 ml

The phenoxetol and glycol must be mixed together before the water is added.

Other preservatives

In the absence of anything better, almost any form of alcohol (e.g. whisky, gin, surgical spirit or methylated spirit) may be used as a short-term preservative but, inevitably, the material will become fragile. Specimens should be transferred to a proper preservative, as described above, as quickly as possible and preferably within 24 hours. Formalin should **never** be used for Arachnida, it is a fixative rather than a preservative and causes specimens to become rigid. It is also a hazardous, carcinogenic substance.

Obtaining alcohol

Whereas propan-2-ol can be bought by anyone, the receipt and use of IDA requires authorisation from the National Registration Unit of HM Revenue & Customs. Further information can be found in their *Notice 473: Production, distribution and use of denatured alcohol*. This document can be downloaded from their website: http://customs.hmrc.gov.uk (Type "IDA" into the search box to find the relevant page). Alternatively, you can phone 0845 010 9000 to obtain a paper copy. It includes an application form for authorisation which requires you to give your name, address etc, to state the purpose for which the IDA will be used and your annual requirement in litres. Provided you apply for authorisation of no more than 20 litres per annum (more than adequate for BAS members) an application to use IDA "for the preservation of biological specimens for scientific study only" will normally be approved. If this is the case you will receive a formal letter of authorisation. Having received authorisation you should be aware that HMRC officers have the right to inspect the premises where you use IDA at any time although this very rarely happens. When purchasing IDA your supplier will need a copy of your authorisation letter. You can obtain IDA through specialist laboratory chemical suppliers although not all are willing to deal with members of the public and/or to provide small quantities. Alternatively, local high street pharmacies such as Boots will probably be able to help, although they may need to order it in specially. Many entomological equipment suppliers do not sell IDA because it is illegal to send it through the post.

4.2.2 Exhibition techniques

Being rather soft-bodied creatures, spiders and harvestmen cannot normally be preserved dry using entomological techniques such as pinning or carding. They will shrivel, become fragile and may develop mould if the environment is humid. However, they can be successfully freeze-dried for exhibition purposes, in a museum for example. Specimens freshly killed in ethyl acetate vapour are arranged on plastazote and held in position with pins. They are then placed in a freeze-drier for periods ranging from 24 hours to over a week depending on the size and sclerotization of the specimen. Moore (1977) gives further details of the technique. Skillfully presented, specimens can look life-like, but the colours will only be preserved when stored away from the light.

For display purposes, Kaiserling's colour preservation technique is an interesting method (Wanless 1969), but the procedure is complicated and success depends on the containers for the specimens being leak proof. The technique is really a novelty but it should preserve colours for several years (in the dark).

4.3 Storage and Curation of Specimens

Introduction

The traditional method of housing a collection is by means of glass tubes stored in jars of alcohol. Today it is common to employ polypropylene tubes in place of glass, which are reliably airtight and do not risk loss of specimens to evaporation of alcohol over long time periods. Glass tubes with plastic caps are commonly used, but a proportion of tubes always allow evaporation and unless tubes are stored inside jars of alcohol there is a high risk of loss of important voucher material. Screw-topped jars with plastic or bakelite lids are almost useless as the lids will inevitably crack with age. Cork bungs are not recommended; they discolour the fluid and are poor at preventing drying-out. Similarly, rubber stoppers are not ideal: they may lower the pH of the preservative and develop a poor seal.

In museum spirit collections it is standard practice to use jars with ground-glass tops. The ground-glass surfaces are smeared with vaseline to minimize evaporation, but some curators have found that this attracts dust and may harden, preventing the lids from opening. Ground-glass jars are expensive but among the many other kinds available, *Mason* or *Le Parfait* fruit-preserving jars (various sizes) with glass lids held on by metal clamps, are very reliable. Their rubber gaskets swell and make tight seals.

Tubes and closures

Many kinds of tube for specimen storage are available, although few are entirely satisfactory. Important decisions have to be made here about the reliability and ease of long term storage of specimens and, for this reason, polypropylene tubes have become very popular amongst arachnologists. They have the advantage of being light, leak proof and almost unbreakable though their slight opacity is less pleasing. They are available with either push-fit stoppers or screw caps sealed with an O-ring. The latter seem to be truly leak-proof unlike the majority of screw-cap tubes. Clear polystyrene tubes are NOT SUITABLE for storage, since the plastic is brittle and easily splits, leaks, and alcohol quickly evaporates.

Although traditional, slender glass tubes (12 × 50 mm and 25 × 75 mm) are commonly used due to the high visibility of both labels and specimens. For such tubes, the most satisfactory polythene closures are those which have a kind of screw thread, although the larger sizes, in particular, are still prone to leakages. Also popular are glass tubes which resemble diminutive bottles, with necks, capped by snap-shut polythene lids. Any tube, or for that matter bottle, with a screw-threaded cap which relies on a kind of washer to make the seal should be treated with caution. If a collection is to be accommodated in glass tubes, without jars, security will be improved by placing smaller tubes inside larger ones. This will guard against problems such as poor seals, cracks in the glass and lids lifting because of changes in temperature, etc. Whether the tubes are stored in jars or not, they should be checked at frequent and regular intervals. Tubes showing unusually high rates of evaporation should be replaced and any tube showing the slightest loss of preservative should be topped up.

Microvials and slides

Sometimes it will be necessary to secure excised parts, e.g. genitalia, in a small vial inside the main specimen tube. Glass or polythene microvials can be used but must be prevented from moving about to avoid the risk of damaging the specimen. A plug of kitchen paper can hold the microvial in place but cotton wool should never be used. Alternatively, small gelatine capsules can be used to store small parts and are less likely to damage specimens. Some collections of Arachnida will include material mounted on slides. Structures such as spider vulvae, harvestman penises, and pseudoscorpions, in whole or in part, will need to be examined on slides under the high power of a compound microscope. The material may be mounted temporarily using glycerine or 80% lactic acid and then returned to microvials, or mounted permanently using Canada balsam or gum chloral (gum arabic, glycerine and chloral hydrate). In many cases it is advisable to make only temporary slide mounts because material on permanent slides can deteriorate with age.

Labelling

Full and accurate labelling of the collection is essential; a specimen without a label has virtually no scientific value. Labels should be clearly written on quality paper or thin card and inserted into the tube with the specimen immediately. The label should be written using pencil or alcohol- and water-proof, permanent black ink (e.g. a fine *Rotring* or *Pilot* water-resistant drawing pen). Printed labels are used by some arachnologists but the long-term reliability of the inks has yet to be tested. Labels should be placed inside every tube (labels on the outside are much more likely to be lost or rendered illegible) and on every permanent slide. It is bad practice to confine data to notebooks through a system of reference numbers, because the notebook may become separated from the collection and lost. Data on each label should include the minimum of locality, grid reference, habitat, collector's name, date and serial number if indexed. The name of the species and of the determiner should normally be given as well but if the species has not been identified this can always be added later. Ideally, the specimens from separate localities, seasons and habitats will be stored in separate tubes.

Storage space

The space available for storage may be used most efficiently by splitting the collection into two parts: e.g. a reference collection containing a limited number of specimens arranged for ease of access, and a main collection holding the larger series of comparative material. If the reference collection is to contain, say, three or four examples from 75% of the British spider list, then storage will be needed for up to 1500 tubes. A *Stor* cabinet (Crocker 1969) of 10 or 15 trays is ideal for a collection involving tubes only. Each tray, foam based, will hold numbers of tubes standing upright in a horizontal grid made from some sort of drilled sheet, small mesh chicken wire or fluorescent light-diffusing panel. Alternatively, plastic racks designed for holding tubes of various sizes are available from laboratory equipment suppliers.

4.4 Cataloguing and Record-Keeping

Notebooks

The collector will probably find it useful to maintain at least one notebook. A primary notebook containing entries made in the field can be used for details of field trips, dates, habitats, specimens collected, collecting techniques, specimens photographed and any other relevant or interesting observations noted down before they are forgotten. Although every arachnologist will have their own favoured style, the field notebook is best organised around sub-divisions of each collecting site visited on a particular date. Examples of such sub-divisions would be different habitats on the site, different microsites within a habitat or the use of different collecting techniques. Space should be left to add details of taxon name, determiner and any other relevant data (including the location of voucher specimens) when eventually the specimens are identified. (In the past a second laboratory or determination book was often kept to record this information but this practice is much less common nowadays.) An A6 format is probably the most convenient for slipping into a pocket and a case-bound notebook is more hardwearing than the spiral-bound equivalent. Some designs incorporate a strip of elastic for holding the book closed when not in use but a strong elastic band will do the job equally well. Field notebooks produced from waterproof paper are now available although not yet in common usage amongst arachnologists.

In addition to the field notebook, a loose-leaf file, which includes a section indexed by genus, may be useful for items such as descriptions of new genera and species, nomenclatorial changes and bibliographical references. It, or another notebook, may also be used as a collection notebook wherein the tubes of specimens in the collection are listed and numbered.

Computing

Specimens should be easily retrievable by virtue of arrangement, card index, notebook or computer. Simple spreadsheets or databases may be used to store and retrieve the data, names and serial numbers involved in a collection. Entries can then be sorted and listed according to a number of criteria e.g. genus, sex, locality, date, etc. The ability of computers to produce an immediate print-out is an obvious advantage and there is little excuse for working exclusively with paper nowadays.

The Spider Recording Scheme encourages the use of the *Mapmate* biological recording software package for the collection and submission of data. Although, the software is primarily aimed at collating distributional and ecological data, information on voucher specimens held in reference collections should be included when inputting records. The existence of such a specimen supporting a biological record is considered an important piece of information.

Final note

If planning to offer a collection to a museum, please ensure that it is well organised, well maintained and, above all, clearly labelled. It should not be assumed that a museum will automatically accept such an offer nowadays. Lack of resources to curate collections, lack of space to store collections, and concerns over the legality of material collected abroad are all issues that may influence decisions. Also, the more conditions a donor wishes to place on an offer the less likely it is that the museum will feel able to accept.

4.5 Important British Arachnid Collections in the UK

Janet Beccaloni

Introduction

There are many institutions around the UK that possess British arachnid collections. The following chapter provides details of the major holdings around the country but are by no means exhaustive! The order in which they are covered does not denote order of importance.

London

The Natural History Museum (NHM) in South Kensington, London, is housed in a building which was designed by Alfred Waterhouse, and opened to the public in 1881. The foundations of the NHM were the collections of the Ulster doctor Sir Hans Sloane (1660–1753) which included animal, human and plant specimens purchased by the British Government. In the 1850s it was recognised that the collections needed re-housing, and that is how they came to South Kensington. The NHM now houses over 70 million natural history specimens, and is a centre of research which specialises in systematics and identification. As well as their scientific importance, many of the specimens also have great historical value, having been collected by Joseph Banks, Alfred Russell Wallace and Charles Darwin.

Arachnida collections

The Museum's entomology collections are the most comprehensive in the world. The National Collections of insects and other terrestrial and freshwater arthropods, including Arachnida and Myriapoda, comprise an estimated 28 million specimens. The Arachnida and Myriapoda spirit collections were recently re-housed in Phase 1 of the new Darwin Centre, adjacent to the main Waterhouse building. All the collections are databased to species level.

Because the Arachnida collections have global coverage, there has been no provision for separate British collections and nearly all the British arachnid material is incorporated into the main collection. However, there is a small core of British spiders, formed mainly by the Jackson collection from the 1940s, which is kept separately. It comprises around 600 jars of identified spiders, arranged in alphabetical order under genus. The J. H. Murgatroyd collection from the 1950s also forms part of this core and comprises around 100 jars of identified material, also arranged in alphabetical order by genus.

The D. J. Clark collection comprises both identified and unidentified British spiders. However, these are mixed together in jars, and need to be sorted. Other known collectors of British material include A. E. Le Gros, Peter Jerrard, Dick Vane-Wright, Fred Wanless, Tony Russell-Smith and Rowley Snazell, together with other BAS members. These specimens are incorporated into the main collection. Additionally, there are several jars of mixed identified material that needs to be separated out, plus a few jars of unidentified material from places such as Cornwall, the Isle of Wight, Ireland, Jersey and Sherwood Forest.

Oxford

The Oxford University Museum of Natural History dates from the 1850s, when the University's Honour School of Natural Science was founded. The University's collections were scattered around the city of Oxford, so a neo-Gothic museum building was constructed in 1855–60 to bring them all together. The largest part of the museum's natural history collections are from the Ashmolean Museum, with many specimens collected by John Tradescant (the elder), William Burchell and William Buckland. The Hope Department containing the entomological collections was founded by the English entomologist Frederick William Hope.

Arachnida collections

The Hope Department is famous for possessing the Reverend O. Pickard-Cambridge collection of Arachnida. There are about 5000 jars of all sizes, many holding from 2 to 20 separate tubes, comprising an almost complete British collection. This important collection contains many hundreds of type specimens, not only those described by Pickard-Cambridge but also by John Blackwell and a considerable number from eminent specialists such as Simon, Koch and Thorell. There is also some material from F. O. Pickard-Cambridge, nephew of O. Pickard-Cambridge, included within the collection which was presented *circa* 1917. Additionally, there are parts of John Blackwall's collection, which was presented by O. Pickard-Cambridge along with his own material.

Manchester

The origins of the Manchester Museum at the University of Manchester, date from 1821, when the collection of John Leigh Philips, a Manchester manufacturer, was purchased by a small group of wealthy men and turned into the museum of the Manchester Natural History Society. In 1868, the museum was transferred to the University of Manchester. The Manchester Museum collections reached their present resting place in a building designed by Waterhouse, opened in 1890.

Arachnida collections

The Manchester Museum has holdings of British spiders and harvestmen. Not all the collections have currently been fully documented and counted, so the following is provisional data.

- G. Locket Spider Reference Collection: 492 species, 5336 specimens; this is the main collection, but there are some unrecorded areas (a couple of hundred tubes).
- Crocker Spider Reference Collection (mostly from Lancashire): 108 jars, 2843 tubes. Information on this collection was published in the BAS *Newsletter* in 2004.
- H. W. Freston and D. W. Mackie Reference Collections of Spiders and Harvestmen: relatively small collections, each of several hundreds of tubes. Freston mostly collected in the vicinities of Manchester, but Mackie also worked in Ireland and Scotland.
- D. V. Logunov Collection of Spiders and Harvestmen from the vicinity of Manchester (Alderley Edge): 114 spiders and 11 harvestmen; 1283 specimens altogether.

Dr Dmitri Logunov, curator of arachnids, has specifically mentioned that he is happy to deal with any spider-related enquiry and to help with identification, and that the Manchester Museum's arachnid collection is fully accessible for anyone who needs to (re)examine any retained specimen.

Liverpool

The National Museums Liverpool, or World Museum Liverpool as it was renamed in 2005, was opened in 1860. The originally collection was formed from the Earl of Derby's bequest, but rapidly grew over the years with donations from private collectors such as Joseph Mayer. The World Museum Liverpool holds large zoological and botanical collections.

Arachnida collections

World Museum Liverpool's arachnid collection has undergone a period of rapid development during the last 15 years and includes arguably the finest contemporary collection of British spiders. The British collections of other arachnids are comprehensive, although mites are less well-represented. WML's spirit collection returned to a new store within the museum building in 2007 and is easily accessible again for visitors. Work is also underway on an updated catalogue of British spider holdings. The WML enjoy very close links with the British Arachnological Society and administer its reprint library.

British spiders & harvestmen

British spiders and harvestmen include approximately 160,000 specimens and provide 85% species coverage. The W. E. Falconer collection was principally from northern England and assembled during the first three decades of the twentieth century. The remainder of the material is incorporated into a single main reference collection. The extensive modern collection of C. Felton primarily contains specimens from north-west England and Wales. Material from D. R. Cowden and R. Leighton, and recently acquired collections of C. G. Butler and A. G. Scott are also present.

Pseudoscorpions

The P. D. Gabbutt collection of *c.* 14,000 spirit and slide-mounted specimens of British pseudoscorpions, including adults and all instars for the majority of the British species comprise the most extensive pseudoscorpion collection in Britain.

Mites

Coverage of mites is less comprehensive. Economically important species (MAFF collections) are represented, and the museum has recently acquired the M. Luxton collection of British Oribatida and its associated reprint library.

Cardiff

The National Museum Cardiff was opened to the public in 1927. It forms part of the Edwardian civic complex of Cathays Park, and is part of the wider network of the National Museum Wales.

Arachnida collections

The Arachnida collections at Cardiff contain over 10,000 specimens, which represent nearly all British families and approximately 65–70% of all British spider species. Cynthia Merrett formed the basis of the collection in the 1970s and 1980s. She also collected specimens from particular habitats, e.g. the Welsh peatland survey in the 1990s. G. W. Garlick collected extensively across the Welsh vice-counties between the 1960s and the early 1980s. There is a computerised checklist to species, with the specimens arranged under families. There are currently around 3000 records on *Filemaker*, and anything of notable standing is on the database. The collections are used quite regularly by amateur groups, such as the South Wales Arachnid Group (SWAG) for workshops etc.

Edinburgh

The University of Edinburgh was founded in 1582, and is one of the largest in the UK. The School of Biological Sciences at the University of Edinburgh is one of the largest and most comprehensive in Britain.

Arachnida collections

The University holds a very large collection of British ticks. They have a complete database, which lists the taxonomic name, sex, number of specimens, preservation method, location in collection, date of collection, collector, country, host, and any extra details.

Warrington

The Museum and Art Gallery at Warrington, was founded in 1848. The natural history collections relate to Warrington and the surrounding areas, with some that have national significance.

Arachnida collections

The most recent audit of the British arachnids at Warrington was undertaken by Chris Felton during the mid 1980s, when he wrote a truly extensive report on the Linnaeus Greening collection of British spiders, which was mainly formed between 1895–1902. The collection consists of an estimated 864 tubes containing around 2565 specimens. The nucleus of the collection is very much a local one, reflecting chiefly the spider fauna of Warrington and its immediate surroundings. Additionally, a feature of the collection is the interesting selection of typically southern spiders from Chatteris, East Grinstead, Ashdown Forest and Titchfield. There are also many specimens from other parts of Britain, including Wales, Scotland and Ireland. Most of these were collected by Greening personally, while others were contributed by friends, in particular G. A. Dunlop.

Other institutions that house smaller collections

It is impossible to know every single institution in Britain that has British arachnid holdings. However, the following places have smaller collections.

- Bangor University
- Dorchester Museum
- Brighton Museum
- Norwich Castle Museum
- Glasgow Kelvingrove

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5. SPIDER PHOTOGRAPHY

Craig Slawson & Chris Spilling

5.1 Introduction

Photography is a very large subject, and this is only intended to be a general guide for those wishing to photograph spiders. Anyone wishing to go into more depth, should refer to a good book on macro-photography, which will include many techniques suitable for photographing the smaller spiders. The skill required to actually find your subject is also not covered, since this is important in all aspects of field arachnology.

5.2 Equipment

Initially, you will need to decide what type of camera to use. This is important, because it is probably the most expensive purchase you will make. There are two different types of camera available at present, those that use film and the more recently developed digital cameras.

5.2.1 Film Cameras

Film comes in two forms, negative (producing prints on paper) and positive (producing slides or transparencies). Because it has been available for many years, there is a wide variety of brands/sizes/speeds, etc. Its main advantage has been in the quality obtained. The definition of an image on film has been much better than that obtained with most digital cameras, but the gap is closing rapidly. In addition, the cameras are generally cheaper than the equivalent digital models and often do not require batteries. Some people prefer 35 mm slides when presenting slide shows for talks as they require simpler projection equipment than that needed for digital images. The main disadvantage of film cameras is running costs; film is not a reusable medium and so must continuously be purchased.

5.2.2 Digital Cameras

Digital cameras have the advantage of being cheap once the initial costs of the camera have been met, but image quality may be slightly lower than film for all but the most powerful models. Other advantages are that the results of each shot can be viewed immediately and images can be copied indefinitely without loss of quality. Disadvantages include the fact that digital cameras are relatively expensive to purchase once you leave the realms of 'point-and-shoot', all cameras require batteries and the fact that you need a digital projector for shows. However, as their popularity increases, prices are continuously falling, and both the image quality and price gap between film and digital cameras have narrowed considerably over the past few years.

5.2.3 Camera Format

There is now a very large variety of both film and digital cameras available and it is important to understand the advantages and limitations of the different formats. There are four main types of camera suitable for spider photography, each with its own pros and cons.

Medium format cameras

This category includes any camera producing an image size greater than that of 35 mm film (36 × 24 mm). The popular sizes are 6 × 4.5 cm and 6 × 6 cm. At present, large format digital cameras are extremely expensive. The principal advantage of these cameras is that image quality is the best of all four categories. Disadvantages include cost (both cameras and lenses are expensive), their weight and bulk which make them cumbersome to use, and that film is only available in 12 or 15 shots per roll.

35mm single-lens reflex (SLR) cameras (interchangeable-lens type)

This has been the most popular and versatile type of camera since its invention. The advantages are availability of a very wide range of makes and an even greater array of lenses and other accessories to aid the photographer and the fact that film cameras are relatively inexpensive. Digital cameras with a 35 mm equivalent image size have only recently appeared on the market and are still relatively expensive, although prices have fallen sharply over the past few years.

Small format SLR Cameras (interchangeable-lens type)

Film cameras smaller than 35 mm are not ideal for spider photography as they tend to be either point-and-shoot or very specialist. However, this is the growth area in digital cameras. Most look very like their 35 mm film equivalent, but their image size varies depending on the number of pixels (dots) making up the picture. This ranges from 3 megapixels (mp) up to around 12 mp. Digital small-format cameras are more expensive than their 35 mm SLR equivalents but many can use lenses and accessories from existing 35 mm SLR ranges.

Bridge cameras

This term is almost exclusively applied to digital cameras. These cameras have a relatively large image size 3–10mp, a variety of built in controls over exposure, etc., but most importantly, they have a fixed, i.e. non-interchangeable lens. This is usually a zoom lens from 3× to over 10× zoom with a macro facility. The term bridge camera comes from the idea that these cameras bridge the gap between point-and-shoot and SLR interchangeable-lens cameras.

Many of these cameras have an SLR-type of viewfinder (i.e. allowing you to see the view you are shooting, through the lens) consisting of a tiny LCD screen, but the majority also have a preview screen on the back of the camera. Advantages include a reasonable image quality for most purposes and relative affordability compared to digital SLR cameras. Disadvantages are that the screen on the back of the camera cannot be used to preview the image and that you are limited to only one lens.

Cameras which are not suitable for spider photography include:

Point-and-shoot. These cameras are very cheap, but usually have a relatively limited lens, often focus-free or fixed focus. They are fine for landscapes and family photographs, but are totally unable to photograph close-up or small items.

Large format film cameras. The image quality of these is beyond comparison, but their problem is that they are too large to be portable enough for field work and are only suitable for studio or landscape photography.

Recommended camera

For versatility and portability, the best option is the interchangeable-lens 35 mm SLR or digital SLR. However, digital SLR cameras are expensive and if your budget is limited you might think of purchasing one of the better quality bridge cameras, at least initially.

5.2.4 Lenses

This section applies mainly to SLR cameras, though accessory lenses can be bought for some bridge cameras. Specialist lenses are necessary to photograph spiders because of their small size. There are several alternative types of lens, depending on the subject matter.

Standard lens with accessories

Using your camera's standard lens (50 mm with a 35 mm SLR camera) is obviously the cheapest alternative. The standard lens is perfectly acceptable for photographing webs, but it cannot focus close enough to photograph the spiders themselves. There are several methods of achieving the close-up effect, but remember: once the close-up attachment is in place, the lens will no longer focus on distant objects. Two types of accessory are available:

Close-up lenses – these are magnifying lenses which screw on to the filter thread of the lens, allowing it to focus in closer. They come in different strengths but all tend to cause degradation in the quality of the image, particularly towards the edges, often producing colour-fringing on objects unless very expensive multi-element lenses are used.

Extension tubes – these are fitted between the lens and the camera so moving the lens further away from the film/sensor allowing it to focus more closely. Extension tubes have no optical elements, so do not degrade the image. However, if you exceed a life-size image (1:1) then quality is reduced, i.e. standard lenses are designed to produce small images from large subjects. This can be corrected by reversing the lens, so the front points towards the film. Extension tubes come in a variety of fixed lengths, but there is an infinitely variable system which uses a concertina of opaque material known as a bellows. Most cameras lose most of their automatic controls when used with bellows or with the lens reversed.

Macro-zoom lens

Macro-zoom lenses are normal zoom lenses which have a close-focus option, usually at one extreme of the zoom, whereby the lens will focus relatively close. This can usually attain quarter (1:4) or half (1:2) life size. These have the advantage of still focussing to infinity, but are only really suitable for the larger spiders such as *Araneus* and *Tegenaria*.

True macro lens

A true macro lens is usually designed to focus from infinity to life-size (or better) without any additional accessories. This is really the minimum requirement for spider photography and the ideal is a lens reaching four times life size on the sensor. However, this can be achieved by adding extension tubes to the macro lens (although it will then lose its ability to focus to infinity). A longer focal length is better than a shorter, because this allows a longer subject-to-camera working distance. The spider is then less likely to be disturbed by the camera and photographer and it is also easier to position the flash.

Recommended lens

For larger spiders (say, 5 mm body length or longer), the macro lens is quite adequate, producing a life-size image. However, for the majority of money spiders (Linyphiidae) and other small spiders, the use of a small extension tube is advisable, to give images around 2–4 × life-size. This is more difficult to use, because of its reduced depth of field and inability to focus to infinity, but is necessary for high quality images.

5.2.5 Accessories

Close-up photography often requires a variety of accessories, because of its specialist nature. The most important is probably a flashgun or some other additional lighting. This is because close-up work often produces a dim image, even with a wide lens aperture. In addition, close-up also generally produces a shallow depth of field. This depth can be increased by using a small lens aperture, further decreasing the amount of light available to form the image.

Lighting

There are a number of different types of lighting. Flood-lights have the advantage of allowing you to see exactly what the finished photograph will look like (including shadows, etc.) but, for fieldwork, these have too many disadvantages. First, they need a permanent power supply, either mains electricity or large batteries, neither of which is really convenient in the field. Second, they heat up the subject, which makes the subject more active and, in many cases, more likely to move away from the heat source.

A number of modern cameras have a built-in flash. These can work, but a single flash close to the camera can create very dark shadows and can be obscured by the front of the lens at very short camera-to-subject distances. Similar problems are associated with a single off-camera flashgun. Multiple flashguns, properly arranged, give a good lighting effect, with reduced shadows and a natural look to photographs. The disadvantage of this system is that it can be cumbersome to carry around. The flashguns can either be attached on the ends of a bar fastened under the camera or to a ring mounted on the lens filter ring. The latter is usually only available from the main camera manufacturers, whilst the former can be built from off-the-shelf items, making it much cheaper.

The most convenient form of lighting is a ring flash, which mounts on the filter ring of the lens with the flashtube surrounding the lens. The two disadvantages of this are (1) almost completely shadowless lighting giving a very flat effect (although this can also be an advantage in certain circumstances) and (2) any reflections of the flash appear as tiny doughnuts rather than just dots, which can be distracting.

Camera support

In most cases, it is most convenient to hand-hold the camera to allow easy tracking of the subject. However, for certain subjects it is better to have the camera mounted on a sturdy fixture. The most popular is a tripod, but for spider work it is often necessary to fix the camera at almost ground level. This is achieved either by reversing the centre column or by using a versatile tripod like the *Benbo* which allows the centre column to point in any direction.

5.3 Techniques

5.3.1 Field photography

A prerequisite for spider photography in the field is patience and perseverance. In addition, it helps to know something about the biology and behaviour of your subject. Knowing where and when the species can be found can save an enormous amount of time searching for it. Many spiders can be too active to be photographed, but if the spider is guarding an egg-sac, caring for young, engaged in courtship display or consuming prey it is more likely to remain still. Often spiders sun themselves on leaves, walls and other surfaces. In this position, they should be approached slowly because they will escape rapidly if disturbed when warm, but take care to avoid shadows from yourself or the camera falling on the spider. A number of other factors will combine to make photography difficult e.g. wind, rain, distraction by other invertebrates (flies, wasps, etc), inadequate lighting or operator instability. There are a number of techniques and pieces of equipment which can be used to alleviate some of the difficulties.

Camera settings

As mentioned in a previous section, the best option for spider photography is to use a 35 mm or digital SLR camera and the notes here refer to such cameras. Using the autofocus setting in the field does not necessarily bring key areas into focus and use of manual focussing can provide more control. Setting speed and aperture manually when using electronic flash is also advisable. The shutter speed will be dictated by the requirements of the flash unit (usually no faster than 1/250th second), whereas the flash duration will be very much briefer and will occur at the mid point of shutter opening. For macro-photography use lens apertures of f11–f32 depending on the depth of field required. If you are getting a more blurred image when using your lens with a small aperture, try opening up the aperture to f11 or f16. For more information on this sort of problem refer to a technical reference.

Some cameras have other facilities. For example, with through-the-lens metering the camera and flash unit combine to compute the length of flash duration required for your particular speed and aperture setting. Some of the modern digital SLR cameras have an image stabilisation feature and this can also be helpful. Refer to your camera manual for details.

Controlling camera and subject movement

Use of a tripod or a beanbag on which to rest the camera can reduce camera movement, but in the field these can be cumbersome unless you have a very still, co-operative subject. The use of a cable release to take your shot and the mirror lock facility (if you have one) can help to control vibrations transmitted via the operating button and mirror swing-up mechanism respectively. Your camera operating manual should give you details on how to set this up. A high shutter speed is another way of freezing movement but this will often be unavailable to you because normally you will need slow shutter speed to gain adequate exposure of your subject.

Movement of plants on which your subject is located can sometimes be reduced by staking the plant, grass stem or twig in place with a small cane or knitting needle and Velcro ties. When taking your photo make sure that you don't have any stems going across your subject. It is easy not to see them in the viewfinder when you are focussing on the spider.

Lighting

As mentioned previously, the ability to obtain adequate light is often a problem. Many spiders will shun the light when disturbed and so will be difficult to photograph in sunlight. This problem can be overcome by using an electronic flash, either hand-held or camera-mounted. Purpose built units (usually a small powerful, adjustable flash unit mounted each side of the lens) are more expensive but can have the advantage over manually set programmable flash output. If using hand-held units, try them out at various angles and distances from your subject to get the required effects. It is best to do this before you go out into the field so that you know what settings are required when the opportunity arises. A disadvantage of electronic flash is that it can give rise to deep shadows and an unnatural look especially if the background is several inches away from the subject.

5.3.2 Photography in Captivity

It is often not practical to photograph spiders in the field and specimens need to be brought back home to be photographed. Most techniques used indoors are similar to those used in the field. However, there is no wind and rain to contend with, you have the ability to control light more effectively and the time and steadiness to focus more accurately. Again, electronic flash can be used to good effect and does not have the problem of heat build-up that you can get with other lighting sources. The use of a reflector (e.g. pieces of white card or foil) to produce a softer light or the use of a background card is also easier at home.

The creation of the correct habitat in which to photograph your spider is important. Try and emulate the habitat in which the spider was found, or look up its habitat requirements in a reference book. Suitable twigs can be set up in an open fronted vivarium where orb weavers can weave their webs.

Very high precision in focussing can be obtained by the use of focussing rails mounted on an optical bench. Such a bench can be made from a thick piece of plywood with rubber feet screwed underneath. Focussing rails are also useful when using bellows extensions on your camera for high magnification. However, your specimen may have complete disregard for your high technology and prefer to amble around, hide or generally try to escape being photographed. Although a short period in the fridge may slow the subject down a bit, this does not always work, and it may leave the animal covered in fine water droplets, spoiling any picture taken. Again, patience is required, and the spider will normally eventually settle down to rest.

Because lack of depth of field is often a problem, some new computer software has been designed to overcome this. The technique involves taking a sequence of photos focussing down through the depth of the subject. These photos are then processed into one image on the computer using software which, hopefully creates a photo where the spider is in focus throughout the depth of its body. Such a sequence is more easily taken under controlled conditions indoors. Photographic processing software packages and their use are not addressed in this handbook but details can be found in many digital imaging books and are constantly being updated and changing. *Adobe Photoshop Elements* is a very useful software package and relatively cheap to buy.

Most important, having completed your indoor photography please remember to return your specimen unharmed to its appropriate habitat in the wild.

Photography down a microscope

To photograph highly enlarged specimens and even individual organs such as the epigyne or palp as an aid to identification requires more than the standard camera or even macro-lens. Using a microscope can give this extra magnification and it can be achieved quite cheaply. Unlike identification, photography does not differentiate between stereo (binocular) and monocular (high-power) microscopes with only one lens. The difference is irrelevant to the camera; both types of microscope produce the same result. The main difference is lighting: binocular microscopes usually use reflected light to view opaque subjects, whilst monocular microscopes use transmitted light to view transparent ones. Again, for photography, these distinctions blur, and both types of microscope can utilise either light source. The main distinction between stereo and high-power microscopes is the fact that stereo dissection microscopes correct the orientation of the image so everything is the right-way-up. High-power monocular microscopes do not, so that everything is upside down and left and right are reversed. For static subjects this is not a problem but it can be very difficult to follow a moving subject on a high-power microscope without a lot of practice.

There are three main methods of photography down a microscope (photomicrography). The three types of photomicrography are differentiated by the number of lenses used:

5.4.1 Three-lens Set-up

Although utilising the most lenses, this is in fact the simplest and cheapest but also the one producing the poorest quality images. The camera, complete with its lens, is simply placed against the eyepiece of the microscope. No other special equipment is required (the third lens is the microscope objective). The simplicity of this method allows relatively inexpensive digital cameras to be used for quite acceptable results.

There are several important factors to be taken into account:

- The camera must be mounted rigidly with the microscope. This can be either using a rigid tripod or an attachment direct to the camera – the latter is essential if the microscope body moves when focussing.
- The camera should be aligned accurately with the microscope to reduce any distortion of the final image
- The camera's autofocus must be switched off and, ideally, the camera should be focussed to infinity. All focussing should be done from the microscope
- It is best to form a baffle around the camera lens to avoid extraneous light entering from around the microscope eyepiece.

- MOST IMPORTANT: it is essential that the microscope eyepiece does not touch the camera lens. This can scratch the front of the camera lens, reducing its quality for all future photography; either use a filter to protect the lens, or a spacer to prevent the lens touching the eyepiece.

This option is only really feasible with digital bridge cameras; it is impossible to use non-SLR film cameras because you do not see through the lens and cannot frame or focus the photograph. In addition, SLR cameras have a relatively large lens which will gather very little light from the eyepiece of the microscope.

5.4.2 Two-lens Set-up

This set-up requires a camera with a removable lens, either a digital or film SLR. The camera body is attached to the eyepiece by a rigid tube. The two lenses are the microscope's eyepiece and objective.

5.4.3 Single-lens Set-up

This is basically the same as 4.2 above but the tube is mounted directly above the microscope objective, dispensing with the eyepiece. The microscope objective can even be replaced by a micro lens which has the advantage of an adjustable aperture.

5.4.4 Lighting

With photomicrography, lighting is the key to good results. When using a microscope, there is no aperture setting or f-number (unless you are using a micro-lens), so changes in exposure can only be made by altering the light intensity when using flash, or intensity and shutter speed when using flood lights. The microscope transmits very little light when compared to a normal camera lens so even a quite powerful flashgun might only have a working distance of a metre, hence the light on the subject can be quite intense. For reflective lighting, some method of reducing shadows is also essential, using either a second slave flashgun or a reflective surface, otherwise shadows can be absolutely black and distracting. For live specimens flood lights are probably too hot for use in photomicrography. This also applies to specimens under alcohol where the heat can cause distracting convection currents. Fibre-optic cold light sources work well with specimens in alcohol and have the advantage of giving a light to focus by.

Transmission microscopes normally only require one light source. This can be focused by the microscope to produce a high intensity spot on the subject if it is fitted with a condenser (usually only fitted to high-power microscopes). Transmission lighting can be adjusted to light only the subject and give a black background, known as dark-ground or dark-field illumination. This can improve the clarity of very transparent subjects, but setting it up can be quite complex and is beyond the scope of this article. Readers should refer to a book dedicated to photomicrography if they wish to proceed beyond the basics outlined here.

5.4.5 Conclusions

There is little to choose between the single- and dual-lens set-ups. Usually because there is less glass, the single lens will give better results, but the dual-lens can give more control by changing eyepiece lens magnification. The three-lens system obviously gives the poorest results because of the number of lenses and the difficultly of accurate alignment and exclusion of extraneous light but, even then, with practice, very good results can be obtained. It is most important to prepare the subject carefully and light it well, when very good results are possible even with the simplest set-up. There is no substitute for experimentation, and it is important to record the settings used for future occasions, to avoid repeating the same tests again.

6. KEEPING SPIDERS IN CAPTIVITY

Bill Blumsom

6.1 Introduction

This chapter is aimed at those who wish to keep spiders in captivity at home, whether for general study of behaviour, photography, or simply to raise specimens to maturity for identification purposes. The information and suggestions are based on my own personal experience of keeping British species over a number of years together with much valuable information gleaned from reading material by W. S. Bristowe, Frances Murphy and others whose articles have been published in the British Arachnological Society *Newsletter*.

There are no hard-and-fast rules when it comes to keeping spiders and much has still to be learnt about this fascinating subject but, hopefully, what follows will provide a good foundation on which to build and will encourage people to develop new methods. Frances Murphy put it very succinctly in some guidance notes she produced for the BAS *Members' Handbook* some years ago: "I consider that the secret for keeping spiders successfully is to have enough interest or even devotion to make one watch the spider carefully. One will then get to know how a healthy spider behaves, the appearance of a thirsty spider, a spider that wishes to be left in peace to moult, and so on. "

6.2 Do you Always Need to Confine the Spider in Captivity?

The starting point for many naturalists is probably observing spiders in their natural environment. In fact, for some of our more familiar and commonplace spiders found around the home, it is not really necessary to keep them in captivity at all. As Geoff Oxford's interesting article on *Tegenaria* in a recent BAS *Newsletter* pointed out, you can observe spiders regularly in a garage, outhouse or conservatory and learn far more about their behaviour than when they are kept within a cage (Oxford 2007).

It is simple and easy to create spider-friendly habitats in a garage or shed with carefully placed airbricks and flower pots or simply a pile of newspapers or old birds nest left undisturbed on a shelf. In a conservatory or greenhouse, strategically placed houseplants can provide excellent opportunities to observe the spiders without having to feed and care for them. If accessible areas and some shelves are left undisturbed, the spiders soon move in and, if there are particular species you wish to observe, you can introduce them to suitable locations. My own house has a shoe cupboard that has a small colony of *Pholcus* (the only area in the house my wife grants them amnesty). The grape vine in the conservatory has a rapidly growing colony of *Uloborus plumipes* which have now survived two winters in the unheated room and successfully bred. The shed, with carefully placed airbricks, flower pots and corrugated cardboard, has plenty of *Amaurobius similis*, *Steatoda bipunctata*, *Harpactea hombergi* and *Tegenaria* spp. in residence. The outside of the property and sheds can also be adapted to encourage more spiders into locations where they are easily observed. Provision of assorted tubes, crevices and retreats at appropriate heights can make night-time observation and photography of species such as *Nuctenea umbratica* and *Zygella x-notata* much easier. At night, it is possible to observe spiders in a completely natural state with a torch.

6.3 Where was the Spider Found?

It will be impossible to observe many species regularly without capturing and confining specimens. Understanding as much as possible about the spiders habitat requirements before keeping it in captivity will help to recreate them and increase the chances of keeping the spider alive. If it is a spider you are unfamiliar with, it is worth taking note of the habitat and conditions you have found it in. If possible, watch the spider *in situ* for a while before catching it, study its web, the structures the web is attached to, and the surrounding general conditions. Is the spider in the centre of a web? Low down in grass or vegetation? Inside a curled leaf at the top of a plant or hidden in a tubular retreat? If found in a shed or house, its requirements will be very different to one you found in woodland or leaf litter.

If possible, use some of the natural material from the site you found the spider in the cage at home, but be careful to minimise any damage to the location. It is also valuable to study the available literature to find out as much as possible about the species and its general habitat and feeding needs. A wide range of literature on the topic exists, some of which is listed at the end of this chapter.

6.4 Why Keep the Spider Alive?

The reason for keeping a particular spider in captivity will also be a key factor in the type of housing provided. The reasons for keeping spiders vary but, generally speaking, fall into three categories:

- Immature specimens to be kept until they reach maturity for identification purposes. These are normally kept for relatively short periods in test tubes or small pots with just a little cover as they will be killed and identified as soon as they complete the final moult. If it is a large spider or a web builder, ensure the container does allow enough room for the final moult to actually take place. It is also well worth feeding immatures a little more regularly as there is some evidence that well-fed spiders mature more rapidly.
- Long-term study of a species where you wish to learn more about its biology and behaviour and therefore want to keep it in conditions as close to the natural habitat as possible. This tends to mean more elaborate housing and care in creating a good balance between the wellbeing of the spider and ease of observation.
- Photography was explored in greater depth in Chapter 5. In general, specimens needed for photography will only be kept for short periods.

6.5 Housing

Having captured the spider, the next important requirement is to provide the right sort of housing. There are many different containers available but I have found that a glass vivarium or old aquarium with proper ventilation is the most satisfactory in terms of ability to create as realistic a habitat as possible. With larger, well ventilated containers, it is also easier to provide different micro-habitats which vary in humidity, temperature and amount of cover. There are six main considerations when designing or buying containers for spiders:

- Is the container an appropriate size in relation to the size of the spider?
- Is the container suitable for the behaviour and requirements of the spider?
- Is the container secure enough to prevent escape?
- Can prey be introduced without having to remove the whole lid (this is preferable)?
- Is the container adequately ventilated?
- Does it provide good views of the spider?

The late Frances Murphy had some useful suggestions in this respect (Murphy 1980). She recommended the *Pal Pets* range of well-ventilated plastic vivaria available cheaply at most pet shops, and also the storage boxes made of strong plastic in a wide range of sizes and shapes available from many DIY outlets. Among the simplest, most effective and flexible containers available are the small plastic strawberry boxes that can be bought for a couple of pounds per dozen, or recycled from supermarkets. These are cheap, durable, give all round visibility and are easily adapted to meet specific requirements

It is easy to buy cheap glass or plastic aquaria or terraria and adapt the lids by removing the central part and replacing it with very fine netting held in place by strong duct tape. I use a variety of sizes from a few square inches to over 40 inches long. The smaller containers are generally used for young spiders and those being kept until mature for identification. The larger containers are used to try and re-create specific habitats and establish species for longer term study and breeding. .

Small containers for rearing spiders to maturity can vary from test tubes to yoghurt pots, jam jars or small cardboard boxes. Use fine netting or old net curtain with the lid held in place by an elastic band. This enables air to circulate and prevents the container getting too wet and humid. Frances Murphy (1980) recommended black netting wherever possible, because this is easier to see through, albeit harder to obtain. She also suggested using string rather than a piece of elastic to hold the netting in place since using elastic bands have the drawback that they eventually perish and break, allowing spiders to escape. She suggested a simple solution to access and feeding by cutting a small cross into the netting and working a small cork into the hole created. The cork can then be removed and the prey tipped into the container from a specimen tube. For *Tegenaria* species, which require little or no water and are large spiders that remain within their sheet webs much of the time, Frances Murphy used cardboard shoe boxes as containers. Alternatives that have also been successful for these spiders include both glass vivaria and an old glass-fronted bookcase.

Another factor to bear in mind is that spiders are generally good climbers and habitually escape. It is well worth always double-checking containers and ensuring they are secure to prevent loss of treasured specimens and to avoid heated arguments about the attractions of spiders with arachnophobic partners! Whenever removing a lid, be sure that you have replaced it properly afterwards.

A variety of innovative and reasonably priced containers and cages can be found at events such as the Amateur Entomologists' Society's Fairs, usually held in spring and autumn. Many of the specialist pet shops dealing with spiders and reptiles have a great variety of housing and some may even make containers to order. On the internet, a variety of entomological equipment suppliers also provide a wide range of containers which can be used to keep live specimens. A list of these suppliers is provided in Chapter 9.

One important point to note is that, whatever container you select, never leave it in direct sunlight since spiders die very quickly if overheated.

6.6 Creating the Habitat

The next step is to try and recreate the natural habitat as closely as possible but also to strike a balance between the requirements of the spiders and the need to actually observe them. A good rule of thumb is to use plenty of cover in three quarters of the vivarium, leaving the front area as free as possible to enable clear viewing. This is particularly useful with night-active spiders such as *Dysdera*, *Drassodes*, *Coelotes* and *Clubiona*. These will emerge at night and search the container for prey, providing good views of their hunting behaviour. More detail about these is provided below in the section on wandering spiders.

It is important to control the humidity in the container and moss can be used for this purpose. Moss absorbs and retains water well but can also be allowed to dry out if you want to decrease humidity. The amount of moss used can be easily adjusted to suit the requirements of different species and it provides excellent cover for the spiders as well as potential prey. Cotton wool has also been used but this tends to become mouldy. It is worth keeping humidity gauges in the larger containers and maintaining humidity within the normal range. Relatively inexpensive gauges can be purchased from garden centres. To maintain moisture, spray with rainwater as necessary, using a fine mister.

For web-weaving spiders, either dried branches or sprigs of yew or holly can be provided to form a framework for web weaving and refuges for those spiders that do not sit in the web itself. The yew and holly stays fresh and does not wilt for a long time. Wherever possible, have branches at either end of the container for web attachment and leave the central portion of the container clear for the web itself. Another approach for large containers is to select living plants and plant them either into the substratum within the container or into small pots that can be placed within the vivarium. Plants provide humidity naturally and avoid the air becoming too dry. Ground ivy does particularly well in these circumstances as do many cacti and succulents, which can also enhance the general look of the habitat created. These require little watering, which can be important to avoid excess moisture in the vivarium

Web-weaving species often ignore the vegetation provided and weave their webs in the top corners of the container or attach it to the lid, making access and observation difficult. An invaluable tip, provided by Dr Emma Shaw, is to grease the top inch of the insides of the container with Vaseline to prevent spiders attaching the web to the lid. Make sure that the highest vegetation in the container is still below the Vaseline level.

For flooring, a variety of substrata can be used, from sand to gravel to soil planted with, for example, ivy, grass or deadnettle. Again, try to relate the ground cover to that the spider was found in. Use leaf litter for woodland species and sand for species from drier places such as heathland or beaches. It is often useful to have an inch or two of coarser gravel at the bottom of the container and then place sand or soil on top of this to avoid the upper layer getting too wet and soggy.

Whenever in the field, it is worth collecting any useful materials seen, even if it is not needed immediately. These include small logs and branches with natural holes and crevices for retreats or with loose bark or moss. Similarly, collect stones and large land-snail shells which can provide effective shelter. Small pieces of slate and flat stone are also useful. In one large vivarium, I created a small replica dry-stone wall which has proved effective for keeping *Segestria* and *Amaurobius*. Airbricks with various hole diameters are also effective for providing retreats and can be found in builder's yards and garden centres. Good supplies of various twigs, bark and branches for creating bushes and retreats for orb-web builders are essential, as are fresh *Sphagnum* moss and heather.

Finally, it is well worth planning ahead, especially for the larger more permanent containers. It often pays to create an environment and plant it well before introducing the species of spider you wish to study. This gives the habitat a chance to settle down and mature and become a genuine living environment in which to house and study your specimens. This has proved particularly effective with *Tegenaria silvestris*, *Agelena labyrinthica*, *Pisaura mirabilis* and ground dwellers such as *Coelotes* and *Pardosa* spp.

6.7 Feeding and Providing Water

Other than actually sitting and observing your spiders, keeping them well fed and supplied with the right kind and size of prey will be the most time-consuming and challenging activity. When determining what to feed spiders, it is important to understand that different species have very varied prey and capture methods. No single prey item will suffice for all species nor will the way food is presented be the same. The size of prey and the willingness of the spider species to tackle it varies enormously from one species to another.

Before attempting to feed a spider, it is usually important to let it settle into its new quarters for a week or so, build a web or retreat, and generally establish itself. This is less important with ground hunters but certainly essential with web builders as the vibration of the web by prey is often what triggers the attack; without a web the spider is incapable of capturing prey. The type of prey taken by web builders varies with the type of web they build. Orb webs are predominately aimed at catching flying prey, sheet webs tend to catch crawling or falling prey whereas vertical trip-wire webs or sheets such as those *Amarobius* build catch mainly crawling prey, as do ground hunters and jumping spiders.

Many insects and other invertebrates have defence mechanisms that make them distasteful to spiders and there is a considerable literature documenting some of these findings. Some detail is provided in the sections below but it is useful to make your own observations about reactions to prey, as there is much new information to be discovered. While some spider species will avoid one type of prey others will attack and eat it readily. .

All spiders prefer live prey, and although there has been some success with presenting dead or recently killed prey to spiders using forceps, it is time-consuming and prevents observation of a spider responding to and capturing insects and other prey naturally.

Size of prey is important: "Limits to the size of prey relative to that of the spider vary in different species, but generally speaking, it can be said that the staple diet of spiders consists of insects appreciably smaller than themselves, and that their prey is confined to insects varying in bulk from that approximately of the spider itself to insects with a body length not less then approximately one-sixth that of the spider." (Bristowe 1939).

However, there are some notable exceptions to this. *Nuctenea umbratica*, a common spider around sheds and fences has taken a Peppered moth, 7 or 8 times its own size above my moth trap and its poison has paralysed the moth in less then one minute. Others that will tackle larger prey are *Thomisus onustus*, a crab spider that ambushes prey in flower heads, *Zygella x-notata* a common spider around window frames and *Pholcus phalangioides*, which uses its long legs to hold and wrap prey, has been known to tackle and overcome full-grown *Tegenaria*. In general, the venoms of crab spiders (Thomisidae), comb-footed spiders (Theridiidae) and daddy long-legs spiders (Pholcidae) seem to be particularly potent and immobilise even very large prey extremely rapidly. Bristowe found that due to the nature of the snares and prey capture techniques used by *Theridion* and *Dictyna* that they will readily tackle prey many times their own bulk whilst *Tetragnatha* and *Linyphia* rely on prey smaller than themselves (Bristowe 1971).

Frequency of feeding is another variable which depends on the hunger level of the spider, the size of prey and the time of year. Generally speaking, feeding once a week in spring and summer and once a fortnight or each month in winter is sufficient. Another approach that can be adopted is to try and mirror the abundance of prey available naturally from season to season. Mercury vapour moth traps produce a by-catch of flies and other insects which can be fed to the spiders. When large amounts are caught the spiders are well fed and when catches are low, due to bad weather or low temperatures, the spiders receive less food. In this way, typical flying prey availability in any year can be duplicated.

6.8 Finding Prey

A variety of methods are available from running a light trap on suitable evenings to using a sweep net or hand collecting with a pooter to get seasonably available prey. If using a light trap, it is preferable to use only the older, worn moths as prey items because these will have usually mated and laid eggs; fresh-looking specimens should be released. Light traps catch a wide variety of different-sized prey and midges and very small flies are of particular value for feeding spiderlings. Moth traps are less successful in winter, when many spiders will still feed if presented with food, even though prey is less widely available.

It is relatively cheap and easy to buy a wide variety of prey items from pet shops, especially specialist reptile and tarantula dealers. Mealworms at all stages from larvae to adult beetle are taken by many of the larger spider species, as are crickets and locusts, which are available from the smallest size to those suitable for tarantulas. Maggots or gentles are available from fishing-tackle shops and are useful for ground-hunting spiders which will take maggots readily. Additionally, a few maggots can be introduced into containers and left to pupate, releasing flies for later capture in webs or by hunting.

Fruit-fly cultures are available and, if looked after and kept at the appropriate temperature, will provide a ready supply of small flies throughout the year. You can in fact start your own culture by putting fruit like banana out in a container exposed for a few days in warm weather and then sealing it up. Usually a colony of fruit flies will emerge soon after. Fruit flies are particularly useful for feeding young spiders. It is also often worth keeping colonies of prey items such as woodlice and earwigs as these can be fed as needed to appropriate spiders.

Many spiders also eat other spiders smaller than themselves. Spiderlings can be kept together when very young as typically they will not eat their siblings in the early stages. The larger survivors should be separated so that they can more easily fed subsequently. When feeding spiders to other spiders, take care, as occasionally there may be a surprise and the smaller spider will kill the larger one.

6.9 Water

Use a fine plant mister to spray one or two leaves, the web itself, or the container side once every week or two. Do not saturate the container; it is easier to add a little more water later if you feel it is necessary, than to try and dry out a container that has excess moisture. Many spiders require little or almost no water, deriving much of their fluid requirements from the prey they consume. This is particularly true of those spiders found in dry or man-made environments such as *Tegenaria*, *Steatoda* and *Pholcus*.

6.10 Individual Groups

With one or two exceptions, I have always concentrated on British spiders so the following sections provide information on families or spider groups that have been kept with varying degrees of success.

Sheet-web spiders (Agelenidae)

These include the large *Tegenaria* house spiders and *Agelena labyrinthica*. If readers are new to keeping spiders, *Tegenaria* house spiders are a good group to start with. I always have a few either in captivity or at large in the shed and garage because they are a fascinating group and easy to observe due to their size. They are hardy, grow large and live relatively long lives (typically 2–4 years). They require little in the way of specialist accommodation. They will breed readily in captivity, if you introduce the male at the right time, when both he and the female are fully mature. The young spiderlings can be raised to adulthood successfully with a little care and patience.

Geoff Oxford has written some interesting articles on *Tegenaria* in past *Newsletters* and I have found them extremely useful. Bristowe spent some time discussing *Tegenaria* in his book *The world of spiders* which is a mine of information on spider's habits and feeding behaviours (Bristowe 1971).

Tegenaria species (except *Tegenaria silvestris*) are usually found indoors in sheds and outbuildings. They can be kept at any age but, as they are long-lived, it is best to catch one or more juveniles, perhaps around 4–5 mm body length, and rear them to adulthood. Search the window sills in your shed or garage or behind any pile of reasonably dry and sheltered bricks and you will find small *Tegenaria* and perhaps even some large ones. Starting with the juvenile size means they should live some time, are at a size relatively easy to feed and will provide opportunities to observe how they establish and develop a web over time, observe a number of moults and note growth rates.

As with most spiders, it is best to house each individual separately. They like a retreat, so a house airbrick with large holes, a broken flowerpot or a tube of some sort in a corner of the container is ideal. *Tegenaria* build sheet webs which get bigger as the spider adds to it over time. The spider will build out from its retreat so ensure the retreat is to the back and one corner of the container so that the sheet web is built towards the front giving you clear views of the spider's behaviour. *Tegenaria* moult by hanging upside down from the web so it is important to ensure there is sufficient room within the container for the spider to do so.

Once the sheet web has been well established and the spider is settled, you can actually leave the lid off the container if you wish as the spider will not usually leave, unless it is a mature male searching for a female. However, *Tegenaria* have been known to take over each other's webs and prey upon one another, so the spider you end up with may not be the one you started with. Depending on the size of the spider, *Tegenaria* will take almost any prey but do not require frequent feeding. Once a week in the spring and summer and once a month in winter appears to be adequate.

Once you have a large mature female with an established web you can introduce a male in the autumn, when they mature. The male should not be placed directly into the web but within the container so he can approach the web at his own leisure, when he will make the appropriate signals on the web to avoid being eaten. Once accepted, the male will often live with the female close to the web for some weeks until he finally dies. In my experience, it is rare for the female to attack the male once she has accepted his courtship. A large egg-sac is made and the young hatch together, often around July. If seeking to rear the young, it is easier to keep them together in the early months until only a few larger ones are left. Then these larger youngsters can be more easily fed in separate containers.

Tegenaria silvestris is more difficult to keep but if you provide a rotten log or two in a container with ivy or a similar plant draped over the logs or preferably planted and growing and providing an overhang, they will do well. They build a small sheet web usually from under the overhanging plant. You won't see them very often though as they tend to remain well hidden during the day.

Agelena labyrinthica is a very interesting spider to keep in terms of its behaviour and the intricacies of web building. It is best to search suitable habitats such as heathland or well vegetated grass banks in April or May and capture small juvenile *Agelena* since mature spiders will not usually construct a full web. *Agelena* always needs a large container as the web system or labyrinth she creates is very extensive. Plenty of vegetation, preferably planted in pots and of varying heights up to 12 inches, is important to enable good web construction. *Agelena* seems to eat most insects but grasshoppers are a common natural prey in the wild.

Mesh- and tube-web spiders (*Segestria*, *Amaurobius* and *Dictyna*)

These spiders all have tubular retreats and tend to construct their webs on vertical surfaces with the exception of *Dictyna*, which tends to prefer the dry heads of plants and other vegetation with forked stems.

For *Amaurobius* and *Segestria*, airbricks obtainable from builders yards provide excellent retreats. If the container is sufficiently large and sufficient prey is provided, you can easily establish a small colony of *Amaurobius similis*. It is possible to create a small dry stone wall effect in a vivarium using slate and this again allows easy construction of tubular retreats by the spiders. When creating this type of habitat, you must ensure that there are small gaps of varying size between the different layers of the wall.

Mature males of *Amaurobius* are often found wandering around, typically in autumn or early winter. When introducing a male to a captive female, it is usually fine to simply introduce him to the container away from her web and observe how he approaches it and signals his presence. Although mating takes place late in the year, eggs are not laid until the following June or July. These spiders are all nocturnal and, when attacking, bite the leg of the prey before dragging it back towards the retreat. Their prey are largely crawling insects but also flies and Bristowe noted that *Amaurobius* species will attack and kill pompilid wasps, which other spiders will not do.

Dictyna species are all small spiders which can usually be collected in their web with the plant head and this can then be placed in a small pot of earth in a container so that any disturbance is minimal. *Dictyna* can be difficult to feed as they are small and, although they will take prey larger then themselves, the main diet seems to be small flying insects. In captivity it is easiest to catch prey items in a pooter and blow them directly into the web.

Orb-web spiders

While I have kept these a few times, generally I leave a few juveniles each spring in the conservatory to weave webs amongst the grapevines and other plants in order to get close to them and observe them there. As with all web-weavers, the main requirement is space to allow them to weave webs. This is hard to provide in a cage but, if provided with suitable dead branches or shrubbery, they will often do so. Wherever possible, branches should be placed at either end of the container for web attachment and the central portion left clear for the web itself. Another option is to place a plant within a large flower pot and build a framework around it which is then covered with netting. This can then be placed in the garden and the spider observed at leisure but remember to leave appropriate access for feeding. Frances Murphy suggested building a framework with netting that could be removed once the web was built to enable photographs to be taken.

In their paper on laboratory methods for web-building spiders, Zschokke and Herberstein (2005) found that spiders varied greatly in their propensity to build webs in captivity. Those that built webs most readily include *Araneus* spp., *Argiope* spp., *Nephila* spp. and uloborid spiders. They found that *Gasteracantha* spp., *Tetragnatha* spp., *Meta* spp. and *Metellina* spp. were more reluctant to build webs but could be encouraged to initiate web building by introducing a fly. They also found that introducing a fly and feeding a spider soon after it had built a web encouraged it to build again. Finally, they found it important to ensure that a proper day/night cycle of light levels and temperature was maintained.

Greenhouses can also be good places for web-weavers if you want to observe them regularly without having to feed them. If you place the spiders inside for a day or two with the doors shut until they weave a web, they will then normally stay in place for some time, allowing you to observe behaviour such as web construction and prey capture at any time. Ensure the greenhouse has enough flies within it to keep the spiders well fed. Larger greenhouses with some shade and well established plants are best. This has worked well in the past with *Nuctenea umbratica*, a nocturnal orb-web spider that is very easy to keep. It likes a retreat where it can spend the day, preferably somewhere narrow that it can wedge itself firmly into, and also needs a largish area to weave a full web.

Larinoides cornutus is another orb-web spider that spends its day in a retreat, in the heads of reeds or plants such as cow parsley and other vegetation. You can remove the retreat with the spider inside and it can then be placed in a suitable container when the spider will emerge to weave a new web. *Zygiella x-notata* is a species which is active all year round and is readily found around houses and sheds where it builds relatively small but distinctive webs with one missing segment. It spends the day in a retreat, often with just the ends of the front legs showing. Provide a narrow container with a glass front and a suitable retreat in the top corners and this spider will readily build webs and feed on flies and other small insects attracted to lit windows at night.

Wandering spiders (e.g. *Dysdera*, *Harpactea*, *Scytodes* and Clubionidae)

Many wandering spiders are largely nocturnal. *Dysdera* spp. are striking spiders with large chelicerae adapted for catching woodlice. They are easy to keep and feed but require good cover in the form of flat stones and vegetation under which to shelter during the day. They require reasonably damp, humid conditions like their prey. At night, they will emerge and slowly patrol their containers seeking woodlice. Both their feeding and mating habits are well worth studying. *Harpactea* is related to *Dysdera* and will thrive well in a small flowerpot filled with dry heather or moss from which she will emerge at night to hunt prey. *Harpactea* feeds on small invertebrates and has a curious way of measuring potential prey before attacking.

Scytodes, the spitting spider, is easy to keep but can be slightly more difficult to feed as they can be fussy about the size of prey tackled, seeming to prefer prey at least half its own size. As they are small themselves, this can present problems unless a ready supply of fruit-flies is available. These spiders thrive in small containers in dry conditions and, unusually, seem comfortable to share containers with other *Scytodes* as long as they are similar in size and well fed.

Clubiona corticalis is a relatively large sac spider, often found in and around homes although it normally occurs in the wild under the bark of dead trees, fence posts etc. As with *Harpactea*, it will live well in a flowerpot with heather or moss and emerge at night to hunt within its container. It is often found in old birds nests in bird boxes and I once kept a colony successfully simply by placing the nest inside a glass container. The nest had spiders, prey and habitat all in one and required no further support.

Crab spiders (Thomisidae)

These spiders are relatively easy to keep and are often voracious and ready feeders in captivity. They will do well in small containers with a sprig of leaves or a flower-head they can patrol and use to ambush their prey. They also thrive on house plants and often, but not always, will remain on a houseplant in a conservatory or a room. It may take a little patience finding them again on the plant each day, but females, in some species at least, will remain in an area, especially if provided with suitable prey items. Frances Murphy simply kept them in small glass tubes and found that they fed and matured readily.

Wolf and jumping spiders (Lycosidae and Salticidae)

Both wolf spiders and jumping spiders are diurnal hunters with excellent eyesight.

Wolf spiders require a great deal of room to thrive and are also major predators of their own young, making them difficult species to keep. They will mature in tubes readily enough but, to date, I have failed to keep them successfully for long-term study.

The nursery-web spider *Pisaura mirabilis*, which belongs to the closely related family Pisauridae, is a spider that is large and easy to keep, provided it has a big container with lots of grassy clumps and vegetation in which it can hunt. The female carries her egg-sac in her chelicerae and, if captured before she constructs her nursery dome, it is possible to watch her build and guard the egg-sac until the young hatch. This can be done by either placing her on a potted plant within a container or wrapping the whole pot and plant around with netting. If in a large container, ensure that it has some vegetation with a good structure and forked branches and leaves in which she can construct a dome, usually at a height of 18 inches above the ground or more.

Jumping spiders seem to like space and sunlight so are very similar in needs to the lycosids. They are a rewarding group to keep, with elaborate courtship behaviours which are highly species specific. A tall roomy container is needed and it should be well sealed as these spiders are very good at escaping. Provide vertical surfaces such as a number of bricks and rocks, together with branches and twigs to provide climbing surfaces. Loose bark on branches is important as many of these spiders like to construct a silk cell under bark when not active. Salticidae like sunshine but you should not leave the container in direct sunlight. A bright sunny room with the container placed away from the window is most suitable.

With some salticids, such as *Salticus* and *Euophrys* species, it is possible to keep several males and females together without them predating each other, so long as plenty of space and suitable retreats are provided together with plentiful food. However, as with most spiders, it is best to keep the majority of species in individual cages except when males are introduced to females to observe courtship and mating. Fruit-flies are readily taken by most members of this group.

Comb-footed spiders (Theridiidae)

These are amongst the easiest spiders to keep and the variety in size and habitat of the different species means there are a great deal of different behaviours to observe.

Steatoda nobilis is a good example of a theridiid, it is a relatively large species that is easy to keep and females are long-lived, reaching up to 4 years of age in captivity to date. They build large, complicated and often tangled-looking webs with a tubular, gourd-shaped retreat. The webs are designed to catch flying insects, predominately moths, based on observation of catches in webs in their natural habitat. They are ferocious predators and always approach prey from under the web and wrap it before dragging it into their retreat. After they have finished with the prey they will take the wrapped bundle and drop it out of the web. They are largely nocturnal, often sitting under the web at night, but are always within the retreat during the day. These and one or two other of the larger *Steatoda* spp. are strong enough to bite and pierce human skin. Egg-sacs have been produced in the third year with spiderlings hatching although the female only mated in the first year of captivity.

When setting up a vivarium for *Steatoda* spp., the retreat should be placed in one corner at the front of the container and then dry, dead branches and twigs placed around it. The web and retreat will be rapidly constructed and, once built, no lid is required as the female will not leave her web as long as she is well fed.

6.11 Conclusion

Keeping spiders, especially for longer-term study, can be both interesting and rewarding. There are no standard methods and many species have not been kept or studied in captivity at all. There are therefore many opportunities to provide important new insights into life cycles, habits and behaviour, particularly for smaller species. This account is only a brief introduction to the subject and, for further reading and information, the following references will prove helpful.

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7. RECORDING SCHEMES

7.1 The Spider Recording Scheme (SRS)

Peter Harvey

7.1.1 Introduction

Spiders are a fascinating group of invertebrates, which have only started to receive the attention they deserve during the past 50 years. The publication of *British Spiders* (Locket & Millidge 1951, 1953; Locket, Millidge & Merrett 1974), the formation of the Flatford Mill Spider Group, and the subsequent development of the British Arachnological Society provided a firm basis for the study of arachnology in Britain in the twentieth century. The publication of a photographic field guide by Dick Jones (Jones 1983) and then the massively important modern identification work by Mike Roberts (Roberts 1985, 1987) provided arachnologists with the tools to reliably identify most species of spider to be found in Britain. Dr Peter Merrett initiated the mapping of the distribution of British spiders on an administrative county basis in Locket, Millidge & Merrett (1974) and has periodically published new county record updates in the *Bulletin of the British Arachnological Society*, but it was the formation of the Spider Recording Scheme in 1987 and the remarkable enthusiasm and energy of the late Clifford Smith that has been instrumental in encouraging the active support of arachnologists and increasing the numbers of recorders.

7.1.2 History and Objectives

The British Arachnological Society collaborated with the Biological Records Centre to develop a revised Spider Recording Scheme which was launched in April 1987. This replaced a scheme that was started in 1964 but which had fallen into abeyance. The British Arachnological Society and the Biological Records Centre (based at the Centre for Ecology & Hydrology, Monks Wood) jointly administer the scheme. Membership of the British Arachnological Society is not essential for a recorder in the scheme, but is strongly recommended. No membership fees or subscriptions are involved but occasionally the cost of posting specimens for verification may have to be met.

The scheme is coordinated by a National Organiser who is supported by a subcommittee of the British Arachnological Society and a number of Area Organisers, each of whom is responsible for one or more vice-counties. Records from an area covered by an Area Organiser are sent to him/her on a Spider Record Card (RA65) or, if possible, in computerised format. After checking, cards are forwarded to the National Organiser and computerised data to Stan Dobson. A committee of experienced arachnologists in the British Arachnological Society decide whether further verification is required, and then these records are added to the national dataset, and periodically forwarded to the Biological Records Centre. For any area not covered by the network of Area Organisers records are sent directly to the National Organiser Peter Harvey, 32 Lodge Lane, Grays, Essex RM16 2YP; email: grays@peterharvey.freeserve.co.uk.

The first phase of the scheme had the following objective:

- To define the geographical distribution at 10 × 10 km resolution of each species of spider found in Britain and the Channel Islands (currently approx. 650 species), recording distribution information at 1 × 1 km, or even 10 × 10 m where possible, and to establish the broad habitats from which species were recorded.

The second phase of the scheme extended these objectives to include:

- The establishment of a better and more comprehensive system to record the ecology, phenology and effects of different land management on spiders.
- To establish a profile of the ecological characteristics of each British spider species.
- To establish a data bank which will form a base line against which future ecological work can be compared, and provide quantified information on spider ecology which will aid future research and stimulate new studies.
- To extend our knowledge of the biology of spiders, with special consideration of their habitats, seasonal occurrence and population dynamics, e.g. by recording distributions afresh on a regular basis so as to track changing distributions over time, and collect and collate records with full dates, numbers of males and females and habitat details to allow increased understanding of the adult activity periods and life-cycles of British spiders.
- To identify those habitats where species richness and/or presence of notable species makes them of special conservation interest, and to establish how well these are represented in protected areas.
- To identify the hot-spots of biodiversity of spiders in the British landscape.
- To record the spider fauna of selected sites of particular concern to nature conservation, and other areas whose habitat potential might be threatened. From time to time the Scheme and the British Arachnological Society organise surveys of specific sites.
- To lead in the assessment of rarity and conservation status for spiders in Britain.

7.1.3 Newsletter

The Spider Recording Scheme Newsletter is issued three times a year (March, July and November) and contains articles and notes submitted by recorders on all aspects of the recording of spiders, as well as regular updates on the progress being made in the recording scheme. From November 2002 the newsletter has been incorporated with that of the British Arachnological Society as *SRS News*, but is available separately to recorders who are not members of the Society.

7.1.4 The *Provisional Atlas*

One of the main aims of the recording scheme is to provide up to date data on the distribution of spiders in Britain. In the first 14 years of recording (1987–2000) over 1500 volunteers have contributed more than 517,000 records. Overall coverage is good although, not surprisingly, it is patchy in some areas, with a number of counties intensively recorded whilst other areas remain more poorly covered.

Provisional maps with species accounts and phenology charts were published by Harvey *et al.* (2002). The two volumes are available from CEH Publications, Monks Wood, Abbots Ripton, Huntingdon, Cambridgeshire PE17 2LS at £20 incl. p&p.

The *Provisional Atlas* provides a very great amount of new information on every British species. The 647 species accounts have been written by volunteer authors, without which the text could not have been produced in the timescale available. The draft text and maps were available to arachnologists on the NBN Gateway with the expert guidance of Dr Peter Merrett. The large amount of feedback has helped to improve the accuracy and usefulness of the atlas. Checklists for many European countries were consulted and most accounts contain a summary of the distribution in western and central Europe, based on the information available.

Maps and distribution patterns

It is satisfying to see how well the distribution maps for various common spiders illustrate the success of the first phase of the recording scheme. Species such as the comb-footed spider *Robertus lividus*, the money spiders *Lepthyphantes tenuis* and *L. zimmermanni*, the garden spider *Araneus diadematus*, the wolf spider *Pardosa pullata* and the crab spider *Xysticus cristatus* are all widespread across Britain. What is surprising is the more limited distribution of some other species previously described as common and widespread or widely distributed throughout Britain. The nursery-web spider *Pisaura mirabilis*, the wolf spider *Pardosa prativaga* (Fig. 18) and the jumping spider *Euophrys frontalis* are species which the maps show are widespread only in the southern half of Britain but which become extremely scarce further north. The maps for the colourful crab spider *Misumena vatia* and the orb-web spider *Araneus triguttatus* show a very pronounced southern and south-eastern distribution respectively.

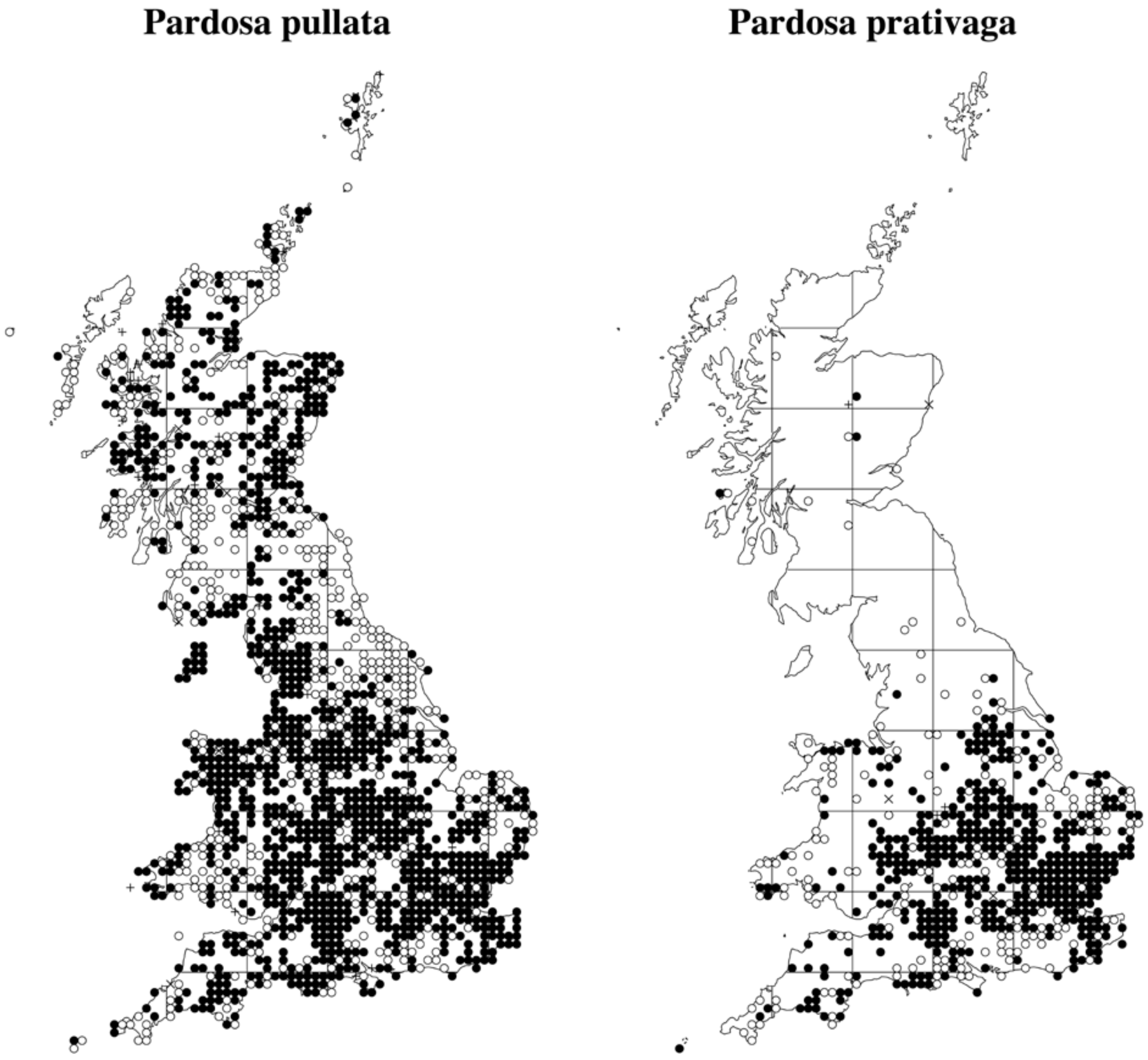

Figure 18. Maps showing distribution of *Pardosa pullata*, a widespread species and *P. prativaga*, a southern species in Britain.

Many spiders are associated with higher altitudes and northern latitudes. Good examples include the money spiders *Hilaira pervicax*, *Meioneta nigripes* and *Scotinotylus evansi*. Some have western and northern distributions and are absent or extremely scarce in the south-east, such as the dictynid spider *Cryphoeca silvicola* and the agelenid spider *Textrix denticulata*.

Other species show interesting patterns of distribution, some well known to arachnologists and some more surprising. The attractive little jumping spider *Talavera petrensis* is a rare, southern heathland species which otherwise only occurs under stones on mountains in a few widely scattered localities in the north. Another rare species, the cryptically coloured running crab spider *Philodromus margaritatus*, occurs on trunks of trees, especially where these are covered with lichens. It also has a disjunct distribution, occurring in scattered localities in the south of England and in central Scotland. A number of species, such as the orb-web spiders *Mangora acalypha* and *Zilla diodia*, show an interesting distribution, stretching in a band across southeastern and central southern England and then reappearing in the Severn Vale. One spider, *Nigma walckenaeri*, has long been known from the Thames valley and London area but has more recently been discovered in Gloucestershire and Warwickshire. There are similarities here with the distribution of several other species of spider and some other invertebrates such as the ant *Lasius brunneus* (Alexander & Taylor, 1997). It should be noted that the Thames and Severn valleys have certain climatic similarities which include average daily maximum and minimum temperatures in January and July (figures from The Climate of England, some facts and figures, The Met Office 1996).

The maps frequently raise more questions than they provide answers but this is in the nature of invertebrate recording and make the results all the more exciting. Whilst there is much to do to fill in gaps and record counties more thoroughly, we already have enough information to act as a baseline for detecting future changes in spider distribution and to stimulate more research into their ecological requirements, habitat management and phenology.

Male/female phenology data

Phase one of the recording scheme did not aim to record male and female data, and therefore the dataset does not provide information on the sex and growth stage of individuals or of the numbers collected. However, at a late stage in the preparation of the *Provisional Atlas*, it became apparent to the National Organiser that published information on the adult season of species was not always in agreement with his own data and the data of the Essex Spider Group. Male/female phenology data were subsequently provided at very short notice by 10 SRS recorders, providing more than 130,000 records from many areas of the country. All these data were worked into a usable form and graphed with the help of Martin Askins. The results have been used to provide phenology data in the majority of the species accounts and to provide summary charts for most species. An interesting example is the wolf spider *Trochosa ruricola*, usually described as mature all year or throughout the year, especially in autumn and spring, whereas our data show a strong peak from spring through to mid-summer.

7.1.5 Where do we go from here?

Adult phenology data on the basis of latitude and longitude

The fruits of the 14-year-old recording scheme are clearly provisional. The phenology data from phase one the recording scheme are potentially even more interesting when analysed on the basis of latitude and longitude in Britain (using the 100 km OS grid for northings and eastings). The limited data available suggest that, for a number of species, the maturity period changes from south to north and from east to west. The collection of wider ranging and comprehensive phenological data during the next phase of the recording scheme would allow us to understand the effects of geographical location on adult activity periods.

Spiders and conservation of invertebrates

Arachnids, insects and other invertebrate groups play a vital role in ecological systems and the importance of invertebrates is increasingly being recognised in nature conservation and management. The complex structural mosaic of a habitat and its vegetational diversity are very important to many invertebrates. Spiders are no exception, although their general lack of prey specialism means that floral and faunal diversity is unlikely to be as important as the structural spaces presented by the ground topography and vegetation, affecting features such as microclimate and web construction. These are the very factors likely to be most influenced by different management regimes, and spiders should therefore be valuable indicators on the success or otherwise of such management and the health of the countryside.

Patterns of change

The recording of spiders on a detailed national basis is at too early a stage to monitor changes in the populations of species, although changes have undoubtedly occurred at an alarming rate especially in southern England, where modern agriculture and other developments have taken an enormous toll on wildlife habitats and their associated flora and fauna. However, a few species have clearly increased their distribution, including *Argiope bruennichi*, a spider expanding its range in southern England, possibly as a result of climate change, in particular longer milder autumns.

The crab spider *Philodromus praedatus* is a species with an interesting history in Britain. The spider is typically found on mature oak trees in open situations, in wood pasture, at the edge of woodland rides or in old hedgerows. It is also sometimes found on other trees such as field maple. Although males are easy enough to distinguish from other members of the *P. aureolus* group, females are difficult to identify without dissection and require reference to reliably identified specimens. It was first recorded from Dorset in the nineteenth century by Pickard-Cambridge (1879). In 1974 the species was still only known from old specimens taken at Bloxworth (Dorset), New Forest (Hampshire) and Shrewsbury (Shropshire). However, by the time Dr Peter Merrett published his review of nationally notable spiders in 1990 the spider had been recorded in 10 counties, mainly in southern England, but also Inverness, and the spider is now known from 25 counties. This remarkable change does not seem to be due to any increase in range or abundance, but rather the result of the recognition of good characters for its separation from other closely related species and a clarification of its ecological preferences.

New species to Britain continue to be discovered, such as *Megalepthyphantes* sp. nov. from North Kent, *Wabasso replicatus* from the Insh Marshes in 1999, *Neriene emphana* from the Isle of Wight in 2000, and *Macaroeris nidicolens* from Mile End in East London in 2002. One species, a six- or eight-eyed oonopid belonging to *Orchestina* or a closely related and possibly new genus, is still only known from six specimens collected by Ray Ruffell north of Colchester between 1992 and 1994. The rate of discovery since the early 1950s has remained almost constant at an average of about 1.5 species per year (Merrett & Murphy 2000).

The qualifications needed to become a Recorder in the SRS

The following skills are required by all SRS recorders:

- To be able to identify spiders to species level with a high degree of competence. Like many invertebrate groups, many spiders are difficult to identify correctly without experience. Beginners will need to have their identifications verified, and rarer or taxonomically difficult species should be checked even when recorded by experienced arachnologists. Area Organisers and the National Organiser are available to advise on identification problems. Note that special courses are arranged by the British Arachnological Society, and by the Field Studies Council, for those wishing to learn how to identify spiders. These courses are either for a day, a weekend, or a whole week and are held at various field centres throughout Great Britain.
- The ability and opportunity to undertake fieldwork that will provide records for the database.
- To complete record cards, provided free of charge by the Biological Records Centre, or to submit data computerised in the agreed format.

A newly registered Recorder is supplied with detailed literature on how to complete record cards or submit computerised data. Record cards and computerised data are then sent to the local Area Organiser who is responsible for the vice-county in which the collection is made. In the absence of an Area Organiser record cards should be sent directly to the National Organiser and computerised data to Stan Dobson.

7.2 The Opiliones (Harvestman) Recording Scheme

Paul Hillyard

The Opiliones Recording Scheme began in 1973 when the first recording cards (RA27) were produced. The Scheme is jointly administered by the British Arachnological Society (BAS) and the Biological Records Centre (BRC) which is based at the Institute of Terrestrial Ecology, Monks Wood, Abbots Ripton, Huntingdon, Cambs PE17 2LS. The purpose of the ORS is to record the geographical distribution of harvestmen in Britain and to publish this information in the form of maps based on the 10km squares of the Ordnance Survey National Grid. The 2nd edition of the *Opiliones Atlas* should eventually be published and, ultimately, the records will form a database that can be accessed to provide species information on particular sites and also present useful data for studies on climate change and biodiversity.

New cards are available free of charge from the BRC. Recorders should complete the cards themselves and send them to National Organiser Paul Hillyard for checking. It is also important to send in any doubtful specimens. The BRC is able to receive records on disk from any database that can output in ASCII format, but the records first need to be validated by the National Organiser.

7.3 The Pseudoscorpion (False-scorpion) Recorders' Group

Gerald Legg

The Pseudoscorpion Recorders' Group has been established to improve our knowledge of this small enigmatic group of arachnids. In the UK there are fewer than 30 species. Historically, records were accumulated at ITE Monks Wood Experimental Station and, in 1980, Jones published the first *Provisional Atlas*. In 1982 the author established a recording scheme and computerised the records. A periodic newsletter, *Galea*, is produced. Details, including copies of *Galea*, images, and the current distributions, can be found on the website below. Contact: Dr Gerald Legg, Booth Museum of Natural History, 194 Dyke Road , Brighton BN1 5AA; email: prg@britishspiders.org.uk or pseudoscorps@chelifer.com; website: http://www.chelifer.com/pseudos. The PRG produces a newsletter annually, distributed to British Arachnological Society members in the UK. These are also available on-line at the BAS website.

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Pickard-Cambridge, O. 1879. The Spiders of Dorset. *Proceedings of the Dorset Natural History and Antiquarian Field Club* **1**: i–xli, 1–235.

Roberts, M. J. 1985/1987. *The spiders of Great Britain and Ireland*. 3 vols. Harley Books.

8. SAFETY MATTERS

A. Russell-Smith

Studying arachnids is no more risky than any other recreational pursuit involving fieldwork. The numbers of deaths or serious accidents each year to those involved in outdoor activities in the UK is fortunately very small. Nevertheless, they do occur and, in many cases, lack of knowledge of the risks or lack of preparation for them appear to be contributory factors. Most of the points listed here could be described as common sense, but ignoring them frequently contributes to accidents in the field or the office.

- Be particularly careful on treacherous ground. This includes boggy or other waterlogged terrain, lake or pond margins with floating mats of vegetation which may give way suddenly, coastal landslips with mudflows, soft-rock sea cliffs and wet rocks or wet, smooth wood. Make sure you have adequate footwear for the surfaces you will encounter, with soles that provide good grip and uppers that provide adequate protection for the ankles. In areas with treacherous ground which can be concealed (such as bogs and some marshes), take the advice of experienced local people about where to venture and where not.
- Make adequate provision for weather conditions. The weather in Britain can change very rapidly, particularly in the West and on mountains. Make sure you have adequate clothing and footwear to keep you both warm and dry even if the day starts fine.
- If you are walking in upland areas, take suitable maps and a compass and always let someone else know where you are planning to go and when you plan to return. Carry a whistle to use as a distress signal in case of emergency. If possible, carry a mobile phone (provided you have reception in the area concerned). The British Mountaineering Council (177–179, Burton Road, Manchester M20 2BB; Tel: 08700 104878; website: http://www.thebmc.co.uk) publish a useful booklet with sound advice: *Safety on Mountains*.
- When working alone in a remote area it is well worth having an outer garment that is brightly coloured and easily spotted. Alternately, a brightly coloured backpack can be carried.
- If you are going into remote areas and plan to be away for more than a few hours, make sure you have adequate food and, particularly, water with you. In the unlikely event of an accident that immobilises you, energy rich food to maintain body heat and water to avoid dehydration can be critical.
- Always carry a torch in your equipment as you may inadvertently find yourself out far longer than you had planned. A torch can enable you to see paths etc. clearly and to signal if necessary.
- Be cautious when collecting in inter-tidal areas such as salt marshes. The sea can creep up extremely rapidly, particularly at spring tides, leaving you stranded. Make sure you know the times of high tide (in most coastal areas, tide time-tables can be bought in local newsagents) and keep a careful eye on the tide level, the position of deep dykes and the distance to the nearest higher ground.
- Be careful with glass equipment such as pooters and glass tubes. Broken glass can cause deep cuts that result in profuse bleeding. It is sensible to carry a small first aid kit in your field bag or rucksack for emergencies.
- When using a pooter, be careful about what you inhale. Avoid pooting spiders or insects off carrion, dung or spore-producing fungi. Be particularly careful in caves where there are bats since there are micro-organisms associated with bat dung that can cause serious lung infections.

There are relatively few medical conditions specifically associated with working in the field and you are probably at much greater risk of acquiring communicable diseases in a crowded bus or train than in the countryside. However it is worth:

- ensuring that your anti-tetanus immunisation is up-to-date
- being aware of the risk from Weil's disease from contaminated water used by rodents
- ensuring you check for and remove ticks after working in areas with high stock or wildlife numbers. Ticks transmit the bacteria that cause Lyme's disease which can be serious if left untreated. Wearing long trousers and tucking the hems into thick socks is a sensible precaution in such areas
- carrying some anti-histamine tablets in your first aid kit. Stings from wasps or hornets can occasionally result in anaphylactic shock which can result in death if not rapidly treated.

When working at home in the office, it is worth keeping the following points in mind:

- Most commonly used laboratory chemicals are toxic if ingested (including industrial methylated spirit) and in some cases caustic. Special care should be taken with sodium hydroxide (a dilute solution of which is sometimes used for clearing genitalia) which is extremely caustic.
- Alcohols and ethyl acetate are flammable. Stock bottles should be stored in a flame-proof cabinet. Bench-top bottles should always be kept away from heat sources and particularly from naked flames. These liquids are also volatile, so when working with them ensure that the room is ventilated and avoid breathing in the fumes.
- All chemicals should be kept locked away out of reach of children when not in use.
- Using a microscope for long periods at a time can cause serious eye-strain. Take frequent breaks to avoid this. Also ensure the seat you use is correctly adjusted for height and position to avoid back problems.

None of these simple precautions should detract from the pleasure you get from collecting and studying arachnids. They could, on the other hand, save you from injury or worse and planning ahead before taking to the field is always time well-spent.

9. LIST OF SUPPLIERS

A. Russell-Smith

9.1. General Laboratory Suppliers

ARACHNE, Oakdene, The Heath, Tattingstone, Ipswich, Suffolk. IP9 2LX. Tel: 01473 327835. Email: paullee@arachne.fsnet.co.uk. Glass and polypropylene specimen tubes. Other entomological and arachnological supplies including glass blocks, forceps, pin vices and reagents.

Barloworld Scientific Ltd, Beacon Road, Stone, Staffordshire ST15 0SATel: 01785 812121 Fax: 01785 813748. Email: bsl@barloworld-scientific.com. Website: http://www.barloworldscientific.com. Suppliers of the Bibby-Sterilin line of laboratory equipment, particularly disposable plastic specimen tubes and petri dishes.

Camlab Ltd, Norman Way Industrial Estate, Over, Cambridge CB24 5WE. Tel: 01954 233100. Email: sales@camlab.co.uk. Website: http://www.camlab.co.uk. Plastic storage boxes (e.g. 100-slot tube box, product number RTP/72400-N @£7 each – takes tubes up to 12 mm diameter.)

Fisher Scientific UK Ltd, Bishop Meadow Road, Loughborough, Leicestershire LE11 5RG. Tel: 01509 231166. Fax: 01509 555111. Email: sales@fisher.co.uk. Website: http://www.fisher.co.uk. Dissection instruments, glass blocks, glass & plastic specimen tubes, ethanol and other laboratory chemicals, petri dishes, wash bottles etc. Catalogues only available to business addresses.

Lab3 Ltd, 1, Ross Road Business Centre, Northampton NN5 5AX. Tel: 01604 581111. Email: sales@lab3.co.uk. Website http://www.lab3.co.uk. Supply plastic containers and petri dishes. Also Meiji microscopes (see below).

Mackay & Lynn Ltd, 18/7 Dryden Road, Bilston Glen, Loanhead EH20 9LZ Tel: 01314 480819. Fax: 0131 448 0865. Email: Sales@MackayandLynn.co.uk. Website: http://www.mackayandlynn.co.uk. Note: Website currently under construction. Dissection instruments, glass & plastic specimen tubes, hand lenses, ethanol and other laboratory chemicals.

Philip Harris Education, Hyde Buildings, Ashton Rd., Hyde, Cheshire SK14 4SH. Tel: 08451 204520. Fax: 0800 138 8881. Email: enquiries@philipharris.co.uk. Website: http://www.philipharris.co.uk. Catalogues only available to educational establishments.

Raymond A. Lamb, Units 4 & 5, Parkview Industrial Estate, Alder Close, Lottbridge Drive, Eastbourne EN23 6QE. Tel: 01327 737000. Email: sales@ralamb.com. Website: http://www.ralamb.co.uk. Dissection instruments, glass blocks, microscopy accessories, wash bottles. Catalogues available as PDFs from website.

R. L. Slaughter Ltd, 11 & 12, Upminster Trading Park, Warley Street, Upminster, Essex RM14 3PJ. Tel: 01708 227140. Fax: 01708 228728. Email: sales@slaughter.co.uk. Website: http://www.slaughter.co.uk. Dissecting instruments, glass and plastic specimen tubes, petri dishes, wash bottles, ethanol and other laboratory chemicals. On-line catalogue.

Sarstedt Ltd, 68 Boston Road, Beaumont Leys, Leicester LE4 1AW. Tel: 01162 359023. Fax: 01162 366099. Email: info@sarstedt.com. Website: http://www.sarstedt.com. Specialise in the supply of high quality polypropylene sample and specimen tubes suitable for storing arachnids and other invertebrates in alcohol.

Scientific & Chemical Supplies Ltd, Carlton House, Livingstone Road, Bilston, West Midlands WV14 0QZ. Tel: 08451 650845. Fax: 01902 402343. Email: customerservices@scichem.co.uk. Website: http://www.scichem.co.uk. Dissection instruments, glass & plastic specimen tubes, hydrometers, ethanol and other laboratory chemicals, petri dishes, wash bottles etc.

Sigma-Aldrich Co. Ltd, The Old Brickyard, New Road, Gillingham, Dorset SP8 4XT. Tel: 0800 717181. Fax: 0800 378785. Email: ukorders@europe. sial.com. Website: http://www.sigmaaldrich.com. Dissection instruments, glass beads (ballotini), plastic specimen tubes, ethanol and other laboratory chemicals, petri dishes, wash bottles etc.

Solmedia Ltd, 6 The Parade, Colchester Road, Romford, Essex RM3 0AQ. Tel: 01708 343334. Fax: 01708 381790. Ethanol and other laboratory chemicals.

S. Murray & Co Ltd, Holborn House, Old Woking, Surrey GU22 9LB. Tel: 01483 740099. Email: laboratory.sales@smurray.co.uk. Website: http://www.smurray.co.uk. Glass vials 35 × 22 mm (code: T103/V1). Push-on plastic caps (code: T003/C1).

9.2 Entomological Suppliers

Anglian Lepidopterist Supplies, Station Road, Hindolveston, Norfolk NR20 5DE. Tel: 01263 862068. Email: jon.clifton@btinternet.com. Website: http://www.angleps.com. Nets, chemicals, microscopes.

Alana Ecology Ltd, The Old Primary School, Church Street, Bishop's Castle, Shropshire, SY9 5AE. Tel: 01588 630173. Fax: 01588 630176. Email: sales@alanaecology.com. Website: http://www.alanaecology.com. Supply sweep nets, beating trays, pooters, dissection equipment and specimen tubes.

B & S Entomological Services. 37, Derrycarne Rd., Portadown, Co. Armagh BT62 1PT. Tel: 07767 386751. Fax: 02838 336922. Email: enquiries@entomology.org.uk. Website: http://www.entomology.org.uk/contact.htm. B & S Entomological Services are, from January 2005, the new owners and suppliers of the Marris House range of entomological nets.

D. J. & D. Henshaw, 34 Rounton Road, Waltham Abbey, Essex EN9 3AR. Tel: 01992 717663. Fax: 01992 717663. Email: djhagro@aol.com. Entomological, arachnological and microscopy supplies, magnifiers, telescopic handles for Marris House nets, specimen tubes, entomological pins, glass beads (in small quantities), data label printing service and reagents.

EFE & GB Nets, PO Box 1, Bodmin, Cornwall PL31 1YJ. Tel: 01208 873945. Email: sales@efe-uk.com Website: http://www.efe-uk.com. Sweep nets, entomological nets, hand lenses, plastic aquaria.

Lydie Rigout, 1, Hillside Avenue, Canterbury, Kent CT2 8ET. Tel: 01227 769924. Fax: 01227 456013. Email: lr@insects.demon.co.uk. Website: http://www.insects.demon.co.uk. Provision of entomological supplies including butterfly nets, pooters, continental insect pins, beating trays, pinholders, forceps, etc.

Watkins & Doncaster, PO Box 5, Cranbrook, Kent, TN18 5EZ. Tel: 08458 333133. Fax: 01580 754054. Email: sales@watdon.com. Website: http://www.watdon.com. Specialise in the manufacture and supply of equipment for collecting, storing and observing insect life. Includes sweep nets, beating trays, pooters, microscopical accessories and specimen tubes.

9.3 Microscopes & Microscopy

Those companies marked with asterisk supply used microscopes only.

Broadhurst, Clarkson and Fuller Ltd*, 63 Farringdon Road, London EC1M 3JB. Tel: 02074 052156. Fax: 02074 302471.

Brunel Microscopes Ltd, Unit 2, Vincents Road, Bumpers Farm Industrial Estate, Chippenham, Wiltshire SN14 6NQ. Tel: 01249 462655. Fax: 01249 445156. Email: mail@brunelmicroscopes.co.uk. Website: http://www.brunelmicroscopes.co.uk.

Carl Zeiss Ltd, PO Box 78, 15–20 Woodfield Road, Welwyn Garden City AL7 ILU. Tel: 01707 871200. Fax: 01707 330237. Email: micro@zeiss.co.uk. Website: http://www.zeiss.co.uk.

GX Microscopes, division of GT Vision Ltd, Hazel Stub Depot, Camps Road, Haverhill, Suffolk CB9 9AF. Tel: 01440 714737. Fax: 01440 709421. Email: eurosales@gxmicroscopes. Website: http://www.gxmicroscopes.com.

Herts. Optical Services*, 102A, Victoria Street, St Albans, Herts. AL1 3TG. Tel: 01727 859392.

John Millham, 82, Brasenose Road, Didcot, Oxford OX11 7BN. Tel: 01235 817157**.**

Lakeland Microscopes, Holly Bank, Windermere Road, Lindale, Grange-over-Sands, Cumbria LA11 6LB. Tel: 01539 534737. Fax : 01539 35026. Email: info@lakeland-microscopes.co.uk. Website: http://www.lakeland-microscopes.co.uk. Free catalogue available.

Leica Microsystems UK Ltd, Davey Avenue, Knowhill, Milton Keynes MK5 8LB. Tel: 01908 246246. Fax: 01908 609992. Website: http://www.leica-microsystems.com.

Lindsey Optical*, 2a, Walden Road, Sewards End, Saffron Walden, Essex, CB10 2LE. Tel: 08709 080205. Fax: 08701 634672. Email: Sales@LindseyOptical.co.uk Website: http://myweb.tiscali.co.uk/aargau/optika.htm.

Meiji Techno UK Ltd, Hillside, Axbridge, Somerset B526 2AN. Tel: 01934 733655, Fax 01934 733660. Email: enquiries@meijitechno.co.uk. Website: http://www.meijitechno.co.uk.

Meta Scientific Ltd, 7 Fosters Grove, Windlesham GU20 6JZ. Tel: 01276 475407. Fax: 01276 472070. Email: mail@metascientific.newnet.co.uk. Website: http://www.metascientific.com.

Microscopes Plus Ltd, Unit 20, The Enterprise Centre, Cranbourne Road, Potters Bar, Herts. EN6 3DQ. Tel: 08452 724007. Fax: 08452 724008. Email: info@microscopesplus.co.uk. Website: http://www.microscopesplus.co.uk.

Nikon Instruments, Nikon House, 380 Richmond Road, Kingston-upon-Thames, Surrey KT2 5PR. Tel: 08712 001964. Website: http://www.nikon.co.uk.

Olympus UK Ltd, Olympus UK Ltd, Great Western Industrial Park, Dean Way, Southall, Middlesex UB2 4SB. Tel: 02072 532772. Email: Microscope@olympus.uk.com. Website: http://www.olympus.co.uk/microscopy.

Stockport Binocular & Telescope Centre, Mercian Way, Edgeley, Stockport SK3 9DF. Tel: 01614 298002. Tax: 01614 740440. Email: tloptics@aol.com. Website: http://www.telescopes-binoculars.co.uk.

9.4 Booksellers

C. Arden, Radnor House, Church Street, Hay-on-Wye, Herefordshire HR3 5DQ. Tel: 01497 820471. Fax: 01497 820498. Email: info@ardenbooks.co.uk. Website: http://www.ardenbooks.co.uk. New and secondhand books on natural history. Also buy books and will do book searches.

Hillside Books, 1, Hillside Avenue, Canterbury, Kent CT2 8ET. Tel: 01227 769924. Fax: 01227 456013. Email: lr@insects. demon.co.uk. Website: http://www.insects.demon.co.uk. Specialist in new and secondhand entomological books.

NHBS Environment Bookstore, 2–3 Wills Road, Totnes, Devon TQ9 5XN. Tel: 01803 865913. Fax: 01803 865280. Email: nhbs@nhbs.co.uk. Website: http://www.nhbs.com. Probably one of the largest stock of new natural history books in Britain.

Pemberley Natural History Books, Ian Johnson, BSc FRES, PO Box 2081, Iver, Bucks. SL0 9YJ. Tel: 01753 631114. Fax: 01753 631115. Email: ian.johnson@pemberleybooks.com. Website: http://www.pembooks.demon.co.uk. Specialises in new and second hand books in entomology and arachnology.

Abebooks. This is a website that searches the stock lists of over 13,000 booksellers worldwide for the item you require. Once found, you are directed to the bookseller's website through which you can purchase the book. Website: http://www.abebooks.co.uk. A very useful resource when searching for difficult to find books or scientific articles.

9.5 Bookbinders

Members requiring books or journals bound generally seek a binder in their local area. A very useful list of names and addresses is provided by The Society of Bookbinders at: http://www.societyofbookbinders.com/links/list_of_binders. html

10. RECOMMENDED READING

Ian Dawson & Tony Russell-Smith

10.1 Identification

Hillyard, P. D. 2005. *Harvestmen*. Synopses of the British Fauna. Field Studies Council.

This excellent summary of our harvestman fauna provides accounts of the structure and biology of our 25 species together with easy to use illustrated keys and distribution maps.

Jones, D. 1983. *The Country Life guide to spiders of Britain and northern Europe*. Country Life. Softback. [Out of print]

Jones, D. 1989. *A guide to spiders of Britain and northern Europe*. Hamlyn, Hardback. [Out of print].

Colour photographs of most of our larger species of spider in life – around 300 species included. Excellent photos are combined with equally valuable text describing features useful for identification which can be seen in the living spider (many of these features actually become less easy to see when spiders are preserved in alcohol). The two versions of this title are essentially the same, though it is worth tracking down the later, hardback printing, as the colour reproduction is better. NB. If these prove difficult to obtain, a French-language edition with exactly the same photographs and layout is still available, with the added advantage of a supplementary section covering some typical Mediterranean spiders:

Jones, D., Ledoux, J-C. & Emerit, M. 2001. *Guide des araignées et des opilions d'Europe*. Les Guides du Naturaliste. Delachaux et Niestlé.

Jones-Walters, L. M. 1989. *Keys to the families of British spiders*. AIDGAP. Field Studies Council.

A very useful key when first starting out with spiders. Placing a spider in the correct family from general appearance becomes easier with experience, but is an essential first step in naming the species.

Legg, G. & Jones, R. E. 1988. *Pseudoscorpions*. Synopses of the British Fauna. Field Studies Council.

As with its sister volume on Harvestmen, this provides information on structure, biology of the British pseudoscorpions as well as keys and distribution maps.

Locket, G. H. & Millidge, A. F. 1951/1953. *British spiders*. Volumes 1 & 2. Ray Society. Reprinted as one volume in 1975 by British Museum (Natural History). [Out of print]

Locket, G. H. Millidge, A. F. & Merrett, P. 1974. *British spiders*. Volume 3. Ray Society. [Out of print].

These three volumes were the standard identification work for spiders before Roberts (see below). Although the illustrations of diagnostic features are not as detailed, and there have been a number of additions to the British fauna since publication, there is an enormous amount of useful information in the text. An essential companion to Roberts if you wish to pursue a serious interest in the identification of British spiders.

Roberts, M. J. 1985/1987. *The spiders of Great Britain and Ireland*. 3 vols. Vol. 1 Atypidae to Theridiosomatidae; Vol. 2 Linyphiidae; Vol. 3 Colour plates. Harley Books. Hardback. [Out of print].

Roberts, M. J. (1993) *The spiders of Great Britain and Ireland*. 2-part Compact edition (Part 1: Text, comprising vols. 1 & 2 of hardback edition; Part 2 Colour plates). Harley Books. Softback.

The standard current identification works for British spiders. The drawings of the male pedipalps and female epigynes are, with few exceptions, stunningly accurate, making identification relatively straightforward once past the initial stages, given a methodical and patient approach. The colour illustrations are also useful with preserved specimens, though less so with living spiders. The text is geared primarily towards identification using the pedipalps and epigynes, so Locket & Millidge is also needed for other features.

Roberts, M. J. 1995. *Collins field guide: spiders of Britain and northern Europe*. HarperCollins.

Mike Roberts re-drew all the pedipalps and epigynes and repainted the colour plates for this single-volume field guide. The species coverage differs from the "Big Roberts" in that a small number of additional species from the adjacent continent are included, while the majority of Linyphiidae or money spiders are excluded – only those relatively few species (around 40) with a distinctive abdominal pattern, some of which can be recognised using a hand lens, are included.

10.2 Distribution

Harvey, P. R., Nellist, D. R. & Telfer, M. G. 2002. *The provisional atlas of British spiders*. Biological Records Centre, on behalf of British Arachnological Society and JNCC.

This two-volume publication has 10 km^2 distribution maps, together with text summarising the known ecology, for every British species. For most species there is also a bar chart showing when males and females are mature. For the first time ever we now have a good baseline from which to work, but there is still a long way to go: the maps should help to stimulate new recording. The distribution maps have now been updated to the end of 2005 and are available for each species from the checklist entry on the BAS website at: http://www.britishspiders.org.uk/html/bas.php?page=cl&menu=bas

Crocker, J. & Daws, J. 1996. *Spiders of Leicestershire and Rutland*. Kairos Press.

Crocker, J. & Daws, J. 2001. *Spiders of Leicestershire and Rutland*: *Millennium atlas*. Kairos Press.

These two volumes comprise tetrad (2 × 2 km squares) dot-distribution maps for a single county and show just how much can be achieved by a few individuals working intensively. The earlier volume summarises habitat and ecological data with each species map, whereas the later volume simply presents updated maps for each of the 340 or so species recorded in the county.

10.3 General Texts

Bristowe, W. S. 1958. *The world of spiders*. Collins (New Naturalist no. 38). [Out of print].

Although now long out of print, this remains probably the best introduction to spiders for the beginner which conveys brilliantly the sheer fascination of this diverse group of organisms. Essential reading which is well worth hunting down through your local library.

Chinery, M. 1993. *Spiders*. Whittet Books.

Aimed at the general naturalist, a popular general overview of spider natural history.

Hillyard, P. 1994. *The book of the spider: from arachnophobia to the love of spiders*. Hutchinson/Pimlico. [Out of print].

A general read all about spiders and their relationship with man.

Murphy, F. 1980. *Keeping spiders, insects and other land invertebrates in captivity*. Bartholomew. 2nd edn. 2000.

The title is self-explanatory. Keeping spiders is an excellent way to learn something of their behaviour.

10.4 Spiders Worldwide

Bellmann, H. 1997. *Kosmos-Atlas Spinnentiere Europas*. Kosmos.

Excellent colour photographs of living spiders of Central Europe, many of the species illustrated also occurring in Britain. The text is of course in German.

Jocqué, R. & Dippenaar-Schoeman, A. S. 2006. *Spider families of the world*. Musée Royal de l'Afrique Centrale.

For the first time ever, all 107 described families of spiders are keyed and described with illustrations of the important features of each. The colour photos by Frances Murphy illustrating representatives of many of the families are an important feature of the book. An essential resource for those who wish to attempt identification of spiders from families not found in Britain.

Heimer, S. & Nentwig, W. 1991. *Spinnen Mitteleuropas*. *Ein Bestimmungsbuch*. Paul Parey. [Out of print].

Although the text is in German and the figures often over-reduced, this small pocket book remains the only complete guide to the spider species of the near continent. It provides keys to families and genera as well as figures of pedipalps and epigynes for 1100 species found in the middle European zone. An essential book for those who visit Europe (other than the Mediterranean) and wish to identify the species they collect. This is the source on which the *Central European Spiders – Determination Key* website (listed below) is based.

McGavin, George C. 2000. *DK Handbook: Insects*, *spiders and other terrestrial arthropods*. Dorling Kindersley.

A general pictorial introduction for younger readers to insects and spiders worldwide.

Preston-Mafham, R. & K. 1984. *Spiders of the world*. Blandford Press. (1993 edn.)

Preston-Mafham, R. 1991. *Spiders: an illustrated guide*. Blandford Press. [Out of print].

Preston-Mafham, K. & R. 1996. *The natural history of spiders*. Crowood Press.

All three Preston-Mafham books are liberally illustrated with superb colour photos.

10.5 Scientific Texts

Barth, F. G. 2001. *A spider's world: senses and behaviour*. Springer.

How the sensory organs of spiders have evolved under the influence of their environment, and their link with spider behaviour.

Foelix, R. 1996. *Biology of spiders*. 2nd ed. Oxford University Press.

The only detailed and comprehensive account of all aspects of spider biology, from anatomy to behaviour. Essential reading for the more advanced students of the group.

Shear, W. A. (ed.). 1986. *Spiders*. *Webs*, *behavior*, *and evolution*. Stanford University Press. [Out of print].

A detailed scientific account of silk, webs and all aspects of spider behaviour and evolution related to web building. Written by a series of experts, the chapters vary in their readability but it is well worth dipping into, particularly as it covers all spider families, not just the British fauna.

Wise, D. H. 1993. *Spiders in ecological webs*. Cambridge University Press.

A comprehensive coverage of all aspects of spider ecology. Although useful as a source of information, the somewhat academic approach and abrasive style of writing do not make for easy reading. Best taken in small doses!

10.6 Websites

The spider checklists of many European countries are now available online, in some cases linked to distribution maps. The British Arachnological Society website is at http://www.britishspiders.org.uk Useful pages include the latest British checklist and a link to the catalogue of the BAS library which contains more than 10,000 papers.

Some sites worth bookmarking are:

- *Platnick's World Spider Catalog*: http://research.amnh.org/entomology/spiders/catalog/index.html which catalogues every known species, together with their synonyms, with references to the relevant taxonomic literature and sources of illustrations.
- *Central European Spiders – Determination Key*: http://www.araneae.unibe.ch/index.html which is a useful supplement to the identification works listed above.
- *British Pseudoscorpions*: http://www.chelifer.com/pseudos/pseudoscorpions.htm Gerald Legg's website on British pseudoscorpions provides information on their biology and ecology as well as photographs of many of them and distribution maps for all the species.

11. GLOSSARY OF TERMS

A. Russell-Smith

Abdomen (adj. abdominal). The posterior of the two major divisions of the body of a spider, also called the opisthosoma.

Accessory claws. Serrated and greatly thickened hairs near the true tarsal claws in some spiders.

Aculeate. Pointed; spiny.

Acuminate. Tapering to a point

Adnexae. A collective term for the spermathecae and ducts forming the internal reproductive organs of female spiders; sometimes variously referred to as the vulva, internal genitalia, internal epigyne or dorsal epigyne. (See also Dorsal, Vulva, and Epigyne).

Allometric growth. A differential rate of growth such that the size of a part of the body changes in proportion to another part, or to the whole body, at a constant exponential rate. Allometry may be positive or negative (where, respectively, a part grows relatively faster or slower than the rest of the body).

Allotype. A term, not regulated by the ICZN, for a designated specimen of the opposite sex to the holotype.

Alveolus. The hollowed-out part of the cymbium, or male palpal tarsus, from which the palpal bulb arises and which partially contains it.

Anal tubercle. A small process, dorsal to the spinnerets, carrying the anal opening.

Analogous. Similar in function or appearance, but differing in origin.

Annulation(s). Ring(s) of pigmentation around leg segments.

Anterior. Nearer the front or head end. May be used in combination, e.g. anterolateral, anteroventral, anterodorsal. (cf. Posterior).

Anterior genital sclerite. A sclerite forming the roof of the epigynal cavity. (See also Lateral genital sclerite, Sclerite, and Subgenital sclerite).

Apodeme. A sclerotized infolding of the body wall that forms an attachment for muscles.

Apomorphic. A relative term meaning derived from and differing from an ancestral or generalised condition. (cf. Plesiomorphic).

Apophysis (pl. apophyses). Usually applied to the type of sclerotized process arising from the tibia and patella of male palps and from palpal sclerites, but may occur elsewhere on the limbs and body.

Arthrodial membrane. A flexible membrane connecting adjacent body sclerites and joints of limbs and other appendages.

Atrium. An internal chamber at the entrance to the copulation duct in female spiders

Autapomorphic. Derived character unique to a given species or other monophyletic group.

Autophagy. The eating of part of the body or limb after its severance (cf. Autospasy, Autotilly, and Autotomy).

Autospasy. The casting of a limb when pulled by some outside agent such as a predator or the forceps of an investigator. The term autotomy is frequently misused in this context (cf Autotilly, Autotomy, and Autophagy).

Autotilly. The removal of an appendage by the animal itself, as when an injured leg is seized In the chelicerae and pulled off. (cf Autospasy, Autotomy, and Autophagy).

Autotomy. The reflex self-mutilation or automatic severance of a limb or appendage from the body. This term is frequently misused because the phenomenon does not occur in the Arachnida, but is a feature of Crustacea (cf. Autospasy, Autotilly, and Autophagy).

Axis. A central line of symmetry of an organ or organism.

Ballooning. The aeronautical activity of spiders, achieved when long strands of silk produced by the spinnerets are caught by air currents; spiders may thus travel through the air, sometimes at great height and over large distances.

Basal membrane. A thick membrane which borders the internal face of the epidermis.

Biserially dentate. Provided with two rows of teeth

Book lung. An air-filled cavity containing thin vascular lamellae arranged like book leaves, opening on the ventral side of the abdomen and through which spiders breath.

Boss. A smooth, rounded or slightly conical prominence.

Branchial operculum (pl. branchial opercula). A sclerotized plate overlying the book lung.

Bulbus or bulb. The complex part of the male palp inserted on the hollowed ventral surface of the cymbium

Bursa copulatrix A pouch-like structure of the epigyne which houses the copulatory pore.

Calamistrum (pl. calamistra). A comb- or brush-like series of hairs on metatarsus IV of cribellate spiders.

Carapace. The exoskeletal shield covering the dorsal surface of the cephalothorax or prosoma.

Cardiac mark. A lanceolate midline mark on the abdomen, anterodorsally, overlying the heart.

Catalepsy. The action of feigning death induced by sudden disturbance.

Cephalothoracic junction. A furrow extending forwards and to the sides from the centre of the carapace, marking the junction of the head and thoracic regions; sometimes called the cervical groove.

Cephalothorax. The anterior of the two major divisions of the body of a spider, also called the prosoma.

Chaetotaxy. The arrangement of the leg spines, particularly in relation to classification.

Character. Any characteristic or its attributes used for recognizing, describing, defining, or differentiating taxa.

Chelicera (pl. chelicerae). One of a pair of jaws, each comprising a large basal portion (paturon) and a fang.

Cheliceral furrow. A narrow groove on the basal portion of the chelicera that accommodates the fang in the resting position.

Chitin. A linear polysaccharide of N-acetyl-D-glucosamine found as the characteristic component of the cuticle of arthropods.

Clade. See Cladogram

Cladistic method. A method of classification employing the most parsimonious distribution of apomorphic characters (states). Resulting cladograms specify hierarchical relations of taxa.

Cladogram. A branching diagram which depicts a pattern of relationships; each branch is a clade (or monophyletic group). The basic unit is a dichotomy of two clades which share a hypothetical ancestor. No attempt is made to identify a particular ancestor, though it can be assumed to be represented by the branching point, or node.

Claw. A strong, curved, sharp-pointed process (often toothed) on the distal extremity of a leg and sometimes the palp, particularly the female.

Claw tuft. A bunch of hairs at the tip of the leg tarsus in those spiders with only two claws.

Clavate. Club-shaped.

Cline. A graded sequence of character expression (morphological or behavioural) across a series of neighbouring populations. Variously qualified as ecocline, ethocline, genocline, geocline, morphocline, nothocline, ontocline, phenocline, topocline.

Clypeus (adj. clypeal). The area between the anterior row of eyes and the anterior edge of the carapace.

Colulus. A small midline appendage or tubercle arising from just in front of the anterior spinnerets in some spiders.

Combination. The association of a generic name and a specific name to form the name of a species. A new combination is the first combination of a generic name and a previously published species-group name.

Conductor. A semi-membranous structure in the male palp which, when functional, serves to support and guide the embolus in copulation or to support it when at rest.

Condyle. A smooth, rounded protuberance sometimes present laterally near the base of the chelicera.

Congeneric. Belonging to the same genus.

Congruence. The degree of correspondence between different character sets or different classifications.

Conspecific. Belonging to the same species.

Copulatory opening. Opening(s) in the epigyne through which the embolus (in entelegynes) or male palpal organs (in haplogynes) are introduced.

Coriaceous. Leathery, tough.

Cotype. A term, not recognized by the ICZN, formerly used for either syntype or paratype, but which should not now be used in zoological nomenclature.

Coxa (pl. coxae; adj. coxal). The segment of leg nearest the body. In the palp, this segment is modified to form the maxilla.

Cribellate. Possesing a cribellum and calamistrum.

Cribellum. A sieve-like spinning organ in the form of a transverse plate, just in front of the spinnerets in some (cribellate) spiders.

Cuticle. The non-cellular part of the integument (q.v.), secreted by the epidermis, with a complex composition but always including chitin and protein.

Cymbium (pl. cymbia; adj. cymbial). The broadened, hollowed-out tarsus of the male palp to which the palpal bulb is attached.

Desmitracheate. The structure of the median tracheal tubes in which two bundles of narrow tracheae pass through the pedicel from abdomen to cephalothorax. (cf. Haplotracheate)

Diaxial. Of chelicerae, vertically oriented and with the fangs opposing each other. All British spiders apart from *Atypus affinis* have this feature. (cf. Paraxial).

Dimorphism. The presence of one or more morphological differences that divide a species into two groups. Many examples come from differences between the sexes (sexual dimorphism) but others represent different forms within one sex (e.g. *Oedothorax gibbosus/tuberosus* males).

Distal. Pertaining to or situated at the outer end: that part of a limb/limb segment/appendage which is the farthest away from the body or its attachment. (cf. Proximal).

Diverticulum (pl. diverticula). 1. A pouch-like protrusion or sac branching off from a hollow organ. 2. A blind tube branching from a tube or cavity.

Dorsum (adj. dorsal). The back or upper surface.

Dorsal view. Viewed from above. May be used in combination e.g. Dorsolateral view.

Ecribellate. Without a cribellum and calamistrium.

Ecdysis. Moulting, the periodic act of casting off the cuticle.

Ectal view. The view of a (usually) paired asymmetrical structure (e.g. male palp) from the outside. May be used in combination e.g. Dorsoectal view. (cf. Lateral view and Mesal view).

Embolus (pl. emboli; adj. embolic). The structure containing the terminal portion of the ejaculatory duct and its opening in the male palp. In some linyphiids it is divided into several sclerites, together forming the embolic division (embolus; radix; lamella; terminal apophysis).

Endite. See Maxilla.

Entelegyne. Of spiders, the females of which have external genitalia in the form of an epigyne having two symmetrical halves. (cf. Haplogyne).

Epidermis. The cellular layer immediately below the cuticle which secretes it and controls its activities; usually single-layered.

Epigastric fold (or **Epigastric furrow**). A fold and groove separating the region of the book lungs and epigyne from the more posterior portion of the ventral surface of the abdomen.

Epigyne (or **Epigynum**). A more or less sclerotized and modified external structure associated with the reproductive openings of the adult females of most spider species.

Ethospecies. Species distinguished mainly by behavioural traits.

Exoskeleton. The hard external supportive covering of all arthropods.

Exuvia (pl. exuviae). The part of the integument (cuticle) cast at ecdysis.

Fang.The claw-like distal segment of the chelicera; near its tip opens the duct from the poison gland.

Femur (pl. femora; adj. femoral). The third segment of the leg or palp counting from the proximal end.

Fertilization ducts. Ducts leading from the female's spermathecae through which stored spermatozoa are passed to fertilize the eggs.

Fissidentate. Of teeth: possessing more than one point.

Flocculent. Woolly.

Folium. A pattern of pigment on the dorsum of the abdomen which is often leaf-shaped.

Fovea (adj. foveal). A short median groove on the thoracic part of the carapace, overlying the internal attachment of the gastric muscles.

Genital opening. In female spiders, the opening of the uterine duct in the epigastric furrow; in males the opening of the duct from the testes in the epigastric furrow.

Genotype. A term not recognized by ICZN, formerly used for the type species, but which should not now be used in zoological nomenclature.

Gnathocoxa. Basal segment (coxa) of palp, also called the maxilla or endite.

Gossamer. A light gauzy film of spider's silken threads, which may sometimes be spread over considerable areas; also silken threads, singly or in groups, as seen (or felt) floating in the air.

Guanin. Fatty tissue, chalky in appearance, associated with the intestinal diverticula in spiders. It is sometimes very abundant beneath the dorsal cuticle of the abdomen, showing through the unpigmented cuticle as conspicuous white markings.

Gynandromorph. A spider exhibiting gynandry (see below).

Gynandry. An abnormal state in an adult spider in which parts of the body and genitalia are female and parts male, and in which the female and male components are themselves normally developed. (cf. Intersexuality)

Haematodocha (pl. haematodochae). A balloon of elastic connective tissue between groups of sclerites in the male palp, which distends with blood during copulation causing the palpal sclerites to separate and rotate. (See also Palpal bulb).

Haplogyne. Of spiders, the females of which have little or no external genitalic structure or epigyne. (cf. Entelegyne).

Haplotracheate. The structure of the median tracheal tubes in which they are unbranched and confined to the abdomen. (cf. Desmitracheate)

Head (or **Cephalic region**). That part of the cephalothorax anterior to the cephalothoracic junction.

Hemimetabolous. A pattern of development (as in spiders) characterized by gradual changes, without metamorphosis or distinct larval, pupal and adult stages.

Holotype. A single specimen designated as the name-bearing type of a species or sub-species when it was established, or the single specimen on which such a taxon was based when no type was specified.

Homologous. Relating to similar structures which have a common origin.

Homonym. 1. In the family group: each of two or more available names having the same spelling, or differing only in suffix, and denoting different taxa. 2. In the genus group: each of two or more available names having the same spelling and denoting different taxa. 3. In the species group: each of two or more available names having the same spelling, or spellings deemed under Article 58 of the ICZN to be the same, and established for different taxa, and either originally (primary homonymy) or subsequently (secondary homonymy) combined with the same generic name.

Homoplasy. Structural resemblance due to parallelism or convergent evolution rather than common ancestry.

ICZN. Abbreviation for *International Code of Zoological Nomenclature*, the fundamental aim of which is to provide the maximum universality and continuity in the scientific names of animals compatible with the freedom of scientists to classify animals according to taxonomic judgements. The fourth edition of the Code was published in 1999.

Inferior. Below; situated lower down; ventral.

Instar. Any stage in the development of an arthropod between moults.

Integument. The outer covering material of arthropods comprising cuticle, epidermis and basal membrane.

Intersex. A spider exhibiting intersexuality (see below).

Intersexuality. An abnormal state in an adult spider in which parts of the body and genitalia are predominantly female and parts predominantly male, but in which the male and female components are themselves not fully expressed or developed. (cf. Gynandry).

Introgression. The spread of genes of one species into the gene pool of another by hybridization and back-crossing.

Labium (pl. labia; adj. labial). The lip, ventral to the mouth opening, lying between the maxillae and attached to the anterior border of the sternum.

Lamella (pl. lamellae). A thin, flattened process or leaf-like plate present in the book lungs and also in the male palps of some spiders.

Lamina. A flat, sometimes translucent, sclerotised excrescence, e.g. on retro- margin of cheliceral groove in some spiders.

Lateral genital sclerite. A sclerite forming the lateral walls of the epigynal cavity. (See also Anterior genital sclerite, Sclerite, and Subgenital sclerite).

Lateral view. The view of a bilaterally symmetrical structure (e.g. spider body) from the side. May also be used in combination e.g. dorsolateral view, ventrolateral view. (cf. Ectal view and Mesal view).

Laterigrade. Refers to spiders that show crab-like movement, with legs directed to the side

Lanceolate. Tapering to a point, lance-shaped.

Lectotype. A syntype designated as the single name-bearing type specimen subsequent to the establishment of a species or subspecies.

Lyriform organ. A sensory organ found commonly near the distal end of limb segments and taking the form of a group of parallel slit organs.

Maxillae (s. maxilla; adj. maxillary). The mouthparts ventral to the mouth opening and lateral to the labium, which are the modified coxae of the palps. On spiders, also known as endites.

Median. In the middle or midline.

Median apophysis. A sclerite arising from, or associated with, the tegulum and forming part of the middle division of the palpal bulb. (Sometimes called the suprategular apophysis in linyphiids).

Mesal view. The view of a (usually) paired asymmetrical structure (e.g. male palp) from the inside. Also combined, e.g. anteromesal, mesoventral (cf. Ectal view and Lateral view).

Metatarsus (pl. metatarsi; adj. metatarsal). The sixth segment of the leg, counting from the proximal end; absent in palps.

Monophyletic group. 1. A group of taxa descended from a single ancestral species. 2. The ancestral species and all descendant species.

Monotypy. A genus or subgenus for a single taxonomic species.

Morphospecies. Species distinguished by morphological characters.

Neotype. The single specimen designated as the name-bearing type of a species or subspecies for which no holotype, lectotype, syntype(s), or prior neotype is believed to exist.

Node. See Cladogram

Nomen dubium (pl. nomina dubia). A descriptive term meaning name of unknown or doubtful application.

Nomen novum (pl. nomina nova). Equivalent to new replacement name.

Nomen nudum (pl. nomina nuda). A name that fails to conform to Article 12 or Article 13 of the ICZN. Because a nomen nudum is not an available name, the same name may be used for the same or a different taxon.

Onychium. An extension of the tarsus between the paired claws in some spiders.

Opisthosoma. The posterior of the two major divisions of the body of a spider, usually referred to as the abdomen.

Palp (or **Palpus**, **Pedipalp**). The second appendage of the cephalothorax, originating behind the chelicerae but in front of leg. Its coxa also forms the maxilla; it lacks a metatarsal segment In adult male spiders where it is modified, often greatly, for sperm transfer.

Palpal bulb (or **Genital bulb**). A collective term for the structures comprising the male palpal organ, arising from and partially contained within the alveolus of the palpal cymbium. Normally it comprises three groups of sclerites; the Subtegulum, the Tegulum and Median apophysis, and terminally the Embolus (or embolic division)and Conductor,

Paracymbium (pl. paracymbia; adj. paracymbial). A structure branching from, or attached loosely to, the cymbium.

Paralectotype. Each specimen of a former syntype series remaining after the designation of a lectotype.

Paraphyletic group. A group of taxa derived from a single ancestral taxon but one which does not contain all the descendants of the most recent ancestor; a category based on the common possession of plesiomorphic characters.

Paratype. Each specimen of a type series other than the holotype.

Paraxial. Of chelicerae, horizontally oriented and with the fangs lying parallel to each other. The cephalothorax must rise to give clearance for the downward strike of the fangs. *Atypus affinis* is the only British spider having this feature. (cf. Diaxial).

Parsimony. A logical criterion that the simplest sufficient hypothesis is to be preferred even though others are possible.

Patella (pl. patellae; adj. patellar). The fourth segment of the leg or palp, counting from the proximal end.

Paturon. The basal segment of the chelicera.

Pedicel (or **Petiolus**). The narrow stalk connecting the cephalothorax and abdomen.

Pedipalp. The correct term for the second appendage of the cephalothorax, but in spiders usually shortened to 'palp' or 'palpus' (q.v.)

Phenology. The study of the impact of climate on the seasonal occurrence of fauna and flora, and of the periodical change in form of an organism, especially as this affects its relationship with its environment.

Pheromone (adj. pheromonal). A chemical, secreted by an animal in minute amounts, which produces a behavioural response in another animal, frequently the opposite sex of the same species and sometimes over considerable distances.

Phylogenetic. Pertaining to evolutionary relationships between and within groups.

Plesiomorphic. A relative term meaning ancestral or primitive character(s) (states). (cf. Apomorphic).

Plumose. Feathery.

Pluridentate. Of chelicerae, possessing more than one tooth.

Polymorphism. The existence of two or more forms that are genetically distinct from one another but contained within the same interbreeding population. The polymorphism may be transient or, if persisting over many generations, balanced.

Polyphyletic group. A group determined by non-homologous relations (similarities or characters) and hence its members do not share a single ancestor.

Posterior. Nearer the rear end. May be used in combination, e.g. posterolateral, posteroventral, posterodorsal. (cf. Anterior).

Preening brush/comb. A dense cluster or transverse row of setae near or at ventral tip of metatarsus.

Process. A projection from the main structure.

Procurved. Curved as an arc having its ends anterior to its centre. (cf. Recurved).

Prograde. Of spiders with legs directed forward (legs I & II) and backward (legs III & IV). See Laterigrade

Prolateral. Of leg spines, on the side directed forwards, in an imaginary state as if the leg were straight out to the side, at right angles to the long axis of the body.

Promargin. Anterior margin of cheliceral furrow.

Prosoma. The anterior of the two major divisions of the body of a spider; usually referred to as the cephalothorax.

Proximal. Pertaining to, or situated at, the inner end; that part of a limb/limb segment/appendage closest to the body or its attachment. Sometimes used in combination e.g. proximolaterally (cf. Distal)

Punctate. Covered with small dots or depressions.

Race. An intraspecific unit, the members of which exhibit common biological, ecological, physiological or geographical characteristics which differ slightly from other members of the species.

Radix. The basal part of the embolic division in the male palp.

Rastellum. Rake-like structure at extremity of chelicerae of mygalomorph spiders, used for digging.

Rebordered. With a thickened edge, as of the labium of some spiders.

Recurved. Curved as an arc having its ends posterior to its centre. (cf. Procurved).

Reticulated. Like a network; netted.

Retrolateral. Of leg spines, on the side directed backwards, in an imaginary state as if the leg were straight out to the sides at right angles to the long axis of the body.

Retromargin. Posterior margin of cheliceral furrow.

Rugose. Rough; wrinkled.

Scape. A finger- tongue-, or lip-like appendage, free at one end, arising from the midline of the female epigyne.

Sclerotized. Hardened or horny; not flexible or membranous. See also Chitin.

Scape (or **Scapus**). An elongate, tongue-shaped appendage of the epigyne.

Sclerite. A discrete sclerotized structure. (See also Anterior genital sclerite, Apodeme, Lateral genital sclerite, and Subgenital sclerite).

Scopula (pl. scopulae). A brush of hairs on the ventral aspect of the tarsus and metatarsus in some spiders; also occurs on the maxillae.

Scutum (pl. scuta). A sclerotized plate occurring on the abdomen of some spiders.

Septum. A partition separating two cavities or parts.

Serrate(d). Saw-toothed.

Serrula. A ridge of fine teeth on the anterolateral margin of the maxilla.

Seta (pl. setae) A hair, bristle, or slender spine.

Sigillum (pl. sigilla). An impressed, sclerotized spot, usually reddish-brown in colour, pairs of which are often present on the abdomen, marking points of internal muscular attachment.

Sister groups. Two monophyletic groups borne from the same node of a dichotomy. (See also Cladogram)

Slit organ (or **Slit sensillum**, pl. slit sensilla). A stress receptor in the exoskeleton which detects vibrations. Found in large numbers over the body surface, but especially on the legs. (See also Lyriform organ).

Somatic. Pertaining to the soma or body of the animal as distinct from the genitalia.

Spatulate. Flattened club-shaped; spoon-shaped.

Species inquirenda. A doubtfully identified species needing further investigation.

Sperm duct. 1. A duct in the female epigyne through which sperm travels from the copulatory pore to the spermatheca. 2. A duct in male palp through which sperm travels to the embolus.

Spermatheca (pl. spermathecae). A sac or cavity in female spiders, used for the reception and storage of spermatozoa.

Spiderling. The nymphal or immature stage of a spider, resembling the adult in general form, but smaller, able to move about and feed and no longer dependent upon the yolk for nourishment.

Spigot. A spinning tube, projecting from the tip of the spinneret, through which the silk is extruded from the silk glands.

Spine. A thick, stiff hair or bristle.

Spinneret. Paired appendage at the posterior end of the abdomen, in front of (below) the anal tubercle, through the spigots of which silk strands are extruded.

Spinule. A short spine, almost as thick as long.

Spiracle. The opening of the tracheae on the ventral side of the abdomen.

Squamiform. Scale-like.

Squamose. Scaly.

Stabilimentum (pl. stabilimenta). A band or bands of silk, decorative in appearance but of disputed function, across the orb-webs of some spiders.

Sternum (pl. sterna; adj. sternal). The heart-shaped or oval exoskeletal shield covering the ventral surface of the cephalothorax, lying posterior to the labium and between the leg coxae.

Stria (pl. striae). A linear mark, streak, ridge, or furrow; usually applied plurally to such marks radiating from the central fovea on the carapace to its margin, or as part of a stridulating organ (see below)

Stridulating organ. An area with numerous sclerotized parallel striae which is rubbed by hairs or a tooth on an opposing structure thus creating a sound; such file-and-scraper apparatus may be variously located on chelicerae. palps. legs, abdomen, and carapace.

Subadult. Almost adult; the last instar before maturity.

Subcutaneous. Situated just below the cuticle or skin.

Subequal. Nearly equal.

Subgenital sclerite. A sclerite occurring as a median plate which forms the floor of the epigynal cavity. (See also Anterior genital sclerite, Lateral genital sclerite, and Sclerite).

Subtegulum. The sclerite that forms the most proximal of the three divisions of the male palpal bulb. Often a ring- or cup-like structure, it is attached to the cymbium by the proximal haematodocha. (See also Palpal bulb and Tegulum).

Sulcus (pl. sulci). A groove or furrow.

Superior. Above; situated higher up; dorsal.

Suprategular apophysis. See Median apophysis.

Symplesiomorphy. The shared possession of a relatively ancestral character.

Synanthropic. Living in or close to human habitation.

Synapomorphy. The shared possession of a relatively derived homologous character.

Synonym. Each of two or more scientific names of the same rank used to denote the same taxon. Of the two synonyms, the junior synonym is the later established and the senior synonym the first established.

Syntype. Each specimen of a type series from which neither a holotype nor a lectotype has been designated.

Tarsus (pl. tarsi; adj. tarsal). The most distal segment of a leg or palp.

Tarsal organ. A sensory receptor, usually in the form of a tiny depression, on the dorsal surface of the tarsus.

Taxon (pl. taxa). Any taxonomic unit (e.g. a family, subgenus or species) whether named or not.

Taxonomy (adj. taxonomic). The theory and practice of classifying organisms; part of systematics, the study of the kinds and diversity of organisms.

Tegulum (adj. tegular). The sclerite that forms, with the median apophysis, the middle of the three divisions of the male palpal bulb; often a broad ring-like structure. (See also Palpal bulb and Subtegulum).

Terminal apophysis. Part of the embolic division of the male palp, lying distal to the radix.

Thorax (adj. thoracic). That part of the cephalothorax posterior to the cephalothoracic junction.

Tibia (pl. tibiae). The fifth segment of the leg or pulp counting from the proximal end.

Tibial spine formula. An indication of the number of dorsal spines (1 or 2) on the tibiae of legs I to IV, from front to back (e.g. 2-2-1-1); used extensively in the identification of the Linyphiidae.

Topotype. A term for a specimen originating from the type locality of the species or subspecies to which it is thought to belong, whether or not the specimen is part of the type series.

Trachea. (pl. tracheae; adj. tracheal) Tubes through which air is carried around the body and which open at the spiracle(s).

TmI. The representation of the relative position of the trichobothrium along the length of metatarsus I expressed as a decimal fraction. This, and the presence or absence of a trichobothrium on the fourth metatarsus (TmIV), is used extensively in the identification of the Linyphiidae.

Trichobothriotaxy. The arrangement and position of the trichobothria, particularly with reference to classification.

Trichobothrium. (pl. trichobothria) A long, fine hair rising almost vertically from a hemispherical socket on the leg; the socket appears as a distinct circle on the limb surface. Trichobothria detect air vibrations and currents.

Trochanter. The second segment of the leg or palp, counting from the proximal end.

Tumid. Swollen.

Type. A term used alone, or forming part of a compound term, to denote a particular kind of specimen or taxon. (See also Allotype, Cotype, Genotype, Holotype, Lectotype, Neotype, Paralectotype, Paratype, Syntype, and Topotype).

Type genus. The genus that is the name-bearing type of a family group taxon.

Type locality. The geographical (and, where relevant, stratigraphical) place of collection of the name-bearing type of a species or subspecies.

Type series. The series of specimens which either constitutes the name-bearing type (syntypes) of a species or subspecies, or from which the name-bearing type has been or may be designated.

Type species. The species that is the name-bearing type of a genus or subgenus. (Formerly sometimes called genotype, q.v.).

Type specimen. A term used for the holotype, lectotype, or neotype; also used generally for any specimen of the type series.

Unidentate. Of chelicerae, having a single tooth.

Velum. A veil- or curtain-like membrane.

Venter. The under-surface of the body.

Ventral view. Viewed from below. Sometimes used in combination e.g. ventrolateral, anteroventral (cf. Dorsal view).

Vicariance biogeography A method of interpreting the distribution of organisms primarily as a result of land disjunctions, e.g. continental drift.

Voucher specimen. Any specimen identified by a recognized authority for reference purposes.

Vulva. Sometimes used as a term for the internal genitalia of the female spider. This is strictly incorrect since vulva is properly the external genital opening of a female mammal. (See also Adnexae).