British Arachnological Society
Advancing Arachnology

Arachnologists' Handbook

Third edition

Edited by Geoff Oxford, Tony Russell-Smith and Helen Smith
on behalf of the British Arachnological Society

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Cover pictures: Top left: *Megabunus diadema*, a widespread harvestman in the UK, with a characteristic ocularium (eye-turret) (© Peter Nicholson). Top centre: the Garden Spider, *Araneus diadematus*, one of our most widespread and familiar species, especially in autumn (© Chris Spilling). Top right: spider web on the dead heads of Teasel (*Dipsacus fullonum*), probably a *Metellina* species (© Geoff Oxford). Middle: *Euophrys frontalis*, a jumping spider (© Peter Harvey). Bottom left: *Thomisus onustus*, a scarce species of crab spider restricted to mature heathland in southern England (© Peter Harvey). Bottom centre: *Chthonius ischnocheles*, a widespread pseudoscorpion (© Gerald Legg). Bottom right: female *Pisaura mirabilis*, the Nursery-web Spider, carrying her egg sac in her chelicerae (© Geoff Oxford).

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Note: This *Handbook* is also available to BAS members as a free, downloadable PDF file on the Members' Area of the BAS website. Updates, including any changes to website links and contact details, will also be made available on the Members' Area.

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

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Geoff Oxford, Tony Russell-Smith and Helen Smith

October 2019

A note on nomenclature

Scientific names of spiders used in this edition of the *Handbook* are from the latest British and Irish checklist (Lavery 2019: see Section 2.3) and are based on those accepted by the *World Spider Catalog* (wsc.nmbe.ch). Where these differ from those used in the last version of the *Handbook* the previous name is shown in parentheses e.g. *Eratigena* (*Tegenaria*) *saeva*.

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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 the Society was renamed the British Spider Study Group. At its tenth annual meeting, in 1968, it was agreed that the Group would become a registered charity and change its name to the British Arachnological Society. In 2019, it updated its charitable status to become a Charitable Incorporated Organisation. The prime objective of the Society is to use science and education to advance the wider understanding and appreciation of arachnids and to promote their conservation. By the end of 2018 the Society had around 490 British members and 120 overseas members.

Membership of the British Arachnological Society is open to anyone interested in the study of spiders and other arachnids. Advantages of membership include:

- Receiving the Society's scientific journal *Arachnology* and the *Newsletter*, published three times a year, and free to members. *Arachnology* publishes peer-reviewed scientific papers on all aspects of arachnology worldwide. The *Newsletter* contains short articles and notes, principally of interest to UK members, and incorporates news from our various recording schemes (see below).
- Access to the Society's library of over 24,000 arachnological papers from scientific journals, many of which are available as PDF files (see Section 10.1).
- Access to the Members' area of the BAS website, which includes downloadable issues of the *Newsletter* and of *Arachnology*, information on the small grant schemes, a PDF copy of this *Handbook*, the ability to search the BAS library, and other resources for arachnologists.
- If required, access to a personal mentor, who can guide inexperienced members through the techniques of identification, advise on equipment etc.
- Access to small grants to assist with arachnid research projects, including those underpinning conservation policy and action, and for travel and other expenses to attend conferences or identification workshops.
- Access to a lively email discussion group: the ideal place for advice, help, or just a chat.

The Society also:

- Runs the Spider and Harvestman Recording Schemes, and supports the Pseudoscorpion Recording Scheme, in order to develop our knowledge of the distribution, habitat preferences and phenology of the British faunas. Full details of the schemes are provided in Chapter 7.
- Provides impartial scientific advice and guidance to individuals, non-governmental organisations and government bodies, on the conservation of the British arachnid fauna.
- Holds, and encourages the holding of, courses, meetings, and other gatherings to foster the study of arachnids. These include short, one-day or weekend meetings, as well as occasional week-long courses, often in association with the Field Studies Council. Many of these activities are organized at a more local level by a network of Regional Co-ordinators.
- Produces an Occasional Publications series dealing with different aspects of arachnid systematics and biology.
- Promotes education and awareness of arachnids by appropriate national and local events and by publishing information and advice.

To become a member of the Society, visit www.britishspiders.org.uk and click on the Membership tab. Follow us on Twitter (@BritishSpiders) and Instagram (www.instagram.com/britishspiders).

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 more than 48,000 known species worldwide, rank second only to insects among terrestrial animals. However, in Britain we have a fairly restricted spider fauna (currently 670 known species) compared to that of other countries of north-west Europe. This is probably 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 added almost every year and the distribution of many established species shifting, 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 that 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 that are unique among animals. Although this is not the place to go into the details of anatomy and physiology of spiders (see, for example, Foelix 2010; Nentwig 2013), they differ completely from insects in their locomotion, prey capture and feeding, respiration, reproductive systems and behaviour. Above all, they differ from the insects in their production and use of silk for a variety of purposes and in injecting venom through their fangs to immobilise 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 lives. Most obvious to the casual observer are the various types of web that many use in prey capture. However, 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 vegetation and then release a long strand of silk into the breeze. Eventually the spider takes off on the end of the 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 the making of the sperm-web, which is spun by male spiders prior to mating. This is a minute sheet of silk on which the male deposits a small drop of sperm, which is drawn into the pedipalp (functionally equivalent in spiders to the penis in mammals). The pedipalp is 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 orbweb 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 orbweb, one for the production of dragline silk, one for both swathing prey items in silk and producing the sperm web and the last to produce the egg cocoon. The silk from each type of gland differs in its chemical and physical properties, ensuring that it is optimally fitted to its function. The silk made by insects is, by comparison, much less diverse. The silks produced by spiders are liquid proteins that 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, 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 suborder Mesothelae, found only in south-east Asia and Japan), individuals 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. This ancestral web type is also found in the British tube-web spiders (Segestriidae). It seems likely therefore that the potential for becoming free-living hunters or sedentary web-builders 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 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 predominantly by sight. Good examples of the first group in the British fauna include the sac spiders (Clubionidae) and the ground spiders (Gnaphosidae). Species in both of these groups have two rows of four small eyes on the carapace that 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 air currents, to detect the movement of prey.

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 capable of forming a proper image of objects. They are most highly developed in the jumping spiders, which have enormous anterior median eyes pointing forward on the carapace (Cover, middle; Fig. 24). These eyes are capable of accommodation: the ability to focus on objects at different distances, which allows the spider to estimate accurately its distance from a prey item. 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 salticids are the most species-rich spider family, with over 6000 species worldwide, although in Britain we have a rather impoverished fauna of some 41 species.

In web-building spiders, a considerable range of prey-capture devices have evolved, each of which is associated with different prey-capture behaviours. Some webs are specifically designed to catch flying insects while others trap insects and other invertebrates crawling on the ground or on the trunks of trees.

An example in the latter category is the funnel web of the Labyrinth Spider *Agelena labyrinthica*, common in the southern half of Britain in hedgerows, bushes and grassy areas. Insects that jump 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 horizontal 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 web. 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 orbweb, 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 orbweb weavers, particularly those belonging to the families Araneidae and Tetragnathidae, which 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—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 elements (Cover, top right; Fig. 10). Although to the casual eye all orbwebs 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 usually 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, from where it 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 ants. This is unusual among invertebrates since ants have formidable defences and live in colonies, thus making them risky prey. Theridiids may have evolved the ability to feed on ants in part because they possess extremely potent venom, which paralyses and kills their prey very rapidly. In *Cryptachaea* (*Achaearanea*) *riparia,* 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 parts of which have drops of gum along them. The threads are stretched extremely taut 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 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*, the web is highly reduced and consists of an H-shaped framework, built just above the ground surface, within which the spider hangs head downwards by its hind pair of legs. These spiders are often found in heathland where they construct the web beneath a heather bush or a similar small shrub. The 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 *Cryptachaea*. 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 behaviours of British spiders, an excellent starting point is Bristowe's (1958) *The world of spiders*. His vivid and well-illustrated accounts have been the stimulus for many to take an interest in this fascinating group. An accessible, up-to-date account of spider silk and webs is Brunetta and Craig's (2012) book, *Spider silk*.

1.5 Courtship Behaviour

As already mentioned, most 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 item or a potential mate before it approaches too closely. 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 trichobothria, special sensory hairs that 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 addition, chemical cues (pheromones) are usually present on the silk of a web or dragline, and provide sexually active males with information on the sex of the occupant, their stage of maturity and, if female, whether they are virgin or not. Air-borne pheromones may also be used by some species. The reception of these chemical messages is often revealed by a sudden change in the male's behaviour from exploratory mode to that of courtship.

In many web-building spiders, the males court the female in the web by plucking the edge of the web with a leg and/or drumming on it with the abdomen, legs or palps. In the genus *Amaurobius*, for example, it has been shown that each of the common European species 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 (Krafft 1978). Similar courtship behaviours are found in male orbweb spiders, which pluck the edge of the female's 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 (Lycosidae), lynx spiders (Oxyopidae) and jumping spiders (Salticidae), 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. Males locate potential mates at a distance by detecting pheromones deposited on dragline silk. 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. Each species has a different and specific pattern of palp and leg movements. The displays have an uncanny resemblance to the semaphore system with flags, used by the Royal Navy in the past to signal from ship to ship. Such visual courtship is also used by other lycosid genera, 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, which 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 coloration of the male palps, which allowed them to be distinguished 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 mentioned earlier, the salticids have the best-developed eyes and most acute vision of all spiders. It is now known 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 (Cover, middle; Fig. 24). As with lycosids, the male performs an elaborate courtship dance which involves palp 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 these behavioural elements. It is thought likely that the precise pattern of coloured hairs on the facial region also plays a role in species-specific recognition of the male by the female.

1.6 The Diversity of Spiders in Britain

Money spiders (Linyphiidae) dominate the British spider fauna, in common with that of most countries of north-west Europe, and account for over 40% of our spider species. This is in 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 large 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 that lack a dorsal pattern. While they are found in virtually all possible habitats, including on trees and shrubs, the majority of species live 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 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 at high densities. However, as might be expected in such a diverse family, many species are rare and known from very few localities. About 13% of our money spiders are considered to be under threat of extinction and 51% are either Nationally Rare or Nationally Scarce (Nationally Rare species have been recorded in 15 or fewer 10 × 10 km OS grid squares (hectads) and Nationally Scarce species in between 16 and 100 squares) (Harvey *et al*. 2017).

As with many other families, there are several different reasons for their rarity. In some cases, this may be a result of very specialised and uncommon habitat or microhabitat requirements. 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 deciduous 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, with adverse climate restricting their spread northwards. These include such species as *Trichoncus saxicola* (Vulnerable and Nationally Rare), *Agyneta* (*Meioneta*) *simplicitarsis* (Nationally Scarce), and *Neriene furtiva* (Nationally Scarce). Finally, there is an unfortunately large group of species that are apparently rare but, because we know so little of their detailed habitat requirements, our understanding 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 as a result is 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 around 8.5% of our species, and the Araneidae, one of the orbweb weaving families, which include about 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*, more often inhabit the ground layer. Again, many species, such as *Phylloneta sisyphia* (Fig. 8), *Enoplognatha ovata* (Fig. 9) and *Paidiscura pallens* are very widespread and common in Britain. In contrast to the linyphiids, only a small proportion (17%) of theridiids are Red Listed and 42% of the 58 species are Nationally Rare or Scarce. Among the Red Listed species are a number of interesting and rather poorly known spiders. Members of the genera *Dipoena*, *Lasaeola* and *Phycosoma* are apparently all specialist feeders on ants and occur predominantly in dry habitats in the south. Among the seven *Dipoena* species recorded in Britain, four are Red Listed (one, last recorded in 1913, as Regionally Extinct), one is Near Threatened (Harvey *et al*. 2017) and the other two are Nationally Scarce. *Robertus insignis*, 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.

British orbweb weavers in the family Araneidae are also characteristically found in the field and shrub layers but additionally extend into the higher tree zone. Among our 32 species, only two are Red Listed (although two more are Near Threatened (see IUCN 2012) and one is considered extinct in Britain), and eight are Nationally Scarce. Almost everyone will be familiar with the common Garden Spider, *Araneus diadematus* (Cover, top centre; Fig. 10) 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 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 started to spread northwards and, by 2019, it had become common throughout the south-east with a few isolated records as far north as Lincolnshire and Shropshire, and increasingly from Devon and Cornwall. It is to be found in tall grass where the female weaves a characteristic web with a zig-zag structure of iridescent silk (the stabilimentum) in the centre, and matures 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 around 5% of the British fauna. The majority of gnaphosids are medium-sized, fairly dark spiders with an elongated body form. As the common name suggests, they live almost exclusively on the ground surface. They are 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 13 of these more or less confined to an area south of a line drawn from the Thames estuary to that of the Severn. Worldwide, ground spiders are typical of hot and dry climates and it is therefore 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. Two thirds of the species are Nationally Rare or Scarce and eight of these are Red Listed, with a further two considered Near Threatened.

The two other common night-hunting families of spiders in Britain are the sac spiders (Clubionidae) and the prowling spiders (Cheiracanthiidae). Although they resemble the gnaphosids in body form, they are distinguished by the conical (rather than cylindrical) anterior spinnerets and their pale coloration. 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 26 species, 60% are widespread in Britain and only five species have a predominantly southern distribution. Indeed, unlike gnaphosids, these families in general are much more diverse in temperate climates than in hot or dry areas. Eight species are Red Listed or Near Threatened and a further two are Nationally Scarce. Most of these rarities are associated with our most threatened habitats; *Clubiona frisia* and *C. pseudoneglecta*, *Porrhoclubiona* (*Clubiona*) *genevensis* (Fig. 20) and *Cheiracanthium virescens* from coastal areas, *Cheiracanthium pennyi* from southern heathland, *Clubiona juvenis* and *C. rosserae* from fens, and *Clubiona subsultans* from Caledonian pine forest.

The wolf spiders (Lycosidae) are characteristically diurnal hunters with well-developed eyes and excellent eyesight. Our 38 species (6% of the total) include many that are familiar inhabitants of not only 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 confined 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* and *Piratula* of which *Pirata piraticus* is the commonest and most widespread, as well as *Pardosa purbeckensis* (salt marshes), *Arctosa leopardus*, and the rare *Hygrolycosa* *rubrofasciata*, which is found in fenlands mainly in East Anglia. Our wolf spiders include 18 Nationally Rare and Scarce species, eight of which are also Red Listed or Near Threatened. The Red Listed species include *Pardosa paludicola* (Endangered) known from half a dozen sites in southern Britain where it usually occurs in long grass near water, *Alopecosa fabrilis* (Critically Endangered) known from two heathland sites in Dorset and Surrey, *Arctosa fulvolineata* (Near Threatened) from saltmarsh sites in southern and eastern England and *Arctosa alpigena* (Vulnerable) from some 10 sites on mountains above 1000 m in northern Scotland.

The jumping spiders (Salticidae) are arguably our most distinctive and beautifully marked family of spiders. As already noted, their excellent eyesight allows 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 less than 6% of the British fauna. As with the gnaphosids, a large proportion of our 41 species have a southern distribution, almost half 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* (Cover, middle), are either very much less widely distributed in, or for 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 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* and *Calositticus* (*Sitticus*) *inexpectus*. 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.

The Salticidae is another family that has an unusually high proportion of rarities with 27 Nationally Rare and Scarce species, of which eight are also Red Listed and four are Near Threatened. The Red Listed species include: *Neon valentulus* (Critically Endangered) known from only six fenland sites in East Anglia, *Pellenes tripunctatus* (Vulnerable) known from only two shingle sites on the south coast and *Heliophanus dampfi* (Vulnerable), a species found in bogs, with one site in Wales, two in north-western England and five in Scotland. Among the Threatened and Near Threatened species, only two extend into northern England or Scotland and eight are largely confined to the area south of the Wash–Severn estuary line.

Since 1998, four new species of jumping spiders have been found in this country. The first was the small and rather inconspicuous species *Neon pictus*, initially discovered on shingle at Rye Harbour (Sussex) in 1998 and since then at three more south coast sites. *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 considered at the time to have been imported with the trees. However, it has subsequently been collected in Surrey, Essex and East Sussex and it is now thought it may have colonised the country without human assistance. A small species of *Attulus* (*Sitticus*), *A. 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. These are currently the only known localities in the UK for this species and since both are threatened by development, 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. Finally, *Sibianor larae*, a species first described in 2001, was discovered on Holcroft Moss, Cheshire, in 2018. In the past, it was probably confused with *S. aurocinctus* and might well be more widespread than we think.

The crab spiders represent a totally different type of diurnal hunter. The Thomisidae and Philodromidae are both included here because, although distinct, they are closely related and were formerly considered a single family. The two families combined include 44 species (about 6% of our total). 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 (Figs. 21, 22, 23). 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. Instead, they 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, with 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* (Fig. 23) 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 are confined to the area south of the Wash–Severn estuary line. The family includes 12 species listed as Nationally Rare or Scarce, of which six are also Red Listed. Among the former is *Thomisus onustus* (Nationally Scarce) (Cover, bottom left; Fig. 22), 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. Individuals can adjust their colour from pink through yellow to white to match the background colour of the flower they sit on. Another much commoner species, *Misumena vatia* (Fig. 21), can also change its colour to match that of the flowers on which it waits for prey. Both of these species can catch and subdue insects many times their own size. It is not uncommon to see a butterfly or bumble bee apparently feeding at a flower but which does not fly off when approached. Closer examination reveals that the insect is held in the jaws of a female *Misumena* while she absorbs the body fluids from it.

The 18 species of Philodromidae again include two different ecological groups. The first, represented by the genera *Philodromus, Rhysodromus* (*Philodromus*) and *Thanatus*, have body forms similar to the thomisid species 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 clear. Like the thomisids, a large proportion of philodromids have a southern distribution in Britain with nine of the 18 species found only in the southern half of the country. The family includes seven Nationally Rare and Scarce species, three of which are also Red Listed and one Near Threatened.

While this brief outline includes most of the larger families of spiders, representing over 80% of all our species, there are many smaller families. There is no space to mention them here but they include a diversity of body forms, prey-capture strategies and ecological adaptations. For those wishing 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 illustrations of many of our larger species can be found in the *Collins field guide: spiders of Britain and northern Europe* by Roberts (1995) and photographs in Bee, Oxford & Smith (2017). Finally, individual species accounts with comprehensive and up-to-date information on distributions are provided on the Society's Spider Recording Scheme (SRS) webpages at srs.britishspiders.org.uk (see Section 7.1).

1.7 Human Attitudes to Spiders

Spiders seem to inspire a combination of fascination and fear amongst the general public. 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 venomous bites can really account for such apprehension. Among the 48,000 or so described species of spiders world-wide, only some 20 or 30 are reliably reported to inflict seriously dangerous bites to humans and none of these is found in Britain. Very few species of British spiders have chelicerae strong enough to penetrate human skin and, in the rare cases where this does occur, the most unpleasant symptom likely to be experienced is a relatively mild and short-lived local reaction, equivalent to a bee or wasp sting. Of course, any puncture wound can become infected and medical help should be sought if in any doubt. Other reasons that are sometimes put forward for fear of spiders 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 (see Kelly 2009). It is hoped that this *Handbook* will help those who have decided to learn more of these fascinating animals.

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! However, if this short account has stimulated you to 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 distributions, habitats and biology of our fauna. For example, although the Spider Recording Scheme (srs.britishspiders.org.uk) has added enormously to our knowledge of the overall distribution of all our spider species, there are clearly still considerable gaps. A glance at the Scheme's coverage map at srs.britishspiders.org.uk/portal/p/Coverage, which plots the distribution of all spider records by 10 km square (hectad), shows that there are still some areas, particularly in north-east England and Scotland, for which there are no records at all. There is much work for the keen arachnologist to do in both filling these gaps and in recording at a more detailed level (tetrad—see the SRS regional coverage maps—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. Recorders are now asked to note details of the habitat and micro-habitat in which specimens are collected, thereby contributing to a more precise understanding of the requirements of our species (see Section 7.1). As a result, formerly very generalised descriptions of habitat preferences are being superseded by the growing body of much more detailed information available in the Habitats charts that follow the SRS species accounts.

Finally, our understanding of the biology of many of our spiders, whether it be life cycles, web construction and use, feeding habits, or courtship and mating behaviour, is woefully inadequate, even for our commonest species. For those who have the time and patience to observe live spiders, either in the field or in captivity, there are important discoveries to be made. For uncommon species, these can help us understand their conservation needs and safeguard their populations for the future. 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 (see Section 6) could add substantially to our knowledge of this important family. Likewise, among our 41 salticid species, courtship behaviour has only been studied properly in about a third and much could be learned about their inter-relationships. The keen amateur could potentially make an important contribution to all of these aspects of spider biology.

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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 Roberts's (1995) book *Collins field guide*: *spiders of Britain and northern Europe* and Bee, Oxford & Smith's (2017) *Britain's spiders – a field guide*, which provide coloured plates and photographs, respectively, of all the species mentioned below.

Pholcidae

The Daddy-longlegs Spider *Pholcus phalangioides* is an indoor species, which is commonest in the southern half of Britain (but rapidly spreading) and is often seen lurking in a tangle of thin threads in the angle of wall and ceiling (Fig. 4). 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 of other spiders, including even the large house spiders (*Eratigena* (*Tegenaria*) spp.) which share our homes.

Segestriidae

The six-eyed *Segestria senoculata* is very common in the stone walls of the north and west of Britain; elsewhere, the web may be found on walls and tree trunks with careful searching. It builds 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 (see page 63), but moves very rapidly and realising the deception darts back to safety. *Segestria senoculata* has a beautifully adapted tubular body with the first three pairs of legs directed forwards, and a clear zigzag 'adder' pattern on the abdomen. The much larger, introduced, *S. florentina*, characterised in adults by green iridescent jaws (Fig. 5), is currently expanding its range northwards. A third species, *S. bavarica*, is confined to western coasts.

Dysderidae

The spectacular and rather menacing-looking pink or red woodlouse spider, *Dysdera crocata* (Fig. 6), is widespread in gardens, though not often seen. It is found mostly in the southern half of Britain. As its common name suggests, it is a specialist predator of 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. In common with the next family, these spiders have only six eyes, not the usual eight.

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 is *Oonops domesticus*, which is found on the internal walls of buildings. Apart from its minute size and pale pink coloration, 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 themselves.

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 to as far north as Cumbria. A third species, *Uloborus plumipes*, is a relatively recent colonist and hence is not found in Roberts (1995). Thought to be native to southern Europe and Africa, it was first found in Britain as recently as the early 1990s and is now very common in garden centres. This family is unique among the world's spiders in lacking poison glands. *Uloborus plumipes* is a small, cryptically coloured spider, which weaves a horizontal orbweb, 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 difficult 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. There are, however, several species of *Steatoda* which, to the uninitiated, may vaguely resemble the Black Widow. *Steatoda bipunctata*, a shining, nut-brown, globular or slightly flattened spider usually with a white or cream crescent around the front of the abdomen, is common in and around houses. *Steatoda nobilis*, commonly (and unfortunately) referred to as the Noble False Widow Spider, also occurs around buildings in the south of the country but, unlike *S. bipunctata*, usually has a pattern of paler markings on the abdomen. It was introduced to Britain in the late nineteenth century and, by 2019, had spread north to the Midlands with some isolated introductions much further north e.g. Orkney. This species is beloved of the media looking for a spider scare story and has been implicated in many bite incidences, but almost always without a shred of evidence. Whilst its bite is often more painful than that of the few other British spiders that can penetrate human skin, verified cases of anything worse are extremely rare. This media coverage has been both highly inaccurate and irresponsible, and led to the temporary closure of four London schools in 2018 (and more since).

Other members of the family include *Paidiscura pallens* which, despite its tiny size, is instantly recognizable with the female guarding a bizarre, spiky egg sac several times larger than herself. In June and early July look on the underside of oak leaves and you have a good chance of finding this spider. Various other *Theridion* species are frequent in a variety of habitats. *Anelosimus vittatus* is a lovely golden-brown spider with a dark central band, commonly beaten from oak. *Episinus angulatus*, not uncommon in low vegetation, has an abdomen the shape of an isosceles triangle. The candy-stripe spider *Enoplognatha ovata* (Fig. 9) comes in three main colour forms: plain yellow, with two red stripes down the sides of the abdomen, or with a single broad carmine central stripe. It is very common in low vegetation in mid- to late summer with females guarding their curiously coloured, sky-blue egg sacs within a rolled leaf.

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, spinier 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 *Tenuiphantes tenuis*. This species can tentatively be 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 and so field identifications always need checking under the microscope. For those who progress to looking at spiders under the microscope, *T. 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 that are often found hanging in their orb webs, often 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, as adults, have abandoned web making and are ground hunters. Like *Tetragnatha*, they have disproportionately large jaws.

Araneidae

These classic orbweb weavers include some of our largest and most familiar spiders. Although the colours are often very variable within a single species, the abdominal patterns enable most araneids to be identified in the field (Figs. 12, 13).

The Garden Spider *Araneus diadematus* (Cover, top centre; Fig. 10) is widespread in gardens and low vegetation across the whole of Britain, and matures in late summer. When they hatch from the egg, spiderlings are bright yellow with a black triangle on the rear of the abdomen (Fig. 11), quite unlike the familiar adult with its white abdominal cross. *Larinioides cornutus* is a white and greenish-patterned spider that can be found in tall grass and herbage in all sorts of wet and damp habitats. Opening silken retreats in dead flower or reed heads often reveals this spider. Its darker and larger relative, the Bridge Orbweb Spider *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.

Cucumber spiders, *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, usually 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 zone 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 also widespread. The Four-spotted Orbweb Spider *Araneus quadratus*, with its characteristic four white spots, is reputedly Britain's heaviest 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 of Britain. One to look out for is the yellow and black Wasp Spider *Argiope bruennichi,* which, since the 1970s, has spread from its stronghold on the Hampshire and Dorset coasts and is known from a good many sites north of the Thames. By 2019 it had also been reported from Lincolnshire and Shropshire. It weaves its web low down in rough grassland in late summer, feeding largely on grasshoppers. Most araneids sit in a hidden retreat with a silk line leading to the web, but *Argiope* waits in the middle of her web.

Lycosidae

The wolf spiders, like the jumping spiders (see Salticidae, below), are mostly daytime hunters and also have good vision. In lycosids, the posterior median eyes are enlarged, whilst it is the anterior median eyes that are enlarged in salticids. There are several common species in the genus *Pardosa*, 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 (Fig. 14).

Several other genera are also common. These include the water-loving *Pirata* and *Piratula* 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 resembling a wolf spider, the eyes are smaller and the overall shape and pattern on the carapace and abdomen are unique to this species, enabling even the tiniest spiderlings to be identified. The female carries her large egg sac in her jaws (unlike the wolf spiders which attach their egg sacs to their spinnerets) until the eggs are about to hatch (Cover, bottom right). 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 emerge (Fig. 15). The two members of the genus *Dolomedes*, the so-called raft spiders, are rare, semi-aquatic species found in bogs and fens (Fig. 16). Both are very large, brown or black spiders usually with yellow or white lateral bands along the cephalothorax and abdomen and are capable of catching prey, including small fish, under water.

Agelenidae

The Labyrinth Spider *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 an 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* (Fig. 19) or *Eratigena* (*Tegenaria*), which include the familiar, hairy, large house spiders, which often turn up in the bath. Although several species are common, the vast majority in the east and Midlands of England are *Eratigena duellica* (*Tegenaria gigantea*) and in the west and Wales, *Eratigena* (*Tegenaria*) *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. *Iberina* (*Hahnia*) *montana* is probably the commonest of our six species and occurs in litter and moss, primarily in woodlands. *Antistea elegans* is found in marshy places and is distinguished by the overall reddish coloration of the body.

Dictynidae

These are small, web-building spiders. The 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*, whereas *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. This family now contains the unique Water Spider *Argyroneta aquatica*—our only spider that lives under water, and is widespread and not uncommon in suitable unpolluted weedy ponds, lakes and dykes. This species was previously placed in the family Cybaeidae.

Amaurobiidae

The abdominal pattern and rather large, slightly raised, parallel-sided head region are unmistakable. 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 a hole or crevice. Two very 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 usually be enticed out of their retreats by using a tuning fork or sonic toothbrush (see section 3.1.3) 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. However, two other much more localized species of *Anyphaena* have recently been found, which look closely similar to *A. accentuata*.

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.

Phrurolithidae

The ant-mimicking spider *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 characteristically folded leaves, sealed with a chalky-white silk.

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.

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* (and other very closely related genera previously regarded as *Zelotes*) are jet black in colour and are fast moving, night-time hunters often found under stones in dry habitats.

Miturgidae

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 coloration provides 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 (*philos* = loving; *dromos* = running) 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 species in Britain to identify confidently. Also in this family are our two *Tibellus* species, elongated straw-coloured spiders that 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 Crustacea. 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* (Fig. 21) is able to change colour to match the white or yellow flower heads where it awaits the arrival of prey. *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 crab spider 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, with their huge forward-facing, anterior median eyes, have been shown to have excellent binocular vision. Indeed, by observing 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. Worldwide it is the most species-rich family, concentrated in tropical regions. *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* (Cover, middle), has a mottled abdomen and bright orange hairs around the eyes. It occurs in a wide range of habitats.

Other families

Twelve other families are represented in Britain by one or more usually distinctive species, which are mostly uncommon or rare: Atypidae (Purse-web Spider *Atypus affinis*); Scytodidae (Spitting Spider *Scytodes thoracica*); Eresidae (Ladybird Spider *Eresus sandaliatus*); Nesticidae (*Nesticus cellulanus* and *Kryptonesticus eremita*); Theridiosomatidae (*Theridiosoma gemmosum*); Mysmenidae (*Trogloneta granulum*); Oxyopidae (*Oxyopes heterophthalmus*); Cybaeidae (*Cryphoeca silvicola* and *Tuberta maerens*); Cheiracanthiidae (*Cheiracanthium erraticum*, *C. pennyi* and *C. virescens*); Zoropsidae (*Zoropsis spinimana*); Sparassidae (*Micrommata virescens*) (Fig. 18). And, finally, we have four species in the family Zodariidae, of which only *Zodarion italicum* is at all likely to be found, and then only in the Thames Gateway region.

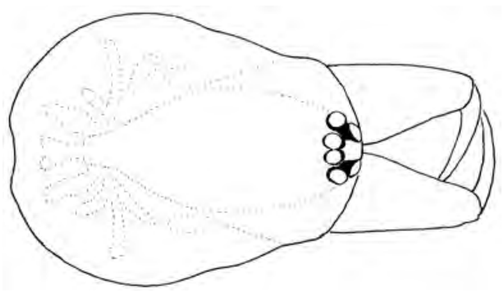
2.2 Illustrated Key to the Families of British Spiders

(*adapted from Roberts 1985/1987*)

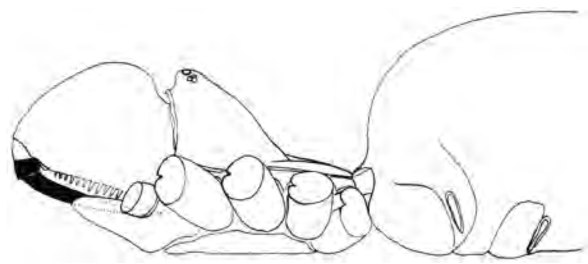
All families in the new British checklist that are established in natural or semi-natural habitats are included (List 1A: see Section 2.3).

1 Chelicerae massive and projecting in front of the carapace, articulated for upward and downward movement. Suborder ORTHOGNATHA (Mygalomorphae) .... ATYPIDAE
Notes: *Atypus affinis*, the only British mygalomorph spider, lives in silk-lined burrows. It is largely confined to southern Britain. 10 mm or more in length, including the chelicerae.

labidognath, top view

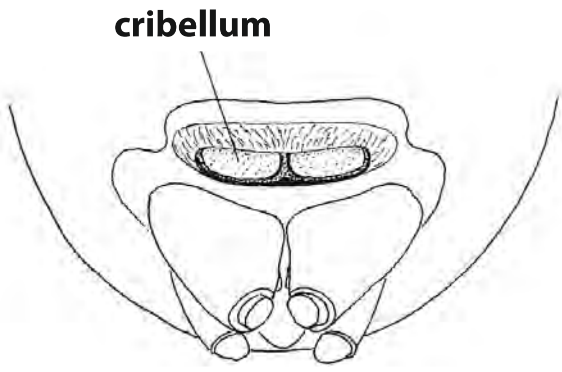


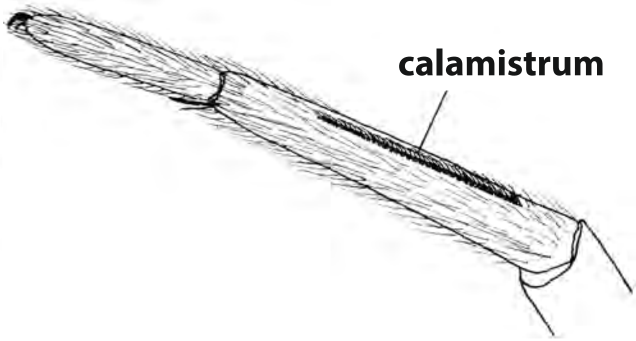
orthognath, side view



– Chelicerae attached under anterior border of carapace, articulated for side to side (pincer action) movement. Suborder LABIDOGNATHA (Araneomorphae) (all other British spiders) 2

2(1) Cribellum present anterior to spinnerets (reduced in ♂), ♀ with calamistrum on metatarsus IV... 3
Notes: The cribellum and calamistrum in smaller species of the family Dictynidae can be difficult to see. It is essential to have adequate lighting and angle specimens correctly when viewing them under the microscope. A photograph of the cribellum of *Amaurobius* can be found in Bee, Oxford & Smith (2017).



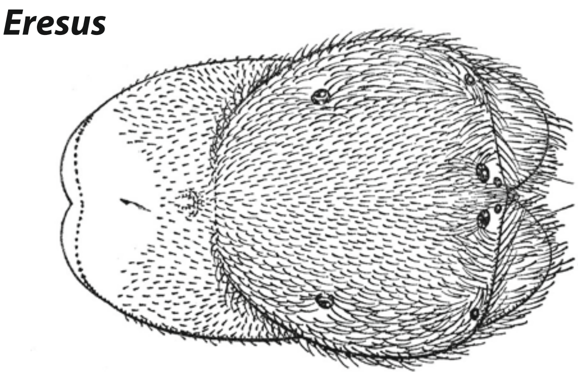


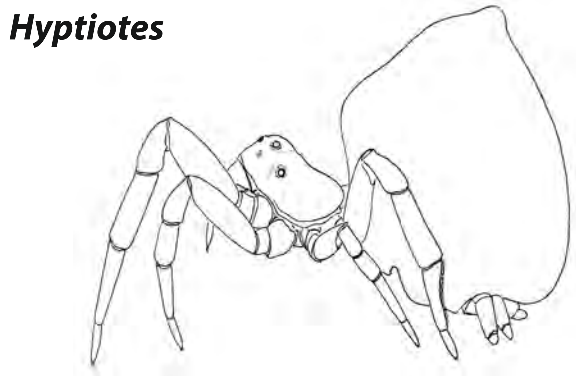
– Cribellum and calamistrum absent.. 6

3(2) Large spiders (6–16 mm) with square-fronted carapace. Male strikingly marked in black and red, female uniformly velvety black.. ERESIDAE
Note: *Eresus sandaliatus* is a rare spider confined to a few heathland sites in Dorset. See: ednieuw.home.xs4all.nl/Spiders/Eresidae/Eresidae.htm

– Smaller spiders (less than 4 mm) with a markedly triangular-shaped abdomen in side view .... ULOBORIDAE
Note: There are three British species, belonging to the genera *Uloborus* and *Hyptiotes*.

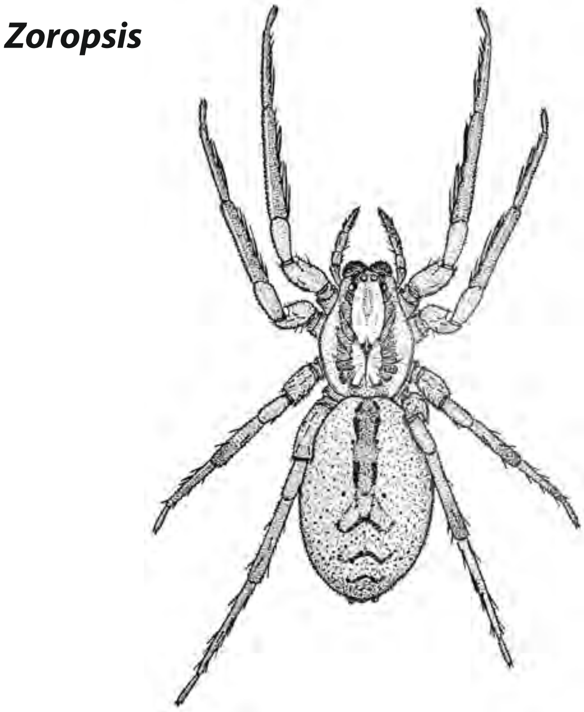
– Not as above 4





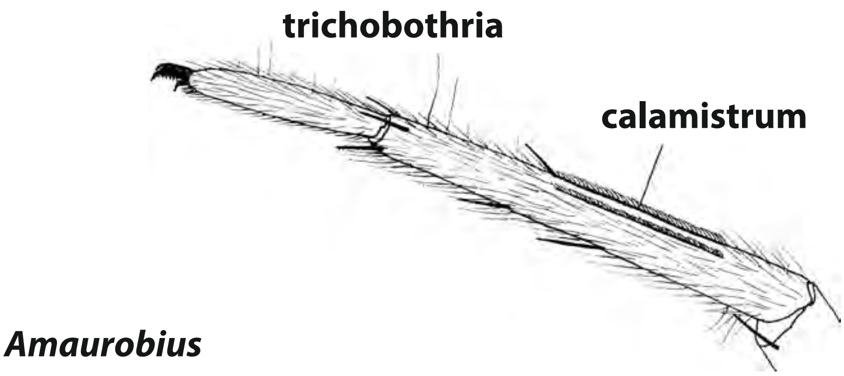
4(3) Very large cribellate spiders (10–19 mm) with eyes in three rows, resembling a large lycosid ....ZOROPSIDAE

Note: A single species, *Zoropsis spinimana*, has recently established in the London area but is now spreading more widely.



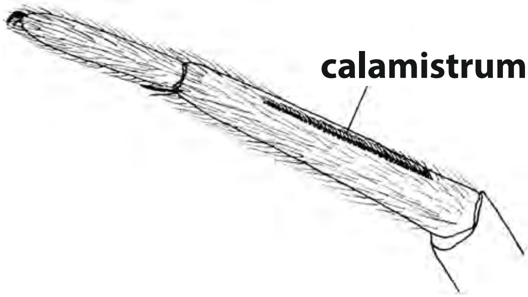
– Not as above, eyes arranged in two rows 5

5(4) Fairly large cribellate spiders (5–12 mm). Calamistrum of ♀ having a double row of bristles. Tarsi with several trichobothria.. AMAUROBIIDAE

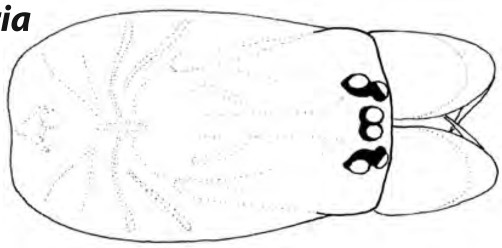


– Mostly small cribellate spiders (1.5–4.0 mm). Calamistrum of ♀ having a single row of bristles. Tarsi with either one or no trichobothria .... DICTYNIDAE

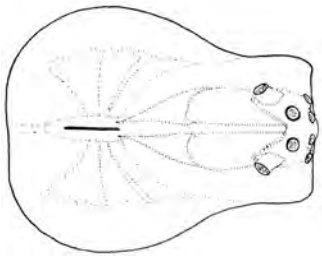
Note: The Water Spider *Argyroneta aquatica* has recently been transferred to the Dictynidae. Its unique habitat and way of life make it unmistakable. Two genera included here, *Cryphoeca* and *Tuberta*, were recently transferred to the family Cybaeidae on the basis of molecular evidence. Since morphological characters to justify the transfer currently appear to be lacking, we have retained them in Dictynidae for the sake of stability.

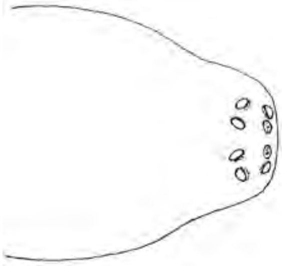


6(2) Spiders with six eyes, all clearly visible from above. Adult female lacking an epigyne and male palp with a simple bulb. (HAPLOGYNE spiders) 7



– Spiders with eight eyes, in some families not all easily seen from above. Adult female with a clearly defined external epigyne and male palp with a more or less complex bulb. (ENTELEGYNE spiders).. 8





7(6) Spiders marked clearly on carapace, abdomen and legs in black on a yellow background. Carapace elevated behind, and approximately as big as the abdomen .... SCYTODIDAE

Note: The single British species, *Scytodes thoracica*, is very distinctive and always found in houses or other buildings.

– Tiny spiders (1.5–2.0 mm), uniformly pale, pinkish when alive, with no dark markings. Chelicerae small .... OONOPIDAE

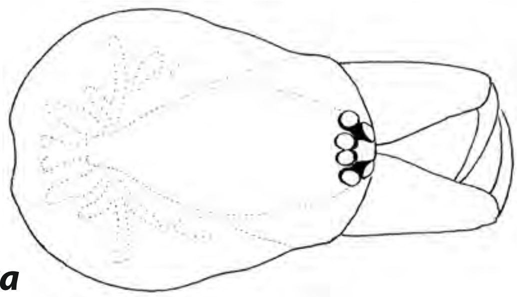
Note: There are only two native British species.

– Larger spiders (5–15 mm), eyes in a compact circular arrangement, abdomen lacking a defined pattern .... DYSDERIDAE

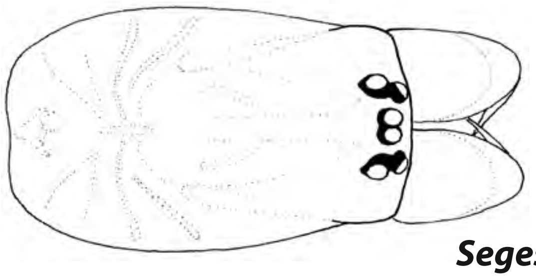
Note: Only four British species belonging to the genera *Dysdera* and *Harpactea*.

– Larger spiders (6–22 mm), eyes arranged as in figure below, abdomen with a clear dorsal pattern or almost black .... SEGESTRIIDAE

Note: There are only three British species belonging to the genus *Segestria*. They build tubular webs in walls, rock faces and tree trunks.



Dysdera

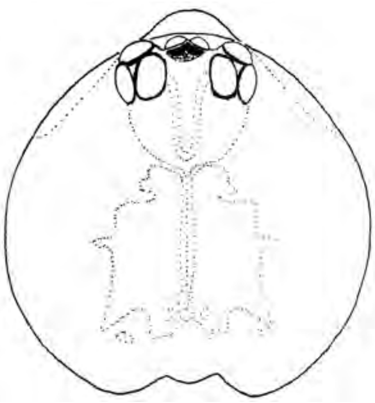


Segestria

8(6) Eight-eyed spiders with a roughly circular carapace and the eyes arranged in a more or less compact group .... PHOLCIDAE

Note: Only two British species, both normally found in houses and other buildings. The exceptionally long, thin legs give rise to the vernacular name 'daddy long-legs spiders'.

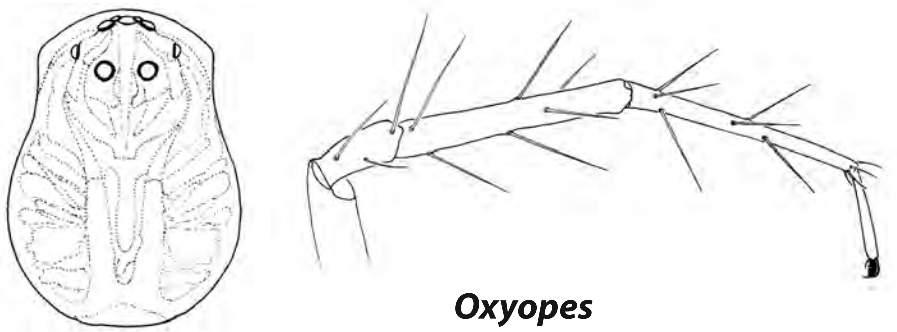
– Carapace and eye arrangement otherwise 9



Pholcus

9(8) Eyes arranged in a roughly circular pattern, the anterior medians extremely small and difficult to see from above. Legs with very long spines .... OXYOPIDAE

Note: The only British species, *Oxyopes heterophthalmus*, is a distinctive spider confined to a few southern heathland areas.



Oxyopes

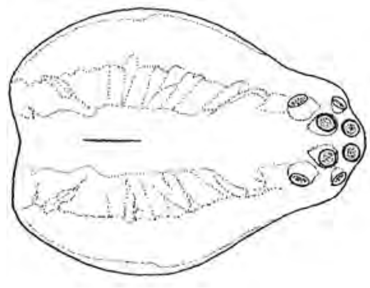
– Eye arrangement otherwise 10

10 (9) Eyes arranged in three more or less distinct rows 11

– Eyes arranged in two rows 15

11(10) Inner eyes of first row and eyes of second row slightly larger than others. Medium sized spiders (3.5–6.5 mm) with a cream carapace with two darker longitudinal bands .... MITURGIDAE

Note: There are only four species belonging to the genus *Zora* in the British fauna; all but *Z. spinimana* are rare.

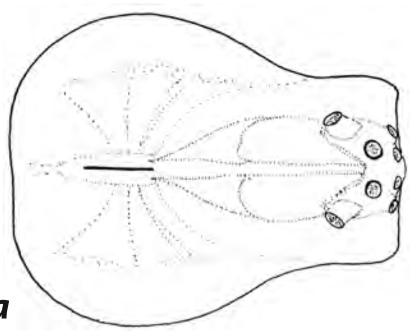


Zora

– Eyes not arranged as in the figure above 12

12(11) Eyes of second and third row more or less equal in size. Second row of eyes noticeably narrower than first. Large spiders (9–16 mm) .... PISAURIDAE

Note: Three very distinctive British species belonging to the genera *Pisaura* and *Dolomedes*.

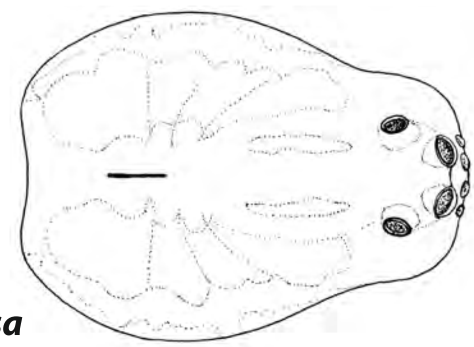


Pisaura

– Eyes not as in the figure above 13

13(12) Anterior margin of carapace more or less curved. Anterior row of eyes small and difficult to see from above. Second eye row as wide as first and the eyes slightly larger than those of posterior row .... LYCOSIDAE

Note: The 38 species of wolf spider in the British fauna include some of our most widespread and common species.

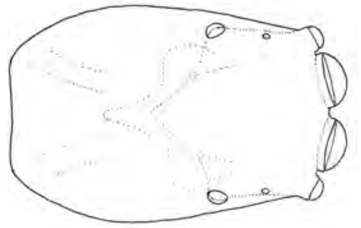


Trochosa

– Carapace shape and eyes not as in the figure above 14

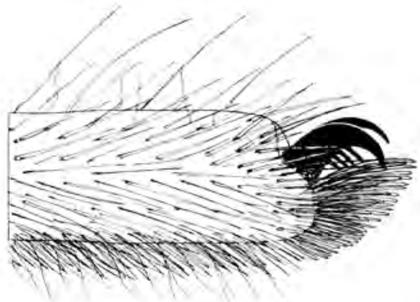
14(13) Anterior margin of carapace square-fronted with two very large and two smaller eyes in the first row. Eyes of second row minute, those of third row larger .... SALTICIDAE

Note: There are 41 species of jumping spiders recorded in Britain. Although some are common and widespread, many are relatively uncommon or rare and mainly confined to southern areas.

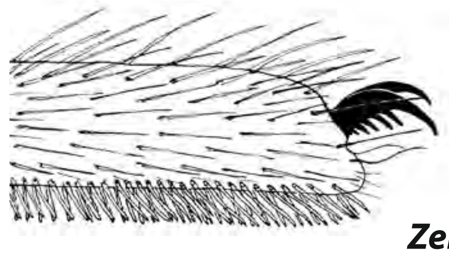


Salticus

15(10) Tarsi with only two claws which may be partially hidden by hair tufts.. 16



Clubiona



Zelotes

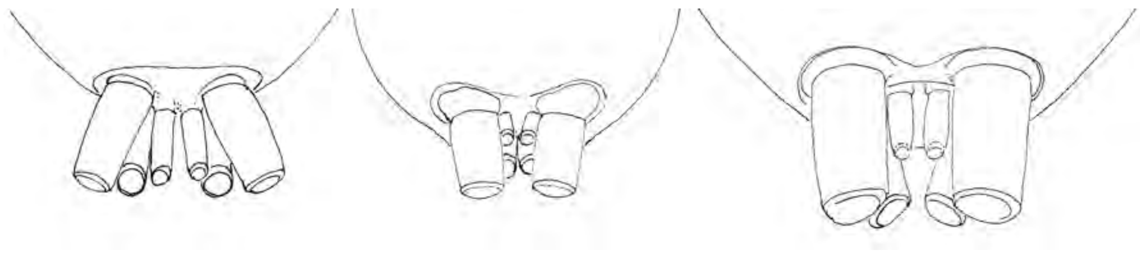
– Tarsi with three, easily visible, claws, the central one often smaller... 23

Araneus

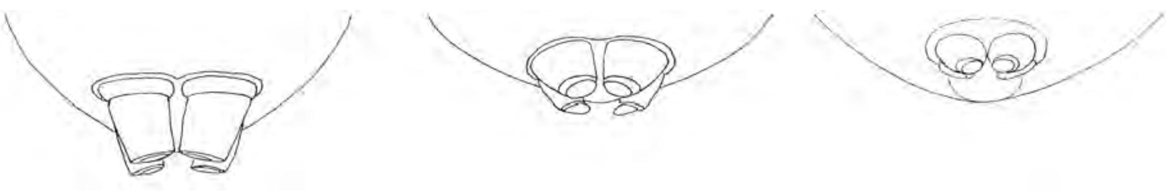
Pachygnatha

16(15) Anterior lateral spinnerets cylindrical (parallel-sided) and well separated so that anterior median spinnerets are easily visible.. GNAPHOSIDAE

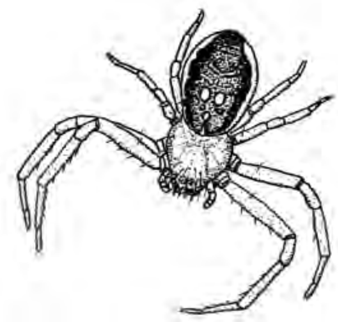
Note: Most of the 33 species of ground spiders in the UK are recorded from warm, dry habitats. Only one third of the species are widespread, the remainder having a more southern distribution.



– Anterior lateral spinnerets conical or occasionally cylindrical but close together so that anterior median spinnerets are not visible... 17



17(16) Legs laterigrade (directed towards side) giving a crab like stance 18



Diaea

Xysticus

– Legs prograde (I & II directed forward, II & IV directed backwards).. 20

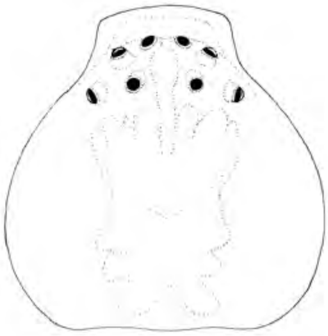
18(17) Tarsi and metatarsi without scopulae. Legs I & II usually much longer than legs III & IV ....
.... THOMISIDAE

Note: There are 26 species of crab spider recorded from Britain. About 40% are widespread but the remainder are largely confined to southern regions.

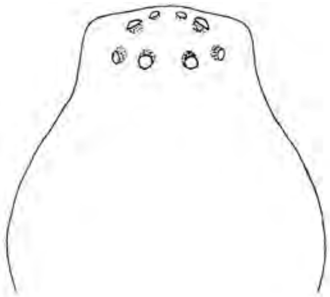
– Tarsi and metatarsi with scopulae, legs more even in length 19

19(18) Posterior row of eyes recurved (curved backwards)... PHILODROMIDAE

Note: The 18 species of running crab spiders in Britain are found on vegetation and include many which are widespread and common throughout the country.



Philodromus



Micrommata

– Posterior row of eyes procurved (curved forward) .... SPARASSIDAE

Note: The single British species, *Micrommata virescens*, is a large and strikingly coloured green spider largely confined to southern counties of England.

20(17) Tracheal spiracles easily visible midway between spinners and epigastric fold. Single British species with a distinctive dorsal mark on abdomen.. ANYPHAENIDAE

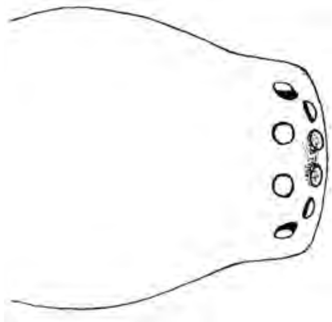
Note: There are three British species belonging to the genus *Anyphaena*.

Anyphaena

– Tracheal spiracles immediately behind epigastric fold 21

21(20) Maximum width of eye rows at least half width of carapace at its widest point. Legs relatively short, male palpal cymbium without a backward directed spur .... CLUBIONIDAE

Note: More than half of the 23 British sac spiders are very common and widespread. The majority are found on the ground or in field-layer vegetation.



Clubionidae

– Legs longer, male cymbium with backward-directed spur .... CHEIRACANTHIIDAE

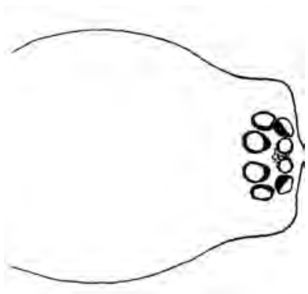
Note: The three British species of *Cheiracanthium* are generally found in the field layer of open habitats.

Cheiracanthium

– Maximum width of eye rows less than half width of carapace.. 22

22 (21) Spiders not ant like, abdomen not patterned .... LIOCRANIDAE

Note: There are 12 species of liocranids found in Britain.



Scotina

– Ant-like spiders, abdomen patterned in black and white .... PHRUROLITHIDAE

Note: There are two British species in the genus *Phrurolithus*.

Phrurolithus

23(15) Spinnerets in a single transverse row. Maximum length 3 mm, usually smaller .... HAHNIIDAE

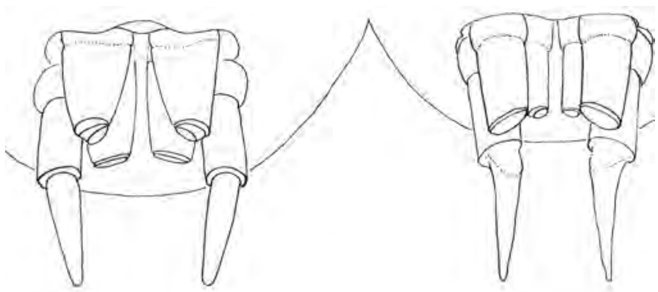
Note: The family includes ten British species in the genera *Hahnia*, *Antistea*, *Circurina* and *Mastigusa*.

Hahnia

– Spinnerets not arranged in a single transverse row 24

24(23) Posterior lateral spinnerets longer, consisting of two segments. Median spinnerets easily visible........ .... AGELENIDAE

Note: The majority of the 14 British species are large spiders, many found in buildings.



Tegenaria ***Textrix***

– Posterior lateral spinnerets not significantly longer than anterior laterals.. 25

25(24) Anterior spinnerets much larger than the others, arising on a large, pale cylindrical projection. Anterior median eyes larger than others.. ZODARIIDAE

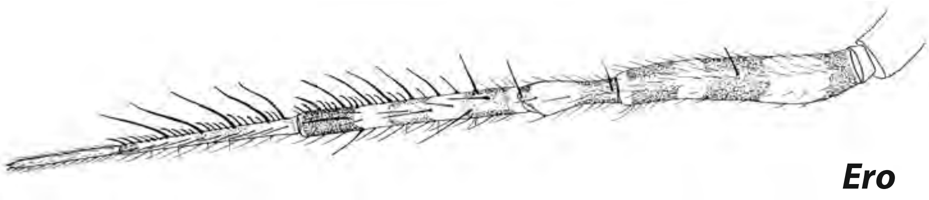
Note: Among the four species of *Zodarion* found in Britain, only *Z. italicum* has been frequently recorded (and then mainly in the Thames corridor).

Zodarion

– Spinnerets not modified as in 23–25 above 26

26(25) Legs I & II with a series of long curved spines on the metatarsus and tibia when viewed from above with small curved spines set at regular intervals between. Abdomen with two or four very obvious swellings. 2.5–4.0 mm .... MIMETIDAE

Note: There are four species of pirate spider in Britain all of which are predators of other web-building spiders.

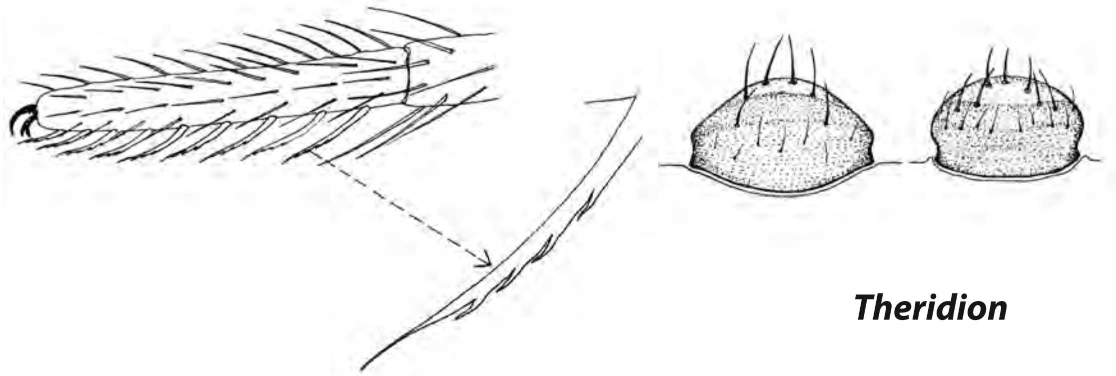


Ero

– Legs not as above 27

27(26) Tarsi IV with a comb of serrated bristles ventrally and labium not swollen distally .... THERIDIIDAE

Note: The tarsal comb is often difficult to see in small species and careful examination is needed. Many of the 58 British species of comb-footed spiders are widespread and common. The abdomen is rather globular in many species, often with a distinctive pattern. Most are medium-sized spiders around 3 to 5 mm. Web composed of irregular criss-cross strands, only occasionally forming a sheet below.

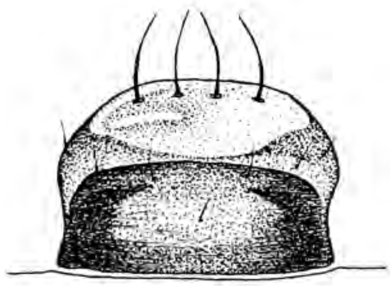


Theridion

– Labium not as above 28

28(27) Tarsi IV with an easily visible comb of serrated bristles ventrally and labium distinctly swollen distally .... NESTICIDAE

Note: There are two British species belonging to the genera *Nesticus* and *Kryptonesticus*.

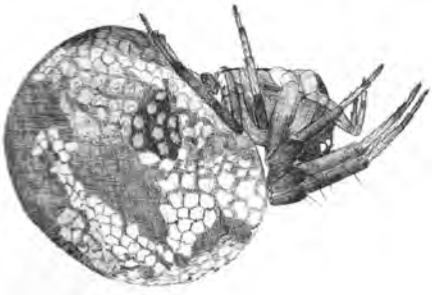


Nesticus

– Tarsal comb on leg IV absent 29

29(28) Small spider (1.5–3.0 mm), both sexes with globular abdomen marked with silver, leg I of male without spur .... THERIDIOSOMATIDAE

Note: The single British species, *Theridiosoma gemmosum* is always found near water where it spins a small orb web



Theridiosoma

– Not as above, leg I of male modified 30

30(29) Minute spider (0.9–1.1 mm), pale abdomen raised and globular, leg I of male with prolateral spine .... MYSMENIDAE

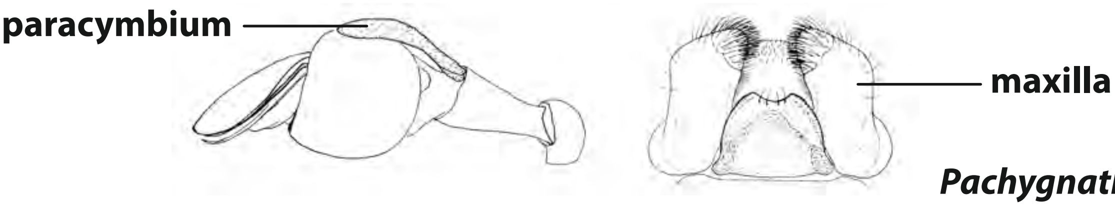
Note: The single British species *Trogloneta granulum* has recently been recorded from two sites in Wales.

Trogloneta

– Abdomen not as above, larger spiders 31

31(30) Maxillae distinctly longer than broad. Female epigynes simple, male palps with elongate paracymbium .... TETRAGNATHIDAE

Note: There are 14 British species in four genera. *Tetragnatha* species have markedly enlarged chelicerae and elongate bodies but the other three genera have less characteristic body forms and are sometimes mistaken for araneids.

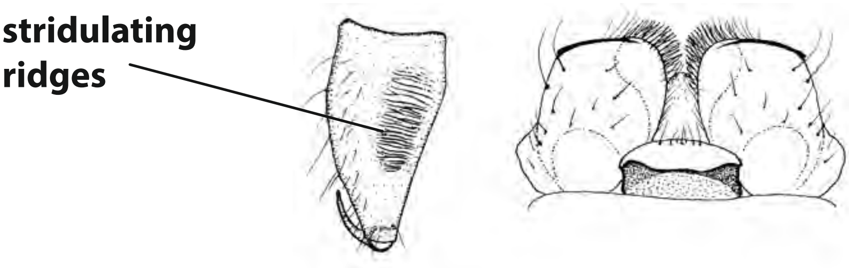


Pachygnatha

– Maxillae not or scarcely longer than broad 32

32(31) Chelicerae normally with stridulating ridges laterally and lacking a lateral condyle (swelling near base of chelicerae). Linyphiids produce sheet webs .... LINYPHIIDAE

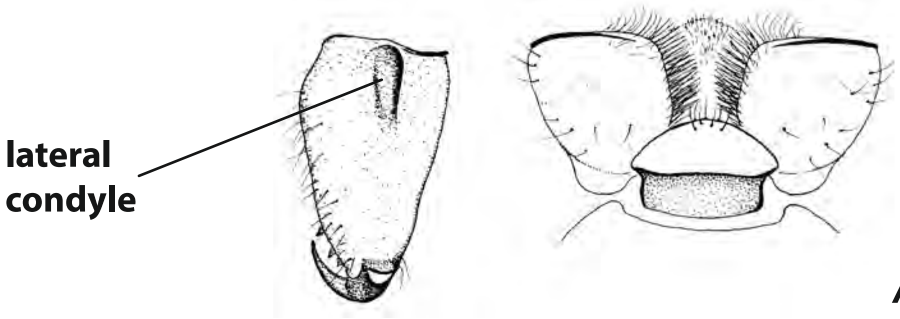
Note: The money spiders are the largest family in the British fauna, with 280 species. The vast majority are small (<4 mm), dark-coloured spiders although a few of the subfamily Linyphiinae are larger and have a dorsal pattern on the abdomen.



Tenuiphantes

– Chelicerae without stridulating ridges and normally with a lateral condyle. Araneids produce orb webs .... ARANEIDAE

Note: The orbweb weavers include 32 British species. The majority are medium to large spiders (5-15 mm) with sub-spherical brightly coloured abdomens, many of which have a marked dorsal pattern.



Araneus

2.3 Checklist of British and Irish Spiders

This new checklist is taken from Lavery (2019), whose paper is available for download from doi.org/10.13156/arac.2019.18.3.196 and is free to BAS members. It replaces a previous list published by Merrett, Russell-Smith & Harvey (2014). In addition to updating the list of spiders found in Great Britain and Ireland with names accepted by the *World Spider Catalog* (wsc.nmbe.ch), it includes:

- presence in constituent nations of the British Isles,
- United Kingdom and International Union for Conservation of Nature (IUCN) conservation statuses from Harvey *et al*. (2017),
- guidance on how to trace current nomenclature to the most frequently used national identification resources (2014 checklist (see above) and Roberts, 1985, 1987 & 1993).

Table 1. Species found in Great Britain and Ireland

List A1. Species established in natural or semi-natural habitats
List A2. Species awaiting taxonomic definition
List A3. Species with insufficient data to determine status

Abbreviations

Nations

E = England: S = Scotland: W= Wales: NI = Northern Ireland: RI = Republic of Ireland.

Anthropic Status (AS)

Ag = exclusively or almost exclusively in greenhouse-like structures: Ah = exclusively or almost exclusively in houses: Ab = exclusively or almost exclusively in buildings other than homes, such as warehouses and industrial structures.

Conservation status

GB = Great Britain designations: NS = Nationally Scarce: NR = Nationally Rare.

IU = IUCN designations: CE = Critically Endangered: E = Endangered: V = Vulnerable: NT = Near Threatened: DD = Data Deficient.

List A1. Species established in natural or semi-natural habitats

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Family ATYPIDAE										
Atypus affinis Eichwald, 1830			E	S	W		RI		NS	
Family SCYTODIDAE										
Scytodes thoracica (Latreille, 1802)			E		W		RI	Ah		
Family PHOLCIDAE										
Pholcus phalangioides (Fuesslin, 1775)			E	S	W	NI	RI	Ah		
Psilochorus simoni (Berland, 1911)			E	S	W	NI	RI	Ah		
Family SEGESTRIIDAE										
Segestria senoculata (Linnaeus, 1758)			E	S	W	NI	RI			
Segestria bavarica C. L. Koch, 1843			E		W				NR	
Segestria florentina (Rossi, 1790)			E	S	W					
Family DYSDERIDAE										
Dysdera erythrina (Walckenaer, 1802)			E	S	W	NI	RI			
Dysdera crocata C. L. Koch, 1838			E	S	W	NI	RI			
Harpactea hombergi (Scopoli, 1763)			E	S	W	NI	RI			
Harpactea rubicunda (C. L. Koch, 1838)		Appendix 2	E						NR	V
Family OONOPIDAE										
Oonops pulcher Templeton, 1835			E	S	W	NI	RI			
Oonops domesticus Dalmas, 1916			E	S	W	NI	RI	Ah		
Family MIMETIDAE										
Ero cambridgei Kulczyński, 1911			E	S	W	NI	RI			
Ero furcata (Villers, 1789)			E	S	W	NI	RI			
Ero aphana (Walckenaer, 1802)			E	S					NS	
Ero tuberculata (De Geer, 1778)			E	S					NS	
Family ERESIDAE										
Eresus sandaliatus (Martini & Goeze, 1778)		*Eresus niger*	E						NR	V
Family ULOBORIDAE										
Uloborus walckenaerius Latreille, 1806			E						NR	NT
Uloborus plumipes Lucas, 1846		not present	E	S	W		RI	Ag		
Hyptiotes paradoxus (C. L. Koch, 1834)			E		W		RI		NS	
Family NESTICIDAE										
Nesticus cellulanus (Clerck, 1757)			E	S	W	NI	RI			
Kryptonesticus eremita (Simon, 1880)	not present	not present			W					
Family THERIDIIDAE										
Episinus angulatus (Blackwall, 1836)			E	S	W	NI	RI			
Episinus truncatus Latreille, 1809			E		W		RI		NS	
Episinus maculipes Cavanna, 1876			E			NI	RI		NS	
Euryopis flavomaculata (C. L. Koch, 1836)			E	S	W	NI	RI		NS	
Lasaeola prona (Menge, 1868)	*Dipoena prona*	*Dipoena prona*	E		W				NR	E
Lasaeola tristis (Hahn, 1833)	*Dipoena tristis*	*Dipoena tristis*	E				RI		NS	
Phycosoma inornatum (O. P.-Cambridge, 1861)	*Dipoena inornata*	*Dipoena inornata*	E	S	W		RI		NS	
Dipoena erythropus (Simon, 1881)			E		W				NR	V
Dipoena melanogaster (C. L. Koch, 1837)			E				RI		NR	E
Dipoena torva (Thorell, 1875)				S					NR	NT
Crustulina guttata (Wider, 1834)			E	S	W		RI			
Crustulina sticta (O. P.-Cambridge, 1861)			E				RI		NS	
Asagena phalerata (Panzer, 1801)	*Steatoda phalerata*	*Steatoda phalerata*	E	S	W	NI	RI			
Steatoda albomaculata (De Geer, 1778)			E		W				NR	
Steadota bipunctata (Linnaeus, 1758)			E	S	W	NI	RI			
Steatoda grossa (C. L. Koch, 1838)			E	S	W	NI	RI	Ab		
Steatoda nobilis (Thorell, 1875)		Appendix 2	E	S	W	NI	RI	Ab		
Steatoda triangulosa (Walckenaer, 1802)		not present	E		W					
Anelosimus vittatus (C. L. Koch, 1836)			E	S	W	NI	RI			
Kochiura aulica (C. L. Koch, 1838)		*Anelosimus aulicus*	E						NS	
Cryptachaea riparia (Blackwall, 1834)	*Achaearanea riparia*	*Achaearanea riparia*	E	S	W		RI		NS	
Cryptachaea blattea (Urquhart, 1886)	not present	not present	E		W					
Cryptachaea veruculata (Urquhart, 1885)		*Achaearanea veruculata*	E							
Parasteatoda lunata (Clerck, 1757)	*Achaearanea lunata*	*Achaearanea lunata*	E		W		RI			
Parasteatoda tepidariorum (C. L. Koch, 1841)	*Achaearanea tepidariorum*	*Achaearanea tepidariorum*	E	S	W	NI	RI	Ag		
Parasteatoda simulans (Thorell, 1875)	*Achaearanea simulans*	*Achaearanea simulans*	E		W					
Phylloneta sisyphia (Clerck, 1757)		*Theridion sisyphium*	E	S	W	NI	RI			
Phylloneta impressa (L. Koch, 1881)		*Theridion impressum*	E	S	W	NI	RI			
Theridion pictum (Walckenaer, 1802)			E	S	W					
Theridion hemerobium Simon, 1914			E		W		RI		NS	
Theridion hannoniae Denis, 1944		not present			W					
Theridion varians Hahn, 1833			E	S	W	NI	RI			
Theridion pinastri L. Koch, 1872			E						NS	

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Theridion familiare O. P.-Cambridge, 1871			E	S					NS	
Theridion melanurum Hahn, 1831			E	S	W	NI	RI			
Theridion mystaceum L. Koch, 1870			E	S	W	NI	RI			
Sardinidion blackwalli (O. P.-Cambridge, 1871)	*Theridion blackwalli*	*Theridion blackwalli*	E	S	W		RI		NS	
Platnickina tincta (Walckenaer, 1802)		*Theridion tinctum*	E	S	W	NI	RI			
Simitidion simile (C. L. Koch, 1836)		*Theridion simile*	E	S	W	NI	RI			
Neottiura bimaculata (Linnaeus, 1767)		*Theridion bimaculatum*	E	S	W	NI	RI			
Paidiscura pallens (Blackwall, 1834)		*Theridion pallens*	E	S	W	NI	RI			
Rugathodes instabilis (O. P.-Cambridge, 1871)		*Theridion instabile*	E		W	NI	RI		NS	
Rugathodes bellicosus (Simon, 1873)		*Theridion bellicosum*	E	S	W		RI		NR	
Rugathodes sexpunctatus (Emerton, 1882)		not present		S						
Coleosoma floridanum (Banks, 1900)	not present	not present	E					Ag		
Enoplognatha ovata (Clerck, 1757)			E	S	W	NI	RI			
Enoplognatha latimana Hippa & Oksala, 1982		Appendix 1	E	S	W		RI			
Enoplognatha thoracica (Hahn, 1833)			E	S	W	NI	RI			
Enoplognatha mordax (Thorell, 1875)		*Enoplognatha crucifera*	E	S	W				NS	
Enoplognatha caricis (Fickert, 1876)	*Enoplognatha tecta*	*Enoplognatha tecta*	E						NR	V
Enoplognatha oelandica (Thorell, 1875)			E						NR	CE
Robertus lividus (Blackwall, 1836)			E	S	W	NI	RI			
Robertus arundineti (O. P.-Cambridge, 1871)			E	S	W	NI	RI			
Robertus neglectus (O. P.-Cambridge, 1871)			E	S	W	NI	RI		NS	
Robertus scoticus Jackson, 1914				S					NR	CE
Robertus insignis O. P.-Cambridge, 1907			E						NR	DD
Pholcomma gibbum (Westring, 1851)			E	S	W	NI	RI			
Theonoe minutissima (O. P.-Cambridge, 1879)			E	S	W	NI	RI			
Family THERIDIOSOMATIDAE										
Theridiosoma gemmosum (L. Koch, 1877)			E		W	NI	RI		NS	
Family MYSMENIDAE										
Trogloneta granulum Simon, 1922	not present	not present			W					
Family LINYPHIIDAE										
Ceratinella brevipes (Westring, 1851)			E	S	W	NI	RI			
Ceratinella brevis (Wider, 1834)			E	S	W	NI	RI			
Ceratinella scabrosa (O. P.-Cambridge, 1871)			E	S	W	NI	RI			
Walckenaeria acuminata Blackwall, 1833			E	S	W	NI	RI			
Walckenaeria mitrata (Menge, 1868)			E						NR	V
Walckenaeria antica (Wider, 1834)			E	S	W	NI	RI			
Walckenaeria alticeps (Denis, 1952)			E	S	W	NI	RI		NS	
Walckenaeria cucullata (C. L. Koch, 1836)			E	S	W		RI			
Walckenaeria nodosa O. P.-Cambridge, 1873			E	S	W	NI	RI		NS	
Walckenaeria atrotibialis (O. P.-Cambridge, 1878)			E	S	W	NI	RI			
Walckenaeria capito (Westring, 1861)			E	S	W		RI		NS	
Walckenaeria incisa (O. P.-Cambridge, 1871)			E	S	W				NS	
Walckenaeria dysderoides (Wider, 1834)			E	S	W	NI	RI		NS	
Walckenaeria stylifrons (O. P.-Cambridge, 1875)			E						NR	V
Walckenaeria nudipalpis (Westring, 1851)			E	S	W	NI	RI			
Walckenaeria obtusa Blackwall, 1836			E	S	W				NS	
Walckenaeria monoceros (Wider, 1834)			E	S	W		RI		NS	
Walckenaeria corniculans (O. P.-Cambridge, 1875)			E				RI		NR	CE
Walckenaeria furcillata (Menge, 1869)			E	S	W				NS	
Walckenaeria unicornis O. P.-Cambridge, 1861			E	S	W	NI	RI			
Walckenaeria kochi (O. P.-Cambridge, 1872)			E	S	W	NI	RI		NS	
Walckenaeria clavicornis (Emerton, 1882)			E	S	W	NI	RI		NS	
Walckenaeria cuspidata Blackwall, 1833			E	S	W	NI	RI			
Walckenaeria vigilax (Blackwall, 1853)			E	S	W	NI	RI			
Dicymbium nigrum (Blackwall, 1834)			E	S	W	NI	RI			
Dicymbium nigrum brevisetosum Locket, 1962	*Dicymbium brevisetosum*	*D. nigrum* f. *brevisetosum*	E	S	W	NI	RI			
Dicymbium tibiale (Blackwall, 1836)			E	S	W	NI	RI			
Entelecara acuminata (Wider, 1834)			E	S	W	NI	RI			
Entelecara congenera (O. P.-Cambridge, 1879)			E		W				NS	
Entelecara erythropus (Westring, 1851)			E	S	W	NI	RI			
Entelecara flavipes (Blackwall, 1834)			E	S	W		RI		NS	
Entelecara omissa O. P.-Cambridge, 1902			E				RI		NS	
Entelecara errata O. P.-Cambridge, 1913			E	S	W		RI		NS	
Moebelia penicillata (Westring, 1851)			E	S	W	NI	RI		NS	
Hylyphantes graminicola (Sundevall, 1830)			E	S	W		RI			
Gnathonarium dentatum (Wider, 1834)			E	S	W	NI	RI			
Trematocephalus cristatus (Wider, 1834)			E						NS	
Tmeticus affinis (Blackwall, 1855)			E	S	W		RI		NS	
Gongylidium rufipes (Linnaeus, 1758)			E	S	W	NI	RI			
Dismodicus bifrons (Blackwall, 1841)			E	S	W	NI	RI			
Dismodicus elevatus (C. L. Koch, 1838)			E	S					NR	V
Hypomma bituberculatum (Wider, 1834)			E	S	W	NI	RI			

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Hypomma fulvum (Bösenberg, 1902)			E		W	NI	RI		NS	
Hypomma cornutum (Blackwall, 1833)			E	S	W	NI	RI			
Metopobactrus prominulus (O. P.-Cambridge, 1872)			E	S	W	NI	RI			
Hybocoptus corrugis (O. P.-Cambridge, 1875)	*Hybocoptus decollatus*	*Hybocoptus decollatus*	E		W				NS	
Baryphyma pratense (Blackwall, 1861)			E	S	W					
Baryphyma gowerense (Locket, 1965)			E		W	NI	RI		NR	V
Baryphyma trifrons (O. P.-Cambridge, 1863)			E	S	W	NI	RI			
Baryphyma maritimum (Crocker & Parker, 1970)			E						NR	NT
Praestigia duffeyi Millidge, 1954		*Baryphyma duffeyi*	E				RI		NR	E
Gonatium rubens (Blackwall, 1833)			E	S	W	NI	RI			
Gonatium rubellum (Blackwall, 1841)			E	S	W	NI	RI			
Gonatium paradoxum (L. Koch, 1869)			E						NR	E
Maso sundevalli (Westring, 1851)			E	S	W	NI	RI			
Maso gallicus Simon, 1894			E		W				NS	
Minicia marginella (Wider, 1834)		Appendix 2	E				RI		NR	DD
Peponocranium ludicrum (O. P.-Cambridge, 1861)			E	S	W	NI	RI			
Pocadicnemis pumila (Blackwall, 1841)			E	S	W	NI	RI			
Pocadicnemis juncea Locket & Millidge, 1953			E	S	W	NI	RI			
Hypselistes jacksoni (O. P.-Cambridge, 1902)			E	S	W	NI	RI		NS	
Oedothorax gibbosus (Blackwall, 1841)			E	S	W	NI	RI			
Oedothorax fuscus (Blackwall, 1834)			E	S	W	NI	RI			
Oedothorax agrestis (Blackwall, 1853)			E	S	W	NI	RI			
Oedothorax retusus (Westring, 1851)			E	S	W	NI	RI			
Oedothorax apicatus (Blackwall, 1850)			E	S	W		RI			
Trichopternoides thorelli (Westring, 1861)		*Trichopterna thorelli*	E	S	W	NI	RI			
Trichopterna cito (O. P.-Cambridge, 1872)			E						NR	E
Pelecopsis mengei (Simon, 1884)			E	S	W	NI	RI			
Pelecopsis parallela (Wider, 1834)			E	S	W	NI	RI			
Pelecopsis elongata (Wider, 1834)				S					NR	NT
Pelecopsis radicicola (L. Koch, 1872)			E						NR	E
Pelecopsis susannae (Simon, 1914)	not present	not present	E							
Parapelecopsis nemoralis (Blackwall, 1841)	*Pelecopsis nemoralis*	*Pelecopsis nemoralis*	E	S	W	NI	RI			
Parapelecopsis nemoralioides (O. P.-Cambridge, 1884)	*Pelecopsis nemoralioides*	not present	E	S	W		RI		NS	
Silometopus elegans (O. P.-Cambridge, 1872)			E	S	W	NI	RI			
Silometopus ambiguus (O. P.-Cambridge, 1905)			E	S	W	NI	RI		NS	
Silometopus reussi (Thorell, 1871)			E	S	W	NI	RI			
Silometopus incurvatus (O. P.-Cambridge, 1873)			E	S			RI		NR	V
Mecopisthes peusi Wunderlich, 1972			E	S	W	NI	RI		NS	
Cnephalocotes obscurus (Blackwall, 1834)			E	S	W	NI	RI			
Acartauchenius scurrilis (O. P.-Cambridge, 1872)			E						NR	NT
Trichoncus saxicola (O. P.-Cambridge, 1861)			E	S			RI		NR	V
Trichoncus hackmani Millidge, 1955			E						NR	V
Trichoncus affinis Kulczyński, 1894			E						NR	
Styloctetor romanus (O. P.-Cambridge, 1873)	*Ceratinopsis romana*	*Ceratinopsis romana*	E		W				NR	
Styloctetor compar (Westring, 1861)	*Ceratinopsis stativa*	*Ceratinopsis stativa*	E		W		RI		NS	
Evansia merens O. P.-Cambridge, 1900			E	S	W		RI		NS	
Tiso vagans (Blackwall, 1834)			E	S	W	NI	RI			
Tiso aestivus (L. Koch, 1872)			E	S	W				NS	
Troxochrus scabriculus (Westring, 1851)			E	S	W		RI			
Minyriolus pusillus (Wider, 1834)			E	S	W	NI	RI			
Tapinocyba praecox (O. P.-Cambridge, 1873)			E	S	W	NI	RI			
Tapinocyba pallens (O. P.-Cambridge, 1872)			E	S	W	NI	RI			
Tapinocyba insecta (L. Koch, 1869)			E	S	W	NI	RI		NS	
Tapinocyba mitis (O. P.-Cambridge, 1882)			E						NR	E
Tapinocyboides pygmaeus (Menge, 1869)			E	S					NR	DD
Microctenonyx subitaneus (O. P.-Cambridge, 1875)			E	S	W	NI	RI		NS	
Satilatlas britteni (Jackson, 1913)			E	S	W	NI	RI		NS	
Thyreosthenius parasiticus (Westring, 1851)			E	S	W	NI	RI			
Thyreosthenius biovatus (O. P.-Cambridge, 1875)			E	S	W		RI		NS	
Monocephalus fuscipes (Blackwall, 1836)			E	S	W	NI	RI			
Monocephalus castaneipes (Simon, 1884)			E	S	W	NI	RI		NS	
Lophomma punctatum (Blackwall, 1841)			E	S	W	NI	RI			
Saloca diceros (O. P.-Cambridge, 1871)			E		W		RI		NS	
Gongylidiellum vivum (O. P.-Cambridge, 1875)			E	S	W	NI	RI			
Gongylidiellum latebricola (O. P.-Cambridge, 1871)			E	S	W	NI	RI		NS	
Gongylidiellum murcidum Simon, 1884			E	S	W		RI		NS	V
Micrargus herbigradus (Blackwall, 1854)			E	S	W	NI	RI			
Micrargus apertus (O. P.-Cambridge, 1871)			E	S	W					
Micrargus subaequalis (Westring, 1851)			E	S	W	NI	RI			
Micrargus laudatus (O. P.-Cambridge, 1881)			E		W				NS	
Notioscopus sarcinatus (O. P.-Cambridge, 1872)			E	S	W				NS	
Glyphesis cottonae (La Touche, 1945)			E		W		RI		NR	V
Glyphesis servulus (Simon, 1881)			E		W				NR	NT

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Erigonella hiemalis (Blackwall, 1841)			E	S	W	NI	RI			
Erigonella ignobilis (O. P.-Cambridge, 1871)			E	S	W	NI	RI		NS	
Savignia frontata Blackwall, 1833			E	S	W	NI	RI			
Diplocephalus cristatus (Blackwall, 1833)			E	S	W	NI	RI			
Diplocephalus permixtus (O. P.-Cambridge, 1871)			E	S	W	NI	RI			
Diplocephalus latifrons (O. P.-Cambridge, 1863)			E	S	W	NI	RI			
Diplocephalus connatus Bertkau, 1889			E						NR	CE
Diplocephalus picinus (Blackwall, 1841)			E	S	W	NI	RI			
Diplocephalus protuberans (O. P.-Cambridge, 1875)			E	S	W				NR	V
Diplocephalus graecus (O. P.-Cambridge, 1872)		not present	E							
Araeoncus humilis (Blackwall, 1841)			E	S	W	NI	RI			
Araeoncus crassiceps (Westring, 1861)			E	S	W	NI	RI		NS	
Panamomops sulcifrons (Wider, 1834)			E		W				NS	
Lessertia dentichelis (Simon, 1884)			E		W	NI	RI		NS	
Scotinotylus evansi (O. P.-Cambridge, 1894)			E	S					NS	
Typhochrestus digitatus (O. P.-Cambridge, 1872)			E	S	W	NI	RI		NS	
Typhochrestus simoni Lessert, 1907			E		W				NR	CE
Diplocentria bidentata (Emerton, 1882)			E	S	W		RI		NS	
Wabasso replicatus (Holm, 1950)		not present		S					NR	V
Erigone dentipalpis (Wider, 1834)			E	S	W	NI	RI			
Erigone atra Blackwall, 1833			E	S	W	NI	RI			
Erigone promiscua (O. P.-Cambridge, 1872)			E	S	W	NI	RI			
Erigone arctica (White, 1852)			E	S	W	NI	RI			
Erigone longipalpis (Sundevall, 1830)			E	S	W	NI	RI			
Erigone tirolensis L. Koch, 1872				S					NS	
Erigone dentigera O. P.-Cambridge, 1874	*Erigone capra*	*Erigone capra*	E	S	W		RI		NR	
Erigone welchi Jackson, 1911			E	S	W		RI		NR	E
Erigone psychrophila Thorell, 1871			E	S					NR	NT
Erigone aletris Crosby & Bishop, 1928			E	S						
Prinerigone vagans (Audouin, 1826)		*Erigone vagans*	E	S	W					
Mermessus trilobatus (Emerton, 1882)		not present	E							
Mecynargus morulus (O. P.-Cambridge, 1873)		*Rhaebothorax morulus*	E	S	W	NI	RI		NS	
Mecynargus paetulus (O. P.-Cambridge, 1875)		*Rhaebothorax paetulus*		S					NR	V
Semljicola faustus (O. P.-Cambridge, 1900)	*Latithorax faustus*	*Latithorax faustus*	E	S	W	NI	RI		NS	
Semljicola caliginosus (Falconer, 1910)			E	S					NR	E
Donacochara speciosa (Thorell, 1875)			E			NI	RI		NS	
Leptorhoptrum robustum (Westring, 1851)			E	S	W	NI	RI			
Drepanotylus uncatus (O. P.-Cambridge, 1873)			E	S	W	NI	RI			
Leptothrix hardyi (Blackwall, 1850)			E	S	W	NI	RI		NS	
Hilaira excisa (O. P.-Cambridge, 1871)			E	S	W	NI	RI			
Hilaira nubigena Hull, 1911			E	S					NR	V
Hilaira pervicax Hull, 1908			E	S	W	NI			NS	
Oreoneta frigida (Thorell, 1872)	*Hilaira frigida*	*Hilaira frigida*	E	S	W	NI	RI			
Halorates reprobus (O. P.-Cambridge, 1879)			E	S	W	NI	RI		NS	
Collinsia inerrans (O. P.-Cambridge, 1885)	*Milleriana inerrans*	*Milleriana inerrans*	E	S	W		RI			
Collinsia distincta (Simon, 1884)	*Halorates distinctus*	*Halorates distinctus*	E	S	W				NS	
Collinsia holmgreni (Thorell, 1871)	*Halorates holmgreni*	*Halorates holmgreni*		S					NS	
Carorita limnaea (Crosby & Bishop, 1927)			E		W	NI	RI		NR	V
Karita paludosa (Duffey, 1971)		*Carorita paludosa*	E			NI	RI		NR	V
Wiehlea calcarifera (Simon, 1884)			E						NR	E
Mioxena blanda (Simon, 1884)			E		W		RI		NR	DD
Caviphantes saxetorum (Hull, 1916)			E	S	W				NR	NT
Asthenargus paganus (Simon, 1884)			E	S	W	NI	RI		NS	
Jacksonella falconeri (Jackson, 1908)			E	S	W	NI	RI		NS	
Pseudomaro aenigmaticus Denis, 1966			E						NR	DD
Ostearius melanopygius (O. P.-Cambridge, 1879)			E	S	W	NI	RI			
Aphileta misera (O. P.-Cambridge, 1882)			E	S	W	NI	RI			
Porrhomma pygmaeum (Blackwall, 1834)			E	S	W	NI	RI			
Porrhomma convexum (Westring, 1851)			E	S	W	NI	RI		NS	
Porrhomma rosenhaueri (L. Koch, 1872)					W		RI		NR	NT
Porrhomma pallidum Jackson, 1913			E	S	W	NI	RI			
Porrhomma campbelli F. O. P.-Cambridge, 1894			E	S	W	NI	RI		NS	
Porrhomma microphthalmum (O. P.-Cambridge, 1871)			E	S	W					
Porrhomma errans (Blackwall, 1841)			E	S	W		RI		NS	
Porrhomma egeria Simon, 1884			E	S	W		RI		NS	
Porrhomma oblitum (O. P.-Cambridge, 1871)			E		W		RI		NS	
Porrhomma cambridgei Merrett, 1994		not present	E						NR	DD
Porrhomma montanum Jackson, 1913			E	S	W	NI	RI		NS	
Agyneta subtilis (O. P.-Cambridge, 1863)			E	S	W	NI	RI			
Agyneta conigera (O. P.-Cambridge, 1863)			E	S	W	NI	RI			
Agyneta decora (O. P.-Cambridge, 1871)			E	S	W	NI	RI			
Agyneta cauta (O. P.-Cambridge, 1902)			E	S	W	NI	RI		NS	
Agyneta olivacea (Emerton, 1882)			E	S	W	NI	RI		NS	

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Agyneta ramosa Jackson, 1912			E	S	W		RI			
Agyneta innotabilis (O. P.-Cambridge, 1863)	*Meioneta innotabilis*	*Meioneta innotabilis*	E	S	W	NI	RI			
Agyneta rurestris (C. L. Koch, 1836)	*Meioneta rurestris*	*Meioneta rurestris*	E	S	W	NI	RI			
Agyneta mollis (O. P.-Cambridge, 1871)	*Meioneta mollis*	*Meioneta mollis*	E		W	NI	RI		NR	NT
Agyneta saxatilis (Blackwall, 1844)	*Meioneta saxatilis*	*Meioneta saxatilis*	E	S	W	NI	RI			
Agyneta mossica (Schikora, 1993)	*Meioneta mossica*	not present	E	S	W	NI	RI		NS	
Agyneta simplicitarsis (Simon, 1884)	*Meioneta simplicitarsis*	*Meioneta simplicitarsis*	E						NS	
Agyneta affinis (Kulczyński, 1898)	*Meioneta beata*	*Meioneta beata*	E	S	W	NI	RI			
Agyneta fuscipalpa (C. L. Koch, 1836)	*Meioneta fuscipalpa*	not present	E						NR	V
Agyneta gulosa (L. Koch, 1869)	*Meioneta gulosa*	*Meioneta gulosa*	E	S	W	NI	RI		NS	
Agyneta nigripes (Simon, 1884)	*Meioneta nigripes*	*Meioneta nigripes*		S	W				NS	
Microneta viaria (Blackwall, 1841)			E	S	W	NI	RI			
Maro minutus O. P.-Cambridge, 1906			E	S	W		RI		NS	
Maro sublestus Falconer, 1915			E	S			RI		NR	E
Maro lepidus Casemir, 1961			E	S	W				NR	E
Syedra gracilis (Menge, 1869)			E	S					NS	
Syedra myrmicarum (Kulczyński, 1882)	not present	not present	E							
Centromerus sylvaticus (Blackwall, 1841)			E	S	W	NI	RI			
Centromerus prudens (O. P.-Cambridge, 1873)			E	S	W	NI	RI			
Centromerus arcanus (O. P.-Cambridge, 1873)			E	S	W	NI	RI			
Centromerus levitarsis (Simon, 1884)			E	S			RI		NR	E
Centromerus dilutus (O. P.-Cambridge, 1875)			E	S	W	NI	RI			
Centromerus capucinus (Simon, 1884)			E						NR	NT
Centromerus incilium (L. Koch, 1881)			E	S					NS	
Centromerus semiater (L. Koch, 1879)			E						NR	E
Centromerus brevipalpus (Menge, 1866)	*Centromerus brevivulvatus*	*Centromerus aequalis*	E	S					NR	E
Centromerus serratus (O. P.-Cambridge, 1875)			E						NR	E
Centromerus albidus Simon, 1929			E				RI		NR	CE
Centromerus cavernarum (L. Koch, 1872)			E						NR	NT
Centromerus persimilis (O. P.-Cambridge, 1912)			E				RI		NR	DD
Centromerus minutissimus Merrett & Powell, 1993		not present	E						NR	DD
Tallusia experta (O. P.-Cambridge, 1871)			E	S	W	NI	RI			
Centromerita bicolor (Blackwall, 1833)			E	S	W	NI	RI			
Centromerita concinna (Thorell, 1875)			E	S	W	NI	RI			
Sintula corniger (Blackwall, 1856)			E	S	W	NI	RI		NS	
Oreonetides vaginatus (Thorell, 1872)			E	S	W		RI		NS	
Saaristoa abnormis (Blackwall, 1841)			E	S	W	NI	RI			
Saaristoa firma (O. P.-Cambridge, 1905)			E	S	W	NI	RI		NS	
Macrargus rufus (Wider, 1834)			E	S	W	NI	RI			
Macrargus carpenteri (O. P.-Cambridge, 1894)			E	S					NS	
Bathyphantes approximatus (O. P.-Cambridge, 1871)			E	S	W	NI	RI			
Bathyphantes gracilis (Blackwall, 1841)			E	S	W	NI	RI			
Bathyphantes parvulus (Westring, 1851)			E	S	W	NI	RI			
Bathyphantes nigrinus (Westring, 1851)			E	S	W	NI	RI			
Bathyphantes setiger F. O. P.-Cambridge, 1894			E	S	W	NI	RI		NS	
Kaestneria dorsalis (Wider, 1834)			E	S	W	NI	RI			
Kaestneria pullata (O. P.-Cambridge, 1863)			E	S	W	NI	RI			
Diplostyla concolor (Wider, 1834)			E	S	W	NI	RI			
Poeciloneta variegata (Blackwall, 1841)		*Poeciloneta globosa*	E	S	W	NI	RI			
Drapetisca socialis (Sundevall, 1833)			E	S	W	NI	RI			
Tapinopa longidens (Wider, 1834)			E	S	W	NI	RI			
Floronia bucculenta (Clerck, 1757)			E	S	W	NI	RI			
Taranucnus setosus (O. P.-Cambridge, 1863)			E	S	W	NI	RI		NS	
Labulla thoracica (Wider, 1834)			E	S	W	NI	RI			
Stemonyphantes lineatus (Linnaeus, 1758)			E	S	W	NI	RI			
Bolyphantes luteolus (Blackwall, 1833)			E	S	W	NI	RI			
Bolyphantes alticeps (Sundevall, 1833)			E	S	W		RI			
Nothophantes horridus Merrett & Stevens, 1995		not present	E						NR	E
Megalepthyphantes nebulosus (Sundevall, 1830)		*Lepthyphantes nebulosus*	E	S	W	NI	RI			
Lepthyphantes leprosus (Ohlert, 1865)			E	S	W	NI	RI			
Lepthyphantes minutus (Blackwall, 1833)			E	S	W	NI	RI			
Mughiphantes whymperi (F. O. P.-Cambridge, 1894)		*Lepthyphantes whymperi*	E	S	W		RI		NS	
Obscuriphantes obscurus (Blackwall, 1841)		*Lepthyphantes obscurus*	E	S	W	NI	RI			
Tenuiphantes alacris (Blackwall, 1853)		*Lepthyphantes alacris*	E	S	W	NI	RI			
Tenuiphantes tenuis (Blackwall, 1852)		*Lepthyphantes tenuis*	E	S	W	NI	RI			
Tenuiphantes zimmermanni (Bertkau, 1890)		*Lepthyphantes zimmermanni*	E	S	W	NI	RI			
Tenuiphantes cristatus (Menge, 1866)		*Lepthyphantes cristatus*	E	S	W	NI	RI			
Tenuiphantes mengei (Kulczyński, 1887)		*Lepthyphantes mengei*	E	S	W	NI	RI			
Tenuiphantes flavipes (Blackwall, 1854)		*Lepthyphantes flavipes*	E	S	W	NI	RI			
Tenuiphantes tenebricola (Wider, 1834)		*Lepthyphantes tenebricola*	E	S	W	NI	RI			
Palliduphantes ericaeus (Blackwall, 1853)		*Lepthyphantes ericaeus*	E	S	W	NI	RI			
Palliduphantes pallidus (O. P.-Cambridge, 1871)		*Lepthyphantes pallidus*	E	S	W	NI	RI			
Palliduphantes insignis (O. P.-Cambridge, 1913)		*Lepthyphantes insignis*	E	S	W		RI		NS	

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Palliduphantes antroniensis (Schenkel, 1933)		*Lepthyphantes antroniensis*		S					NR	CE
Piniphantes pinicola (Simon, 1884)		*Lepthyphantes pinicola*	E	S	W				NR	
Oryphantes angulatus (O. P.-Cambridge, 1881)		*Lepthyphantes angulatus*	E	S	W	NI	RI		NS	
Improphantes complicatus (Emerton, 1882)		*Lepthyphantes complicatus*		S					NR	NT
Agnyphantes expunctus (O. P.-Cambridge, 1875)		*Lepthyphantes expunctus*	E	S					NS	
Midia midas (Simon, 1884)		*Lepthyphantes midas*	E						NR	E
Helophora insignis (Blackwall, 1841)			E	S	W	NI	RI			
Pityohyphantes phrygianus (C. L. Koch, 1836)			E	S					NS	
Linyphia triangularis (Clerck, 1757)			E	S	W	NI	RI			
Linyphia hortensis Sundevall, 1830			E	S	W	NI	RI			
Neriene montana (Clerck, 1757)			E	S	W	NI	RI			
Neriene clathrata (Sundevall, 1830)			E	S	W	NI	RI			
Neriene peltata (Wider, 1834)			E	S	W	NI	RI			
Neriene furtiva (O. P.-Cambridge, 1871)			E		W				NS	
Neriene radiata (Walckenaer, 1841)			E	S					NR	NT
Microlinyphia pusilla (Sundevall, 1830)			E	S	W	NI	RI			
Microlinyphia impigra (O. P.-Cambridge, 1871)			E	S	W	NI	RI			
Allomengea scopigera (Grube, 1859)			E	S	W	NI	RI			
Allomengea vidua (L. Koch, 1879)			E	S	W	NI	RI		NS	
Family TETRAGNATHIDAE										
Tetragnatha extensa (Linnaeus, 1758)			E	S	W	NI	RI			
Tetragnatha pinicola L. Koch, 1870			E	S	W		RI			
Tetragnatha montana Simon, 1874			E	S	W	NI	RI			
Tetragnatha obtusa C. L. Koch, 1837			E	S	W	NI	RI			
Tetragnatha nigrita Lendl, 1886			E		W	NI	RI			
Tetragnatha striata L. Koch, 1862			E	S	W		RI			
Pachygnatha clercki Sundevall, 1823			E	S	W	NI	RI			
Pachygnatha listeri Sundevall, 1830			E	S	W		RI			
Pachygnatha degeeri Sundevall, 1830			E	S	W	NI	RI			
Metellina segmentata (Clerck, 1757)		*Meta segmentata*	E	S	W	NI	RI			
Metellina mengei (Blackwall, 1869)		*Meta mengei*	E	S	W	NI	RI			
Metellina merianae (Scopoli, 1763)		*Meta merianae*	E	S	W	NI	RI			
Meta menardi (Latreille, 1804)			E	S	W	NI	RI			
Meta bourneti Simon, 1922			E		W				NS	
Family ARANEIDAE										
Gibbaranea gibbosa (Walckenaer, 1802)			E	S	W	NI	RI			
Araneus angulatus Clerck, 1757			E						NS	
Araneus diadematus Clerck, 1757			E	S	W	NI	RI			
Araneus quadratus Clerck, 1757			E	S	W	NI	RI			
Araneus marmoreus Clerck, 1757			E	S	W					
Araneus alsine (Walckenaer, 1802)			E	S					NS	
Araneus sturmi (Hahn, 1831)			E	S	W		RI			
Araneus triguttatus (Fabricius, 1775)			E	S	W	NI				
Larinioides cornutus (Clerck, 1757)			E	S	W	NI	RI			
Larinioides sclopetarius (Clerck, 1757)			E	S	W	NI	RI			
Larinioides patagiatus (Clerck, 1757)			E	S	W	NI	RI		NS	
Nuctenea umbratica (Clerck, 1757)			E	S	W	NI	RI			
Agalenatea redii (Scopoli, 1763)			E	S	W	NI	RI			
Neoscona adianta (Walckenaer, 1802)			E		W		RI			
Araniella cucurbitina (Clerck, 1757)			E	S	W	NI	RI			
Araniella opisthographa (Kulczyński, 1905)			E	S	W	NI	RI			
Araniella inconspicua (Simon, 1874)			E						NS	
Araniella alpica (L. Koch, 1869)			E		W				NR	E
Araniella displicata (Hentz, 1847)			E						NR	NT
Zilla diodia (Walckenaer, 1802)			E		W					
Hypsosinga albovittata (Westring, 1851)			E	S	W		RI		NS	
Hypsosinga pygmaea (Sundevall, 1831)			E	S	W	NI	RI			
Hypsosinga sanguinea (C. L. Koch, 1844)			E						NS	
Hypsosinga heri (Hahn, 1831)			E						NR	V
Singa hamata (Clerck, 1757)			E	S	W				NS	
Cercidia prominens (Westring, 1851)			E	S	W				NS	
Zygiella x-notata (Clerck, 1757)			E	S	W	NI	RI			
Zygiella atrica (C. L. Koch, 1845)			E	S	W	NI	RI			
Leviellus stroemi (Thorell, 1870)	*Stroemiellus stroemi*	*Zygiella stroemi*	E						NR	NT
Mangora acalypha (Walckenaer, 1802)			E		W		RI			
Cyclosa conica (Pallas, 1772)			E	S	W	NI	RI			
Argiope bruennichi (Scopoli, 1772)			E		W					
Family LYCOSIDAE										
Pardosa agricola (Thorell, 1856)			E	S	W	NI	RI			
Pardosa agrestis (Westring, 1861)			E	S	W	NI	RI		NS	
Pardosa purbeckensis F. O. P.-Cambridge, 1895		*Pardosa agrestis*	E	S	W		RI			
Pardosa monticola (Clerck, 1757)			E	S	W	NI	RI			

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Pardosa palustris (Linnaeus, 1758)			E	S	W	NI	RI			
Pardosa pullata (Clerck, 1757)			E	S	W	NI	RI			
Pardosa prativaga (L. Koch, 1870)			E	S	W	NI	RI			
Pardosa amentata (Clerck, 1757)			E	S	W	NI	RI			
Pardosa nigriceps (Thorell, 1856)			E	S	W	NI	RI			
Pardosa lugubris (Walckenaer, 1802)			E	S			RI		NS	
Pardosa saltans Töpfer-Hofmann, 2000		not present	E	S	W		RI			
Pardosa hortensis (Thorell, 1872)			E	S	W					
Pardosa tenuipes C. L. Koch, 1847	*Pardosa proxima*	*Pardosa proxima*	E	S	W				NS	
Pardosa trailli (O. P.-Cambridge, 1873)			E	S	W				NR	V
Pardosa paludicola (Clerck, 1757)			E						NR	E
Hygrolycosa rubrofasciata (Ohlert, 1865)			E						NR	E
Xerolycosa nemoralis (Westring, 1861)			E						NS	
Xerolycosa miniata (C. L. Koch, 1834)			E	S	W				NS	
Alopecosa pulverulenta (Clerck, 1757)			E	S	W	NI	RI			
Alopecosa cuneata (Clerck, 1757)			E		W	NI	RI		NS	
Alopecosa barbipes (Sundevall, 1833)		*Alopecosa accentuata*	E	S	W	NI	RI			
Alopecosa fabrilis (Clerck, 1757)			E						NR	CE
Trochosa ruricola (De Geer, 1778)			E	S	W	NI	RI			
Trochosa robusta (Simon, 1876)			E						NR	V
Trochosa terricola Thorell, 1856			E	S	W	NI	RI			
Trochosa spinipalpis (F. O. P.-Cambridge, 1895)			E	S	W	NI	RI		NS	
Arctosa fulvolineata (Lucas, 1846)			E						NR	NT
Arctosa perita (Latreille, 1799)			E	S	W	NI	RI			
Arctosa leopardus (Sundevall, 1833)			E	S	W	NI	RI			
Arctosa cinerea (Fabricius, 1777)			E	S	W		RI		NS	
Arctosa alpigena (Doleschall, 1852)		*Tricca alpigena*		S					NR	V
Pirata piraticus (Clerck, 1757)			E	S	W	NI	RI			
Pirata tenuitarsis Simon, 1876			E	S	W	NI	RI		NS	
Pirata piscatorius (Clerck, 1757)			E	S	W	NI	RI		NS	
Piratula hygrophila (Thorell, 1872)	*Pirata hygrophilus*	*Pirata hygrophilus*	E	S	W	NI	RI			
Piratula uliginosa (Thorell, 1856)	*Pirata uliginosus*	*Pirata uliginosus*	E	S	W	NI	RI			
Piratula latitans (Blackwall, 1841)	*Pirata latitans*	*Pirata latitans*	E	S	W		RI			
Aulonia albimana (Walckenaer, 1805)			E						NR	CE
Family PISAURIDAE										
Pisaura mirabilis (Clerck, 1757)			E	S	W	NI	RI			
Dolomedes fimbriatus (Clerck, 1757)			E	S	W		RI		NS	
Dolomedes plantarius (Clerck, 1757)			E		W				NR	V
Family OXYOPIDAE										
Oxyopes heterophthalmus (Latreille, 1804)			E						NR	V
Family AGELENIDAE										
Agelena labyrinthica (Clerck, 1757)			E		W		RI			
Textrix denticulata (Olivier, 1789)			E	S	W	NI	RI			
Eratigena agrestis (Walckenaer, 1802)	*Tegenaria agrestis*	*Tegenaria agrestis*	E	S	W		RI			
Eratigena atrica (C. L. Koch, 1843)	*Tegenaria atrica*	*Tegenaria atrica*	E	S	W		RI			
Eratigena duellica (Simon, 1875)	*Tegenaria gigantea*	*Tegenaria duellica*	E	S	W	NI	RI			
Eratigena picta (Simon, 1870)	*Tegenaria picta*	Appendix 1 as *T. picta*	E						NR	V
Eratigena saeva (Blackwall, 1844)	*Tegenaria saeva*	*Tegenaria saeva*	E	S	W	NI	RI			
Tegenaria domestica (Clerck, 1757)			E	S	W	NI	RI			
Tegenaria ferruginea (Panzer, 1804)		not present	E							
Tegenaria parietina (Fourcroy, 1785)			E				RI			
Tegenaria hasperi Chyzer, 1897	not present	not present	E					Ab		
Tegenaria silvestris C. L. Koch, 1872			E	S	W	NI	RI			
Coelotes atropos (Walckenaer, 1830)	from Amaurobiidae	from Agelenidae	E	S	W					
Coelotes terrestris (Wider, 1834)	from Amaurobiidae	from Agelenidae	E	S	W				NS	
Family CYBAEIDAE										
Cryphoeca silvicola (C. L. Koch, 1834)	from Dictynidae	from Agelenidae	E	S	W	NI	RI			
Tuberta maerens (O. P.-Cambridge, 1863)	from Dictynidae	from Agelenidae	E						NR	E
Family HAHNIIDAE										
Antistea elegans (Blackwall, 1841)			E	S	W	NI	RI			
Iberina montana (Blackwall, 1841)	*Hahnia montana*	*Hahnia montana*	E	S	W	NI	RI			
Iberina candida (Simon, 1875)	*Hahnia candida*	*Hahnia candida*	E						NR	V
Iberina microphthalma (Snazell & Duffey, 1980)	*Hahnia microphthalma*	*Hahnia microphthalma*	E						NR	DD
Hahnia nava (Blackwall, 1841)			E	S	W	NI	RI			
Hahnia helveola Simon, 1875			E	S	W	NI	RI			
Hahnia pusilla C. L. Koch, 1841			E	S	W		RI		NS	
Cicurina cicur (Fabricius, 1793)	from Dictynidae	from Agelenidae	E	S	W				NS	
Mastigusa arietina (Thorell, 1871)	from Dictynidae	*Tetrilus arietina*	E						E	RE
Mastigusa macrophthalma (Kulczyński, 1897)	from Dictynidae	*Tetrilus macrophthalma*	E						NR	V

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Family DICTYNIDAE										
Dictyna arundinacea (Linnaeus, 1758)			E	S	W	NI	RI			
Dictyna pusilla Thorell, 1856			E	S	W				NS	
Dictyna major Menge, 1869				S					NR	CE
Dictyna uncinata Thorell, 1856			E	S	W	NI	RI			
Brigittea latens (Fabricius, 1775)	*Dictyna latens*	*Dictyna latens*	E	S	W	NI	RI			
Nigma puella (Simon, 1870)			E		W		RI		NS	
Nigma flavescens (Walckenaer, 1830)	not present	*Nigma flavescens*	E							
Nigma walckenaeri (Roewer, 1951)			E							
Lathys humilis (Blackwall, 1855)			E	S	W					
Lathys heterophthalma Kulczyński, 1891	*Lathys nielseni*	Appendix 1	E						NR	V
Lathys stigmatisata (Menge, 1869)			E		W				NR	V
Argenna subnigra (O. P.-Cambridge, 1861)			E		W		RI		NS	
Argenna patula (Simon, 1874)			E	S	W				NS	
Altella lucida (Simon, 1874)			E						NR	CE
Argyroneta aquatica (Clerck, 1757)	from Cybaeidae	from Argyronetidae	E	S	W	NI	RI			
Family AMAUROBIIDAE										
Amaurobius fenestralis (Stroem, 1768)			E	S	W	NI	RI			
Amaurobius similis (Blackwall, 1861)			E	S	W	NI	RI			
Amaurobius ferox (Walckenaer, 1830)			E	S	W	NI	RI			
Family ANYPHAENIDAE										
Anyphaena accentuata (Walckenaer, 1802)			E	S	W	NI	RI			
Anyphaena numida Simon, 1897	not present	not present	E							
Anyphaena sabina L. Koch, 1866	not present	not present	E							
Family LIOCRANIDAE										
Agroeca brunnea (Blackwall, 1833)			E	S	W					
Agroeca proxima (O. P.-Cambridge, 1871)			E	S	W	NI	RI			
Agroeca inopina O. P.-Cambridge, 1886			E		W					
Agroeca lusatica (L. Koch, 1875)			E						NR	E
Agroeca dentigera Kulczyński, 1913		not present			W				NR	DD
Agroeca cuprea Menge, 1873			E	S					NR	NT
Liocranoeca striata (Kulczyński, 1882)	*Agraecina striata*	*Agroeca striata*	E	S	W	NI	RI		NS	
Apostenus fuscus Westring, 1851		Appendix 1	E						NR	V
Scotina celans (Blackwall, 1841)			E	S	W		RI		NS	
Scotina gracilipes (Blackwall, 1859)			E	S	W	NI	RI		NS	
Scotina palliardii (L. Koch, 1881)			E						NR	E
Liocranum rupicola (Walckenaer, 1830)			E		W		RI		NS	
Family PHRUROLITHIDAE	Corinnidae	Clubionidae								
Phrurolithus festivus (C. L. Koch, 1835)			E	S	W	NI	RI			
Phrurolithus minimus C. L. Koch, 1839			E						NS	
Family CLUBIONIDAE										
Clubiona corticalis (Walckenaer, 1802)			E		W					
Clubiona reclusa O. P.-Cambridge, 1863			E	S	W	NI	RI			
Clubiona subsultans Thorell, 1875				S					NR	NT
Clubiona stagnatilis Kulczyński, 1897			E	S	W	NI	RI			
Clubiona rosserae Locket, 1953			E						NR	V
Clubiona norvegica Strand, 1900			E	S	W				NS	
Clubiona caerulescens L. Koch, 1867			E	S	W				NR	V
Clubiona pallidula (Clerck, 1757)			E	S	W	NI	RI			
Clubiona phragmitis C. L. Koch, 1843			E	S	W	NI	RI			
Clubiona terrestris Westring, 1851			E	S	W	NI	RI			
Clubiona frutetorum L. Koch, 1866		not present					RI			
Clubiona neglecta O. P.-Cambridge, 1862			E	S	W	NI	RI			
Clubiona pseudoneglecta Wunderlich, 1994		not present	E						NR	V
Clubiona frisia Wunderlich & Schütt, 1995		*Clubiona similis*	E						NR	NT
Clubiona lutescens Westring, 1851			E	S	W	NI	RI			
Clubiona comta C. L. Koch, 1839			E	S	W	NI	RI			
Clubiona brevipes Blackwall, 1841			E	S	W	NI	RI			
Clubiona trivialis C. L. Koch, 1843			E	S	W	NI	RI			
Clubiona juvenis Simon, 1878			E				RI		NR	NT
Clubiona diversa O. P.-Cambridge, 1862			E	S	W	NI	RI			
Clubiona subtilis L. Koch, 1867			E	S	W		RI			
Porrhoclubiona genevensis (L. Koch, 1866)	*Clubiona genevensis*	*Clubiona genevensis*	E		W				NR	NT
Porrhoclubiona leucaspis (Simon, 1932)	*Clubiona leucaspis*	not present	E							
Family CHEIRACANTHIIDAE	Clubionidae	Clubionidae								
Cheiracanthium erraticum (Walckenaer, 1802)			E	S	W	NI	RI			
Cheiracanthium pennyi O. P.-Cambridge, 1873			E						NR	E
Cheiracanthium virescens (Sundevall, 1833)			E	S	W	NI	RI		NS	

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Family ZODARIIDAE										
Zodarion italicum (Canestrini, 1868)		Appendix 1	E						NS	
Zodarion vicinum Denis, 1935		not present	E						NR	V
Zodarion rubidum Simon, 1914		not present	E						NR	
Zodarion fuscum (Simon, 1870)		not present	E		W				NR	V
Family GNAPHOSIDAE										
Drassodes lapidosus (Walckenaer, 1802)			E	S	W	NI	RI			
Drassodes cupreus (Blackwall, 1834)			E	S	W	NI	RI			
Drassodes pubescens (Thorell, 1856)			E	S	W		RI		NS	
Haplodrassus signifer (C. L. Koch, 1839)			E	S	W	NI	RI			
Haplodrassus dalmatensis (L. Koch, 1866)			E		W				NS	
Haplodrassus umbratilis (L. Koch, 1866)			E						NR	DD
Haplodrassus soerenseni (Strand, 1900)				S					NR	E
Haplodrassus silvestris (Blackwall, 1833)			E	S	W				NS	
Haplodrassus minor (O. P.-Cambridge, 1879)			E		W				NS	
Scotophaeus blackwalli (Thorell, 1871)			E	S	W	NI	RI			
Scotophaeus scutulatus (L. Koch, 1866)		Appendix 2	E					Ah		
Phaeocedus braccatus (L. Koch, 1866)			E						NR	V
Zelotes electus (C. L. Koch, 1839)			E	S	W	NI	RI		NS	
Zelotes latreillei (Simon, 1878)			E	S	W	NI	RI			
Zelotes apricorum (L. Koch, 1876)			E	S	W		RI			
Zelotes subterraneus (C. L. Koch, 1833)		Appendix 1	E	S	W				NS	
Zelotes longipes (L. Koch, 1866)		*Zelotes serotinus*	E						NR	V
Zelotes petrensis (C. L. Koch, 1839)			E						NR	
Trachyzelotes pedestris (C. L. Koch, 1837)		*Zelotes pedestris*	E		W					
Urozelotes rusticus (L. Koch, 1872)		*Zelotes rusticus*	E		W					
Drassyllus lutetianus (L. Koch, 1866)		*Zelotes lutetianus*	E		W		RI		NS	
Drassyllus pusillus (C. L. Koch, 1833)		*Zelotes pusillus*	E	S	W	NI	RI			
Drassyllus praeficus (L. Koch, 1866)		*Zelotes praeficus*	E		W				NS	
Gnaphosa lugubris (C. L. Koch, 1839)			E						NR	V
Gnaphosa occidentalis Simon, 1878			E						NR	NT
Gnaphosa nigerrima L. Koch, 1877		not present	E						NR	V
Gnaphosa leporina (L. Koch, 1866)			E	S	W				NS	
Callilepis nocturna (Linnaeus, 1758)			E		W				NR	V
Micaria pulicaria (Sundevall, 1831)			E	S	W	NI	RI			
Micaria albovittata (Lucas, 1846)		*Micaria romana*	E						NR	V
Micaria alpina L. Koch, 1872				S	W				NR	V
Micaria subopaca Westring, 1861			E	S					NS	
Micaria silesiaca L. Koch, 1875			E						NR	NT
Family MITURGIDAE	Zoridae	Zoridae								
Zora spinimana (Sundevall, 1833)			E	S	W	NI	RI			
Zora armillata Simon, 1878			E						NR	CE
Zora nemoralis (Blackwall, 1861)			E	S	W				NR	V
Zora silvestris Kulczyński, 1897			E						NR	CE
Family ZOROPSIDAE										
Zoropsis spinimana (Dufour, 1820)	not present	not present	E							
Family SPARASSIDAE		Eusparassidae								
Micrommata virescens (Clerck, 1757)			E		W		RI		NS	
Family PHILODROMIDAE		Thomisidae								
Philodromus dispar Walckenaer, 1826			E	S	W		RI			
Philodromus aureolus (Clerck, 1757)			E	S	W	NI	RI			
Philodromus praedatus O. P.-Cambridge, 1871			E	S	W		RI			
Philodromus cespitum (Walckenaer, 1802)			E	S	W	NI	RI			
Philodromus longipalpis Simon, 1870		Appendix 2	E						NS	
Philodromus collinus C. L. Koch, 1835			E							
Philodromus buxi Simon, 1884			E							
Philodromus emarginatus (Schrank, 1803)			E	S		NI			NR	V
Philodromus albidus Kulczyński, 1911			E		W		RI		NS	
Philodromus margaritatus (Clerck, 1757)			E	S					NR	NT
Philodromus rufus Walckenaer, 1826			E							
Rhysodromus fallax (Sundevall, 1833)	*Philodromus fallax*	*Philodromus fallax*	E		W				NR	V
Rhysodromus histrio (Latreille, 1819)	*Philodromus histrio*	*Philodromus histrio*	E	S	W				NS	
Thanatus striatus C. L. Koch, 1845			E		W		RI			
Thanatus formicinus (Clerck, 1757)			E						NR	CE
Thanatus vulgaris Simon, 1870	not present	not present	E	S	W			Ab		
Tibellus maritimus (Menge, 1875)			E	S	W	NI	RI			
Tibellus oblongus (Walckenaer, 1802)			E	S	W	NI	RI			
Family THOMISIDAE										
Thomisus onustus Walckenaer, 1806			E						NS	
Diaea dorsata (Fabricius, 1777)			E		W		RI			

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Misumena vatia (Clerck, 1757)			E		W		RI			
Pistius truncatus (Pallas, 1772)			E						NR	CE
Xysticus cristatus (Clerck, 1757)			E	S	W	NI	RI			
Xysticus audax (Schrank, 1803)			E	S	W		RI			
Xysticus kochi Thorell, 1872			E	S	W					
Xysticus erraticus (Blackwall, 1834)			E	S	W	NI	RI			
Xysticus lanio C. L. Koch, 1835			E		W		RI			
Xysticus ulmi (Hahn, 1831)			E	S	W		RI			
Xysticus bifasciatus C. L. Koch, 1837			E	S	W				NS	
Xysticus luctator L. Koch, 1870			E						NR	E
Xysticus sabulosus (Hahn, 1832)			E	S	W	NI	RI		NS	
Xysticus luctuosus (Blackwall, 1836)			E	S	W		RI		NR	E
Xysticus acerbus Thorell, 1872			E		W				NR	
Bassaniodes robustus (Hahn, 1832)	*Xysticus robustus*	*Xysticus robustus*	E						NR	E
Cozyptila blackwalli (Simon, 1875)	*Ozyptila blackwalli*	*Ozyptila blackwalli*	E						NR	E
Ozyptila scabricula (Westring, 1851)			E		W				NS	
Ozyptila claveata (Walckenaer, 1837)	*Ozyptila nigrita*	*Ozyptila nigrita*	E						NS	
Ozyptila pullata (Thorell, 1875)		not present	E						NR	V
Ozyptila sanctuaria (O. P.-Cambridge, 1871)			E		W		RI			
Ozyptila praticola (C. L. Koch, 1837)			E		W		RI			
Ozyptila trux (Blackwall, 1846)			E	S	W	NI	RI			
Ozyptila simplex (O. P.-Cambridge, 1862)			E		W					
Ozyptila atomaria (Panzer, 1801)			E	S	W	NI	RI			
Ozyptila brevipes (Hahn, 1826)			E		W		RI			

Family SALTICIDAE

Species	2014 Checklist	Roberts	E	S	W	NI	RI	AS	GB	IU
Salticus scenicus (Clerck, 1757)			E	S	W	NI	RI			
Salticus cingulatus (Panzer, 1797)			E	S	W	NI	RI			
Salticus zebraneus (C. L. Koch, 1837)			E						NS	
Heliophanus cupreus (Walckenaer, 1802)			E	S	W	NI	RI			
Heliophanus flavipes (Hahn, 1832)			E	S	W	NI	RI			
Heliophanus auratus C. L. Koch, 1835			E						NR	V
Heliophanus dampfi Schenkel, 1923		Appendix 2	E	S	W				NR	V
Marpissa muscosa (Clerck, 1757)			E	S	W				NS	
Marpissa radiata (Grube, 1859)			E		W				NR	V
Marpissa nivoyi (Lucas, 1846)			E		W		RI		NS	
Sibianor aurocinctus (Ohlert, 1865)		*Bianor aurocinctus*	E	S	W				NS	
Sibianor larae Logunov, 2001	not present	not present	E							
Ballus chalybeius (Walckenaer, 1802)		*Ballus depressus*	E		W				NS	
Neon reticulatus (Blackwall, 1853)			E	S	W	NI	RI			
Neon robustus Lohmander, 1945		not present	E	S	W		RI		NS	
Neon valentulus Falconer, 1912			E						NR	CE
Neon pictus Kulczyński, 1891		not present	E						NR	NT
Euophrys frontalis (Walckenaer, 1802)			E	S	W	NI	RI			
Euophrys herbigrada (Simon, 1871)			E						NR	V
Euophrys petrensis C. L. Koch, 1837	*Talavera petrensis*	*Euophrys petrensis*	E	S		NI	RI		NR	NT
Pseudeuophrys erratica (Walckenaer, 1826)		*Euophrys erratica*	E	S	W	NI	RI		NS	
Pseudeuophrys lanigera (Simon, 1871)		*Euophrys lanigera*	E	S	W		RI			
Pseudeuophrys obsoleta (Simon, 1868)		*Euophrys browningi*	E						NS	
Talavera aequipes (O. P.-Cambridge, 1871)		*Euophrys aequipes*	E	S	W		RI			
Talavera thorelli (Kulczyński, 1891)		Appendix 2 as *Euophrys thorelli*	E						NR	V
Hypositticus pubescens (Fabricius, 1775)	*Sitticus pubescens*	*Sitticus pubescens*	E	S	W		RI			
Attulus distinguendus (Simon, 1968)	*Sitticus distinguendus*	not present	E						NR	CE
Attulus saltator (O. P.-Cambridge, 1868)	*Sitticus saltator*	*Attulus saltator*	E		W				NS	
Calositticus caricis (Westring, 1861)	*Sitticus caricis*	*Sitticus caricis*	E		W		RI		NR	
Calositticus floricola (C. L. Koch, 1837)	*Sitticus floricola*	*Sitticus floricola*	E	S	W	NI	RI		NR	NT
Calositticus inexpectus (Logunov & Kronestedt, 1997)	*Sitticus inexpectus*	not present	E		W				NS	
Evarcha falcata (Clerck, 1757)			E	S	W		RI			
Evarcha arcuata (Clerck, 1757)			E		W				NS	
Macaroeris nidicolens (Walckenaer, 1802)		not present	E							
Aelurillus v-insignitus (Clerck, 1757)			E	S	W				NS	
Phlegra fasciata (Hahn, 1826)			E		W				NR	NT
Synageles venator (Lucas, 1836)			E		W				NS	
Myrmarachne formicaria (De Geer, 1778)			E						NS	
Pellenes tripunctatus (Walckenaer, 1802)			E						NR	V
Dendryphantes rudis (Sundevall, 1832)	not present	not present	E							
Hasarius adansoni (Audouin, 1826)	not present	not present	E			NI	RI	Ag		

List A2. Species awaiting taxonomic definition

Species	2014 Checklist	Family	E	S	W	NI	RI	AS	GB	IU
Orchestina sp.		Oonopidae	E						NR	CE
Megalepthyphantes cf. *collinus*		Linyphiidae	E							
Lathys sp.	not present	Dictynidae					RI			

List A3. Species with insufficient data to determine status

Species	2014 Checklist	Family	E	S	W	NI	RI	AS	GB	IU
Holocnemus pluchei (Scopoli, 1763)	not present	Pholcidae	E					Ab		
Oecobius navus Blackwall, 1859	not present	Oecobidae	E					Ab		
Islandiana falsifica (Keyserling, 1886)		Linyphiidae			W					
Frontinellina frutetorum (C.L.Koch, 1834)		Linyphiidae	E							
Neriene emphana (Walckenaer, 1841)	not present	Linyphiidae	E							
Tegenaria ramblae Barrientos, 1978		Agelenidae	E							
Trachyzelotes fuscipes (C.L.Koch, 1866)	not present	Gnaphosidae	E							
Synema globosum (Fabricius, 1775)	not present	Thomisidae	E							

Table 2. Species recorded but not established in natural or semi-natural habitats

List B1. Species found only in artificial tropical biomes

Species	2014 Checklist	Family
Spermophora kerinci Huber, 2005	not present	Pholcidae
Nesticella mogera (Yaginuma, 1972)	not present	Nesticidae
Pseudanapis aloha Forster, 1959	not present	Anapidae

List B2. Species recorded from imported goods but not established

Species	2014 Checklist	Family
Uroctea durandi (Latreille, 1809)	not present	Oecobiidae
Artema atlanta Walckenaer, 1873	not present	Pholcidae
Steatoda paykulliana (Walckenaer, 1806)		Theridiidae
Cyrtophora citricola (Forsskål, 1775)	not present	Araneidae
Olios sanctivincentii (Simon, 1897)	not present	Sparassidae
Barylestis variatus (Pocock, 1899)	not present	Sparassidae
Heteropoda venatoria (Linnaeus, 1767)	not present	Sparassidae
Philaeus chrysops (Poda, 1761)		Salticidae
Phidippus johnsoni (Peckham and Peckham, 1883)	not present	Salticidae

2.4 Checklist of British Harvestmen

This new checklist is taken from Davidson (2019), whose paper is available for download from doi.org/10.13156/arac.2019.18.3.213 and is free to BAS members.

order OPILIONES Sundevall, 1833
suborder LANIATORES Thorell, 1876
  family Phalangodidae Simon, 1879
    Scotolemon doriae Pavesi, 1878 [1#] relict or possible introduction
suborder DYSPNOI Hansen & Sørensen, 1904
  family Nemastomatidae Simon, 1872
    Nemastoma bimaculatum (Fabricius, 1775)
    Mitostoma chrysomelas (Hermann, 1804)
    Nemastomella bacillifera (Simon, 1879)
      = *Centetostoma bacilliferum* (Simon, 1879) [2#] relict or possible introduction
  family Trogulidae Sundevall, 1833
    Trogulus tricarinatus (Linnaeus, 1767)
    Anelasmocephalus cambridgei (Westwood, 1874)
  family Sabaconidae Dresco, 1970
    Sabacon viscayanus ramblaianus Martens, 1983
      = *Sabacon viscayanum ramblaianum* (Martens, 1983) [3#] relict or possible introduction
suborder EUPNOI Hansen & Sørensen, 1904
  family Phalangiidae Latreille, 1802
    subfamily Oligolophinae Banks, 1893
      Oligolophus tridens (C. L. Koch, 1836)
      Oligolophus hanseni (Kraepelin, 1896)
      Paroligolophus agrestis (Meade, 1855)

Paroligolophus meadii (O. Pickard-Cambridge, 1890)	
Lacinius ephippiatus (C. L. Koch, 1835)	
Odiellus spinosus (Bosc, 1792)	established introduction
Mitopus morio (Fabricius, 1799)	
subfamily Phalangiinae Latreille, 1802	
Phalangium opilio Linnaeus, 1758	
subfamily Opilioninae C. L. Koch, 1839	
Opilio parietinus (De Geer, 1778)	
Opilio canestrinii (Thorell, 1876)	recent introduction
Opilio saxatilis C. L. Koch, 1839	
subfamily Platybuninae Staręga, 1976	
Megabunus diadema (Fabricius, 1779)	
Rilaena triangularis (Herbst, 1799)	
= *Platybunus triangularis* (Herbst, 1799)[4]	
Platybunus pinetorum C. L. Koch, 1839	recent introduction
Lophopilio palpinalis (Herbst, 1799)	
family Sclerosomatidae Simon, 1879	
subfamily Scleosomatinae Simon, 1879	
Homalenotus quadridentatus (Cuvier, 1795)	
subfamily Gyinae Šilhavý, 1946	
Dicranopalpus caudatus Dresco, 1948[5][#]	relict or possible introduction
Dicranopalpus ramosus (Simon, 1909)[5]	recent introduction
Dicranopalpus larvatus (Canestrini,1874)[6]	recent introduction
subfamily Leiobuninae Banks, 1893	
Leiobunum rotundum (Latreille, 1798)	
Leiobunum blackwalli Meade, 1861	
Leiobunum gracile Thorell, 1876	
= *Leiobunum tisciae* Avram, 1971[7]	recent introduction
Leiobunum sp. A[8]	recent introduction
Nelima gothica Lohmander, 1945[#]	relict or possible introduction

Notes

It is possible that some or all of these are previously undetected relict species, consistent with former geographical linkage to the Pyrenees/Iberia. Alternatively, they may be relatively recent introductions, sometimes indicated by rapid range expansion, e.g. in *Sabacon*, but this might equally be due to climate change allowing them to expand from their refugial areas.

1. *Scotolemon doriae* has recently been reported from Plymouth (Bilton 2018).

2. *Centetostoma bacilliferum* is now in the genus *Nemastomella* (Schönhofer 2013). The genus name *Nemastomella* was introduced by Mello-Leitão (1936) for an Iberian species, *N. integripes*, then synonymised with *N. dubia* by Staręga (1986). Staręga (1986) attached to the still valid genus name 11 Iberian species including *bacillifera* (Simon, 1879); Schönhofer (2013) validated this decision.

3. *Sabacon* - change of gender (Schönhofer 2013).

4. *Rilaena triangularis* was moved back to the genus *Platybunus* by Hillyard (2005). The justification for this is unclear and it has been restored to *Rilaena* for consistency with other European checklists and until the relationship between these genera is resolved.

5. The presence in Britain of *Dicranopalpus caudatus* and *Dicranopalpus ramosus* was established by Wijnhoven & Prieto (2015).

6. *Dicranopalpus larvatus* was discovered in the Scilly Isles, Guernsey and the Isle of Wight in February 2019.

7. The *Leiobunum rupestre* species group was reviewed by Martens & Schönhofer (2016). This concluded that *Leiobunum gracile* was the earliest available name for *L. tisciae*.

8. An unknown species of *Leiobunum* has been spreading across Europe since about the year 2000 (Wijnhoven *et al.* 2007). This has so far remained un-named and has been known as *Leiobunum* sp. A in the interim.

2.5 Checklist of British and Irish Pseudoscorpions

This checklist, based on Harvey (2013), is taken from that published by Legg (2019). The paper is available for download from doi.org/10.13156/arac.2019.18.3.189 and is free to BAS members.

order PSEUDOSCORPIONES Latreille, 1825
suborder EPIOCHEIRATA Harvey, 1992
superfamily Chthonioidea Daday, 1889
family Chthoniidae Daday, 1889
tribe Chthoniini Daday, 1889
Ephippiochthonius Zaragoza, 2017
Ephippiochthonius tetrachelatus (Pressler, 1790)
Dimpled-clawed chthoniid
Ephippiochthonius kewi Gabbutt, 1966
Kew's chthoniid
Chthonius C. L. Koch, 1843
Chthonius (*Chthonius*) *halberti* Kew, 1916
Halbert's chthonid
Chthonius (*Chthonius*) *ischnocheles* (Hermann, 1804)
Common chthoniid
Chthonius (*Chthonius*) *tenuis* L. Koch, 1873
Dark-clawed chthoniid
Chthonius (*Chthonian*) *orthodactylus* (Leach, 1817) *s.s.*
Straight-clawed chthoniid
suborder IOCHEIRATA Harvey, 1992
superfamily Neobisiodea Chamberlin, 1930
family Neobisiidae Chamberlin, 1930
subfamily Neobisiinae Chamberlin, 1930
Neobisium Chamberlin, 1930
Neobisium (*Neobisium*) *maritimum* (Leach, 1817)
Shore neobisiid
Neobisium (*Neobisium*) *carpenteri* (Kew, 1910)
Carpenter's neobisiid
Neobisium (*Neobisium*) *carcinoides* (Herman, 1804)
Moss neobisiid
Roncus L. Koch, 1873
Roncus (*Roncus*) *lubricus* L. Koch, 1873
Reddish two-eyed neobisiid
subfamily Microcreagrinae, Balzan, 1892
Roncocreagris Mahnert, 1976
Roncocreagris cambridgei (L. Koch, 1873)
Cambridge's two-eyed neobisiid
Microbisium Chamberlin, 1930
Microbisium brevifemoratum (Ellingsen, 1903)
Bog neobisiid

superfamily Cheiridioidea Hansen, 1894
family Cheiridiidae Hansen, 1894
subfamily Cheiridiinae Hansen, 1894
Cheiridium Menge, 1855
Cheiridium museorum (Leach, 1817)
Book chelifer
superfamily Cheliferoidea Risso, 1827
family Chernetidae Menge, 1855
subfamily Lamprochernetinae Beier, 1932
Lamprochernes Tömösvary, 1892
Lamprochernes savignyi (Simon, 1881)
Savigny's shining claw
Lamprochernes nodosus (Schrank, 1803)
Knotty shining claw
Lamprochernes chyzeri (Tömösvary, 1882)
Chyzer's shining claw
Pselaphochernes Beier, 1932
Pselaphochernes scorpioides (Hermann, 1804)
Compost chernes
Pselaphochernes dubius (O. Pickard-Cambridge, 1892)
Small chernes
Allochernes Beier, 1932
Allochernes powelli (Kew, 1916)
Powell's chernes
Allochernes wideri (L. Koch, 1873)
Wider's tree chernes
subfamily Chernetinae Beier, 1932
Dinocheirus Chamberlin, 1929
Dinocheirus panzeri (L. Koch, 1873)
Terrible-clawed chernes
Chernes Menge, 1855
Chernes cimicoides (Fabricius, 1793)
Common tree chernes
Dendrochernes Beier, 1932
Dendrochernes cyrneus (L. Koch, 1873)
Large tree chernes
Americhernes Muchmore, 1976
Americhernes oblongus (Say, 1821)
American chernes
family Withiidae Chamberlin, 1931
subfamily Withiinae Chamberlin 1931
Withius Kew, 1911
Withius piger (Simon, 1878)
Lazy chelifer
family Cheliferidae Risso, 1827
subfamily Cheliferinae Risso, 1827
tribe Cheliferini Risso 1827
Chelifer Geoffroy, 1762
Chelifer cancroides (Linnaeus, 1758)
House chelifer

tribe Dactylocheliferini Beier, 1932
Dactylochelifer Beier, 1932
Dactylochelifer latreillii (Leach, 1817)
Marram grass chelifer
superfamily Garypoidea Simon, 1879
family Larcidae Harvey 1992
Larca Chamberlin, 1930
Larca lata (Hansen, 1884)
Oak tree chelifer

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COLOUR PLATES

Fig. 1: *Euscorpius flavicaudis* (Euscorpiidae), 35–45 mm. The European Yellow-tailed Scorpion was introduced to docks in south-east England, possibly in the early 19th century. It remains our only species of true scorpion. © R. C. Gallon.

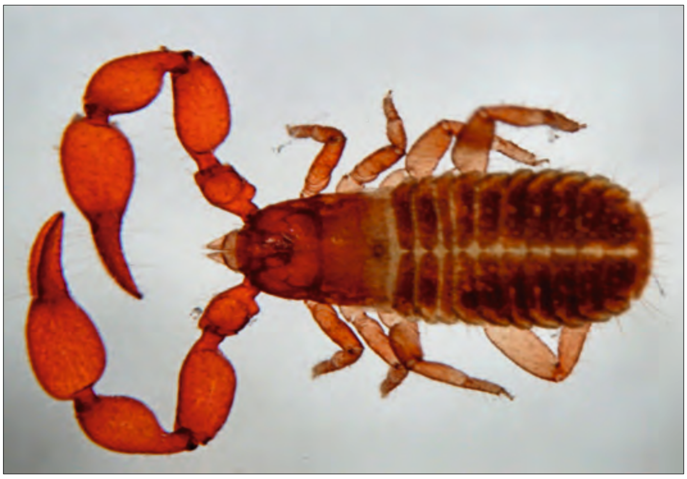

Fig. 2: *Lamprochenes nodosus* (Chernetidae), 2 mm. One of our 28 species of pseudoscorpions. This species is found in a variety of habitats, including birds' nests, and often disperses attached to flying insects (phoresy). © P. J. Nicholson.

Fig. 3: *Opilio canestrinii* (Phalangiidae) female, 9 mm. Although records are scattered, this harvestman is widely distributed across Britain. Mature individuals can be found from July to December. © J. P. Richards.

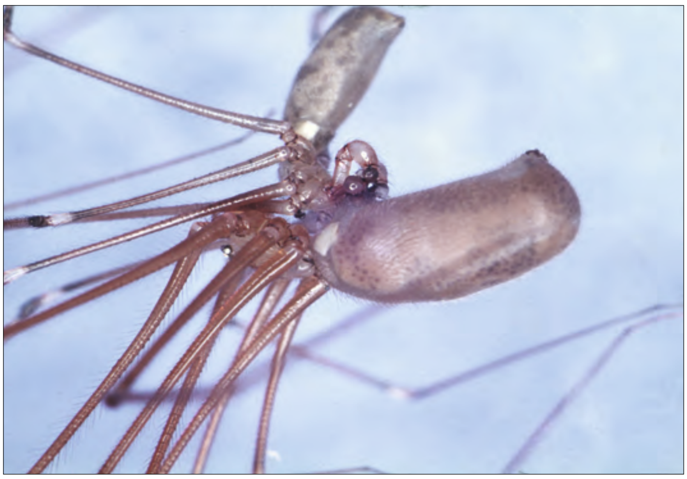

Fig. 4: *Pholcus phalangioides* (Pholcidae), mating pair. Male, 7–10 mm; female, 8–10 mm. This species has rapidly spread across Britain in the last few decades. © J. Murphy.

Fig. 5: *Segestria florentina* (Segestriidae) female, 13–22 mm. This large, six-eyed species lives in holes in walls and is characterised by a green irridescence on its jaws. Although currently largely confined to southern England, it is spreading northwards. © P. R. Harvey.

Fig. 6: *Dysdera crocata* (Dysderidae), male, 9–10 mm. The massively enlarged chelicerae are specially adapted for feeding on well-armoured prey like woodlice. © R. C. Gallon

Fig. 7: *Hyptiotes paradoxus* (Uloboridae), female, 5–6 mm, in her web. The spider bridges the gap in a trip line which, when released, allows the triangular web to collapse around any prey item touching it. © J. Murphy.

Fig. 8: *Phylloneta sisyphia* (Theridiidae), female, 3–4 mm, with young. This species feeds the young with regurgitated liquid food. © J. Murphy.

Fig. 9: *Enoplognatha ovata* (Theridiidae) pair. Female 4–6 mm; male 3–5 mm. The mature male (in focus: yellow form, *lineata*) waits until the penultimate instar female (striped form, *redimita*) moults to maturity. © G. S. Oxford.

Fig. 10: *Araneus diadematus* (Araneidae), female, 10–13 mm, weaving orb web. The spider is dragging silk for the sticky spiral from her spinneret using a hind leg. © J. Murphy.

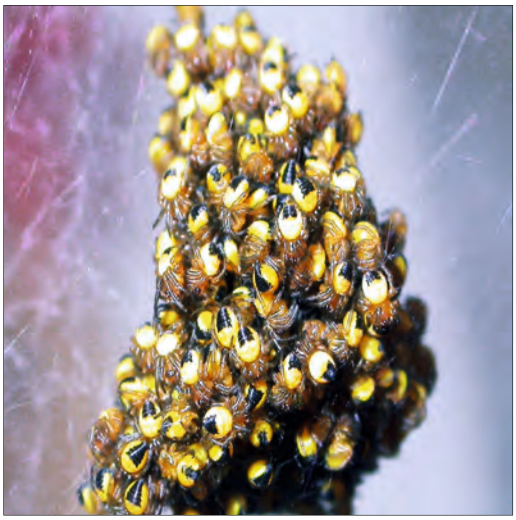

Fig. 11: *Araneus diadematus* (Araneidae). The newly emerged young cluster together for a few days after emerging from the egg sac. If disturbed, the ball of spiderlings expands outwards, only to contract again when danger has passed. © G. S. Oxford.

Fig. 12: *Cyclosa conica* (Araneidae) female, 5–7 mm. A small, orbweb spider that often frequents dark habitats, such as on yew trees. An irregular band of silk (stabilimentum) across the centre usually distinguishes its web. © E. Jones.

Fig. 13: *Araneus alsine* (Araneidae), female, 7–13 mm. A very local species with recent records concentrated in two main areas: south-eastern England and Inverness-shire. The web is spun in low herbage and is associated with a retreat of dry leaves (as illustrated). © E. Jones.

Fig. 14: *Pardosa saltans* (Lycosidae), female, 5–6 mm, with spiderlings on abdomen. Females of all wolf spiders carry the young on the abdomen until they are large enough to fend for themselves. © M. Askins.

Fig. 15: *Pisaura mirabilis* (Pisauridae), nursery web with egg sac and spiderlings. The female, 12–15 mm, guards her nursery for several days until the spiderlings are ready to disperse. © A. Rivett.

Fig. 16: *Dolomedes fimbriatus* (Pisauridae) female, 13–20 mm. A species that is widespread on wet heathlands in the south but very scattered elsewhere in Britain. Like the more familiar *Pisaura mirabilis*, females initially carry their egg sacs in their chelicerae and then construct a nursery web as the young start to hatch. © R. C. Gallon.

Fig. 17: *Nigma walckenaeri* (Dictynidae) female, 4–5 mm. A bright green species often found in the curled leaves of ivy. It is currently spreading rapidly northwards from a stronghold in south-eastern England. A second centre of distribution is in the Severn valley. © N. Nimbus.

Fig. 18: *Micrommata virescens* (Sparassidae) mating pair. Female 10–15 mm; male 7–10 mm. This species shows a marked sexual dimorphism, with the mature male sporting red and yellow stripes and the female bright green. The eggs, too, are a vivid green. © E. Jones.

Fig. 19: *Tegenaria ferruginea* (Agelenidae) female, 11–14 mm. This very attractive species, while common across continental Europe, is found in Britain at just one location: Elvington, Yorkshire. © G. S. Oxford.

Fig. 20: *Porrhoclubiona* (*Clubiona*) *genevensis* (Clubionidae) female, 3.5–4.5 mm. This Nationally Rare species is most recently recorded on the very western extremities of Britain: the Lizard Peninsula, Isles of Scilly, the Pembrokeshire islands and the Lleyn Peninsula. It frequents maritime grasslands and heaths. © R. C. Gallon.

Fig. 21: *Misumena vatia* (Thomisidae) mating pair. Female 9–11 mm; male 3–4 mm. Females hunt on flowers and can reversibly adjust their colour from white to yellow. The more cursorial males show disruptive coloration. © R. Rigby.

Fig. 22: *Thomisus onustus* (Thomisidae) female, 6–7 mm. This species can vary its coloration—white, pink or yellow—depending on its background. The colour change takes place over a matter of days as pigments are synthesised or destroyed. © S. Covey.

Fig. 23: *Xysticus cristatus* (Thomisidae), female, 6–8 mm, feeding on a Cinnabar moth. Crab spiders are capable of capturing prey much larger than themselves. © P. R. Harvey.

Fig. 24: *Marpissa muscosa* (Salticidae) female, 8–10 mm. This is one of our largest jumping spiders and is confined to the south east of England. It frequents crevices of dead trunks and branches. © E. Jones

Fig. 25: Plastic cup pitfall trap *in situ*. Here, protection from rain is provided by a plastic plant pot saucer supported on a wire loop. © P. Smithers.

Fig. 26: A plywood platform pitfall trap deployed in scree. © P. Smithers.

Fig. 27: A subterranean pitfall trap inside a bird feeder. This is then buried in shingle to catch interstitial spiders. © P. Smithers.

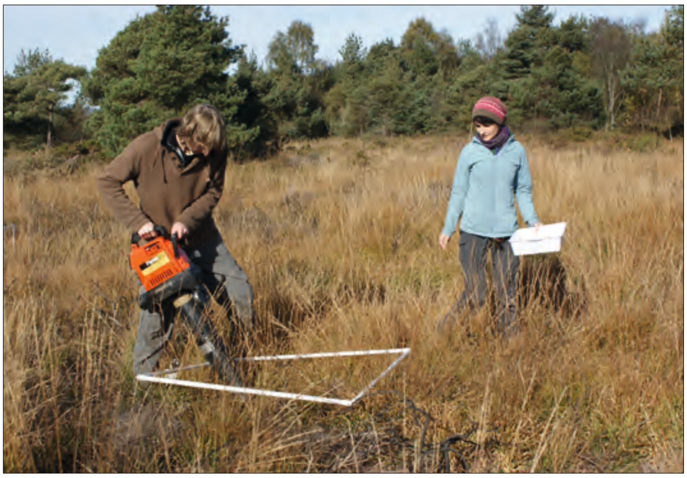

Fig. 28: Semi-quantitative sampling within a square metre quadrat using a G-vac. © P. Smithers.

3 COLLECTING ARACHNIDS

3.1 Techniques

Lawrence Bee & Peter Smithers

This chapter 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 the majority have long, delicate legs. 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 quarry 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) or tray placed underneath the sieve will catch any animals that drop through and enable them to be seen 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; tussocks of living grass can be investigated by grubbing about. This is an excellent method for finding litter-living pseudoscorpions although they tend to be inactive, and therefore very difficult to spot, immediately after the disturbance.

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 non-natural materials that have been lying on the ground for some time. It is always worth investigating these sites, but remember to restore whatever has been disturbed to its original position. Bear 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, tray 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 and stones above lifted off immediately over the net. This is very effective for catching fast-moving spiders such as *Segestria* and *Textrix*. Once again, ensure that stones are replaced on the wall exactly as found. 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

One of the most productive collecting methods for ground-dwelling spiders, particularly those that are active hunters, is to set pitfall traps. They are cheap and easy to install, sample over extended periods, and collect a wide range of ground-living invertebrates. At their simplest, they can be plastic pots 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 yoghurt pots, which are cheap and easily obtained, but any convenient non-breakable container can be used. Translucent pots are best because they become invisible when set in the soil and do not attract the attention of vertebrates as readily as white ones. 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 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, long-handled 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 pot and pile displaced soil at least an arms-reach away. Displaced soil will act as a barrier, which will deflect invertebrates away from the trap. Freshly disturbed soil has also been shown to temporarily increase the activity of invertebrates in the vicinity of the trap, possibly a result of an increase in soil respiration, which elevates local CO_2 levels (Wright & Stewart 1992).

If traps are to be used over a period of weeks or more, disturbance can be reduced by having two plastic pots, 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 tape work well, either attached to nearby vegetation or as a flag. Mark the ends of each row and draw a sketch map noting the positions of the traps together with any prominent features in the micro-landscape; it is amazing how difficult the traps can be to relocate a week or two later. If possible, it is useful to place traps a constant distance (for example, one pace) apart to aid relocation. If GPS is available, record the location of the ends of the trap line or, if it is a long line, the location of individual traps.

Where traps are not checked on a daily basis, 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 (feral and domestic), which find it very attractive. To avoid this problem, we recommend that this preservative is only used in situations where mammals cannot access the traps. Propylene glycol, however, is less toxic than ethylene glycol and can be obtained from camping supply shops where it is sold as an antifreeze for caravan shower and water systems. Another alternative is the bactericide propylene phenoxetol. This is used as a 1% solution in tap water and is available from Blades Biological (see Section 9.1). Propylene phenoxetol will preserve the trap contents for up to a week but is not effective over longer periods.

Where traps can be emptied daily (avoiding the risk of decomposition of the catch) and the spiders 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. If the spiders are required alive, vegetation can be added to the dry cup to minimise encounters between predators and provide retreats in which smaller invertebrates can hide. These traps should be emptied at least daily.

When traps are to be left out for any length of time, rain covers are essential to prevent flooding and loss of the catch (Fig. 25). They can be plastic lids, squares of plastic or plywood, or simply a stone held above the cup on supports made of wire, 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 more than a very short period, a coarse wire mesh should be placed over them to prevent small mammals, reptiles, or amphibians falling in and drowning. If this is a problem, despite precautions, the trapping programme should be stopped.

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. A larger number of smaller traps is usually the best solution because the risk of catching small mammals and reptiles is increased by increasing trap size. However, if a trapping 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 a pair of pitfall traps (○) on either side and at each end of a linear barrier (see diagram below). Plastic lawn edging is a cheap and effective material that works well. Spiders and other invertebrates that 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.

Pitfall traps can also be useful in sampling the arachnid fauna of hollow, ancient trees. Locating the trap in the accumulated rotted material within a hollow trunk, in a similar way to that used when positioning traps in the ground, can be a productive method of collecting surface-active arachnids within this particular microhabitat.

Subterranean pitfalls can be used to sample the invertebrate fauna of any loosely packed medium, such as soil or shingle (Fig. 27). They can be made from plastic bird feeders which are available from garden centres. Select one with a mesh of 5–10 mm to allow larger invertebrates entry. A plastic cup or other container is cut to size so that it fits inside the feeder. This is filled with preservative (as this will be away from mammals ethylene glycol can be used), and the cup is slid to the bottom of the feeder. Stout string is tied to the top of the feeder and a marker of some sort attached (label or coloured ball) to ensure it can be relocated. A hole is dug in the substrate to be sampled and the trap lowered in and carefully buried (Owen 1995). The traps should be left in place for a week or two before being removed to inspect the contents. If investigating shingle beaches, traps should be positioned above the limits of expected high tides. Those that are close to the intertidal area need regular inspection as a good storm could remove all trace of your markers.

Platform pitfalls are extremely useful where there is no surface in which to bury normal pitfalls, such as scree slopes, boulder fields, and caves (Fig. 26). They comprise a square or circular platform of plywood or thin plastic with a central hole. A suitable container is fixed beneath the platform. They can be propped so that the platform touches as many of the surrounding rocks as possible, and invertebrates wandering over the surrounding terrain will move naturally onto the platform and fall into the pitfall container (Růžička 1982). They can also be used to sample vertical surfaces such as cliffs and tree trunks by hanging them from nails driven in to the vertical surface (Růžička & Antus 1997). Alternatively, they can be placed in bushes or hoisted into the canopies of trees so that the platform makes contact with the leaves and finer branches. Platform pitfalls can also be modified to sample aquatic environments by fixing cork or polystyrene to the underside of the platforms, thus enabling them to float and sample the surface faunas of water bodies.

Care should be taken when interpreting the results from pitfall surveys. The relative abundance of organisms in pitfall traps rarely reflects accurately 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 their 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. Factors affecting the efficiency of pitfall trapping for spiders have been considered by, among others, Topping & Luff (1995) and Siewers *et al*. (2014).

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 topped up 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, beating tray, 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, umbrella, or 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 debris to ensure that nothing has 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. 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. This method is not recommended when the vegetation is wet. Sweep net material, when wet, is heavy and can do considerable damage to the catch.

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, particularly, an inverted umbrella held tight against the tree, can usefully be employed to catch the sweepings.

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 double strip of 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 interstices. 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 and pseudoscorpions, 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 when any potential escapees can be caught before they disappear. Artificial nests can be created by packing thin twigs, wood, wool, small wood shavings, or dry grass into a plastic mesh bag (for example, those used by supermarkets to package oranges and onions). 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 (i. e. non-colonised) leaf litter may be left out for weeks or months to allow arachnids to move in. This technique is useful when natural litter is scarce. Alternatively, leaves heaped up on polythene sheets can be used to produce a large pile of leaf litter, even when the natural layer is thin, and can be taken away eventually by lifting and tying the sheet corners.

3.1.3 Other collecting techniques

Checking webs and retreats

It is always worth taking a close look at any webs encountered while 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. Webs are best seen when dew-covered, but a similar effect can be achieved by spraying them with water from a plant mister. Spiders that do not weave webs may construct some type of silken retreat, most commonly found in seed heads, in rolled-up leaves and under bark. On walls, rocks, tree trunks etc., threads left by wandering spiders often show their presence when viewed with sunlight reflecting off them.

Leaf litter sorting

It may be more convenient to collect plastic bags full of leaf litter and sort through them at home, rather than in the field. Empty your bags of litter, a few handfuls at a time, onto a large, white tray and carefully sort through the material. 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 litter is to use a Tullgren funnel. This consists of a sieve containing the litter sample with a desk lamp placed over the top. 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 that leads to a container of alcohol to preserve the extracted animals, or damp, screwed-up kitchen roll if the fauna is to be kept alive.

Winkler bags. An alternative to the Tullgren funnel, 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 or polythene funnel, held open with a wire hoop. The top of the funnel can be closed to prevent organisms escaping. The bottom of the funnel leads to a container of 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, falling out into the funnel and through to the container below. Tullgren funnels and Winkler bags can be very effective for extracting very small spider and pseudoscorpion species that are easily overlooked when hand collecting.

Simulating insect vibrations in webs

Luring crevice-dwelling spiders out of their lairs can be difficult. One way is to 'fish' for them with a maggot (Oxford & Croucher 1997). Maggots can be purchased at fishing shops and keep for months in a ventilated but well-lidded plastic box in a fridge. A quarter-pint measure of pinkies (greenbottle larvae) is enough to fish hundreds of webs. Drop a maggot into the web (flattish webs work best, e.g. of *Agelena*, *Tegenaria*, *Eratigena* and *Amaurobius*) and when the occupant emerges, wait for it to bite the prey. There is a split-second pause while the spider extricates the maggot from the web; this is the time to strike. Place a suitably sized specimen tube over the spider, trapping it against its web, and at the same time bring the lid up from beneath the web to secure the spider in the tube. Fishing in this way does not work very well after rain because the maggots easily squirm their way through wet silk.

Some spiders, e.g. *Segestria*, *Amaurobius*, *Tegenaria* and *Zygiella* spp. respond to a vibrating tuning-fork being placed on the web. *Segestria florentina* (Fig. 5), for example, can be encouraged to come to the entrance of her retreat by placing the tip of the tuning-fork on one of the silk lines 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.

Greg Hitchcock has suggested a modern alternative to the tuning fork: the sonic toothbrush. Battery-operated sonic toothbrushes that vibrate in one plane 20,000 times a minute (about 330 Hz) are widely available from chemists and supermarkets. The vibrations set up in the web are similar to those produced by the wings of a medium-sized fly. A toothbrush is extremely effective at luring spiders from their retreats and has the advantages of being cheaper than a tuning fork, does not require constant striking and, unlike maggots, works well on wet webs (Oxford 2013). The brush is applied under a web (if possible) and the spiders caught as described for the maggot technique above.

Night collecting

Many species are active mainly or only at night: and so it is well worthwhile going out with a hand-held or head-torch. Particular care should be taken when collecting at night; someone should always be told where you are going and when you expect to return (see Chapter 8).

Vacuum sampling

This technique uses a garden vacuum to suck invertebrates off short vegetation and the surface of the ground. The apparatus (formerly known as the D-vac) used to be bulky and expensive, which meant it was a technique only available to professionals. The development of petrol-driven garden leaf-vacuums and, more recently, rechargeable ones, has made suction samplers available to many more arachnologists and entomologists. A cordless garden leaf-vacuum can be easily modified by inserting a fine net bag in the intake of the machine secured in place with gaffer-tape or wide jubilee clips. Net bags can be hand-made from net curtain material, but brewers' wort bags, which just happen to be the correct size, are a ready-made alternative and are available from home brew shops. Purpose made vacuum sampler bags are also available from some entomological net retailers. Such modified machines are now known as G-vacs.

For the best results, the intake is held against the surface of the ground and swept from side to side to cover a known area delineated by a quadrat (Fig. 28). Vacuum 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 are shaken through a garden sieve (to remove loose plant material) onto a high-sided, white tray, where the invertebrates can be sorted and collected. The majority of your catch will be undamaged and active. Alternatively, the contents of the bag can be placed in containers to be sorted later. G-vacs are not without their limitations because, like sweep nets, wet vegetation reduces their efficiency. Battery powered G-vacs currently have a considerably shorter run-time than petrol equivalents, but tend to be quieter, lighter and more pleasant to use. Petrol G-vacs however are more powerful and reliable in wetland sites being more tolerant of accidentally sucking-up water.

Care should also be taken when interpreting catches as measures of population density per unit area because the high speed air flows at the edges of the intake tube tend to drag in items from outside the immediate area sampled. This effect decreases as the diameter of the intake tube is increased (Bell *et al.* 2002). But while they may not produce absolute data, they are excellent for comparative studies.

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 carry adequate storage space in the form of a strong and spacious shoulder bag or rucksack with plenty of separate compartments and pockets and/or a jacket with multiple pockets for collecting tubes and other small items of equipment.

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. Labels are best written in pencil and inserted into the tubes with the specimen and preserving fluid. Information noted should include date, habitat, grid reference, locality, and collector's name. Carry spare pencils and/or a pencil-sharpener/knife.

GPS

Recording the exact location of samples is essential for mapping purposes. It also enables the relocation of a collecting spot and better recording of microhabitats. In rather featureless habitats, such as heather moorland or sand dunes, a GPS unit or phone app is vital and will record a grid reference, potentially to within one metre. In more structured environments, several websites allow similarly precise grid references to be obtained retrospectively by locating the collection site relative to specific landmarks on aerial photographs; see References.

Collecting tubes

These 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 ones. 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 and disappear from view.

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 chamber of a pooter (see below) or collecting tube. The hand lens can be in virtually constant use so it is advisable to have it near at hand; wearing it on a neck strap is strongly recommended. Many fairly cheap, but reasonably good, lenses with an inbuilt light are now available. They can be extremely useful when examining spiders in a spi-pot (Section 4.1) or similar, as it is not always easy to position the enclosed specimen in good natural light, particularly on cloudy days. Even on sunny days, the shadows of the observer and the lens may cut out a portion of the available light.

Pooter

A pooter is a simple device used for collecting/catching spiders for examination. There are a number of designs. One of the best for spider collecting is the so-called Cooke pooter, which consists of a length of glass or plastic tubing with different sizes of glass tubing attached to one end (see diagram on next page). The spider is sucked into the glass or plastic barrel 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 prevent the spider being sucked beyond the barrel. Again, it is sometimes useful to attach your pooter to a neck strap to have it immediately available.

An even simpler pooter consists of two lengths of flexible plastic tubing, one with an approximately 10 mm internal diameter and the other with an approximately 10 mm external diameter. The piece with the larger internal diameter should be about 10 cm long and the other piece about 30 cm in length. The long

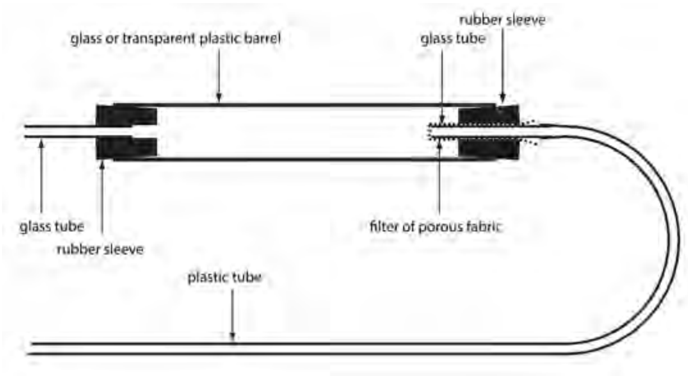

Diagram of a Cooke pooter shown in cross-section. The plastic tube has been truncated in this figure and would normally be three to four times the length of the barrel.

section has a piece of gauze placed over one end which is then push-fitted into one end of the shorter section and, if necessary, bound in place with tape.

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 when 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; but white sheeting can be too dazzling in bright sunshine. Old fertiliser sacks or polythene obtainable as cheap garden-pool liner from garden shops are ideal. 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

Collecting in areas of long grass, heather etc. is best done with a sweep net. 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 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. A robust, very economical sweep net can be made from an old tennis racquet from which the strings have been removed and a cheap pillowcase fixed to the rim with drawing pins.

3.2.2 Other useful items of equipment

Beating tray

A beating tray is used for collecting from shrubs 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 and pseudoscorpions 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 employed with care since indiscriminate use can cause considerable damage to the environment.

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; this can be bent into any convenient shape. Alternatively, an inverted umbrella may be used, and fits snugly against the bark.

Torch

A robust hand or head torch is a must for caves and 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 finish later in the day than planned (see Chapter 8).

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.

Holding devices

A small spi-pot or one of the other holding devices (Section 4.1) is often useful in the field, not necessarily for initial identification purposes, but to check whether a specimen is mature or not. This prevents the unnecessary collection of unidentifiable juveniles, although it is obviously impractical where mass-collection techniques such as vacuum sampling are used.

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 currently the most effective repellant is the DEET-containing 'Repel 100' obtainable from outdoor shops and online. Although there are reports of toxicity with sustained use, and care is needed to avoid contact with optical equipment, which it can damage, occasional use on collecting trips should present little risk and the benefits are considerable. A much more recent product 'Incognito' claims to be more powerful than DEET-containing repellants without the associated toxicity risks. To date, it is very well reviewed. A fine-mesh head net, worn over a brimmed hat and tied at the neck, is also very useful, particularly if soaked in repellent.

3.3 Code of Conduct for Collecting

Paul Lee

Introduction

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 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* (www.royensoc.co.uk/invertebrate-links) issued by Invertebrate Link (formerly Joint Committee for the Conservation of British Invertebrates), and 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 (SSSI) 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 followed strictly.

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 landowner and other appropriate authorities, as well as to the Spider Recording Scheme (see Section 7.1).

Protecting the environment

- While 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 every precaution to ensure that larger creatures such as frogs, shrews, and lizards 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 species of invertebrates in addition to arachnids. Every effort should be made to contact experts on other groups so that this by-catch material is not 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; most are of a size that mean a microscope is needed to examine the morphological features which determine the species. 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 is 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 subsequently to extract the best performance from it.

3.4.2 The components of the microscope

There are two main 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 arachnids. 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 (see Section 4.1.5). 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 while they are lying in preservative (see Chapter 4) 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 understood more easily. Third, the image is upright, in contrast to the two-dimensional image provided by a compound microscope in which 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 manoeuvring 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 less. 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 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 if 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 the eye tubes make 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 a range of both fixed and variable angles. 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.

In general, microscopes are supplied with the column supporting the lens housing at the back of the instrument 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 is 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 frequently 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 (graticule) is normally used to make such linear measurements. This is a glass disc with 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 can then be measured against the scale. 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, so 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; this should be checked with the supplier. Measurements can also be made using photographic images taken through the microscope as long as a scale line is included in the image. For example, the free software package ImageJ allows measurements to be made very easily and can also be used with images of live spiders taken with or without a microscope (Baillie & Smith 2012).

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, some slight refocusing is invariably 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 or online). 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 microscope should always be covered with the dust hood suppled when not in use.

The top light

It is standard practice to examine specimens whilst they are lying on fine glass beads immersed in preservative and illuminated by one or two high-intensity top lights. Top lights are available in two forms: those using a cold halogen light (generally in combination with fibre-optic light guides) and those with LEDs (light-emitting diodes). Halogen systems are much more expensive than LEDs: several hundred pounds versus tens of pounds. Devotees of halogen sources 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 lamps do have one distinct disadvantage: the light has a higher blue component which is scattered by colloidal material in the preservative (e.g. particles having a diameter less than 0.002 mm). It is extremely difficult to keep preservative free of colloidal material and it will thus appear slightly more opalescent when illuminated with a halogen bulb. This 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.

LED lights have all the advantages of halogen lighting systems but at a tiny fraction of the cost. They can be bought in many shapes and sizes from good hardware shops or online to suit specific needs. LED torches with varying numbers of LED elements (and therefore light intensity) just require a suitable stand/clamp so that they can be directed at the specimen; other lights come with a stand or desk clips and have flexible necks. Some models have adjustable swan necks allowing fine adjustments to the light direction, like the fibre-optic light guides of the halogen systems. LEDs also generate a bluish light and may suffer from the light scattering noted above.

Spirit

A discussion of different preservatives is provided in Section 4.2.2. Here it is sufficient to say that it is wise to avoid dirty preservative as that will increase light scattering and obscure details of the specimen, as outlined above.

Glass beads/sand/supportive gels

Specimens lying in preservative are often supported on 80-mesh glass beads in an excavated glass block (solid watchglass). The specimen can then be pushed into the beads and manipulated to any desired position to allow 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. 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. One disadvantage of fine glass beads is that if they are spilt in quantity on a hard floor they present a significant slip hazard—think of walking on tiny ball bearings.

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. For routine purposes, very fine, cleaned sand can be substituted for glass beads, although sand grains may be more likely to adhere to the specimen.

Small bottles of hand sanitiser (a clear gel containing alcohol) are now readilby available in supermarkets and pharmacists (e.g. Superdrug antimicrobial hand gel). They contain 60% or 70% ethanol, the same or nearly the same concentration as used to preserve spiders (see Section 4.2.2). This can be used as an alternative to glass beads or sand for holding specimens steady. Squeeze a small amount of sanitizer gel into the bottom of an excavated glass block, position the specimen and then gently fill the block with preservative. The gel is transparent and so the block can be placed on a white background if photographs are required (see Section 5.4) or against a black background to view trichobothria or hairs. Care should be taken not to totally immerse the specimen in the gel. When replaced in its tube, any gel adhering to the specimen will dissolve; it is not known whether traces of gel have any long-term detrimental effect on storage or DNA preservation.

Drawing images (by Rowley Snazell)

Biological drawing methods, such as those used to prepare figures either for scientific papers or purely for one's own use, have little to do with art. They are a series of techniques which, with practice, can be learnt by most people. Given very basic equipment, it is possible to produce excellent results. First, it is necessary to draw accurately the main shape of the object and place on it all the major features. To do this, I use a squared graticule in the eyepiece of the microscope in conjunction with graph paper. That initial image is drawn with a fairly soft pencil (HB or F) and then transferred to the final drawing surface using drafting film or tracing paper. It is also possible to achieve this initial image using a camera lucida or drawing tube, although these pieces of equipment are usually quite expensive. Once the basic layout of the drawing is captured on the final drawing surface, I work with Rötring pens to apply stippling and various other techniques to achieve the finished figure. If using pens, a very high quality card or heavy cartridge type paper with a plate finish is necessary. I then scan the drawings to file in order to add numbers, guide lines etc. using Photoshop. Other people use rather different methods to great effect, as seen in Bernhard Huber's excellent website www.pholcidae.de. The beginner can learn a great deal by using a magnifying glass to study the methods the best illustrators use to achieve different effects, such as surface textures and degrees of translucence. A much more comprehensive discussion can be found in Drawing techniques for publication written by Bowestead and Eccles, available at: www.zin.ru/ANIMALIA/coleoptera/pdf/bowestead_eccles_drawing_techniques.pdf. Even in this age of readily accessible digital photography, autographic drawings are vital, especially to the taxonomist. Photographs make useful adjuncts to scientific descriptions but cannot replace high quality drawings. Also, there are too many examples of potentially useful taxonomic papers which have been spoilt by poor figures; authors of papers should realise that a well observed, well executed drawing is more useful than any amount of text. Taking photographs of specimens under the microscope (photomicrography) is covered in Section 5.4.

3.4.4 USB digital microscopes

For those just beginning to identify spiders, the outlay on a good stereo microscope can be a serious impediment. Would a USB digital microscope do instead? These devices are now readily available and simply plug into the USB port of your computer, with the image viewed on screen. They start at as little as £30, but higher quality ones cost up to £700 or more (way beyond that of a reasonable quality, second-hand, optical microscope which can be purchased for about £200). Although we have limited experience of using these microscopes to examine palps and epigynes in preservative, the magnification offered (usually zooming to 200× or 400×) should be adequate if the resolution and lighting are sufficient.

Some of the pros and cons of digital microscopes for identifying spiders are given below, and are based on the experiences of Katty Baird using a Dino-Lite AM2111 unit costing just over £100, and Nigel Cane-Honeysett using a £29 Aldi scope with 10×, 60× and 200× magnification, a £100 DigiMicro Mobile which runs off a battery and a £130-ish Dino-Lite Edge. Each has its limitations, but a detailed review of makes and models is beyond the scope of this chapter; up-to-date reviews can be found online.

Pros

- More portable than conventional microscopes (so potentially useful on field trips).
- Have their own built-in light sources powered by the computer (or battery in the case of the DigiMicro Mobile).
- The software allows screen capture of images, which can then easily be shared to check identification. Video recording is possible.
- Stands are available, allowing specimens to be manoeuvred beneath the lens.

Cons

- Inbuilt light sources may be inadequate at high magnification, but a variety of external LED lights (mains/battery) are readily available if the light can be directed to the parts of interest.
- The position of the built-in lights (often immediately above the object) can cause reflectance and focussing problems if the specimen is examined within a container (e.g. a Petri dish). This can also apply to palps/epigynes viewed in preservative.
- Zooming in, for at least some models, brings the microscope closer to the specimen, so at high magnifications there is little room to allow manipulation.
- The screen captures can be very grainy and the depth of focus is shallow.
- With the supplied software there is no control over such things as exposure, and so supplementary lighting is compensated for by the software and therefore fails to enhance the final image in the way intended.

For some of the larger species with highly distinctive epigynes, e.g. *Pardosa pullata*, identification is possible using these devices, even when alive and constrained in a Spi-pot (Section 4.1). However, lighting difficulties and the quality of the image mean many of the crucial fine details of palps and epigynes of most species are impossible to resolve. So, on the basis of this limited experience, if species identification is the primary aim it would seem that the outlay on a digital microscope would be better spent on, or put towards, the purchase of a conventional stereo microscope.

Digital microscopes come into their own when used for educational or training purposes because images can be shown directly to an audience using a data projector. Children, in particular, often have difficulties looking down a conventional microscope, and so projected images are the best way to ensure they actually appreciate what they are looking for. Live spiders can be viewed in this way if restrained in a small transparent container or a Spi-pot; tracking a rapidly moving specimen in a larger container while keeping it in focus can be extremely frustrating. Digital microscopes have been used successfully for demonstrating key features of spider families, e.g. body shapes, eye patterns, during identification workshops. For many beginners, even observing the movement of the heart is a magical moment.

3.4.5 Suppliers

The companies listed in Chapter 9 supply microscopes and high-intensity lights of all types. They are provided simply as a guide, without any particular recommendations or endorsements. 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 every autumn at Kempton Park Racecourse, and this provides an excellent opportunity for their various models to be examined. Meiji Techno UK Ltd 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 and a wide and bright field of view without shadows resulting from dirt on internal glass surfaces. An object in focus in the centre of the field should also be in focus at the edges; there may be some slight degradation at the edge but the field should not appear to be concave. 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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Some geolocation packages

SRS website: srs.britishspiders.org.uk/portal/p/Locate

Grab a Grid reference: www.bnhs.co.uk/focuson/grabagridref/html/index.htm

UK Grid Reference Finder: gridreferencefinder.com

Vice county look-up: herbariaunited.org/gridrefVC

4 STARTING AND USING A REFERENCE COLLECTION

Paul Lee

4.1 Identification

4.1.1 Introduction

Although a photographic guide to Britain's spiders is now available (Bee, Oxford & Smith 2017) it remains the case that only a minority can be identified in the field by the novice, and even experts will only attempt identification of a limited number of species this way. Nevertheless, under some circumstances, there is merit in examining spiders alive with a hand lens or microscope before, or instead of, preservation. For those new to spiders, viewing their features in life—eye conformation, coloration, heart beat—leads to a deeper understanding and appreciation of the group. In addition, in most cases juvenile spiders cannot reliably be identified and live examination on capture means that they can be spared an unnecessary death. This may be impractical when mass sampling, for example with pitfall traps (see Section 3.1), but is sensible if a novice just wants to identify occasional spiders in their garden. Devices for restraining live spiders are described below.

For the majority of spiders, definitive identification to species requires that they are collected and killed prior to microscopic examination of the male palps or the female epigyne. Once preserved, specimens are available for future reference and where expert opinion may be required to confirm identification (for example where rare species are involved).

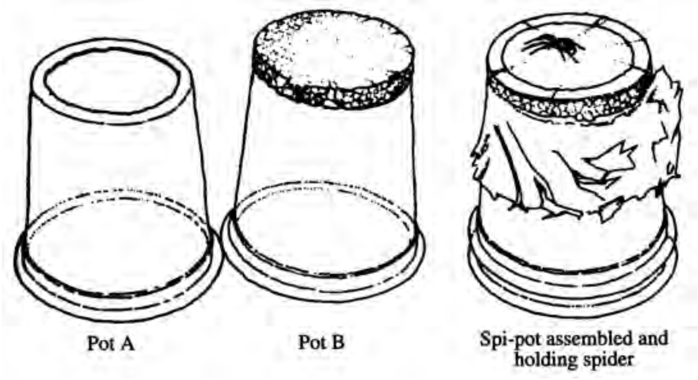
When specimens are killed in alcohol, there is a tendency for the legs to contract, obscuring characteristic features of the legs themselves and of the underside of the animal, particularly the epigyne. In such cases, removal of individual limbs may be necessary. Many arachnologists consider such problems minor drawbacks. 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.

4.1.2 Examination of live spiders

Live spiders can be examined most obviously in a glass tube with a hand lens or stereo microscope, but their movement constantly takes them out of focus. This is especially frustrating if measurements are to be made (e.g. leg lengths or carapace widths as measures of spider size). It is better, therefore, to restrain them using one of the following methods.

Spi-pot

This holding device (Roberts 1995) is made out of readily available household materials. It can be made large or small depending on the size of cup chosen. Two identical plastic or waxed paper cups are modified as shown. The bottom is cut out of pot A and pot B has a disc of expanded polystyrene or similar glued to the base. The cut end of pot A is covered with cling-film. A spider is placed in pot A (inverted from the position shown) and pot B slid in to trap the specimen



Spi-pot; reproduced, with permission, from Roberts (1995) against the cling film. One disadvantage is accommodating the Spi-pot under a stereo microscope, unless the pots used are very small. Spi-pots made out of mini-milk cartons (often used by hotels) are ideal for this purpose and also convenient to take into the field (see illustration in Bee, Oxford & Smith 2017). A variation on the original Roberts Spi-pot, using a rigid toothpaste tube, can be found here: www.tombio.uk/sites/default/files/Spi-pot_0.pdf.

Plastic bag

This is the simplest device of all, devised by Phil Baldwin. The spider is placed in a small, clear plastic bag, which is then gently folded over so that the animal is trapped between the layers of polythene. Both dorsal and ventral surfaces can be examined simply by turning the bag over.

Chilling

For a brief examination, specimens can be put in a fridge for 30 minutes or so to slow down their movements. Viewing time is extended if they are then examined on a cold surface e.g. a chilled excavated glass block.

Carbon dioxide

Some wine bottle openers (e.g. Cork Pops Legacy Wine Bottle Opener) inject carbon dioxide into the bottle, under pressure, to push the cork out. Carbon dioxide can be use temporally to knock out spiders for inspection or photography. The nozzle of the bottle opener is inserted into a pot containing the spider and some gas squirted in. The spider is left for a few second until it stops moving and can be tipped out and examined. Initial photographs may look wrong until the spider starts to revive, because only then does it revert to a natural position. Used carefully, mortality should be very low.

4.1.3 Equipment for the examination of dead spiders (see also Section 3.4)

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 3.4.
- 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 (solid watchglass) for holding specimens in alcohol whilst viewing them under the microscope. A clear glass block is normally used, but a black block (or black card placed beneath a clear block, or a block filled with black wax) can be useful sometimes when pale hairs and trichobothria are proving difficult to see. For larger specimens, deeper containers might be necessary.
- 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 (see also Section 3.4.3).
- 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 70% ethanol, industrial denatured alcohol (IDA) diluted with distilled water to 70%, or propan-2-ol diluted with distilled water to 60% (see Section 4.2.1 for more on preservatives). 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.

4.1.4 Use of keys and other literature

A range of specialist literature is available to assist with the identification of spiders. Details are given in Section 10.2. 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, but the Glossary in Chapter 11 may assist. It can also be helpful to have guidance from an experienced arachnologist when initially interpreting keys. The BAS's mentoring scheme is designed specifically to provide tailored advice on such matters and help is also available on a range of short courses arranged in different locations around the country by BAS members, the Field Studies Council and various other organisations (advertised on the BAS website). It is important to be patient when attempting to identify specimens. If you get to an identification that is clearly incorrect, go back to the start of the key and begin 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 by flicking through the diagrams to find those that appear similar in overall appearance to your specimen. Sketching the main features of the palp or epigyne may help in the search. 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.

The BAS is developing a series of guides for the identification of particularly difficult species. Those available so far can be found at: srs.britishspiders.org.uk/portal.php/p/Difficult+species and have also been published as articles in the *SRS News*, the Spider Recording Scheme section of the BAS *Newsletter*.

4.1.5 Clearing genitalia

Some female spiders differ little in their external genitalia (e.g. *Tetragnatha* spp.) but do so in the 'plumbing' of their internal reproductive organs (adnexae). To examine these it is often necessary to clear muscle and connective tissue in alcohol-preserved specimens (see Section 4.2) so that the disposition of the spermathecae and associated ducts can easily been seen. There are several techniques for doing this. Some are permanent so that the external view, as illustrated in Roberts (1995) for example, is not recoverable. These involve immersing excised genitalia or, for small spiders, the entire specimen in solutions of potassium or sodium hydroxide or glacial acetic acid until the occluding tissues are digested leaving only the chitinised structures (e.g. Cooke 1970). A far easier, and fully reversible, method is that described by Parker & Roberts (1972) in which the whole specimen is dehydrated through a series of immersions in different dilutions of alcohol and clove oil. They note that this gradual dehydration and rehydration is important to prevent rapid shrivelling of the abdomen. However, if the genital area is excised it can be moved directly from 70% alcohol to 100% alcohol for an hour or so and then straight into clove oil for about 20 minutes. It is examined from the dorsal side (i.e. from the inside) in a solid watchglass while immersed in clove oil. This technique works very well, even for large species such as *Eratigena* (*Tegenaria*). To reverse the clearing, the tissue is just replaced in 70% alcohol. The clove oil method has major advantages: (a) it is totally reversible; (b) the oil is not dangerous (cf. sodium hydroxide or glacial acetic acid); (c) it is readily available from pharmacists; (d) it has a wonderful smell! The only warning is that it dissolves some plastics.

4.1.6 Reference collections

The value of a reference collection lies in the opportunity to compare problematic specimens with specimens that have been named reliably. 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, forms 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 general collections of spiders, for example those sampled from a specific site in order to generate a species list. It is important to number each tube (whether in the reference or general collections) so that it can be cross-referenced to your computer database or notebook 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 4.5 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 normal 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 (see Section 4.1.7) before a record is accepted.

4.1.7 Help is at hand

All arachnologists encounter specimens 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 of the Spider Recording Scheme (Section 7.1) 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:

- 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

Advice for sending preserved specimens in the post

Post Office regulations regarding sending preserved specimens through the post within the UK are provided on their *Prohibited and restricted items – advice for personal customers* web page (personal.help.royalmail.com/app/answers/detail/a_id/96). Under 'alcohol' it states: ***Alcoholic beverages and liquids containing more than 24% but not more than 70% alcohol by volume (ABV)*** *(e.g. gin, rum, vodka, whisky) is permitted*. This appears to be straightforward. However, 70% ethanol (and other alcohols) are bought bearing a flammable symbol, which means it is **not** permissible to send it through the post. Zoë Simmons (Oxford University Museum of Natural History) recommends that as much liquid as possible is drained off, the tube is plugged with an absorbent cotton wool bung (which can be wrapped in tissue to prevent entanglement) and then packaged from there. The bung will take up any excess liquid but preserve a saturated atmosphere which should persist while specimens are in the post. This often means placing the bung quite close to the specimens so as to prevent them rattling about, hence a layer of tissue can help.

Specimens should be placed in polypropylene tubes, protected in bubble wrap, sealed in a leak-proof bag and sent in a padded envelope, together with a covering letter and return postage. Tubes should not be sent in an ordinary envelope; these may split or burst if any bulky items are contained. If you are asked about the contents of the package by Post Office counter staff the best responses would be 'plastic tubes' or 'museum specimens'.

If polypropylene tubes are not available and specimens have to be sent in glass tubes, additional protection is needed. Glass tubes, wrapped as above and in a sealed polythene bag, should then be put in a rigid box or tin padded with tissue or further bubble wrap.

Advice for sending live specimens in the post

Post Office regulations for sending live creatures, insects and invertebrates in the post can be found at: www.royalmail.com/sites/default/files/royal-mail-prohibited-and-restricted-items-nov-23-2018---23410530.pdf. These state that: 'live creatures are accepted on Royal Mail services only. Prohibited by Parcelforce Worldwide (including fish fry, coral, bees, spiders and some other insects). Packaging guidelines: must be boxed and packaged to protect the creatures, our staff and our customers from harm. Use 1st Class as the minimum service. Items must be clearly marked "URGENT - LIVING CREATURES – HANDLE WITH CARE".'

For spiders, it is best to send them in plastic specimen tubes (of an appropriate size for the specimen) with a few small air holes in the lid. Include a very small piece of just-damp tissue in the tube for moisture and let the spider settle in for a day or two in order to spin some web. This will cushion it during the journey through the post.

4.2 Killing, Preservation and Display of Specimens

No arachnologist enjoys killing spiders, but it is often necessary for reliable identification and should be done as quickly and humanely as possible. Once killed, the spider's body needs to be preserved for long-term storage. For specific purposes (see below), the specimen might need to be fixed (to denature proteins) before moving to a different, preserving liquid.

4.2.1 Killing spiders

One way of killing spiders humanely is to put them in a freezer, where they will slowly cool down and die, before adding preservative. Another is to store the preservative in a freezer where the alcohol-based ones, at least, remain liquid. Pouring preservative at -20°C onto a live spider will kill it instantly.

4.2.2 Types of preservative

Alcohols

Alcohol (ethanol), diluted with de-ionised water to about 70%, remains the best and most reliable medium for killing, fixing 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 HM Revenue & Customs. Obtaining such authorisation needs only to be done once and is not difficult (see below). 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 60% 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 distortion of the image seen down the microscope as the liquids have different refractive indices.

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 sometimes-suggested addition of 5% glycerine to the alcohol as an insurance against complete dehydration is not recommended. A secondary problem is that when specimens are killed in alcohol, there is a tendency for the legs to contract and obscure the underside. However, limbs can usually be manipulated out of the way in order to examine other features.

For large spiders preserved in 70% alcohol at room temperature, decay may set in before the alcohol fully penetrates the specimen, leading to a burst abdomen. This can be overcome by storing the newly-preserved spider for some time at -20°C, which prevents decay and allows the alcohol time to fully fix the tissues.

To preserve material in a way that allows for future DNA extraction, 70% ethanol (dilution with de-ionised water) is appropriate although 100% ethanol is even better. However, the latter makes specimens brittle and more difficult to examine for morphological characters. IDA, which contains methanol, should not be used. Dried specimens are often as good as ethanol-preserved material, so if a leg (or even an exuvium) is kept dry, DNA should be extractable.

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 coloration, 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 (although not as fast as when using alcohol at -20°C, see above). The material is 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: 10 ml propylene glycol, 1 ml propylene phenoxetol, 89 ml distilled water. The phenoxetol and propylene 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) can 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, as mentioned above, the receipt and use of IDA requires authorisation from the National Registration Unit of HM Revenue & Customs. Further information can be found in their Excise Notice 473: *Production, distribution and use of denatured alcohol*. This document can be downloaded from their website: customs.hmrc.gov.uk (Type 'denatured alcohol' into the search box to find the relevant page). Alternatively, you can phone 0300 200 3700 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.

Some suppliers (e.g. Solmedia Ltd: see section 9.1) can supply small quantities of IDA and deliver it by courier.

4.2.3 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 sclerotisation 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, a technique known as double immersion; this is still standard practice in museums because plastic tubes have a relative short shelf life. For personal collections, however, it is common to employ polypropylene tubes with push caps in place of glass, which are reliably airtight and do not risk loss of specimens to evaporation of alcohol over long time periods.

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 (for example, those sold by Sarstedt). 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.

Traditional, slender glass tubes (12 × 50 mm and 25 × 75 mm) are commonly used because of the high visibility of both labels and specimens. Indeed, despite the utility of plastic tubes (see above) the general trend to use less plastic may well herald a renaissance in the use of glass. For glass tubes, the most satisfactory polythene closures are those with 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.

Double immersion

When glass tubes with plastic caps are used to store specimens, a proportion of tubes always allow evaporation and, unless the tubes are stored inside jars that are also full 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 make tight seals when the clamp is closed.

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. 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, with data labels giving full information about where, when and who collected a particular specimen/sample; 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). Laser 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 one on every permanent slide. Data on each label should include the minimum of: locality, grid reference, habitat, collector's name, date and serial number if indexed; it is bad practice to confine information about the specimen to notebooks or databases through a system of reference numbers, because the notebook may become separated from the collection and lost. 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 should be stored in separate tubes.

Storage space

The space available for storage may be used most efficiently by splitting the collection into two parts: 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 around 2000 tubes. Cabinets designed for storage of entomological specimens are available (see, for example, www.watdon.co.uk/acatalog/entomological-cabinets.html), but are very expensive. A cabinet 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. The collection should ideally be stored in the cool and dark, and at low humidity.

4.4 Cataloguing and Record Keeping

Notebooks or their equivalent

The collector will probably find it useful to maintain at least one field notebook or use a digital equivalent. A primary notebook or digital recording system 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. In addition, mobile phones and other smart devices that include GPS are able to collect environmental data linked to photographs of the animal and/or habitat *in situ*. Although every arachnologist will have their own favoured style, field recording 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 available.

In addition to the field notebook, a loose-leaf file, which includes a section indexed by genus, might 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. Recording is usually achieved using databases.

Databases

Specimens should be easily retrievable by virtue of arrangement, notebook, or computer. Simple spreadsheets or databases (e.g. Microsoft Excel) 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 Spider Recording Scheme and the Harvestman Recording Scheme encourage the use of the Mapmate biological recording software package for the collection and submission of data (see Section 7.1). 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 (for more information see Section 7.1).

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 them, 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. It is important to plan ahead and make appropriate arrangements for the future of your collection by contacting potential recipient establishments at an early stage.

4.5 Important British Arachnid Collections in the UK

Janet Beccaloni

4.5.1 Introduction

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

4.5.2 London

The Natural History Museum's (NHM) history began with Sir Hans Sloane, a London physician and a very wealthy individual, who assembled the largest-ever collection of natural history specimens, coins, medals, books, manuscripts, and artfacts, by anyone in Europe (Stearn 1998). After his death in 1753, Sloane's collection was bequeathed to King George II for the nation. This collection formed the basis of the British Museum which was originally located at Montagu House on the site of the current British Museum in Bloomsbury. In the 1850s, it was recognised that the building was too small to house the ever-expanding collections, so the natural history collections were moved to a purpose-built museum in South Kensington, which was opened in 1881. The NHM now contains 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, amongst others, Joseph Banks, Alfred Russell Wallace and Charles Darwin.

Arachnida collections

The NHM Arachnida collections are housed in purpose-built, environmentally controlled store rooms in the Darwin Centre. They are preserved mainly in spirit (industrial denatured alcohol), but there is also a large number of microscope slides (mainly mites), together with drawers of dry, pinned material, and frozen tissues of a few species in the Molecular Collections Facility (MCF). The spirit collection is strong in worldwide material, particularly areas such as West Africa (collections hotspots) that reflect former British colonisation, with a large volume of type specimens and historic material. Because of this historical emphasis on global coverage, there was, until recently, no specific separate UK collection, and all incoming UK arachnid material was incorporated into the main collection.

When the NHM's Angela Marmont Centre for UK Biodiversity (AMC) was opened in 2009, the focus shifted back onto UK collections. The dry, pinned UK synoptic collection in the AMC is designed to assist with identification but only a few drawers of dried Arachnida species are available. From 2011, a UK spirit collection (based around the A. R. Jackson collection from the 1940s) has been developed. This new collection (comprising approximately 690 jars) contains the vast majority of species on the current BAS checklist of British spiders (see Section 2.3), together with around 30 jars of UK harvestmen (Opiliones). It is arranged into families, with genera and species in alphabetical order. The collection is currently being databased to specimen level and given barcode labels by BAS member Bill Parker. These data will be made available via the NHM's Data Portal. The frozen Arachnida tissues collection in the MCF is also being developed through the UK DNA barcode project, where specimens are now being collected for sequencing. The aim is to have fresh tissue samples from every UK species, which will be available to researchers around the world.

4.5.3 Oxford

The Oxford University Museum of Natural History (OUMNH) was established in 1860, when a purpose-built neo-Gothic museum building was completed to bring together the University's various natural history collections. The Hope Department, containing the entomological and arachnid collections, was founded by the English entomologist Frederick William Hope in 1849 via deed of gift and the specimens transferred over when the building opened. The first Hope Professor and curator of the collections, John Obadiah Westwood, held his inaugural lecture in 1861. The collections contain many types and important historic specimens from famous entomologists and arachnologists of the day. These collections have been built on over the years and they continue to grow with the addition of modern research materials from around the world.

Arachnida collections

The Hope Department is famous for possessing the Reverend Octavius Pickard-Cambridge collection of Arachnida. There are about 5000 jars of all sizes, many holding from two to twenty separate tubes. This important collection contains many hundreds of type specimens, not only those described by Pickard-Cambridge but also by a considerable number of eminent specialists such as E. Simon, L. Koch and T. Thorell. There is some material from F. O. Pickard-Cambridge, nephew of O. Pickard-Cambridge, included within the collection, which was presented in 1917. Additionally, there are the remaining specimens of John Blackwall's collection, which was rescued by O. Pickard-Cambridge after Blackwall's death and presented to the museum along with his own material.

The British reference collection of Araneae has recently been recurated and rehoused. All British materials have now been amalgamated and the collections rearranged to the 2010 edition of the online OUMNH catalogue: oumnh.ox.ac.uk/collections-online#/search. Eighty five percent of British species are represented in this collection and are available to researchers through loan or visit. The collection continues to grow, and the museum actively solicits new donations.

4.5.4 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 Alfred Waterhouse and opened in 1890.

Arachnida collections

The Manchester Museum has a very comprehensive collection of British spiders (over 100,000 specimens of 613 species) and harvestmen, which includes the following notable individual collections:

- H. W. Freston reference collections of spiders: 273 species, 2925 specimens; mostly from around Greater Manchester.
- L. A. Carr spider collection: 263 species, 7188 specimens; from Lichfield.
- D. W. Mackie reference collections of spiders: 436 species, 4535 specimens; mostly from Cheshire, but also includes specimens from Ireland and Scotland.
- G. H. Locket spider reference collection: 543 species, 8684 specimens; from various places in the UK, but the collection includes some specimens with no/poor data labels.
- A. La Touche spider collection: 570 species, 15,799 specimens; from various places in the UK and some from France.
- J. Crocker spider reference collection: 498 species, over 40,000 specimens; mostly from Leicestershire. Brief information on this collection was published by Logunov (2004).
- E. Duffey spider reference collection: 560 species, 12,581 specimens; from various places in the UK.
- R. Allison spider reference collection: 149 species, 454 specimens; from various places in the UK.

- J. A. and F. M. Murphy world spider collection: 3063 species, 45,415 specimens, including 579 British species from various places in the UK.
- R. Jones spider collection: some 4000 sample tubes of a, yet unknown, number of specimens and species (both British and foreign).
- British Harvestman collection (of various authors): 24 species, 360 specimens (materials by D. Mackie, H. W. Freston and D. V. Logunov).

The Manchester Museum also contains a substantial number of overseas spider collections (currently 788 species, 8604 specimens), particularly from the Mediterranean, France, European Russia, Siberia, and some Neotropic materials, collected by A. Russell-Smith, E. Duffey, D. V. Logunov, Yu. M. Marusik, F. and J. Murphy and others. As well as the arachnid collections, there is an archive of material relating to such notable figures as H. W. Freston, R. Jones, E. Duffey, G. H. Locket, J. Murphy and J. Crocker.

Dr Dmitri Logunov, Curator of Arthropods, is happy to deal with any spider-related enquiry, to help with identification and/or to loan spider specimens for research (email: dmitri.v.logunov@manchester.ac.uk). The Manchester Museum's arachnid collection is fully accessible for anyone who is interested in studying any of the deposited specimens; it can be searched online at www.museum.manchester.ac.uk/collection/entomology.

4.5.5 Liverpool

World Museum is part of National Museums Liverpool and was founded in 1860 as the Liverpool Free Museum. The original collection was formed from a bequest by the 13th Earl of Derby and has grown rapidly over the years with major donations from private collectors such as Joseph Mayer. It comprises extensive, world-wide, natural science, physical science, antiquities and ethnology collections.

Arachnida collections

World Museum's arachnid holdings underwent a period of rapid development in the 1990s and early 2000s and include one of the finest contemporary collections of British spiders. The collections of most other British arachnids are comprehensive, although mites are typically less well represented. The collection is housed in a purpose-built spirit store and is easily accessible for visitors by appointment. Work on an online catalogue of British spider holdings is under way. World Museum enjoys very close links with the British Arachnological Society and administers its reprint library.

Spiders & harvestmen. British spiders and harvestmen comprise approximately 160,000 specimens and provide around 85% species coverage. The W. E. Falconer collection was principally from northern England and was 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. The museum also has some West Palaearctic material.

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 is the most extensive pseudoscorpion collection in Britain.

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

Ticks. There is also a collection of *c*. 500 medically important ticks, which was originally held by the Liverpool School of Tropical Medicine.

4.5.6 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 established 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.

4.5.7 Edinburgh (University)

The University of Edinburgh was founded in 1582 and is one of the largest in the UK.

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.

4.5.8 Edinburgh (NMS)

The National Museum of Scotland, Edinburgh, was formed in 2006 with the merger of the new Museum of Scotland, with collections relating to Scottish antiquities, culture and history, and the adjacent Royal Museum (so renamed in 1995), with collections covering science and technology, natural history, and world cultures.

Arachnida collections

The National Museum of Scotland holds a substantial arachnid collection comprising 160 boxes, with up to 30 lots in each, and mostly British. From older statistics there are apparently some 400 British species but they have a synoptic collection with almost all species.

4.5.9 Warrington

The Museum and Art Gallery at Warrington was founded in 1848. The natural history collections largely 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 an 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, chiefly reflecting 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.

4.5.10 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
- Yorkshire Museum, York

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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 wanting to photograph spiders. Anyone wishing to go into more depth should refer to a good book or go on a course on macro-photography, which will include many techniques suitable for photographing the smaller spiders. The social media (e.g. the BAS Twitter feed @Britishspiders) can also be an excellent source of up-to-date inspiration, ideas and advice on macro-photography. The skill required to find your subject is also not covered in this chapter, 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 chapter deals only with digital photography.

5.2.1 Camera format

A huge variety of digital cameras are available (including the increasingly high quality cameras in mobile phones and tablets), 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. In general, what is most useful for spider photography is a camera with fast autofocus and some image-stabilisation facility. Increasing numbers of cameras are available with LCD screens that swivel and tilt away from the camera body; this is a particularly useful feature for photographing invertebrates on the ground or in relatively inaccessible places.

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) and their weight and bulk, which make them cumbersome to use.

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. They are also lighter and less bulky than medium-format cameras.

Compact cameras

This is the growth area in digital cameras (and increasingly in phones and other smart devices). Small-format cameras are not ideal for spider photography because they tend to be either point-and-shoot or very specialist. The final image size varies depending on the resolution: the number of pixels the image sensor captures to produce the image. They range from three megapixels (mp) up to around 12 mp, with some models having even higher resolutions.

Bridge cameras

These cameras have a relatively large image size (12–16 mp), a variety of built-in controls for exposure etc. but, most importantly, they have a fixed (i.e. non-interchangeable) lens. This is usually a long zoom lens from 5× often to over 50× zoom with a macro facility. Many are capable of close focusing, often down to 1 cm from the subject, with accurate focusing achieved through a magnified live-view screen. Some have pivoting and tilting screens that allow images to be taken from a much greater variety of angles. 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 electronic viewfinder, but the majority also have an LCD preview screen on the back of the camera. Advantages include a reasonable image quality (often including RAW shooting), a fast lens (with none of the bulk and inconvenience of multiple lenses), image stabilisation, and relative affordability compared with digital SLR (DSLR) cameras. The main disadvantage is that you are limited to only one lens.

Recommended camera

For versatility and portability, the best option is the interchangeable-lens DSLR camera. However, when you factor in cost of lenses, DSLR cameras are expensive and, if your budget is limited, you might think of purchasing one of the better quality bridge cameras, at least initially.

5.2.2 Lenses

Due to their small size, spiders are best photographed using a specialist macro lens (one that produces an image on the sensor at least life size). However, there are other, less costly, alternatives available depending on your subject matter.

Standard prime lens with accessories

Using a standard prime lens (e.g. in the region of 50 mm with a 35 mm camera) is probably the cheapest option. The standard lens is perfectly acceptable for photographing webs, but it cannot focus close enough to photograph the spiders themselves in any detail. There are three methods of achieving the close-up effect with a standard lens; however, once the close-up attachment is in place, the lens will no longer focus on distant objects.

Close-up lenses. These are magnifying lenses that 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, thus moving the lens further away from the sensor and allowing it to focus more closely. Extension tubes have no optical elements, so do not degrade the image. Extension tubes come in a variety of fixed lengths, but there is also a variable system that uses a concertina of opaque material known as a bellows. Most cameras lose most of their automatic controls when used with bellows.

Reversing the lens. Macro photographs can also be achieved by using a mount that enables the lens to be attached to the camera (or another lens) in reverse so the front points towards the sensor although most cameras will lose their automatic controls under these circumstances.

Macro-zoom lens

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

True macro lens

A true macro lens is able to produce an image on the sensor that is life size (or bigger), and focus to infinity. This is really the minimum requirement for spider photography and the ideal is a lens reaching four times life size. However, this can be achieved by adding extension tubes to the macro lens (although it will then lose its ability to focus to infinity).

Recommended lens

For larger spiders (approximately 5 mm body length or longer), a true 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 extension tubes is advisable, to give images larger than life-size. The high resolution of many cameras now available means you can enlarge parts of the photograph when processing and achieve images many times larger than life.

5.2.3 Accessories

Close-up photography can benefit from a number of accessories. The most important is probably a flashgun or some other additional lighting. This is because with close-up work the small distances between camera and subject can mean insufficient light, even with a wide lens aperture. In addition, close-up work also generally has a shallow depth of field. The depth of field can be increased by using a small lens aperture, but this reduces the amount of light reaching the sensor to form the image.

Lighting

Photographing in natural light often yields the best images with respect to a faithful colour balance and a lack of distracting highlights, such as can be produced by flash. However, on dull days and/or where the camera is very close to the subject, there may not be enough light to be able to use small apertures and thus achieve a sufficient depth of field. Under these circumstances, artificial lighting is required.

There are a number of different types of lighting. Use of floodlights or LED panels has the advantage of seeing 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 convenient in the field. Second, floodlights heat up the subject, which makes the spider more active and, in many cases, more likely to move away from the heat source.

Most cameras have a built-in flash. This can work, but creates very dark shadows, and the flash lighting can be obscured by the front of the lens when the subject is close. Similar problems are associated with a single flashgun mounted on the camera's hot shoe. An arrangement of multiple flashguns gives a better lighting effect, with reduced shadows and a more 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 (see below). 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- or twin-flash system, which mounts on the filter ring of the lens with the flash tube either side of, or surrounding, the lens. The two disadvantages of this are, (a) almost completely shadowless lighting giving a very flat effect (although this can also be an advantage in certain circumstances and in some twin-flash systems the light outputs can be varied for each flash tube), and (b), when using a ring flash, any reflections of the flash can appear as tiny doughnuts rather than just dots, which can be distracting.

Camera support

It is usually 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. For spider work it is often necessary to fix the camera at almost ground level. This can be achieved either by reversing the centre column or by using a versatile tripod which allows the centre column to point in any direction. A small beanbag or cushion can also be used to rest a camera on and help hold it steady.

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 in one place. Often spiders sun themselves on leaves, walls, and other surfaces. In this position, they should be approached slowly because they will, when warm, rapidly escape if disturbed. Also, take care to avoid shadows from yourself or the camera falling on the spider. A number of other factors, such as wind, rain, inadequate lighting, hand shake, or distraction by other invertebrates (flies, wasps, etc.) can combine to make photography difficult. There are a number of techniques and pieces of equipment that can be used to alleviate some of this.

Many spiders are active at night (spinning webs, hunting prey, and seeking mates) and much behavioural activity can be photographed or videoed. The majority of modern cameras, including smart phones cameras, have a video facility, often of very high quality. Again, lighting is the all-important factor. For night photography and for video it can be achieved using suitable LED lamp systems (low power consumption and minimal heat output).

Camera settings

As mentioned above, the best option for spider photography is to use a DSLR camera and the notes here refer to these cameras. Using the autofocus setting in the field does not necessarily bring key areas into focus, and so manual focusing should usually be used because it provides more control. When using electronic flash, the shutter speed and lens aperture should be set manually. The shutter speed will be dictated by the requirements of the flash unit (usually no higher than 1/250th second), whereas the flash duration will be very much briefer and will occur at the mid-point of shutter opening. For most shots using flash, an ISO value of 100 is ideal. However, the sensitivity to light can be increased on most modern digital cameras by adjusting the ISO setting without much degradation of image quality (refer to your camera manual). For macro-photography lens apertures of f11–f32 are appropriate, depending on the depth of field required (see also Section 5.3.3). If you are getting a softer image when using your lens with a small aperture, try opening up the aperture to f11 or f16. For more information on these issues, refer to a technical manual or search online.

With through-the-lens metering, the camera and flash unit combine to compute the length of flash duration required for your particular shutter speed and aperture setting. As mentioned previously, some modern DSLR cameras (and most bridge cameras) have an image stabilisation feature and this can also be helpful. Again, refer to your camera manual for details.

Controlling camera and subject movement

A tripod or 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-lockup facility (if you have one) can help to control vibrations transmitted via the shutter release button and mirror movement respectively. Your camera operating manual should give you details on how to set this up. A high shutter speed is a way of freezing subject movement, but this may not be an option as you may need a relatively 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 stem or twig in place with a small cane or knitting needle and Velcro ties. When taking your photo make sure that you do not have any stems going across your subject. It is easy not to see them in the viewfinder when you are focusing on the spider.

5.3.2 Photography in captivity

It is often not practical to photograph spiders in the field, in which case specimens can be brought indoors to be photographed. Most techniques used indoors are similar to those utilised 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 to focus more accurately. Again, electronic flash can be used to good effect and does not have the problem of heat build-up, which 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.

A very shallow depth of field is often a problem with spider photography. However, new computer software is now available to overcome this. Multiple frames of the subject are taken, each focusing successively further from the camera in minute increments. Changing the focus incrementally can be done either with computer-driven software or by hand. These photos are then processed into one image on the computer using software such as Adobe Photoshop CS6 to CC2020, Helicon Focus or Zerene Stacker. The result is a single image where the spider is in focus throughout the depth of its body. Generally speaking, the more frames that are taken the better the final combined image. This technique, known as photo-stacking is much easier to control when the spider is in captivity. Some cameras have photo-stacking software build in. For example, in macro mode, the Olympus Tough series can take a number of images in different focal planes, which it then combines into a single image. This works well, even in the field, if both the camera and subject keep very still. Using the 'focus bracketing' option, this camera (and, no doubt, others) can also take up to 30 images in incrementally changing focal planes. These are stored as separate files, which can then be downloaded to one of the stacking packages mentioned above. Some digital microscopes, at the more expensive end of the range, can also stack images (e.g. the Dino-Lite EDOF models).

Photographic processing software packages are constantly being updated. Their use is not addressed here but details can be found in many books on digital imaging. Adobe Photoshop Elements and Adobe Lightroom are very useful software packages, widely used and relatively cheap to buy.

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

Very high precision in focusing can be obtained by the use of focusing rails mounted on an optical bench. Such a bench can be made from a thick piece of plywood with rubber feet screwed underneath. Focusing rails are also useful when using bellows extensions on your camera for high magnification. Bought focusing rails can be automated and used to take multiple images for photo stacking (e.g. UltraMacro Auto-Rail or Cognysis StackShot). 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 eventually settle down to rest. Active species, such as *Eratigena* (*Tegenaria*), can be tired out by running them round an arena, made from a cut-off plastic food pot, with a pencil behind them. When passive, the arena can be removed and the spider photographed. This is really only suitable for specimens that naturally occur on flat surfaces.

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

5.4 Photography down a microscope

To photograph specimens at high magnification, and even individual features such as the epigyne or palp as an aid to identification, requires more than a standard camera or even macro lens. Using a microscope can give this extra magnification and adequate pictures can be achieved quite cheaply.

There are three main methods of photography down a microscope (photomicrography), which differ in the number of lenses used.

5.4.1 Three-lens set-up

Although utilising the most lenses, this is the simplest and cheapest method, 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 or mobile phones to be used, but the results, while acceptable, are rarely as good as those from the one- or two-lens systems.

There are several important factors to be taken into account:

- The camera must be mounted rigidly with the microscope. This can be achieved using either a rigid tripod or a direct attachment between the microscope and the camera; the latter is essential if the microscope body moves when focusing.
- 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 focused to infinity. All focusing 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 DSLR cameras; it is difficult to use a non-SLR camera because you do not see through the lens and so cannot frame or focus the photograph. Another problem for the three-lens set-up is the fact that DSLR cameras have a relatively large lens, so will gather very little light from the eyepiece of the microscope.

5.4.2 Two-lens set-up

This set-up requires an SLR camera with a removable lens. The camera body is attached to the eyepiece by a rigid tube, which is normally available as a standard microscope accessory.

5.4.3 Single-lens set-up

This is basically the same as 5.4.2, 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 (unless you are using a micro lens), so changes in exposure can only be made by altering the light intensity. The microscope transmits very little light when compared with a normal camera lens, so the subject must be well illuminated. For reflective lighting, some method of reducing shadows is essential, using either a second, slave flashgun or a reflector. Otherwise, shadows can be absolutely black and distracting. For live specimens, floodlights are probably too hot for use in photomicrography. This also applies to specimens under alcohol where the heat can cause distracting convection currents, which distort the image. Fibre-optic and LED cold light sources are a much better solution.

5.4.5 Conclusions

There is little to choose between the single- and dual-lens set-ups for photomicrography. 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; good results are then 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. Many books and websites are now available that give more detailed guidance on this subject.

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6 KEEPING SPIDERS IN CAPTIVITY

Bill Blumsom

6.1 Introduction

This section is aimed at those who wish to keep spiders in captivity at home, whether for study of behaviour and life-history, photography, or simply to raise specimens to maturity for identification purposes. The information and suggestions are based on my own 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 BAS *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 the development of 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 articles on large house spiders *Eratigena* (*Tegenaria*) in the 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; see also Oxford & Smith 2014a,b, 2015).

It is simple and easy to create spider-friendly habitats in a garage or shed with carefully placed air-bricks and flower pots or simply a pile of newspapers or old bird's 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, or 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 where 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 bred successfully. The shed, with carefully placed air-bricks, flower pots and corrugated cardboard, has plenty of *Amaurobius similis*, *Steatoda bipunctata*, *Harpactea hombergi*, and *Eratigena* (*Tegenaria*) spp. in residence. The outside of the property and sheds can also be adapted to encourage more spiders into easily observed locations. Provision of assorted tubes, crevices and retreats at appropriate heights can make night-time observation and photography of species such as *Nuctenea umbratica* and *Zygiella x-notata* much easier. At night, it is possible to observe spiders in a completely natural state with a hand or head 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 spider's habitat requirements before keeping it in captivity will help to recreate suitable conditions and increase the chances of maintaining the spider successfully. If you are unfamiliar with a spider, it is worth taking note of the habitat and conditions in which it was found. 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. Observe whether the spider is 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 those of a spider taken from woodland or leaf litter.

If possible, use some of the natural material from the collection site in the cage at home, but be careful to minimise any damage to the location. It is also valuable to study the literature and websites to find out as much as possible about the species and its general habitat and feeding needs. Remember that permission is required before removing spiders or other material from private land and/or protected sites (see Section 3.3).

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 objectives for keeping spiders vary but, generally speaking, fall into three categories:

- Immature specimens to be kept until they reach maturity for identification purposes (see the use of restraining devices for identifying immatures: Section 4.1). These are normally kept for relatively short periods in tubes or small pots with just a little cover as they will be identified as soon as they complete the final moult. If it is a large spider or a web-builder, ensure the container allows enough room for the final moult to take place. It is worth feeding immatures a little more frequently because 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 imply more elaborate housing and care in creating a good balance between the wellbeing of the spider and ease of observation.
- Photography (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 are the most satisfactory in terms of the 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 and possibly damaging webs (this is clearly preferable)?
- Is the container adequately ventilated?
- Does it provide good views of the spider?

Frances Murphy had some useful suggestions in this respect (Murphy 1980). She recommended well-ventilated plastic vivaria, available at most pet shops, and also the storage boxes made of strong plastic and in a wide range of sizes and shapes sold in many DIY outlets. A myriad containers are, of course, obtainable online to suit different requirements and budgets. 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 centimetres to over 1 m 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 to re-create specific habitats and establish species for longer-term study and breeding.

Small containers for rearing spiders to maturity can vary from specimen tubes to yoghurt pots, jam jars, or small cardboard boxes. Use fine netting or old net curtain as the lid, held in place by an elastic band. This enables air to circulate and prevents the container getting too wet and humid. Murphy (1980) recommended black netting wherever possible, because this is easier to see through (although harder to obtain), and using string rather than elastic bands to hold the netting in place since elastic eventually perishes and breaks, allowing spiders to escape. She also suggested a simple solution to provide access for feeding: 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 *Eratigena* (*Tegenaria*) species, which require little or no water and are large spiders that remain within their funnel webs much of the time, she used cardboard shoeboxes as containers. Successful alternatives for these spiders include glass vivaria, plastic sweet jars, 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 you remove a lid make sure it is replaced 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, entomological equipment suppliers also provide a wide range of containers that 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 as spiders overheat and die very quickly. For further information on vivaria, see Bruins (1999).

6.6 Creating the Habitat

The next step is to try to recreate the natural habitat as closely as possible and to strike a balance between the requirements of the spiders and the need to 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 dusk and search the container for prey, providing good views of their hunting behaviour. More details about these are provided below in the section on wandering spiders.

It is important to control the humidity in the container, and moss is ideal 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 easily be adjusted to suit the requirements of different species and it provides excellent cover for the spiders as well as their potential prey. Cotton wool can be used but tends to become mouldy. It is worth keeping humidity gauges in the larger containers and maintaining humidity within the normal range. Relatively inexpensive gauges are available 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 webs and refuges for those spiders that do not sit in the web itself. Yew and holly stay fresh and do not wilt for a long time. Wherever possible, have branches at either end of the container for web attachment, leaving 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 it. 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 in avoiding 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 Emma Shaw, is to grease the top couple of centimetres 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 substrates can be used, from sand or gravel to soil planted with, for example, ivy, grass, or deadnettle. Again, try to make the ground cover appropriate to the area in which the spider was found. 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 with sand or soil on top, to avoid the upper layer getting too wet and soggy.

Whenever in the field, it is worth collecting any useful materials seen, even if they are 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 ideal for keeping *Segestria* and *Amaurobius*. Air bricks 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 orbweb builders are essential, as are fresh sphagnum moss and heather (landowner's permission required, and consent from Natural England/Natural Resources Wales/Scottish Natural Heritage on protected sites).

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 in advance of introducing the species of spider you wish to study. This gives the habitat a chance to settle down and mature to become a living environment in which to house and study your specimens. This has proved particularly effective with *Tegenaria silvestris*, *Agelena labyrinthica*, *Pisaura mirabilis* (Cover, bottom right) and ground dwellers such as *Coelotes* and *Pardosa* species.

6.7 Feeding and Providing Water

Other than 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 food to provide, 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 vary enormously between species and their stage of maturity.

Before attempting to feed a spider, it is usually best 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 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 food. The type of prey taken by web builders varies with the type of web they build. Orb webs are designed to catching flying prey, sheet webs tend to catch crawling or falling prey, whereas vertical trip-wire webs or sheets, such as those of *Amaurobius*, 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 readily eat it.

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 than approximately one-sixth that of the spider" (Bristowe 1939).

However, there are some notable exceptions to this. The Walnut Orbweb Spider *Nuctenea umbratica*, a common spider around sheds and fences, has taken a Peppered Moth, seven or eight times its own size above my moth trap and its venom has paralysed the moth in less than one minute. Others that will tackle larger prey include *Thomisus onustus* (Cover, bottom left; Fig. 22), a crab spider that ambushes insects in flower heads, and *Zygiella x-notata*, a common spider around window frames. The daddy long-legs spider *Pholcus phalangioides* (Fig. 4), which uses its long legs to hold and wrap prey, can tackle and overcome fully grown *Eratigena* (*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, because of the nature of the snares and prey-capture techniques used by *Theridion* and *Dictyna*, these genera will readily tackle prey many times their own bulk, whilst *Tetragnatha* and *Linyphia* rely on prey smaller than themselves (Bristowe 1958).

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

6.8 Finding Prey

A variety of methods is available, from running a light trap on suitable evenings, to using a sweep net or hand collecting with a pooter (Section 3.1), 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; 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.

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. Because of the risks of introducing diseases and parasites through the invertebrate food trade, great care should be taken to avoid the escape of bought prey into the wild. Spiders fed on species bought through the pet trade should not be released to the wild.

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. Smaller maggots (Pinkies) turn into greenbottle flies whereas the larger, normal maggots become larger blowflies.

Fruit fly cultures are commercially available and, if looked after and kept at the appropriate temperature, will provide a ready supply of small flies throughout the year. Better still, you can start your own culture by putting fruit, such as banana, sprinkled with a few grains of dried yeast, in an exposed container during warm weather. Seal it up after a few days and wait for a colony of fruit flies to emerge. 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. When very young, spiderlings can often be kept together but as they grow they may start to eat their siblings. The larger survivors should subsequently be separated so that they can be appropriately housed and fed. When feeding spiders to other spiders take care, as occasionally there may be a surprise and the smaller and/or more flimsy spider will kill the larger one (see the *Pholcus* and *Eratigena* (*Tegenaria*) example above).

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 dry out a container that has excess moisture. Many spiders need 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 *Eratigena* (*Tegenaria*), *Steatoda* and *Pholcus*.

6.10 Individual Groups

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

Funnel-web spiders (Agelenidae)

These include the large house spiders *Eratigena* (*Tegenaria*) spp. and *Agelena labyrinthica*. If readers are new to keeping spiders, large house spiders are good 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 because of their size are easy to observe. They are hardy, grow large and live relatively long lives (typically 2–3 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 he is fully mature and the female is in her penultimate instar. The young spiderlings can be raised to adulthood successfully with a little care and patience.

Geoff Oxford has written some interesting articles on large house spiders in past BAS *Newsletters* and I have found them extremely useful (e.g. Oxford 2007). Bristowe spent some time discussing these species in his book *The world of spiders*, which is a mine of information on spider's habits and feeding behaviours (Bristowe 1958).

Eratigena and *Tegenaria* species (except *Tegenaria silvestris*) are often 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 windowsills in your shed or garage or behind any pile of reasonably dry and sheltered bricks and you will find young large house spiders. Starting with a juvenile stage means they should live some time, are at a size relatively easy to feed and will provide opportunities to observe development of the web, moulting and growth rates.

As with most spiders, it is best to house each individual separately. They like a retreat, so a house air brick with large holes, a broken flower pot or a tube of some sort in a corner of the container, is ideal. Large house spiders build a funnel web that gets bigger as the spider adds to it over time. The spider will build out from its retreat so ensure the retreat is in a back corner of the container so that the sheet web is built towards the front, giving you clear views of the spider's behaviour. Large house spiders moult by hanging upside down from the web so it is important to ensure there is sufficient room within the container for this.

Once the sheet web has been well established and the spider is settled, you can leave the lid off the container if you wish as the spider will not usually escape, unless it is a mature male searching for a female. However, large house spiders have been known to take over each other's webs and prey upon one another, and so if the top is off the spider you end up with may not be the one you started with. Depending on size, large house spiders 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 appear to be adequate.

Once you have a large almost-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 leisure, when he will make the appropriate signals on the web to announce his presence. Once accepted, the male will often live within the female's retreat for some weeks, guarding her until she matures and then mating repeatedly 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. These larger youngsters can then be moved to 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 providing an overhang, they will do well. They build a small sheet web usually from under the overhanging plant. You will not see them very often though as they tend to remain well hidden during the day.

Agelena labyrinthica is a very interesting spider to keep for observing 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 juveniles because mature spiders will not usually construct a full web. *Agelena* always needs a large container, as the web system 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 prey in the wild.

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

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

For *Amaurobius* and *Segestria*, air-bricks obtainable from builders' yards provide excellent retreats. If the container is sufficiently large and enough 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; this, again, allows easy construction of tubular retreats by the spiders. When setting up 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 simply to 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 nocturnal and, when attacking, bite the leg of the prey before dragging it back towards the retreat. Their prey comprise largely crawling insects. Flies are also taken, and Bristowe noted that, unlike other spiders, *Amaurobius* species will attack and kill pompilid wasps (spider parasites).

Dictyna species are all small spiders, which can usually be collected in their web with the plant head. 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 than 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.

Orbweb 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 for observation. The main requirement for all web-weavers is sufficient space to allow them to construct webs. This is hard to provide in a cage but, if furnished with suitable dead branches or shrubbery, they will often do so. Branches placed at either end of the container for web attachment leave the central portion clear for the web itself. Another option is to place a plant in a large flower pot and build a spacious netting-covered cage around it. This can be placed in the garden and the spider observed at leisure, but remember to leave appropriate access for feeding. Frances Murphy suggested building a cage 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 & 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* and uloborid spiders. They found that *Tetragnatha*, *Meta* and *Metellina* species were more reluctant, but could be encouraged to initiate web building by introducing a fly. They also found that introducing a fly to 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 both 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. Ensure that there are enough flies in the greenhouse 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 the Walnut Orbweb Spider *Nuctenea umbratica*, a nocturnal orbweb spider that is very easy to keep. It likes a retreat where it can spend the day, preferably somewhere narrow in which it can wedge itself firmly, and a largish area in which to weave a full web.

Larinioides cornutus is another orb-weaving spider that spends its day in a retreat in the heads of rushes or plants such as cow parsley. You can remove the retreat with the spider inside and then placed it in a suitable container where the spider will emerge to weave a new web. *Zygiella x-notata* is a species that is active all year round and readily found around houses and sheds where it builds relatively small but distinctive webs, usually with one segment missing. 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 (Fig. 6). 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. *Scytodes thoracica*, 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 less than half their 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, on 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 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 fairly 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 houseplants 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, nursery-web and jumping spiders (Lycosidae, Pisauridae and Salticidae)

All these families are diurnal hunters with excellent eyesight.

Wolf spiders require a great deal of room to thrive and are major predators of their own young, making them difficult 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* (Cover, bottom right; Fig. 15) is a relatively large species that is 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 web, it is possible to watch her build this structure 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 40 cm or more above the ground.

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 that 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 opportunities. Loose bark on branches is important as many of these spiders like to construct a silk cell under bark when not active. Salticids 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* (Cover, middle) species, it is possible to keep several males and females together without them predating each other, so long as plenty of space, suitable retreats and plentiful food are provided. 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 among the easiest spiders to keep and the variety in size and habitat of the different species means there is a range of behaviours to observe.

The Noble False Widow Spider *Steatoda nobilis* is a relatively large species that is easy to keep; females are long-lived, reaching more than four years of age in captivity. They build large, complicated, and often tangled-looking webs with a tubular, gourd-shaped retreat. The webs are designed to catch flying insects and observations of catches in webs in natural habitats suggest that these are predominately moths. They are ferocious predators and always approach prey from under the web, wrapping it before dragging it into their retreat. After they have finished they will wrap the remains and drop the bundle 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 large *Steatoda* species have jaws strong enough to bite and pierce human skin and so should be handled carefully. Fertile egg-sacs can be produced over a number of years after just one mating.

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 constructed rapidly 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. Such observations could form the basis of very informative articles for the BAS *Newsletter*.

This account is only a brief introduction to the subject; for further reading and information, the following references will prove helpful.

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7 NATIONAL RECORDING SCHEMES

7.1 The Spider Recording Scheme (SRS)

Peter Harvey

7.1.1 Introduction

Spiders are a fascinating group of invertebrates that have only started to receive the attention they deserve since the 1950s. 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 (BAS), provided a firm basis for the study of arachnology in Britain. The publication of a photographic field guide by Dick Jones (Jones 1983) and then the massively important modern identification works by Mike Roberts (Roberts 1985, 1987, 1995) provided arachnologists with the tools to identify reliably most species of spider found in Britain, and thus enable the mapping of their distributions. A more recent photographic field guide *Britain's spiders* (Bee, Oxford & Smith 2017) has introduced an even wider range of people to spider identification and recording.

Dr Peter Merrett initiated the mapping of the distribution of British spiders on an administrative Watsonian vice-county basis in Locket, Millidge & Merrett (1974) and periodically published new county record updates in the *Bulletin of the British Arachnological Society* (now *Arachnology*). However, it was the formation of the Spider Recording Scheme (SRS) in April 1987, originally in collaboration with the Centre for Ecology & Hydrology's Biological Records Centre, together with the remarkable enthusiasm and energy of Clifford Smith, which were instrumental in encouraging the active support of arachnologists and increasing the numbers of recorders.

In 2002, the *Provisional atlas of British spiders* (Harvey, Nellist & Telfer 2002) was published, based on data recorded and submitted to the scheme to the end of 2000. Some 1500 volunteers contributed over 517,000 records during the Scheme's first 14 years. The 647 species accounts in the *Provisional atlas* provided much new information on every British spider species. The *Atlas* was a landmark in the development of spider recording in Britain and a great tribute to the many arachnologists involved in its production, from the recorders, to the authors and editors of species accounts and of the volume itself.

Since publication of the *Provisional atlas*, Phase 2 of the Scheme has placed much greater emphasis on the recording of autecological, phenological and habitat management information, as described below. All of this information is made available in species accounts on the SRS website (srs.britishspiders.org.uk), which has superseded the *Provisional atlas*.

7.1.2 Organisation and objectives of the Spider Recording Scheme

The SRS is coordinated by a National Organiser who is supported by Area Organisers (AOs), each of whom is responsible for one or more vice-counties. The AOs receive records submitted from recorders in their areas, usually as Mapmate files. For more information on submitting records see srs.britishspiders.org.uk/portal/p/Recording+spiders. After checking, the data are sent to the National Organiser. Unless further verification is required by other national experts (see Section 4.1.7), records submitted using a MapMate sync file or new records provided in an easily importable Excel spreadsheet template format are added to the national dataset and soon uploaded and made available through the SRS website, where they contribute to distribution maps displayed for each species. Registered users of the site, including all BAS members, can also view full details of the records. For any area not covered by the network of AOs, records are sent directly to the National Organiser, currently Peter Harvey at 32 Lodge Lane, Grays, Essex RM16 2YP; email: grayspeterharvey@gmail.com.

The first phase of the Recording Scheme aimed to:

- Define the geographical distribution at 10 × 10 km (hectad) resolution of each species of spider found in Britain and the Channel Islands (currently around 670 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.
- The establishment of a profile of the ecological characteristics of each British spider species.
- The establishment of a data bank that will form a base line against which future ecological work can be compared and provide quantified information on spider ecology that will aid future research and stimulate new studies.
- The extension of 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 changes over time, and collect and collate records with full dates, sex ratios and habitat details to improve understanding of the adult activity periods and life-cycles.
- The identification of habitats where species richness and/or presence of notable species makes them of special conservation interest and establishing how well these are represented in protected areas.
- The identification of hot spots of spider biodiversity in the British landscape.
- The recording of the spider fauna of selected sites of particular concern to nature conservation, and other areas where habitat potential might be threatened (from time to time the Scheme and the BAS organise surveys of specific sites).
- Leading in the assessment of rarity and conservation status for spiders in Britain.

7.1.3 Newsletter

The Spider Recording Scheme newsletter (the *SRS News*) is issued three times a year, and contains articles and notes submitted by recorders on all aspects of spider recording, as well as regular updates on the progress being made in the recording scheme. Since November 2002, the *SRS News* has been incorporated into the BAS *Newsletter*: all back issues are available for members to download from the SRS website.

7.1.4 The SRS database and website

One of the main aims of the recording scheme is to provide up-to-date information on the distribution of spiders in Britain. By 2019 the SRS database held over 1.1 million spider records. Coverage is good for most of Britain although, not surprisingly, some counties are much more intensively recorded than are others.

The SRS website allows everyone to search and view the distribution maps and ecological information on each species, explore regional distributions, download *SRS News* issues, access guidance for difficult species identification, and create and download individual species reports as PDF files. Any member of the public can register and, when logged-on, submit records for a number of easily recognisable species which are not readily confused with other similar species, upload images or species notes, and use the site's forum to discuss identification questions or observations of interest. Logged-on users who are BAS members or members of the SRS can also view full details of the records.

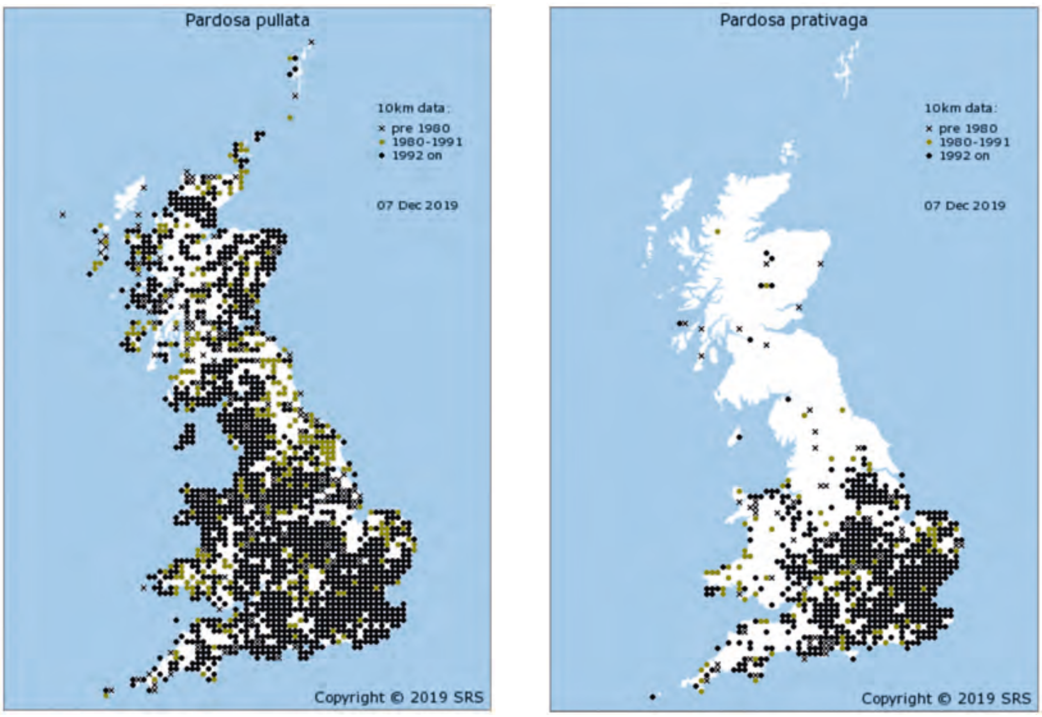
Maps and distribution patterns

The distribution maps for various common spiders illustrate the success of the recording scheme. Species such as the comb-footed spider *Robertus lividus*, the money spiders *Tenuiphantes* (*Lepthyphantes*) *tenuis* and *T. zimmermanni*, the Garden Spider *Araneus diadematus* (Cover, top centre; Fig. 10), the wolf spider *Pardosa pullata* (Map below, left) and the crab spider *Xysticus cristatus* (Fig. 23) are all widespread across Britain. However, the Scheme has also highlighted 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* (Cover, bottom right; Fig. 15), the wolf spider *Pardosa prativaga* (Map below, right) and the jumping spider *Euophrys frontalis* (Cover, middle) are species that are widespread only in the southern half of Britain, becoming relatively scarce further north. The Flower Crab Spider *Misumena vatia* (Fig. 21) and the orbweb spider *Araneus triguttatus* show very pronounced southern and south-eastern distributions, respectively.

Many spiders are associated with higher altitudes and northern latitudes. Good examples include the money spiders *Hilaira pervicax*, *Agyneta* (*Meioneta*) *nigripes* and *Scotinotylus evansi*. Some have western and northern distributions and are absent or scarce in the south-east, such as the cybaeid (formerly 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 *Euophrys* (*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 Lichen Running 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 orbweb spiders *Mangora acalypha* and *Zilla diodia*, and the dictynid *Nigma walckenaeri*, show curious distributions, stretching in a band across southeastern and central southern England and then reappearing in the Severn Vale, although in recent years all three species, especially *Nigma walckenaeri*, have undergone a considerable extension of range.

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. Much remains to be done to fill in gaps



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

and record counties more thoroughly, but we already have enough information to detect changes in the distributions of many species and to stimulate more research into their ecological requirements, habitat management and phenology.

Male/female phenology and habitat data

Phase 2 of the Scheme (post-*Provisional atlas*) has placed much greater emphasis on the recording of autecological, phenological, and habitat management information. The vast majority of the SRS species accounts now provide summary charts with data on adult season for males and females, the habitats in which they have been recorded, and information on occurrence in relation to habitat management. These increasingly comprehensive data are often at variance with published accounts, usually based on small sample sizes. For example, the wolf spider *Trochosa ruricola* is 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 with a much smaller peak in late summer/early autumn.

7.1.5 Current developments

Adult phenology data on the basis of latitude and longitude

The ever-increasing volume of SRS phenology data gives the potential for analyses on the basis of latitude and longitude in Britain (using the 100 km OS grid for northings and eastings). The data already available suggest that, for a number of species, the maturity period changes from south to north and from east to west. As the phenological data set becomes more comprehensive it allows better understanding of the effects of geographical location on adult activity periods and of the impacts on this of climate change.

Spiders and conservation of invertebrates

Arachnids, insects and other invertebrate groups play a vital role in ecological systems and the importance of invertebrates is now well 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 and vegetation topography, affecting features such as microclimate and appropriate scaffolding for web construction. These are the very factors likely to be most influenced by different management regimes, and spiders should therefore be valuable indicators of the success or otherwise of such management and the health of the countryside (e.g. Scott, Oxford & Selden 2006).

The SRS data are used to assess the conservation status of spiders in Britain. Both the Red List for Britain, which assesses extinction risk using international criteria set by the International Union for Nature Conservation (IUCN) (see www.iucnredlist.org), and the evaluation of Great Britain Rarity status, assessed simply by frequency of occurrence (see jncc.defra.gov.uk/page-3425), are based on SRS data. These assessments are brought together in *A review of the status of scarce and threatened spiders (Araneae) of Great Britain* (Harvey *et al*. 2017). Together with SRS distribution maps, habitat, micro-habitat and other ecological information, this document underpins both legislative protection (jncc.defra.gov.uk/page-1377) and conservation planning and action for spiders in England, Wales and Scotland. It is used by both voluntary sector and statutory conservation bodies, at local and wider geographical scales.

Similarly, the SRS data on occurrence of species in relation to habitat management helps to inform substantial improvements in site management for spiders. While this is particularly critical for our least common species, it also helps to underline the importance of structurally diverse habitats for promoting species-rich assemblages of spiders.

Patterns of change

The recording of spiders on a detailed national basis is now at a stage when it can be used to monitor changes in the distribution of species (e.g. Hickling *et al.* 2006; Mason *et al.* 2015), although it is not yet possible to use it for more detailed assessment of smaller-scale changes in population size. It is proving particularly useful in identifying changes in distribution likely to be the result either of the loss of specialist habitats or of climate change; for example, the marked northward expansions in range of *Argiope bruennichi*, *Pholcus phalangioides* (Fig. 4) and *Steatoda nobilis*.

Other changes in distribution documented by the SRS result from improvements in the recognition of good characters for separating closely related species, and clarification of their ecological preferences, rather than from real changes in distribution or abundance. For example, the crab spider *Philodromus praedatus* has an interesting history in Britain. It 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 comparatively easy to distinguish with microscopical examination from other members of the *P. aureolus* group, females are difficult to identify without dissection and require reference to reliably identified specimens. Pickard-Cambridge (1879) first recorded it from Dorset in the nineteenth century. In 1974 the species was still only known from old specimens taken at Bloxworth (Dorset), the New Forest (Hampshire) and Shrewsbury (Shropshire). However, by the time Dr Peter Merrett published the first review of nationally notable spiders in 1990, the spider had been recorded in 10 counties, mainly in southern England but also Inverness, and by 2019 it was known from 40 counties.

New species to Britain continue to be discovered, such as *Macaroeris nidicolens* from Mile End in East London in 2002, *Zoropsis spinimana*, which has established breeding populations in London and the south-east since 2012, and *Rugathodes sexpunctatus*, found on the Glasgow Necropolis in 2012. *Sibianor larae* was first recognized in this country in 2018 and soon afterwards, Falconer's 1924 specimen of *Sibianor aurocinctus* from Kirkby Moss in South Lancashire was confirmed as *Sibianor larae*, showing the importance of museum collections. Sadly, this locality is now intensive farmland and we will have to re-examine all the British specimens to obtain a clear diagnosis and clarify the distribution and habitat information on both species, something only possible from voucher specimens and museum collections.

Surprisingly perhaps, the rate of discovery since the early 1950s has remained almost constant at an average of just over one and a half species per year.

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 the British Arachnological Society, the Field Studies Council and others arrange special courses 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. Up-to-date information on forthcoming courses around the country is available on the BAS website.
- The ability and opportunity to undertake field work that will provide records for the database.
- The submission of data computerised in an agreed format. Ideally this should be as MapMate files although Excel files structured in a way that allows import into the Scheme's dataset-specific format can also be submitted. MapMate is an inexpensive, extensively-used software package designed for mapping, storing and sharing biological recording data. It is available to paid-up BAS members and SRS recorders at a discounted price.
- Guidance on recording spiders and data submission is available on the SRS website at srs.britishspiders.org.uk/portal/p/Recording+spiders and srs.britishspiders.org.uk/portal/p/Recording+Methodology. Records are then sent to the local Area Organiser or, in the absence of an Area Organiser, to the National Organiser (Section 7.1.2). Further information on all aspects of the SRS is available at srs.britishspiders.org.uk.

7.2 The Harvestman Recording Scheme (HRS)

Peter Nicholson and Mike Davidson

The Harvestman Recording Scheme (HRS) began in 1973 when the first recording cards (RA27) were produced. Originally administered jointly by the British Arachnological Society (BAS) and the Centre for Ecology and Hydrology's Biological Records Centre, it is now run by the BAS. It aims to record the geographical distribution of harvestmen in Britain. By 2019 the scheme held over 50,000 records which are published as maps based on the 10 km squares (hectads) of the Ordnance Survey National Grid. The HRS data are now incorporated into the SRS database and so up-to-date distribution maps, and species accounts with information on seasonality and habitat requirements, are available online for all (currently) 31 British species. Although the HRS does not have Area Organisers, several regional experts are willing to check identifications. Articles on harvestmen regularly appear in the *SRS News* section of the BAS *Newsletter*. The HRS recorder can be contacted at hrs@britishspiders.org.uk. Other harvestman information is available at: srs.britishspiders.org.uk/portal.php/p/Harvestmen, srs.britishspiders.org.uk/portal.php/p/Harvestman+Resources, and srs.britishspiders.org.uk/portal.php/p/Harvestman+Recording+Scheme+Newsletters.

7.3 The Pseudoscorpion Recorders' Group (PRG)

Gerald Legg

The Pseudoscorpion Recorders' Group (PRG) was established to improve our knowledge of this small enigmatic group of arachnids that includes 28 species in the UK. Historically, records were accumulated at ITE Monks Wood Experimental Station and, in 1980, Philip Jones published the first *Provisional atlas*. In 1982, he established a recording scheme and computerised the records. Later, the records were transferred to Dr Gerald Legg, then at the Booth Museum of Natural History, Brighton.

The identification of pseudoscorpions was made considerably easier by the 2017 publication of an illustrated FSC key (Legg & Farr-Cox 2017). A useful Facebook group (en-gb.facebook.com/pseudoscorpion) and Twitter feed (@PseudoscorpUK) are run by Liam Andrews. Distribution data are available from the NBN Gateway at data.nbn.org.uk/Taxa/ NHMSYS0000356759. Occasional articles on pseudoscorpions are now included in the BAS *Newsletter* and further details of British pseudoscorpions, images, and current distributions can be found at www.chelifer.com/?page_id=61. A periodic newsletter, *Galea*, was produced between 1998 and 2001 and is available on the BAS website at britishspiders.org.uk/wiki2015/index.php?title=Pseudoscorpion_Recorders_Group. The Scheme organizer, Dr Gerald Legg, is happy to receive specimens for identification: contact him at 20 Manor Gardens, Hurstpierpoint, Hassocks, West Sussex, BN6 9UG; email: prg@britishspiders.org.uk or gerald@chelifer.com.

References

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Harvey, P., Davidson, M., Dawson, I., Fowles, A., Hitchcock, G., Lee, P., Merrett, P., Russell-Smith, A. & Smith, H. 2017: *A review of the scarce and threatened spiders* (*Araneae*) *of Great Britain*: *Species Status No. 22. NRW Evidence Report* **11**, Bangor: Natural Resources Wales.

Harvey, P. R., Nellist, D. R. & Telfer, M. G. (eds.) 2002: *Provisional atlas of British spiders* (*Arachnida, Araneae*). *Vols 1 & 2*. Abbots Ripton, Hunts: Biological Records Centre.

Hickling, R., Roy, D. B., Hill, J. K., Fox, R. & Thomas, C. D. 2006:The distributions of a wide range of taxonomic groups are expanding polewards. *Global Change Biology* **12**: 450–455.

Jones, D. 1983: *The Country Life guide to spiders of Britain and northern Europe*. Littlehampton: Littlehampton Book Services Ltd.

Jones, P. E. (ed.). 1980: *Provisional atlas of the Arachnida of the British Isles. Part 1. Pseudoscorpiones*. Abbots Ripton: Institute of Terrestrial Ecology.

Legg, G. & Farr-Cox, F. 2017: *Illustrated key to the British false scorpions* (*Pseudoscorpions*). Shrewsbury: Field Studies Council AIDGAP Guides Volume OP173.

Locket, G. H. & Millidge, A. F. 1951: *British spiders. Volume 1.* London: Ray Society.

Locket, G. H. & Millidge, A. F. 1953: *British spiders. Volume 2*. London: Ray Society.

Locket, G. H., Millidge, A. F. & Merrett, P. 1974: *British spiders. Volume 3*. London: Ray Society.

Mason, S. C., Palmer, G., Fox, R., Gillings, S., Hill, J. K.,Thomas, C. D. & Oliver, T. H. 2015: Geographical range margins of many taxonomic groups continue to shift polewards. *Biological Journal of the Linnean Society* **115**: 586–597.

Merrett, P., Russell-Smith, A. & Harvey, P. 2014: A revised check list of British spiders. *Arachnology* **16**: 134–144.

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*. Colchester: Harley Books.

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

Scott, A. G., Oxford, G. S. & Selden, P. A. 2006: Epigeic spiders as ecological indicators of conservation value for peat bogs. *Biological Conservation* **127**: 420–428.

SRS (Spider Recording Scheme). 2019: *Distribution maps for* Pardosa pullata *and* P. prativaga, online at srs.britishspiders.org.uk/portal/p/A-Z+Species+Index

8 SAFETY MATTERS

A. Russell-Smith

Studying arachnids is no more risky than any other recreational pursuit involving field work. 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, wet rocks, and 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, a GPS unit and a compass, and always let someone else know where you are planning to go and when you plan to return. Carry a mobile phone and in areas with poor reception carry a whistle to use as an emergency distress signal. The British Mountaineering Council (177–179 Burton Road, Manchester M20 2BB; Tel: 08700 104878; website: 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. Alternatively, carry a brightly coloured backpack.
- If you are going into remote areas and plan to be away for more than a few hours, make sure you have with you adequate food and, particularly, water. 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 intertidal 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 (this information can be found online and 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 channels and the distance to the nearest higher ground.
- Avoid carrying glass equipment such as pooters and glass tubes, take plastic alternatives. 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. This is also important when sampling in caves. However, the occurrence of *Histoplasma capsulatum* or histoplasmosis fungus, generally associated with bat droppings in tropical climates, has not been recorded in the droppings of British bats. Histoplasmosis is a very rare disease in Britain (all occurrences are of foreign origin) and there is no evidence that the fungus occurs naturally in this country. The British climate is generally unsuitable for this fungus, which requires a relatively stable temperature of 20–35°C and a high relative humidity to flourish.

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 of contracting Weil's disease from rodent-contaminated water;
- ensuring you check for and remove ticks after working in areas with high stock or wild mammal numbers. Wearing long trousers and tucking the hems into thick socks is a sensible precaution in such areas, as is the use of an insect repellent on your clothes and skin; products containing DEET are best. Wearing light-coloured clothing means that ticks are easier to spot and brush off. Ticks transmit the bacterium that causes Lyme disease, which can be serious if left untreated. Awareness of symptoms and prompt treatment is essential; see www.nhs.uk/conditions/Lyme-disease/Pages/Introduction.aspx for more information. The virus responsible for tick-borne encephalitis, which is has been spreading west across Europe, has recently (2019) been recorded in GB. Guidance should be sought from the NHS website at www.nhs.uk/conditions/tick-borne-encephalitis and the Encephalitis Society at www.encephalitis.info/TBE;
- carrying some anti-histamine tablets in your first aid kit. Stings from wasps or hornets can occasionally cause 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 denatured alcohol) 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 well 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 (Section 3.4.3).

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. Planning ahead before taking to the field is always time well spent.

9 LIST OF SUPPLIERS

A. Russell-Smith

9.1 General Laboratory Suppliers

See also: britishspiders.org.uk/wiki2015/index.php?title=Suppliers

Arachne Ecology Ltd, Paul Lee, 1 Holly Cottages, Church Road, Tattingstone, Ipswich IP9 2LZ. Mobile: 07545 230367. Email: arachne2222@aol.com. *Price list on application. Polypropylene specimen tubes; entomological and arachnological supplies including glass blocks, forceps, and pin vices*.

Atom Scientific Ltd, Unit 2B, East Tame Business Park, Wrexcine Way, Hyde, Cheshire SK14 4GX. www.atomscientific.com. *Cheap source of 70% Industrial Denatured Alcohol*.

Blades Biological Ltd, Cowden, Edenbridge, Kent TN8 7DX. Tel: 01342 850242. blades-bio.co.uk. *Terraria, other tanks, live fruit fly cultures and instant fly medium*.

Camlab Ltd, Camlab House, Norman Way Industrial Estate, Over, Cambridge CB24 5WE. Tel: 01954 233110. 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 in diameter*).

Fisher Scientific UK Ltd, Bishop Meadow Road, Loughborough, Leicestershire LE11 5RG. Tel: 01509 555500. 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*.

Mackay & Lynn Ltd, 18/7 Dryden Road, Bilston Glen, Loanhead EH20 9LZ. Tel: 0131 448 0819. www.mackayandlynn.co.uk/shop. *Dissection instruments, glass & plastic specimen tubes, hand lenses, ethanol and other laboratory chemicals*.

Philip Harris Education, 2 Gregory Street, Hyde, Cheshire, SK14 4TH. Tel: 03452 204520. www.philipharris.co.uk. *Catalogues only available to educational establishments*.

Scilabware Ltd, Unit 4, Riverside, 2 Campbell Road, Stoke-on-Trent, Staffordshire ST4 4RJ Tel: 01782 444406. www.scilabware.com/en. *Suppliers of the Bibby-Sterilin line of laboratory equipment, particularly disposable plastic specimen tubes and Petri dishes*.

R & L Slaughter Ltd, Unit 14, Carnival Park, Carnival Close, Basildon, Essex, SS14 3WN. Tel: 01708 227140. www.slaughter.co.uk. *Dissecting instruments, glass and plastic specimen tubes, Petri dishes, wash bottles, ethanol and other laboratory chemicals; online catalogue*.

Murray & Co. Ltd, Holborn House, Old Woking, Surrey GU22 9LB. Tel: 01483 740099. www.smurray.co.uk. *Glass vials 35 × 22 ml* (*code: T103/V1*); *push-on plastic caps* (*code: T003/C1*).

Sarstedt Ltd, 68 Boston Road, Beaumont Leys, Leicester LE4 1AW. Tel: 01162 359023. www.sarstedt.com/en/home. *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: 01902 402402. www.scichem.com. *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: 1747 833000. www.sigmaaldrich.com/united-kingdom.html. *Dissection instruments, glass beads (ballotini), plastic specimen tubes, ethanol and other laboratory chemicals, Petri dishes, wash bottles, etc*.

Solmedia Ltd, Unit 2, Vernon Drive, Battlefield Enterprise Park, Shrewsbury, SY1 3TF. Tel: 08448 080900. solmedialtd.com. *Ethanol, IDA and other laboratory chemicals*.

VWR, VWR International, Hunter Boulevard, Magna Park, Lutterworth, Leicestershire, LE17 4XN Tel: 0845 073 6649. uk.vwr.com/cms/avantor_services_technical_services. *Supply plastic containers and Petri dishes, and also microscopes*.

9.2 Entomological Suppliers

Anglian Lepidopterist Supplies, Station Road, Hindolveston, Norfolk NR20 5DE. Tel: 01263 862068. angleps.com. *Nets, chemicals, microscopes*.

B & S Entomological Services, 37 Derrycarne Road, Portadown, Co. Armagh BT62 1PT. Tel: 07760 388463 or 02838 336922. www.entomology.org.uk. *Suppliers of the Marris House range of entomological nets*.

D. J. & D. Henshaw, 34 Rounton Road, Waltham Abbey, Essex EN9 3AR. Tel: 01992 717663, email: djhagro@aol.com. *Specimen bottles and tubes, microscope accessories*.

NHBS Ltd, 1-6 The Stables, Ford Road, Totnes, Devon, TQ9 5LE. Tel: 01803 865913. www.nhbs.com. *Wide range of entomological supplies* (*as formerly stocked by Alana Ecology and EFE & GB Nets*), *including plastic aquaria.*

Watkins & Doncaster, PO Box 114, Leominster, HR6 6BS. Tel: 03338 003133 or 01568 750657. www.watdon.co.uk. *Specialise in the manufacture and supply of equipment for the collecting, storing, and observing of insect life, including sweep nets, beating trays, pooters, microscopical accessories and specimen tubes.*

Wildcare, www.wildcare.co.uk, Stow Agricultural Ltd, Eastgate House, Moreton Road, Longborough, Gloucestershire, Gl56 0QI. Tel: 01451 493 883. *Stock a range of outdoor and ecological survey equipment, including entomological supplies (nets and sampling trays).*

9.3 Microscopes & Microscopy

Brunel Microscopes Ltd, Unit 2, Vincients Road, Bumpers Farm Industrial Estate, Chippenham, Wiltshire SN14 6NQ U.K. Tel: 01249 462655. www.brunelmicroscopes.co.uk.

Carl Zeiss Ltd, Zeiss House, Building 1030, Cambourne Business Park, Cambourne CB23 6DW Cambridge. Tel: 01223 401500. www.zeiss.co.uk/microscopy/home.html.

GX Microscopes, GX Microscopes divn, GT Vision Ltd, Aspen Grove Farm, Assington Green Stansfield, Suffolk, CO10 8LY. Tel: 014284 789697. www.gxmicroscopes.com.

Lakeland Microscopes, Kynance, Charney Well Lane, Grange-over-Sands, Cumbria LA11 6DB. Tel: 01539 534737. www.lakeland-microscopes.co.uk.

Leica Microsystems UK Ltd, Larch House, Woodlands Business Park, Breckland, Linford Wood, Milton Keynes, MK14 6FG. Tel: 0800 2982344. www.leica-microsystems.com.

Meiji Techno UK Ltd, The Vinyard, Axbridge, Somerset BS26 2AW. Tel: 01934 733655. www.meijitechno.co.uk.

Metascientific Ltd, 3 Aldgate Close, Potton, Bedfordshire SG19 2RU. Tel: 01767 260295. www.metascientific.com.

Microscopes Plus Ltd, 72 Brookside South, Barnet, EN4 8LW. Tel: 03334 441950. www.microscopesplus.co.uk/page102.html.

Nikon Instruments, Nikon House, 380 Richmond Road, Kingston-upon-Thames, Surrey KT2 5PR.Tel: 02082 471717. www.microscope.healthcare.nikon.com/en_EU.

Olympus KeyMed (Medical & Industrial Equipment) Ltd, KeyMed House, Stock Road, Southend-on-Sea, Essex SS2 5QH. Tel: 01702 616333. www.olympus-lifescience.com/en/microscope-resource.

One Stop Nature Stop, 9 Dalegate Market, Burnham Deepdale, Norfolk, PE31 8FB, Tel: 01485 211223. www.onestopnature.co.uk. *Suppliers of microscopes and microscope photography systems*.

SBTC Binocular & Telescope, 7, Harrop Drive, Swinton, Mexborough, South Yorkshire, S64 8LB. Tel: 07815 437220. www.telescopesandbinoculars.co.uk.

9.4 Booksellers

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

C. Arden. Darren Bloodworth, The Nursery, Forest Road, Hay on Wye, HR3 5DT. Tel: 01497 820471. www.ardenbooks.co.uk. *New and second- hand books on natural history; also buys books and will do book searches.*

NHBS Ltd, 1-6 The Stables, Ford Road, Totnes, Devon, TQ9 5LE. Tel: 01803 865913. www.nhbs.com. *Probably one of the largest stockists of new natural history books in Britain*.

Pemberley Books, 18 Bathurst Walk, Iver, Bucks, SL0 9AZ. Tel: 01753 631114. www.pemberleybooks.com. *Specialises in new and second hand books in entomology and arachnology*.

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: www.societyofbookbinders.com/about-us/useful-links.

10 RECOMMENDED READING

Ian Dawson & Tony Russell-Smith

Listed here are books on general aspects of arachnology. The first port of call if specialist papers are required is the BAS library, briefly described below. Abstracts of almost all papers are freely available on the web, either through a normal Google (or other search-engine) search or through Google Scholar. If full papers are not in the BAS library, they are often available by emailing the first author, whose email address will be provided with the abstract of the paper. Most people are only too happy to disseminate their work.

10.1 The BAS Library

A major benefit of BAS membership is access to our library of some 25,000 reprints of scientific papers of arachnological interest. The library database is held on our website and accessible only to members. A large number of the reprints are downloadable from the website for members use only. While most are of a technical nature, there are also many papers giving a general introduction to various aspects of arachnology, which are ideal for the beginner. Printed papers cannot be ordered from the website but can be obtained by emailing or writing to the Librarian. They are normally sent out within a couple of weeks or so. The library is housed at the World Museum Liverpool and we rely on the goodwill of staff members to service requests. Very lengthy papers and books are subject to special arrangement and only available to UK members. Members may visit the library by prior arrangement.

The Librarian can be contacted by email at: library@britishspiders.org.uk

10.2 Identification

Bee, L., Oxford, G. & Smith, H. 2017: *Britain's spiders – a field guide*. Princeton: WILDGuides. *A photographic guide designed to be used in the field in conjunction with a spi-pot and a 10× hand lens. It includes species accounts with distribution maps, periods of maturity and level of magnification required for identification (eye, lens or microscope). Since it is a field guide, only a few of the larger linyphiids are featured.*

Hillyard, P. D. 2005: *Harvestmen*. Synopses of the British Fauna. Shrewsbury: Field Studies Council. *This excellent summary of our harvestman fauna provides accounts of the structure and biology of 25 species together with easy-to-use illustrated keys and distribution maps.*

Jones, D. 1989: *A guide to spiders of Britain and northern Europe*. London: Hamlyn. *Out of print. Colour photographs and text for around 300 of our larger species of spider. Although out of print in the UK, 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—see next title*.

Jones, D., Ledoux, J-C. & Emerit, M. 2001: *Guide des araignées et des opilions d'Europe*. Lonay, Switzerland: Delachaux et Niestlé.

Jones-Walters, L. M. 1989: *Keys to the families of British spiders*. AIDGAP. Shrewsbury: 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. & Farr-Cox, F. 2017: *Illustrated key to the British false scorpions* (*Pseudoscorpions*) Shrewsbury: Field Studies Council AIDGAP Guides Volume OP173. *Allows the identification of almost all species of British pseudoscorpions.*

Legg, G. & Jones, R. E. 1988: *Pseudoscorpions*. Synopses of the British Fauna. Shrewsbury: Field Studies Council. *As with its sister volume on harvestmen, this provides information on structure and biology of the British pseudoscorpions as well as keys and distribution maps.*

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

Locket, G. H., Millidge, A. F. & Merrett, P. 1974: *British spiders. Vol. 3.* London: 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. A CD version comprising all three volumes is published by Pisces CD books and is available from NHBS (see Section 9.4 above).*

Richards, P. 2010: *Harvestmen*. Fold-out chart. Shrewsbury: Field Studies Council. *A photographic guide to the identification of almost all species of British harvestmen.*

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

Roberts, M. J. 1993: *The spiders of Great Britain and Ireland. 2-part Compact edition*. Colchester: Harley Books. Softback. Part 1 text, comprising vols. 1 & 2 of the hardback edition; Part 2 colour plates. *Now published by Brill, Leiden. 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 primarily aimed 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*. London: 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 also described. Only a relatively few Linyphiidae or money spiders species (around 40) with a distinctive abdominal pattern, some of which can be recognised using a hand lens, are included.*

Sauer, F. 2012: *The spider families of Europe*. Hirschberg: Jörg Wunderlich Publishing House.

Wijnhoven, H. 2009: *De Nederlandse hooiwagens* (*Opiliones*). Entomologische Tabellen 3, supplement bij Nederlandse Faunistische Mededelingen. *An English translation (but without artwork) is available at srs.britishspiders.org.uk/resource/De_Hooiwagens_1st_revision140615.pdf*

10.2 Distribution

Harvey, P. R., Nellist, D. R. & Telfer, M. G. (eds.) 2002: *Provisional atlas of British spiders* (Arachnida, *Araneae*). *Vols. 1 & 2.* Abbots Ripton: Biological Records Centre. *Out of print. This two-volume publication has 10 km² distribution maps together with text summarised the known ecology for every British species. Now superseded by information on the SRS website (srs.britishspiders.org.uk).*

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

Crocker, J. & Daws, J. 2001: *Spiders of Leicestershire and Rutland: millennium atlas*. Leicester: 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

Bee, L. & Lewington, R. 2002: *A guide to house and garden spiders*. Fold-out chart. Shrewsbury: Field Studies Council. *A pictorial introduction to 40 species of spiders common in and around the home*.

Bristowe, W. S. 1958: *The world of spiders* (New Naturalist no. 38). London: Collins. *This remains probably the best introduction to spiders for the beginner and conveys brilliantly the sheer fascination of this diverse group of organisms. Essential reading, which is well worth hunting down through your local library or purchasing through a print-on-demand service from HarperCollins (www.newnaturalists.com/product-category/print-on-demand-main-series).*

Brunetta, L. & Craig, C. L. 2010: *Spider silk*. Yale: Yale University Press. *The subtitle of this popular science text 'evolution and 400 million years of spinning, waiting, snagging, and mating' covers its contents—all about spider silk.*

Chinery, M. 1993: *Spiders.* Stowmarket, Suffolk: Whittet Books. *Out of print. Aimed at the general naturalist, a popular overview of spider natural history*.

Hillyard, P. 1994: *The book of the spider: from arachnophobia to the love of spiders*. London: Hutchinson/Pimlico. *Out of print. A general read about spiders and their relationship with man*.

Kelly, K. 2009: *Spiders: learning to love them*. Crows Nest, NSW: Jacana Books – Allen & Unwin. *A general introduction to spiders from an Australian science writer through her journey from arachnophobia to arachnophilia. A good read, particularly from anyone wanting to make the same journey*.

Murphy, F. 2000: *Keeping spiders, insects and other land invertebrates in captivity. 2nd edn*. Edinburgh: J. Bartholomew & Son Ltd. *Out of print. The title is self-explanatory. Keeping spiders is an excellent way to learn something of their behaviour*.

10.4 Spiders Worldwide

This is just a very small selection of possible titles. For many pages of arachnological titles see the NHBS website: www.nhbs.com.

Beccaloni, J. 2009: *Arachnids*. London: Natural History Museum. *An excellent international coverage of all arachnid families and with stunning photographs*.

Bellmann, H. 1997: *Kosmos-Atlas Spinnentiere Europas*. Stuttgart: Kosmos. *Excellent colour photographs of living spiders of Central Europe, many of the species illustrated also occur in Britain. The text is, of course, in German*.

Fet, V. & Selden, P. A. (eds.) 2001: *Scorpions 2001. In memoriam Gary Polis*. Burnham Beeches, Bucks: British Arachnological Society. *Out of print. A collection of 32 academic papers on many aspects of scorpion biology, taxonomy and distribution*.

Jocqué, R. & Dippenaar-Schoeman, A. S. 2006: *Spider families of the world.* Tervuren: 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*. Berlin: 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 book is superseded by the website: araneae.nmbe.ch*

McGavin, G. C. 2000: *DK Handbook: insects, spiders and other terrestrial arthropods*. London: Dorling Kindersley. *A general pictorial introduction for younger readers to insects and spiders worldwide*.

Murphy, J. 2007: *Gnaphosid genera of the world*. St Neots, Cambs: British Arachnological Society. *A two-volume atlas illustrated by Mike Roberts and describing species in over 100 genera. Volume 1 contains identification keys and Volume 2 plates with black-and-white line drawings and distribution maps*.

Murphy, J. & Roberts, M. J. 2015: *Spider families of the world and their spinnerets*. York: British Arachnological Society. *Typical genera of every family of spiders are illustrated, together with details of eye pattern, tarsal claws, setae types and, especially, spinneret and spigot morphology. The authors also present a groundbreaking new classification scheme based on the evolution of the spinning organs*.

Preston-Mafham, R. & K. 1993: *Spiders of the world*. London: Blandford Press.

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

Preston-Mafham, K. & R. 1996: *The natural history of spiders*. Ramsbury, Wiltshire: Crowood Press. *All three Preston-Mafham books are liberally illustrated with superb colour photographs*.

10.5 Scientific Texts

Again, this can only be a very incomplete list of works.

Barth, F. G. 2001: *A spider's world: senses and behaviour*. Berlin: Springer. *How the sensory organs of spiders have evolved under the influence of their environment, and their link with spider behaviour*.

Foelix, R. 2011: *Biology of spiders. 3rd edn*. New York: 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.*

Herberstein, M. H. (ed.) 2011: *Spider behavior: flexibility and versatility*. Cambridge: Cambridge University Press. *An up-to-date and readable compendium covering academic research on the amazing variety and flexibility of spider behaviours*.

Nentwig, W. 2013: *Spider ecophysiology*. Heidelberg: Springer. *A compendium of the latest research combining functional and evolutionary aspects of morphology, physiology, biochemistry and molecular biology with ecology.*

Shear, W. A. (ed.) 1986: *Spiders. Webs, behavior, and evolution*. Stanford: 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: 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 number of websites devoted to spiders is ever increasing. The BAS website gives links to many of them. The spider checklists of many European countries are now available online, in some cases linked to distribution maps.

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 **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).

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 evolutionary origin.

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

Anterior. Nearer the front or head end. Can 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 sclerotised infolding of the body wall that forms an attachment for muscles.

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

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

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. **Autophagy**, **Autotilly** and **Autotomy**).

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

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. **Autophagy**, **Autospasy** and **Autotilly**).

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 bordering the internal face of the epidermis.

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 breathe.

Boss. A smooth, rounded, or slightly conical prominence.

Branchial operculum (pl. **branchial opercula**). A sclerotised 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 that 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.
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, the characteristic component of the hardened cuticle of arthropods.
Claw. A strong, curved, sharp-pointed process (often toothed) on the distal extremity of a leg and sometimes the palp, particularly of the female.
Clade. See **Monophyletic group**.
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 species geographical range.
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.
Conductor. A semi-membranous structure in the male palp that, 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.
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.
Coxa (pl. **coxae**; adj. **coxal**). The segment of leg nearest the body. In the palp, this segment is modified to form the maxilla.
Cribellate. Possessing 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 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.
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 (or colour) 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* males).
Distal. Pertaining to or situated at the outer end; that part of a limb/limb segment/appendage that 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 calamistrum.
Ecdysis. Moulting, the periodic act of casting off the old 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 sclerotised and modified external structure associated with the reproductive openings of adult females of most spider species.
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; the duct from the poison gland opens near its tip.
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 pass to fertilize the eggs.
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.
Gnathocoxa. Basal segment (coxa) of palp, also called the maxilla or endite.
Gossamer. A light gauzy film of 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.
Guanine. The principal nitrogenous excretory product in spiders. In some species, it is deposited on the surfaces of the intestinal diverticula where it acts as a white or silver pigment in its own right, or as a background for overlying pigments.
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).**
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 that have a common evolutionary origin.

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; a result of hybridization and subsequent 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 in the male palps of some spiders.

Lamina. A flat, sometimes translucent, sclerotised excrescence, e.g. on the retro-margin of the 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. Can 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. In males, 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 (or **Clade**). Taxonomic group which includes the most recent common ancestor of all its constituent species and (therefore) all the descendants of that most recent common ancestor.

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.

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 the first 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.

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**).

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.

Penultimate instar. The instar immediately before the moult to sexual maturity.

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.

Plumose. Feathery.

Polymorphism. The existence of two or more forms that are genetically distinct from one another but contained within the same interbreeding population.

Posterior. Nearer the rear end. Can 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 the ventral tip of the 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). (cf. **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.

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.

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

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.

Rugose. Rough; wrinkled.

Scape (or **Scapus**). A finger-, tongue-, or lip-like appendage, free at one end, arising from the midline of the female epigyne in some species.

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

Sclerotised. Hardened or horny; not flexible or membranous. (See also **Chitin**).

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 sclerotised 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, sclerotised spot, usually reddish-brown in colour, pairs of which are often present on the abdomen, marking points of internal muscular attachment.
Slit organ (or **Slit sensillum**, pl. **slit sensilla**). A stress receptor in the exoskeleton that 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.
Sperm duct. 1. A duct in the female epigyne through which sperm travels from the copulatory pore to the spermatheca. 2. A duct in the 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 immature stage of a spider, able to move about and feed and no longer dependent upon the yolk for nourishment. Resembles the adult in general form, but smaller.
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, across the orbwebs of some spiders. Its function(s) seems to vary with species.
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 sclerotised 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, or carapace.
Subadult. Almost adult; the penultimate 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**.
Synanthropic. Living in or close to human habitation.
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 palp 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**). Tube through which air is carried around the body and which opens at a spiracle.

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.

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 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.

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

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).**

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**).

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