BLOOMSBURY NATURALIST

HOVERFLIES

OF BRITAIN AND NORTH-WEST EUROPE

A PHOTOGRAPHIC GUIDE

Sander Bot &
Frank Van de Meutter

Hoverflies

of Britain and
North-west Europe

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Cover photos © Frank Vassen
Front cover top: *Leucozona glaucia*; bottom (left to right): *Helophilus trivittatus*, *Sphaerophoria scripta*, *Xanthogramma pedissequum*
Spine: *Microdon myrmicae*
Back cover (left to right): *Caliprobola speciosa*, *Paragus haemorrhous*, *Criorhina floccosa*

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Acknowledgements

The basis of this book is the Dutch field guide *Veldgids Zweefvliegen*, published by KNNV publishing in the Netherlands in 2019. The current book, however, is not just a simple translation. The text has been completely revised, and the book has been expanded to include full species accounts of an additional 12 species. Furthermore, the text has been rewritten to include identifications of British species, and descriptions of the ecology of the British fauna, and distribution maps and flight-time diagrams for a much larger area, including the British Isles, have been added.

We would not have been able to write this book without funding from the Meester Prikkebeen Fund, part of the Prins Bernhard Cultuurfonds, and the Uyttenboogaart-Eliasen Foundation. A special thanks goes to Christophe Brochard. First, he taught us how to make a camera set-up and how to take good photographs with it. Over the years, Christophe continually worked with us at improving the set-up. In addition, the images with the finest detail were taken by him using his personal microscope set-up; they could not have been produced with a standard camera. Without his immense help, we would not have images of such high quality.

To be able to take all the 1,797 photos for this book, we needed not only a specialised camera set-up, but also the flies themselves. Obtaining them was quite a challenge. The material needed to be preserved well (i.e. appropriately pinned), and we wanted to include so-called *mythical*, very rarely observed species, while using specimens preferably from within the geographical scope of this book. Our own collections were not sufficient for this, so help from colleagues and museums was essential. Consequently, in recent years, there has been a lively traffic of rare hoverflies from collections to our office, with people and museums entrusting us with these precious fly specimens for a long time. We sincerely thank them for their trust. These are, in order of number of flies supplied: Naturalis Biodiversity Center Leiden (collection manager Pasquale Ciliberti), Jeroen van Steenis, Gerard Pennards, Elias de Bree, Jonas Mortelmans, John Smit, Chris Palmer, Wouter van Steenis, Leendert-Jan van der Ent, Steven Vantieghem, Wout Opdekamp, André van Eck, Bastiaan Wakkie, Tim Faasen, Jan Versigghel, Menno Reemer, The Natural History Museum London (Nigel Wyatt), The Royal Belgian Institute of Natural Sciences (Wouter Dekoninck and Pol Limbourg), Menno van Zuijen, Franz Malec, Lenze Hofstee, Zoological Research Museum Alexander Koenig Bonn (Ximo Mengual), Biological Museum Lund (Rune Bygebjerg), Steven Falk, Peter Lindenburg, Rune Bygebjerg, Aat Barendregt (now deceased), Jan Edelsjö, Niklas Johansson, Xavier Lair, Łukasz Mielczarek, Naturhistorisches Museum Bern (Hannes Baur), Lisa Fisler and Wil van der Hoven.

A novel feature of this book also is that it provides common names for all species. This was a difficult task, as some names were already in circulation and many British species have been given a name by Steven Falk on his excellent Flickr website (www.flickr.com/photos/63075200@N07/collections/72157629600153789/...). However, common names are also given in the *Field Guide to the Flower Flies of Northeastern North America* by Jeff Skevington and Michelle Locke. Because several species are found in both America and Europe and, in fact, most of the genera are shared, there was a risk that two parallel naming systems for hoverflies would evolve. To avoid this, we worked with Steven Falk (artist, naturalist and photographer) and Jeff Skevington to devise a system that we hope will unify the thus-far diverging naming systems. Steven and Jeff, thanks a lot for the discussions and ideas that led to the common names we have used. Before sending the manuscript to the publisher, we asked Steven Falk to review the manuscript. Thanks a lot Steven for the considerable corrections and for sharing your immense knowledge of British hoverflies. Ximo Mengual and Jeff Skevington, thanks for the taxonomic advice.

Many people and institutions shared data on the hoverflies of their region (see the section on creating the maps): without their help and cooperation, we would not have been able to include the maps and phenology bars. This brings us to Stuart Ball. He has been truly amazing in producing the maps and the phenology bars. He's a wizard with data handling and programming, but what's been most exceptional has been his willingness to instantly address the dozens of comments or requests for changes we fired at him. Stuart, we hope you know how much we appreciated this.

And finally, Sander would like to thank Janne Ouwehand, the love of his life: Janne, you're incredible! Frank tries to fathom how it is to live together with a husband or a father that is distracted with every buzzing of a fly, and strays into the bushes at many an inconvenient time. The closest he gets to understanding is that it must be very, very hard sometimes, and that utter patience is required! Thanks Olga, Annebel and Mauro, my beloved ones!

Introduction

Is it a hoverfly?

In the field, most hoverflies will be readily identifiable as hoverflies owing to their typical behaviour and their bright colours. Some, however, are very dull and look like regular flies, while others are champions of mimicry and not only are near-perfect wasp or (bumble)bee imitations, but also behave like them. The following hints should help you to recognise every hoverfly you encounter.

A hoverfly that resembles a bee or wasp can be recognised by looking at its wings. Hoverflies belong to the insect order Diptera (flies), the name being derived from the Greek *two* and *wing*, meaning they have one pair of wings. Bees and wasps belong to the Hymenoptera and have two pairs of wings. The second pair of (hind)wings that would have been present in the ancestors of flies has not totally disappeared: it is now reduced to the halteres that today's flies use for balance while flying. Furthermore, hoverflies have only three antennal segments (bees and wasps have many), which means that the antennae are usually much shorter, but not always. In species that rely on wasp mimicry, the appearance of a longer, wasp-like antenna is thanks to different modifications: some species have a spine on the head, on top of which the antennae stand; some have elongated antennal segments; while in others the colour and shape of the fore legs mimics those of wasp antennae and the flies wave them along the head as wasps do with their antennae!

Within Diptera, the hoverflies can be recognised by their wing venation (and this feature is also used to identify the different tribes and genera of hoverflies). Hoverflies are the only flies with a false vein (Photo 1). As for every rule, there are some exceptions: the false vein is missing in *Eristalinus sepulchralis* and in *Psilota* it is hard to discern. Other typical (but not unique) features of hoverflies are the closed wing cells r_{4+5} and cua (Photo 1), 'closed' meaning that the cell is completely surrounded by veins. The wing cell cua is long and pointed, and ends near the wing margin. Regarding the antenna, in some similar-looking fly families the arista is terminal, but in hoverflies it is usually on the dorsal side of the third antennal segment. Among the few exceptions to this are mainly species that mimic wasps; in these species, the arista is terminal and makes the antenna look longer (*Ceriana, Sphiximorpha, Callicera*) to improve their likeness to wasps. Hoverflies also lack the black, bristle-like hairs on the thorax and legs that are found in house flies or tachinid flies (the exception: *Ferdinandea*).

Once a fly is recognised as a hoverfly, the next step is to identify it to species. For this, a good knowledge of hoverfly anatomy is essential, and the following photographs show all the body parts relevant for identification. All hoverflies share the same basic body plan. However, the arrangement of their body parts, and especially the relative proportions, may vary considerably. We believe it is best to memorise the basic components, and then try to find them in the different species.

It can be important to know whether your fly is a male or a female. The easiest feature to use for finding that out are the eyes. In females, the eyes are separated; if the eyes meet each other, the fly is always a male. However, there are quite a few species in which the eyes of the male are also separated (e.g. *Anasimyia, Helophilus, Pelecocera*) but usually less so than in the females. Therefore, also check the underside of the tip of the abdomen. The male has a small to very large complex asymmetrical structure that is curled ventrally, or under (the genitalia), creating a bulging tip to the abdomen; while the abdomen in the female ends symmetrically in an – often retracted and barely visible – ovipositor, with no bulge at the end. Occasionally, intersex specimens occur, in which the eyes are separated but the gap is less than in typical females. Such flies share features of both males and females, or may be darkly coloured, and they cannot be identified with the keys (or they will not be correctly identified). Be aware of this possibility when confronted with a strange-looking female fly. Compare the distance between the eyes with that of other females of the same species, and check for slightly wrinkled abdomens and swollen or deformed legs and tarsi, which often indicates an intersex individual.

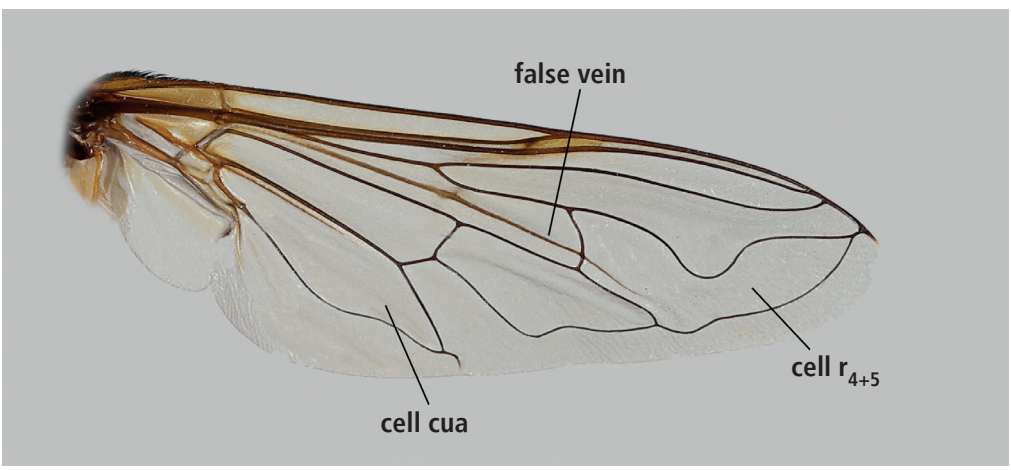
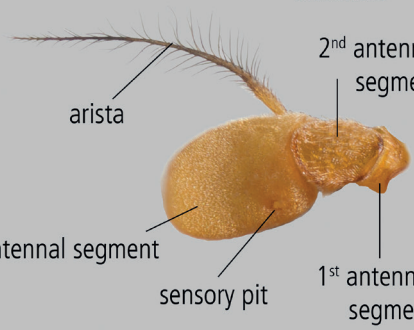


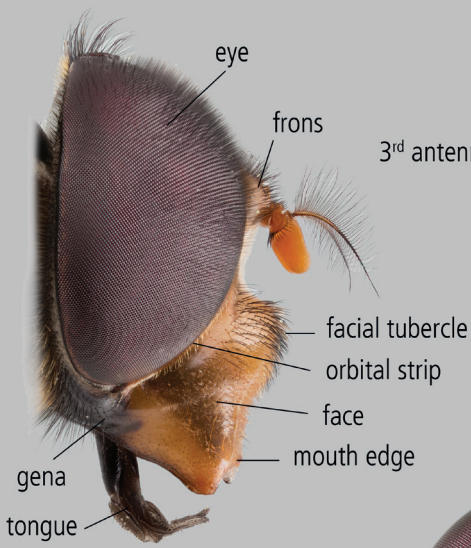
Photo 1 The wing of a hoverfly.

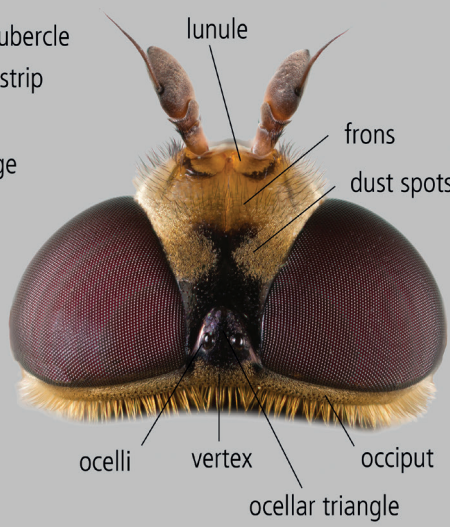
imago dorsal view
Spineleg *Myolepta*

antenna



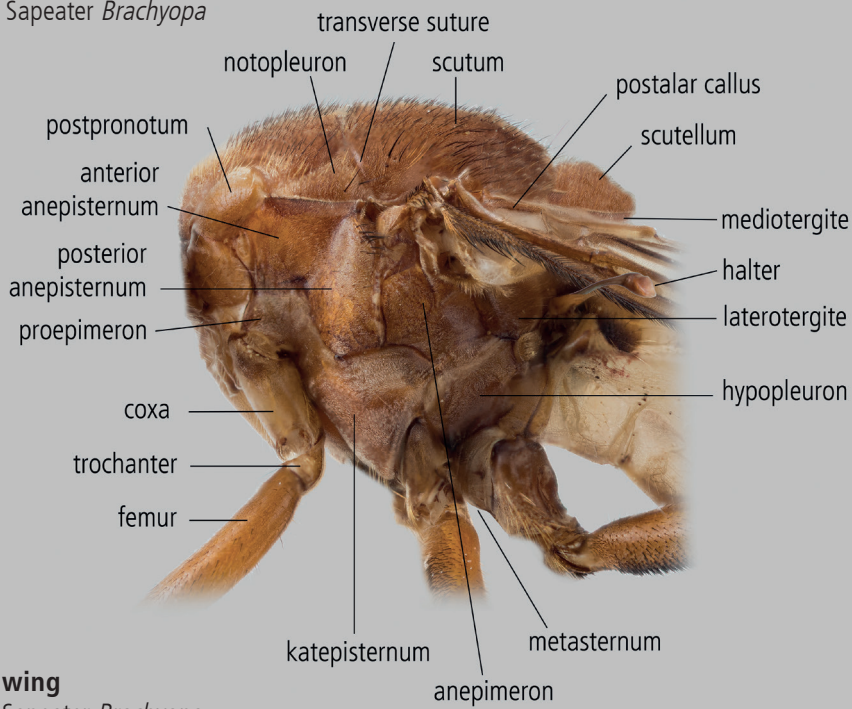
head lateral view
Plumehorn *Volucella*



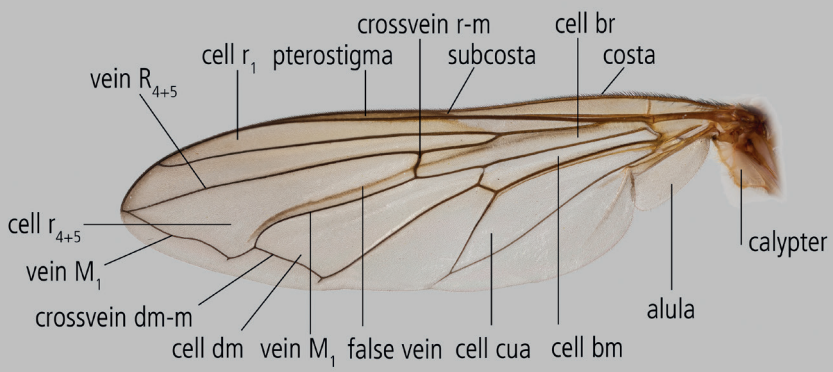


head dorsal view
Aphideater *Eupeodes*

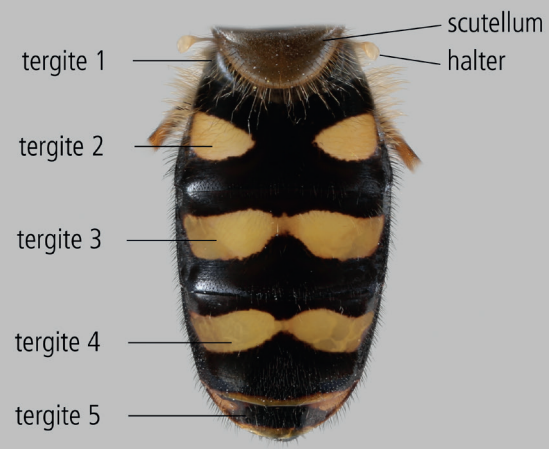
thorax
Sapeater *Brachyopa*



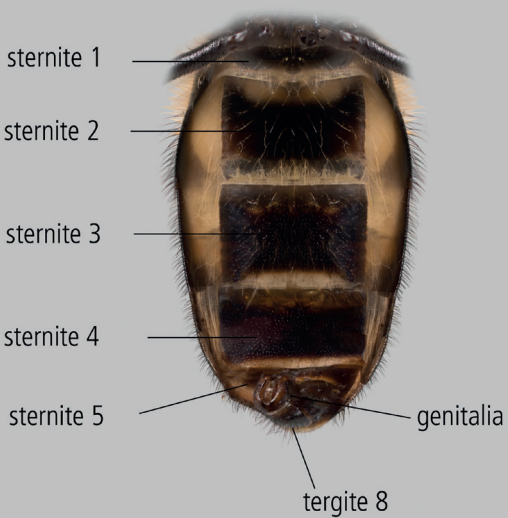
wing
Sapeater *Brachyopa*



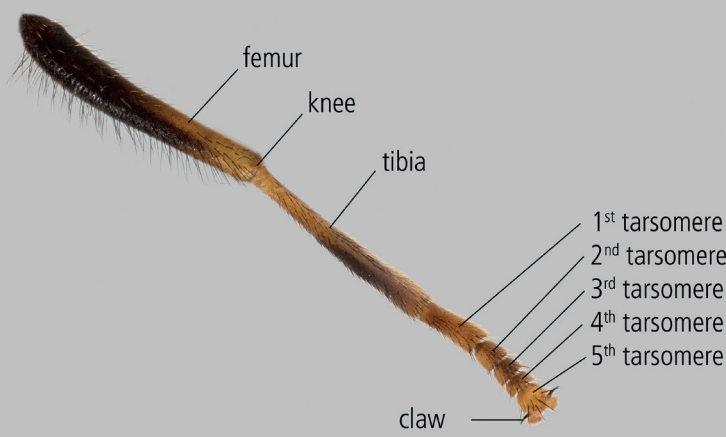
abdomen dorsal view
Aphideater *Eupedes*



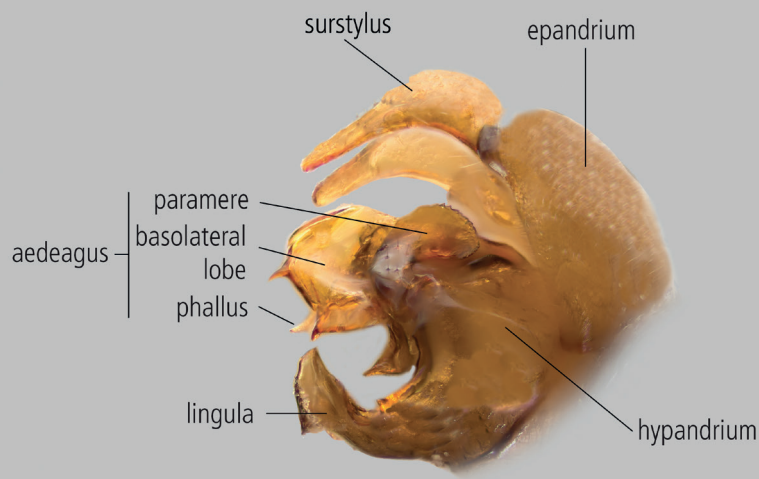
abdomen ventral view
Aphideater *Eupedes*



leg



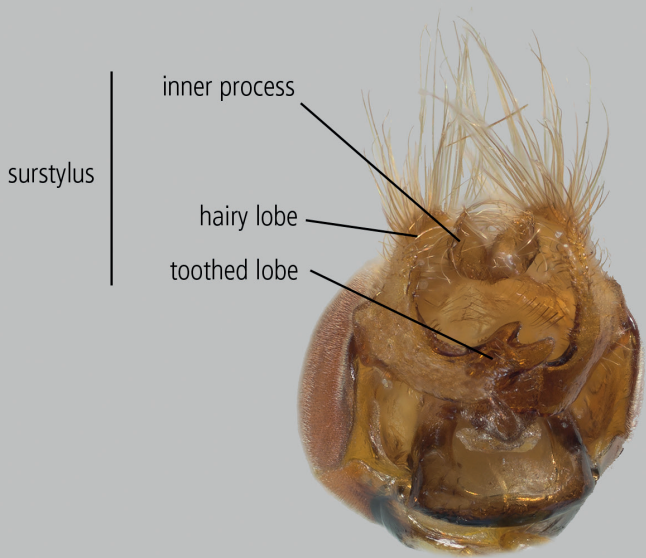
genitalia ♂ Grass skimmer *Paragus*



genitalia ♂ Globetail *Sphaerophoria*

dorsal view

top view



Finding hoverflies

In the spring and summer, you can see hoverflies everywhere. Yet many species are hard to find unless you know how to search for them. Inexperienced observers often see few species and encounter the same species over and over again. Detecting hoverflies in your environment efficiently requires knowledge and experience. The best way and places to find hoverflies vary from district to district, from day to day and even during the day. Here are some tips and tricks that can help you find many species of hoverflies.

Flowery places

Okay, we realise this may sound rather obvious: hoverflies visit flowers for foraging. But there is more to this. Not all flowers are equally favoured, and flower preferences differ between species. If your aim is to find as many species as possible in a single place, it is best to combine two strategies: finding a patch of flowers that attracts many and a large variety of hoverflies and finding specific flower types known to be visited by specialised species. Flowers that attract many hoverflies usually produce much and easily accessible nectar because most hoverflies require nectar for feeding and have a short tongue. Very attractive hover-fly flowers include umbellifers (e.g. Cow parsley, hogweeds, angelicas, parsnips), willows, maple trees (including Sycamore), hawthorns, *Prunus* trees (including Blackthorn and cherries), brambles and Ivy. Some species specialise in flowers with nectar that is more difficult to reach. For example, *Rhingia* prefers the flowers of the mint family, with their deep, narrow calyx. Some hoverflies have a pronounced preference for a particular flower colour. *Sericomyia bombiformis* and *S. superbiens* like large blue and purple flowers (Devil's-bit scabious, Field scabious, thistles) while *Cheilosia canicularis* prefers large, yellow flowers of the aster family and is especially fond of Canadian hawkweed. Also, the height and size of the flowers is important. Have a close look at small and low flowers (stitchworts, eyebrights, Common tormentil, bedstraws) for *Sphaerophoria, Paragus, Eumerus* and *Neoascia*. Conversely, there are also species that live high in the canopy of forests and visit flowers in trees and at the top of shrubs (e.g. *Mallota, Criorhina, Pocota*). Binoculars can come in handy here! And, maybe unexpectedly, on flowering grasses, sedges, rushes and plantains you can find a wealth of sedgesitters. Finally, some species are rarely seen on flowers at all. *Xylota* walk over leaves of trees in the forest edge, looking for windblown pollen and the sugary excretions of aphids. Their food disappears during heavy rain and they may turn up at flowers for a short time afterwards. Finally, *Microdon* species do not appear to visit flowers at all!

The same flower may prove less or more attractive to hoverflies, depending on where the plant grows. Most of our hoverflies are linked to forest or tree habitats so are mainly found on flowers near forest. All hoverflies hate breezes, and this certainly applies to the specialised forest hoverflies (e.g. *Brachyopa*). A small, sheltered woodland ride or a forest glade with the right flowers can be extremely rewarding. Some forest hoverflies take it a step further and rarely come out in full sun: the delicate *Baccha elongata* and *Sphegina* are often found in dappled sunlight or full shade, the latter nearly always near small streams in forest.

Foodplants: know your ecology!

Our knowledge of the ecology of hoverflies is rapidly increasing, which is extremely helpful when trying to find them. Knowing more about larval ecology, and especially the host plants of herbivorous species, helps us to predict which species can be seen where based on the composition of the vegetation. As soil type, current and historical land use, landscape composition and management, and climatic variables together shape the local vegetation, these variables also may indicate which (especially herbivorous) hoverfly species can be expected where.

Some examples. *Cheilosia*, *Eumerus, Merodon* and *Portevinia* are all herbivorous hoverflies and often depend on one or a few plant species for their development. *Portevinia maculata* and *Cheilosia fasciata* can be found only in the vicinity of Wild garlic. *Cheilosia pubera* lives in Water avens. The distribution of these plants in Great Britain is well known and can be discovered with a few clicks: then, you can know exactly where to look for these hoverflies. This knowledge can even help you to fill in apparent gaps in the distribution of a hoverfly in areas where there are few records. Be aware, though, that some species are quite mobile, and where they complete their larval stage may be kilometres from where they forage as adults. For example, the larva of *Cheilosia canicularis* lives in Butterbur in river valleys, but the adults are mostly found at some distance on the drier higher banks, e.g. on Canadian hawkweed. Again, knowing this, you can optimise your catch. And although the foodplants of many *Cheilosia* species remain unknown, it just takes a bit of luck to get a break. If you see a female ovipositing on a plant – maybe a pre-flowering leaf rosette – try to make a note of species of fly and plant. This information could be significant and add to our understanding of that species. You will soon learn that female *Cheilosia* species spend some of their time sitting on their foodplants; e.g. *Cheilosia albipila* often rests on rosettes of Marsh thistle.

It is not just adult herbivorous hoverflies that have a special attachment to certain plant species. The larvae of many species prey on aphids, psyllids or caterpillars that themselves live only on one or a restricted set of plant species. As a case in point, the larva of *Platycheirus perpallidus* is fond of the aphid *Trichocallis cyperi,* which is almost only ever found in large stands of Bottle sedge. Moreover, *P. perpallidus* appears to be restricted to the places where Bottle sedge grows in permanently inundated conditions. In the genus *Dasysyrphus*, there are species whose larva eats aphids only on deciduous trees, and others eat aphids only on coniferous trees. This specialisation, of consuming aphids of either deciduous or coniferous trees, can also be found in various other groups of hoverflies (e.g. *Parasyrphus*, *Melangyna*, *Didea*), resulting in completely different species communities in, for example, a spruce forest compared to an oak forest. We have tried to include as much of this critical information as possible in the species texts, to help you find and/or identify these species.

Weather

Hoverflies in our temperate region like sunny weather and mild temperatures. On a sunny day, they are most active at a temperature of about 16–22°C (in the shade). When it is colder, they will often sun themselves to warm up. After a cold spring night, you can find large numbers of hoverflies in the morning on the east-facing forest edges and hill slopes, sitting on the leaves or the grass. The bonus for us is that all hoverflies participate in this: you could find a *Criorhina,* a *Brachyopa,* a *Mallota,* a *Syrphus* and a *Melangyna* next to each other in the morning, but later in the day they all will have returned to their specific habitats and some may become near-impossible to find. On rather cold but sunny days, you can also see this behaviour in the evening on the western edge of forests. When it is overcast, there are often still a few species active; some are even more active than at full sun, such as *Platycheirus* and *Melanostoma*.

In heatwaves, hoverflies are more likely to visit flowers in the shade (especially in the forest interior) or to restrict foraging to early morning before the heat builds up. At midday, most species seem to have disappeared. They have gone to a cool resting place (often the underside of leaves) to avoid desiccation and overheating. Moreover, flower nectar production and availability is much reduced during the heat of the day so foraging is less efficient. Some of the flies will start searching for water, especially during prolonged dry periods and with strong dry winds. Damp forest paths, edges of streams, and other similar watery habitats can attract large numbers of flies at such times. As when hoverflies are sunning, the species palette that descends for drinking is different from that on flowers. Species of the genera *Parasyrphus* and *Neocnemodon* are remarkably more common at drinking sites, and also the enigmatic *Callicera* species are often seen this way. Presumably they are largely arboreal species that come down to drink during droughts. If you can't find a natural drinking spot, you can easily make one by lightly spraying the lawn with a garden hose. This works best in a semi-shaded spot. In a garden in Belgium, over 80 species – including all five species of *Neocnemodon* – were observed this way, while the *Neocnemodon* species were otherwise rarely or never seen there.

When the sun breaks through the clouds after a few days of bad weather, the numbers of hoverflies on flowers may be unusually high. Their energy reserves are low, and they need to refuel. In contrast, when suitable weather continues for a longer period, the number of hoverflies on flowers may decrease after a few days. The flies' energy reserves are optimal and the animals are focusing now on reproduction.

A last but very important point is that hoverflies generally hate breezes. On your search for hoverflies, try to seek out flowers that are sheltered from the wind. And if the day is cool and breezy, by concentrating on sheltered flowers your search can be very productive.

The combination of all the above factors means that the number and diversity of hoverflies can change greatly from day to day, and that the best spot to look for them may change constantly.

What is the best season?

Adult hoverflies can be seen almost all year round, but the best season is spring. The vast majority of hoverflies overwinter as pupa or as larva. Only a few species overwinter as adults (e.g. *Eristalis tenax, Eristalinus aeneus* and *Episyrphus balteatus*). They start to fly as soon as the weather permits, and are therefore also seen on warm sunny days during the winter months. Species that overwinter as pupae or larvae may fly from March to October (nowadays often February–November), depending on species. Hoverflies can be divided into two large groups based on the number and timing of their cohorts throughout the year. The first group flies in both spring and summer. They have several generations in a year: usually two, but some species produce successive cohorts as long as the weather permits (e.g. in some *Paragus* species there are up to four generations in a year). Usually the cohorts vary in size: some species have a large spring generation and only a small generation later in the summer, but in other species the summer population is much larger (especially in species that migrate). The second group consists of the species with only one generation per year. Almost all of them fly exclusively in spring, which is why spring is the best period of the year to find many species of hoverflies. Species tallies for each day of the year have a bimodal distribution, with the highest peak mid-May to mid-June (depending on height and latitude), and a second but much lower peak at the beginning of August.

Specific behaviours

Many hoverflies gather at high points in the landscape, in a behaviour called 'hill-topping'. Any small hill will do, as long as it rises above its surroundings. Hills with some bushes or small trees near the top yield more species than treeless hill-tops. Each species has its own behaviour at hill-tops, and this may vary during the season or between hill-tops. Many hoverflies form loose swarms at wind-sheltered spots near the top (*Epistrophe*, *Epistrophella*, *Melangyna*, *Platycheirus*, *Rhingia, Xanthogramma*), while others sit on bark (*Chalcosyrphus*, *Ferdinandea*), near tree bases (*Criorhina*), on leaves of bushes and trees (*Chrysotoxum*, *Pipizella*, *Pipiza*) or on the ground (*Eupeodes*, *Eristalis*). The more isolated and the pointier a hill-top, the more concentrated and abundant are hoverflies there. Because several very rare species perform hill-topping they are (far more) easily found this way than using other searching techniques. For example, *Callicera rufa* will often sit on or hover next to large trees on hill-tops.

A very interesting group of hoverflies with a specific behaviour are the sap-run species. Species of *Brachyopa*, *Hammerschmidtia* and *Ferdinandea*, and *Volucella inflata*, are some of the hoverflies you can find at sap runs on trees. Sap runs can be looked for but can also be found by their typical scent. Not every sap run will yield the same species: different trees may attract different species of hoverflies, but also size and exposure of the sap run play a role. A large sap run with abundant sap will more likely attract *Brachyopa insensilis*, *Sphiximorpha subsessilis* or *Volucella inflata* than will a small, hardly visible subcortical sap run, where more often *Brachyopa bicolor*, *B. scutellaris* or *B. pilosa* are seen.

Hoverflies as a way to evaluate and appreciate the environment

One of the most fulfilling outcomes of accumulating knowledge on hoverflies is that, at some point, you will be able to read the history of a place and to evaluate its current ecological state only by looking at its hoverflies. Finding yourself in a place that has all the species that it possibly can have is not only very exciting. That knowledge also tells you that the place has a long, stable and benign history, that current threats have not (yet) had their worst effects, that biodiversity is still at its zenith. Such things often cannot be read from how habitats look today. All too often, many species of hoverflies are lacking even from our best-looking natural areas, whereas outwardly degraded systems may be hotspots of hoverfly diversity. A great instrument to detect this hidden quality of natural areas is *Syrph the Net: the database of European Syrphidae (Diptera)* by Martin Speight and collaborators. It provides reference data to calculate the quality and intactness of a natural area based on the presence of hoverflies.

Identifying hoverflies

Photography

With this field guide in your pocket, you can go outside to identify hoverflies. Many observers in the field will only use a camera and will not want to collect specimens. You will soon learn that many hoverflies can be identified by photos alone, especially if you use a good macro lens and photograph the fly from different angles so you can see all necessary features. The great thing about photos is that they can be posted on online forums such as the UK Hoverflies Facebook Group, iNaturalist (https://www.inaturalist.org) and Observation.org (https://observation.org), where experts can often accurately identify even poor photographs, based on the overall impression or appearance of a species (the 'jizz'). Such experts can also quickly tell you if your identification is wrong. Many photographs, however, do not show the fly in such a way that permits identification, so a substantial number of hoverflies can never be identified from photographs.

Catching

With increasing concerns about the fate of hoverflies and our pollinators in general, paradoxically, it has become even more important to regularly catch and even collect hoverflies. Each observation of a hoverfly is valuable, yet – as mentioned above – using photography alone means that a large share of the hoverfly fauna cannot be identified, and information about it will not be stored in databases and will be disregarded in analyses.

For catching and identifying in the field, an insect net and a hand lens are needed. Opinions differ, but a net with a mouth diameter of *c.* 16 inch (40 cm) is the most popular. In spring, a longer pole (and preferably an extendible one) on the net can be useful to catch flies in flowering bushes and trees, but in meadows a shorter stick is usually handier. As to the colour of the net, some prefer darker colours (green, black) as it is believed that they allow for a stealthier approach to the fly, yet a white net may make it easier to find the fly once it is in the net. With a hand lens, you can much better study the fly in the field than with the naked eye. Lenses often have a magnification of 10x to 20x. 10x may be too small a magnification to, for example, determine the colour of the hairs on the top of femur 3 in *Syrphus*, so 15x or 20x magnification might be preferred.

Unfortunately, you will discover that a certain proportion of species still cannot be identified in the field (this number will decrease with experience). There are two options now. Either you let the fly go and accept that you cannot identify everything, or you take the fly home to study it further under the microscope.

Collecting and curating

If a specimen is required for further study, a tube is needed. You can take small tubes and transport flies individually or take a slightly larger plastic bottle with a narrow entrance and stopper, and keep all the catches for a specific area in it. If you visit several areas in one day, make sure you use different bottles and remember which flies come from which area, or take several labelled bottles. Once captured, the fly must eventually be killed. This can be done by putting the jar in the freezer for a few hours, or by adding a small wadge of tissue/toilet paper soaked with a little ethyl acetate to the jar. The ethyl acetate sedates the flies within a minute but can take an hour or so to actually kill them, depending on how much you use (use too little and they may eventually wake up again!). Nail polish remover, which is more readily available, can be used instead of ethyl acetate. Be aware, use the acetone-free variant. A more natural but at least as effective method is putting bruised young shoots of Cherry laurel into the jar.

After killing, the animal must be pinned. One approach is to pin the fly with a long pin. The pin is inserted through the thorax of the fly, approximately two-thirds of its length back from the head, leaving room on the pin above the fly for your fingers to handle it, and room below to add labels and insert the pin into the collection box or tray. There are various sizes of insect pins on the market, but size 0 or 1 will do for most species. The other approach is to pin the fly with a shorter micro-pin onto a Plastazote stage that has a longer pin going through it.

By pinning the fly immediately after killing it, it is still flexible so that it can be set up neatly. The scientific value of the fly and your collection overall increases if you make sure that the characters important for identification are clearly visible. This can be done by pinning the fly temporarily on a thick piece of foam, so that its body (nearly) touches the foam. Then the legs can be folded open and rested on the foam. Next, the wings can be nudged opened with a pin and the head can be turned to face right if necessary. If the fly is a male, it is best to unfold the genitalia, especially in species where this helps

with, or is even necessary for, identification. Here, again use an insect pin, and pull the genitals out a little before you pin the fly close to the foam (Photo 2). *Sphaerophoria* are the easiest to prepare the genitals of: turn the genitals upwards so that they protrude above the abdomen. Usually they will stay in that position, displaying the important features. For other species, the genitalia usually cannot remain unfolded after pinning. Once the fly is just above the foam, fix the abdomen with pins, pull the genitals out again and fix the genitalia with pins (Photo 2). With micro-pins and a trained eye, you can now also stretch the genitalia themselves in such a way that all the diagnostic parts will be visible. Quite a hassle, but after some practice this becomes routine work.

Store your pinned fly in a closed box away from walls or the floor to inhibit pests from entering the box. After a few days, the fly will have hardened and can be added to the collection. But not before putting a label under it (Photo 3). Unlabelled flies are almost worthless because you don't know where and when they were collected. Labels are easy to make yourself, but prepare them to last for long. For example, use a 5pt font size and print, using high-quality ink, on 120g paper, so that you get small, sturdy labels. Always print collection location and date, and the name of the collector, on the label. Try to be specific enough with the location that other users of your collection know exactly where you mean. Stating GPS coordinates helps enormously. The identification can be put on a second label, preferably with the scientific name, year of the identification and the name of the person who made the identification. Using a second label for the identification means that, should the identification change in the future, the location label can be untouched and only the label with the name has to be changed. The identification should be placed under the first label. It is useful to pin this label upside down, so that it is easy to read when you turn the fly upside down.

It is at this point that the fly can finally be moved to your collection. As with your temporary drying boxes, store your collection boxes somewhere dark and dry, away from walls and the floor to avoid mould and pests. Various carpet beetles and booklice will love your collection, and some collectors swear they have a preference for rare species, so buy storage boxes that close properly. In the long run, no collection will be spared. Check regularly for frass lying on the labels or underneath the flies. If you are too late, whole flies may be eaten. One solution is to place the storage box in the freezer for a day or two. A common preventive or curative measure is placing mothballs in the collection box. This is very effective, but has two major disadvantages: mothballs have an unpleasant smell and may be toxic to humans.

After pinning the fly, it can be studied under the stereomicroscope. An inexpensive stereomicroscope usually has a magnification range of 10x to 40x. This range is good to observe virtually all of the features described in this book. Everything you need to catch hoverflies and/or to start a collection is available at Veldshop (www.veldshop.nl/en/), including expert advice from the first author of this book. In the UK, advice is available from the Dipterists Forum (https://dipterists.org.uk) and Hoverfly recording Scheme (http://hoverfly.uk/hrs/; which runs regular workshops).

Photo 2 Example of a freshly pinned *Eristalis tenax*.

Photo 3 Example of a pinned *Chrysotoxum cautum*. There is a second label with the specimen identification information below the location label.

Is collecting still necessary?

Killing and collecting hoverflies is often criticised as being unethical, outdated and unnecessary in this digital era. Collecting hoverflies also is considered to be a threat to the persistence of populations. Indeed, catching butterflies and dragonflies has become unusual and frowned upon, and some people view collecting hoverflies, bees, moths and the like the same way. These arguments may seem to make sense, but we want to stress that collecting hoverflies in general does not harm populations (see below), is still useful and can actually be essential in conserving scarcer species.

Nowadays almost every bird, butterfly or dragonfly that is observed in Western Europe can be identified without the need to kill it. A few decent photos suffice to identify it and corroborate the observation. But the situation is different with hoverflies because: 1) many species can be confirmed only by examining their microscopic features, and 2) we continue to find new species and to redefine how we identify the known ones. Species to be identified under the microscope can be studied only by first killing the fly. This applies to all species for which the genitalia need to be examined, but also to species where only a set of subtle characters can resolve the identification, as for instance in many *Cheilosia*. Should collecting stop, we would get no more information about the occurrence of, for example, *Pipizella, Cheilosia* or *Sphaerophoria* species. An estimated 35% of the species in this book cannot be identified with certainty from regular photos. In reality, this number is often higher because photographs rarely show all the critical characters. In fact, the effect of only 'photographic sampling' in hoverflies is already apparent: recent trend analyses revealed that the numbers of small, dark and difficult to identify species have generally decreased and those of large and easily recognised species is increasing. This illustrates the need for continued collecting of hoverflies. National recording schemes are the backbone of scientific research on status, trends, range shifts and other key population information in hoverflies, but the value of such schemes depends wholly on accurate and unbiased recording.

But even for species that are easy to identify, collecting specimens can still be useful. A collection is a potential archive of genetic variation and thus an important source for scientific research on the ecology, taxonomy, phylogeny or population health of a species. As to the taxonomy of hoverflies, even today, lookalikes of common and easily identifiable species continue to be discovered. A good example is *Epistrophe olgae*, a twin species of *E. nitidicollis*, that was described from the Russian Far East. After people realised it occurred in Europe as well, it was found to be widespread in Europe. Thanks to all the '*Epistrophe nitidicollis*' collected so far, we are able to reconstruct its history and prevalence here. Another example is *Myathropa florea*, an unmistakable species that therefore does not need to be collected. However, recent genetic research has shown that it is actually a multi-species complex, two of which are probably present in Great Britain. For researchers, it is essential that this *M. florea* is collected so that large numbers of it can be studied and a first picture of its distribution and ecology can be drawn.

Furthermore, for the individual, to become really good at identifying hoverflies, having a reference collection is indispensable. It is recommended that you have several specimens of each species, preferably from several regions or different times of the year. Indeed, many species may appear different in spring than in autumn. Often spring (or late autumn) generation individuals are furrier (e.g. *Eristalis pertinax*) and/or darker (*Episyrphus balteatus*, *Eupeodes corollae*), compared to individuals of the summer generation. By archiving this variation in your collection, you learn which characters are variable and to what extent, or you may discover new characters of a species. Eventually, this will help you improve your identification skills and produce more reliable data.

The scientific value of collecting is clear, we believe, but does collecting flies come at a cost to the population of the species? There are no studies that have answered this question, but several facts suggest this is not the case. Hoverflies generally are hard to find compared to many other insects (e.g. butterflies, dragonflies). Many species are arboreal or hide within vegetation. They are also quite mobile and will be spread over a large area. These attributes make it difficult to collect a significant part of a population. That said, where rare species are clearly confined to a small area and can be collected efficiently, it is better not to collect more than one to three individuals, and preferably to collect just males, since this affects the population less than collecting females. Furthermore, do not collect in places where collecting is not allowed, such as protected nature areas. If you want to collect there, you must contact the owner in advance and seek permission. And, above all, always collect with respect for the environment.

Creation of this guide

Photography

For this field guide, we decided to use photos of pinned hoverflies rather than photos taken in the field. Using the latter is destined to failure if your goal is to show the diagnostic features of all species. About 70% of the photos are from flies in the authors' collections. For the remaining, 28 other collections provided specimens, for which we are most grateful. As much as possible we have used flies originating from the geographical scope of this book (mainly Belgium and the Netherlands), but for some 100 species, specimens from outside the area were obtained. Details on the origin of each of the specimens used herein can be obtained from the authors.

The next challenge was to get everything photographed. To show the details of small hoverflies, a good camera set-up is a must. Building a good set-up is a time-consuming job that requires a lot of photographic knowledge. Fortunately, macro photographer Christophe Brochard was willing to share his expertise with us and helped extensively with assembling, and learning to work with, the camera set-up. The set-up consisted of a Canon 6D body, an MPE-65 macro lens, two flash units

and a macro rail along which the fly can be moved. For the tiniest details, such as of the genitalia, the set-up turned out to be inadequate and instead we used a microscope that had been converted into a camera by Christophe. Getting a fly completely in focus from top to bottom is not possible with a single photo because the depth of field is not sufficient. We used focus stacking to solve this. Here, the fly is moved gradually a fraction of a millimetre along the macro rail for successive photos, so the whole depth of the fly is covered. Image-merging software (Helicon Focus) was used to create a single completely sharp picture of the fly. More than 200 photos were taken to produce a single sharp image of the largest flies. All up, the number of clicks of the camera body for this project amounted to more than 150,000. Once the sharp photo was composed, it was further processed using Photoshop to: remove dust, loose hair and pollen grains from the animals, straighten their legs and wings, remove the pin, remove any reflective flash light or restore fresh colours (for discoloured specimens) (Photo 4).

Photo 4 In order to get a good picture of *Spilomyia manicata* for this guide, three different specimens have been photographed. Each photo consists of dozens of photos that together result in one sharp image. Extensive processing of the three photos in Photoshop yielded the final image at the bottom right.

Distribution maps and phenology bars

The maps cover more or less the geographical scope of this book, that is most of north-western Europe, including Belgium, Denmark, Luxembourg, Ireland, the Netherlands, Great Britain, and parts of western Germany and northern France. Observations made by both professionals and volunteers have been collated in recording schemes in some countries. Such data, for this purpose restricted to data gathered since 1990, were available from Belgium (128,815 records), Great Britain (1,115,878 records) and the Netherlands (429,518 records). Figure 1 shows the combined coverage of these three recording schemes in terms of occurrences in 10km by 10km squares (hectads). The numbers of records per square gives an indication of the distribution of recording effort. This clearly tends to follow the pattern of human population density, with recording concentrated in densely populated areas such as the south-east of England and the Netherlands and sparse in thinly populated areas such as the Highlands and islands of western Scotland.

This variation in recording effort had a profound impact on our ability to map the distribution of hoverfly species. On average, the more records, the greater the chance of detecting a species. Thus, species that tend to occur in poorly recorded areas are less likely to be detected and their range is likely to be underestimated. Consequently, when assessing the distribution of a species, it is necessary to take the distribution of recording effort into account. This is an active area of research and increasingly sophisticated statistical methods are being developed. The method used here was FRESCALO. FRESCALO works by calculating the weighted, relative frequency of species in 'neighbourhoods' – a cluster of nearby localities (hectads in this case) – and then rescaling these frequencies according to the amount of recording in that neighbourhood. For each hectad, the list of species occurring in its neighbourhood is drawn up and the weighted relative frequency of each species is calculated as the sum of the number of occurrences falling in each hectad multiplied by the weight assigned to that hectad. Figure 2 shows how this sequence of steps results in a map for an example species, *Tropidia scita*. The middle pane illustrates the rescaled frequencies calculated by the FRESCALO analysis – the greater the rescaled frequency, the darker the green. Finally, these results are contoured to produce only three shades of green, for the final map. They show areas in which the species is abundant (dark green) through to scarce (lightest green).

In the areas where detailed recording scheme data were not available, only whole-country presence (Denmark and Luxembourg) or whole-region presence (France, Germany and Ireland) were mapped. Data were sourced from publications and consultation with local specialists to attempt to assemble up-to-date species lists for these areas. Not all areas have up-to-date species lists available and it was not possible to get hold of some recent data within a short time span. Therefore, some variation in how up-to-date the data were was inevitable. This may affect the distribution for species that have undergone drastic recent changes in range (e.g. *Melangyna pavlovskyi*, *Cheilosia luteicornis*, *Leucozona glaucia*). The coloured areas in Figure 3 show the countries and regions for which species lists were assembled. The grey areas in Figure 3 are outside the scope of this book.

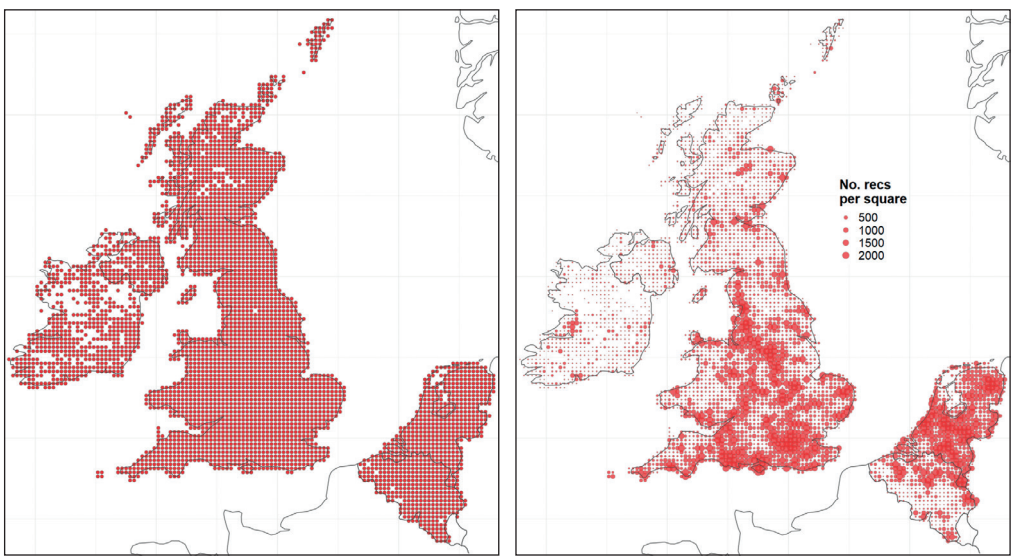


Figure 1: Coverage – the hectads from which at least one record was received, 1990–2021 (left) and the numbers of records received from each square (right), where the area of the symbol is proportional to the number of records (capped at a value of 2,000; actual maximum was 16,071).

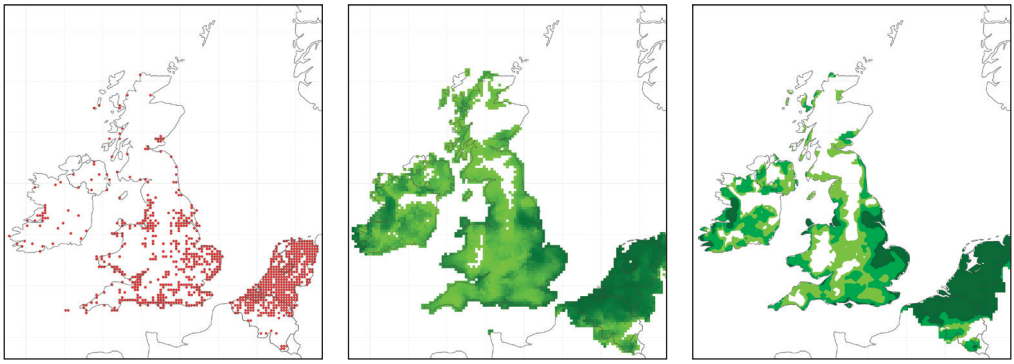


Figure 2: Distribution of *Tropidia scita*. The left-hand pane shows the hectads into which the available observations (1990–2021) fall. The middle pane shows the rescaled frequencies resulting from a FRESCALO analysis and, in the right-hand pane, these are contoured to produce areas in one of three shades as used in the maps included in the text.

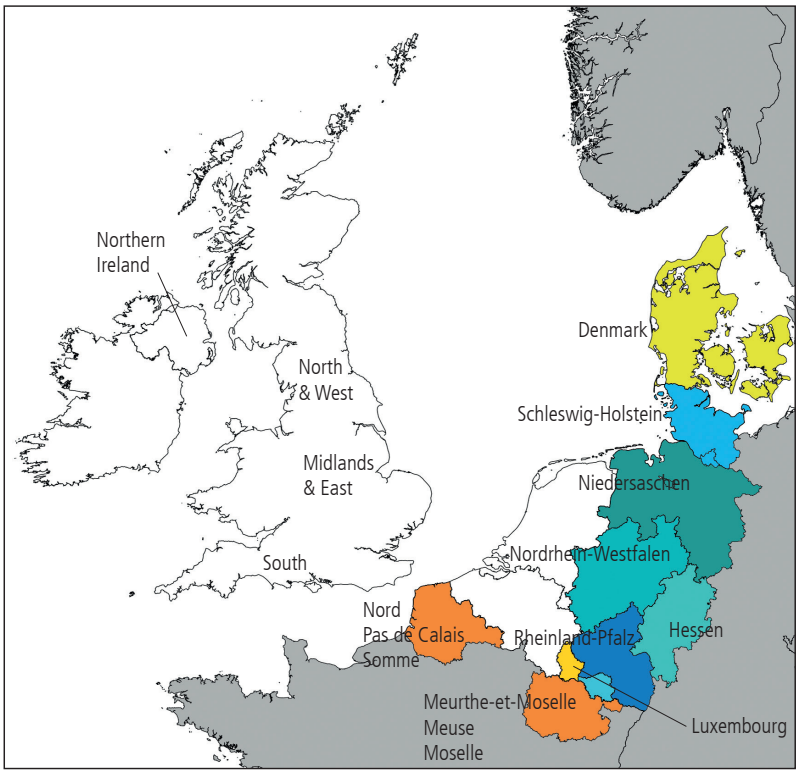


Figure 3: Countries and regions for which species presences were mapped.

For the areas where detailed data were available, data could be used to present the phenology of a species. To represent regional variation, phenology strips for three different regions are shown: Belgium and the Netherlands, southern Great Britain and northern Great Britain. The phenology bars represent the weekly proportion of observations relative to all observations over the whole year. Deeper colours indicate weeks with a higher proportion of observations or, in other words, the peak of a species' flight season. With such a diverse and large set of databases, it was impossible to clean all data of erroneous records. It is likely that for some species, larval records are shown, which may disturb the actual pattern, but this is rare and the overall pattern is clear. A very small lower threshold value was used, which helps to reduce the effect of small outliers for many species, but this also can make a species invisible during periods when it occurs in very low abundances. All in all, the phenology strips give a good view on seasonal fluctuations in abundance for the main flight time of each species.

Scientific names

How useful it would be if scientific names of hoverflies did not continually change, but they do. This book introduces some recently published name changes that have not appeared in previous keys. The fact that names change so often is (at least partly) a reflection of our increasing understanding of biodiversity. Cladistics, the process by which evolutionary trees are created and classifications refined, recently gained momentum through DNA research. In just a few years, DNA has become a prime source of information in the study of phylogenies and variation within and between species. This alternative approach to biodiversity, compared with traditional morphological research, can yield surprising insights. Cryptic species (species that can hardly be told apart morphologically but differ genetically) are revealed, but very different-looking species have also proven to be one species (though care is needed, as some distinct species – e.g. in *Melanostoma* or *Neocnemodon* – can have almost identical DNA, depending on which fragment of the DNA you are using). This type of research is rapidly becoming standard, but we are still in the middle of the process of re-evaluating current taxonomies, so further name changes can be anticipated.

Common names

Previous British field guides did not use common names for hoverflies. We think it is important to introduce them here. It has probably never been more important that hoverflies become popular amongst a wider audience to raise awareness of these flies, and insects in general, and we know common names do help. We have mainly relied on two existing sources that have proposed common names: the *Field Guide to the Flower Flies of Northeastern North America* (Skevington *et al.* 2019) and the excellent Flickr website of British entomologist Steven Falk (http://www.stevenfalk.co.uk). Although the former does not include many European species, it introduces common names for most of the genera found in the area this book covers. To standardise common names at the genus level for Europe and North America, we adopted most of them. The species names, therefore, are often a mix of the names proposed by Steven Falk and the American genus name. We realise common names may already exist regionally and that the new names may cause some controversy, but we feel our approach is the best way to bring about a standardised, consistent and widely used English nomenclature of hoverflies.

Geographical scope

This book contains all species of hoverflies that have been observed in the wild in the British Isles, Denmark, the Netherlands, Belgium and Luxembourg and will also work well for individuals observed in western Germany and northern France. Because the fauna of a region is constantly changing, it is quite a challenge to produce a key that will not become outdated in a few years. For instance, in the small and thoroughly investigated country of Belgium, over the last 12 years, 18 new species of hoverfly have been reported. These include: cryptic species that have been present there for some time but have only just been discovered as we have learned how to identify them; species that have recently expanded their distribution to Belgium; and species that have been newly described. Through intensive dialogue with top researchers in hoverflies, and implementing their latest taxonomic insights, and by gathering knowledge about the occurrence and ongoing range shifts of species from neighbouring regions, we have aimed to produce a guide that is at least current at the date of publication and that hopefully anticipates upcoming changes to the fauna of the geographical scope of this book. That said, we realise that sooner or later someone will make an unexpected discovery that we didn't anticipate and – to be honest – we look forward to that too!
We wish everyone good luck and lots of fun with this guide.

Right: *Melangyna quadrimaculata* (female) feeding on sallow pollen.

Keys

Main key

1a Antennae short, much shorter than head → 2
1b Antennae elongated, as long as or longer than head → **Key 1** p. 24

2a Wing: crossvein r-m located before the middle of cell dm → 3
2b Wing: crossvein r-m located in or after the middle of cell dm → 5

3a Wing: cell r1 open; the fly is large or small → 4
3b Wing: cell r1 closed; the fly is large, at least 11 mm. → **Key 2** p. 28

4a Scutum and scutellum black or blackish; if hind margin of scutellum yellow, then abdomen without clear yellow markings → **Key 3** p. 30
4b Scutellum paler than scutum, usually yellow; if only hind margin of scutellum is paler, abdomen with yellow markings → **Key 4** p. 64

5a Wing: vein R_{4+5} sinuous → **Key 5** p. 86
5b Wing: vein R_{4+5} straight or nearly so → **Key 6** p. 94

Key 1

1a Abdomen black with bright yellow markings → 2
1b Abdomen brown, black or red → 14

2a Abdomen cylindrical, black, with three equal narrow yellow bands → 3
2b Abdomen wide, yellow marks not in the form of three equal narrow bands → 5

3a Antennae placed on a frontal prominence → 4
3b Frontal prominence absent → **Northern Saprun Wasp Fly** *Sphiximorpha subsessilis* p. 223

4a Hind half of scutellum black → **Common Wasp Fly** *Ceriana conopsoides* p. 223
4b Scutellum entirely yellow → **Southern Wasp Fly** *Ceriana vespiformis* p. 223

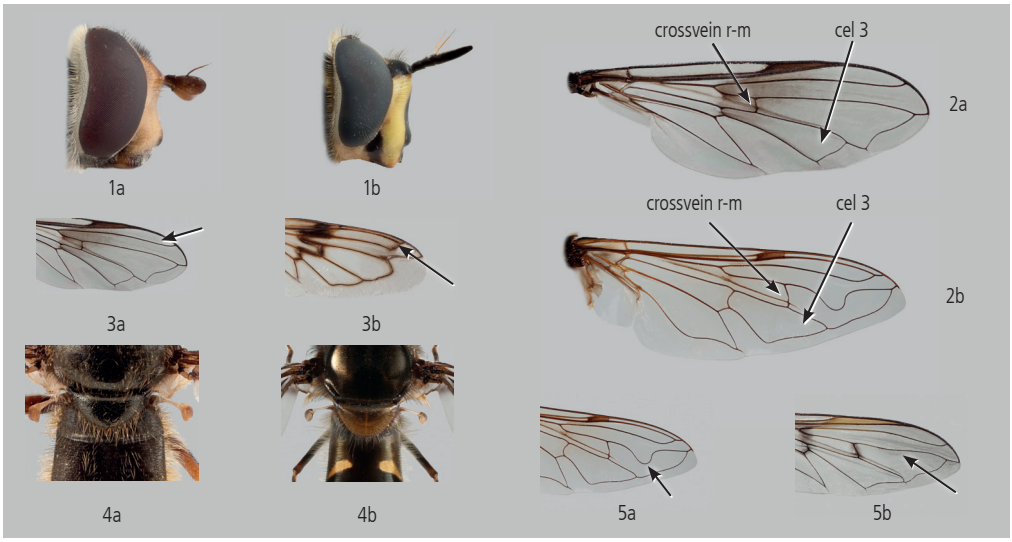
5a Tergites 2-4 with roughly equal yellow markings → 6
5b Tergite 3 black or with yellow markings much smaller than those on tergites 2 and 4 → **Two-banded Meadow Fly** *Chrysotoxum bicinctum* p. 111

6a Yellow pairs of markings on tergites 3 and 4 do not reach the side margin, therefore side margin black; hind margin of tergites 3 and 4 black or narrowly yellow, if narrowly yellow, not connected with the yellow markings above it → 7
6b Yellow pairs of markings on tergites 3 and 4 reach the side margin; hind margin of tergites 3 and 4 yellow, in the hind corner connected with the yellow markings above it → 8

7a Fore and mid femora entirely yellow or only narrowly black at base; katepisternum usually with yellow spot → **Hook-banded Meadow Fly** *Chrysotoxum festivum* p. 111
7b At least basal fifth of fore and mid femora black; katepisternum usually black → **Smooth-banded Meadow Fly** *Chrysotoxum vernale* p. 111

8a Third antennal segment, measured from outer upper edge, as long as or longer than first antennal segment and second antennal segment combined → 9
8b Third antennal segment, measured from outer upper edge, shorter than first antennal segment and second antennal segment combined → 12

9a Male: genitalia not very large. Female: tergite 6 without a median longitudinal keel. Third antennal segment, measured from outer upper edge, longer than first antennal segment and second antennal segment combined → 10
9b Male: genitalia strikingly large. Female: tergite 6 with a median longitudinal keel. Third antennal segment, measured from outer upper edge, as long as first antennal segment and second antennal segment combined → **Large Meadow Fly** *Chrysotoxum cautum* p. 109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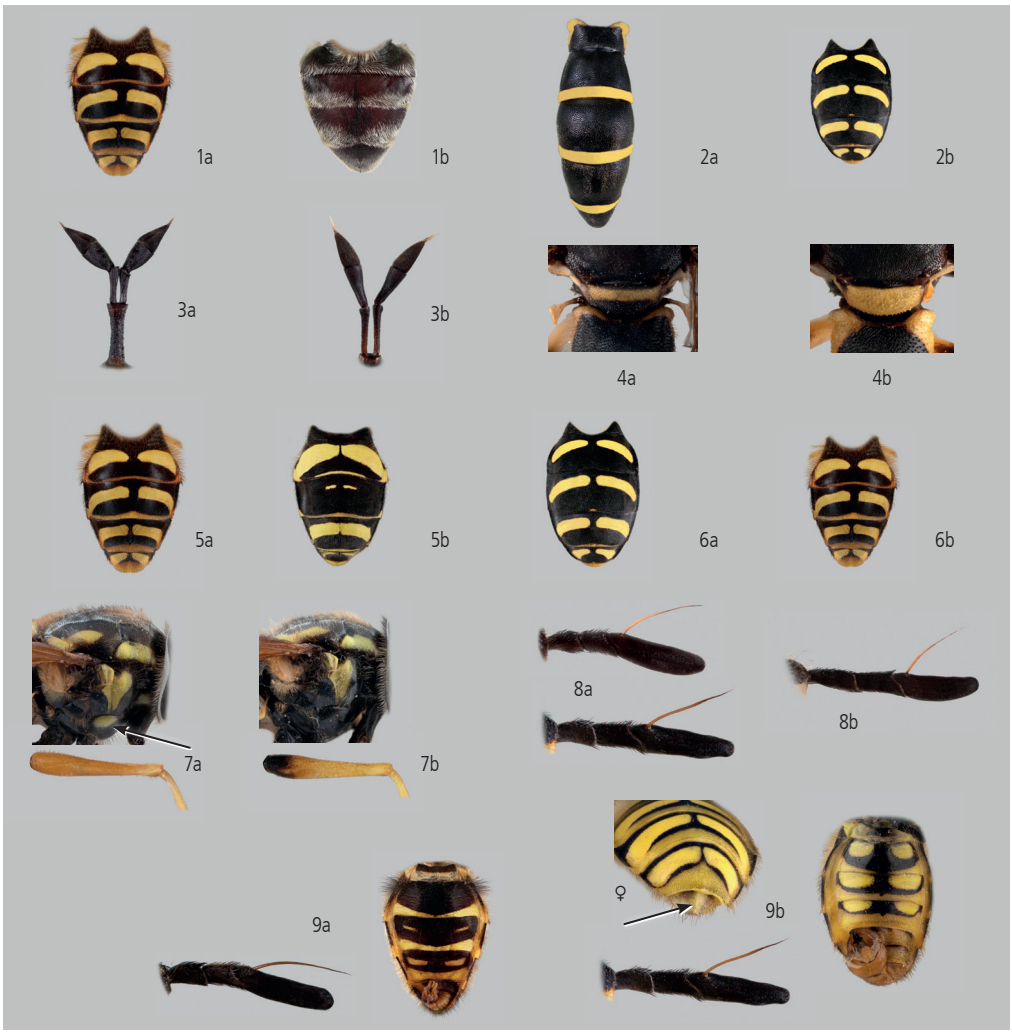
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10a Abdomen with long hairs, hairs as long as or longer than width of hind tarsus → 11
10b Abdomen with short hairs, except for anterior corners of tergite 2; hairs distinctly shorter than width of hind tarsus
→ **Naked Meadow Fly** *Chrysotoxum lessonae* p. 107

11a Smaller species; abdomen short and convex; scutellum yellow with black centre; anterior margin of wing not darkened
→ **Little Meadow Fly** *Chrysotoxum arcuatum* p. 109
11b Larger species; abdomen not so short and convex; scutellum black except yellow anterior part; anterior margin of wing darkened → **Greater Meadow Fly** *Chrysotoxum fasciolatum* p. 109

12a Black anterior band on tergites 3 and 4 runs continuously to side margin → 13
12b Black anterior band on tergites 3 and 4 is broken just before side margin
→ **Broken-banded Meadow Fly** *Chrysotoxum octomaculatum* p. 113

13a Anterior margin of yellow markings on tergite 2 straight and parallel with anterior margin of tergite, except on outer sixth of their width, where they bend abruptly backwards. Therefore, band strongly widens towards side margin
→ **Verrall's Meadow Fly** *Chrysotoxum verralli* p. 113
13b Anterior margin of yellow markings on tergite 2 convex, only roughly parallel with tergite anterior margin for at most half their width, curving gradually backwards. Yellow band not strongly widening towards side margin
→ **Variable Meadow Fly** *Chrysotoxum elegans* p. 113

14a Abdomen copper or bronze, without red markings → 15
14b Abdomen largely red → **Vermillion Fly** *Psarus abdominalis* p. 267

15a Arista implanted on base of upper surface of third antennal segment; eyes bare; wing vein R_{4+5} with an appendix into cell r_{4+5} (**Ant flies** *Microdon*; see Key 1A for identification of puparia) → 16
15b Arista implanted on tip of antennae; eyes with hairs; wing vein R_{4+5} without an appendix → 18

16a Scutum with pale hairs only; posterior margin of scutellum straight or only slightly concave → 17
16b Scutum with a band or two patches of black hairs between wing bases; posterior margin of scutellum between teeth distinctly concave → **Chalk Ant Fly** *Microdon devius* p. 105

17a Scutellum same colour as scutum, dark → **Heath Ant fly** *Microdon analis* / **Large Ant Fly** *Microdon major* p. 105
17b Scutellum usually paler than scutum, reddish → **Bog Ant Fly** *Microdon myrmicae* / **Limestone Ant Fly** *Microdon mutabilis* p. 107

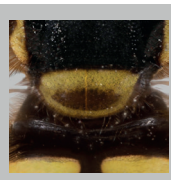
18a Third antennal segment twice as long as first and second antennal segments combined; first antennal segment twice as long as second antennal segment → 19
18b Third antennal segment as long as first and second antennal segments combined; first antennal segment as long as second antennal segment → 21

19a Underside of hind femur with a kink and excavation in which the hind tibia fits; tarsomere 3 of all legs dark; scutum dull → 20
19b Hind femur of normal shape, cylindrical; tarsomere 3 of all legs pale; scutum shiny → **Pine Longhorn** *Callicera rufa* p. 219

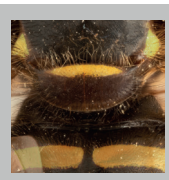
20a Hairs on scutellum longer than first antennal segment; hairs on scutum and abdomen yellow–brown to grey–brown; in male, usually at least tergite 4 largely covered with black hairs; hind tibia often darkened at tip
→ **Dark Longhorn** *Callicera fagesii* p. 219
20b Most hairs on scutellum not longer than first antennal segment; hairs on scutum and abdomen orange to white–yellow; tergites usually without black hairs; all tibiae orange → **Macquart's Longhorn** *Callicera macquarti* p. 219

10a

10b



11a



11b

12a

12b

13a

13b

14a

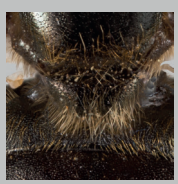
14b

15a

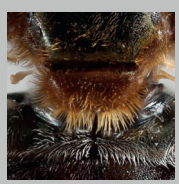
15b

16a

16b



17a



17b

18a

18b

19a

19b

♂

20a

♂

20b

21a Male: frons above antennae bare. Female: base of femora black → 22
21b Male: frons above antennae with hairs along eyes, hairs as long as hairs on eyes. Female: femora yellow or only very base black → **Ivy Longhorn** *Callicera spinolae* p. 221

22a Scutum thinly but distinctly dusted greyish over almost entire surface, with or without two or four distinct longitudinal dust-stripes; scutellum with long hairs, hairs on posterior margin longer than scutellum, hairs on upper surface of scutellum two-thirds or more length of scutellum → **Yellow Longhorn** *Callicera aenea* p. 221
22b Scutum shiny, except anterior which has two longitudinal grey stripes of dusting that stop abruptly between wing bases; scutellum with short hairs, hairs on posterior margin shorter than scutellum, hairs on upper surface of scutellum not longer than half length of scutellum → **Golden Longhorn** *Callicera aurata* p. 221

Key 1A Ant flies *Microdon* puparia

1a Reticular pattern of ridges confined to sides, upper surface of puparium smooth, bald → 2
1b All surfaces of puparium covered in reticular pattern of ridges → 3

2a Reticulations at sides poorly defined; anterior spiracle longer than width at base; found in ant nests of the genus *Myrmica* → **Bog Ant Fly** *Microdon myrmicae* p. 107
2b Reticulations at the sides well defined; anterior spiracle shorter than width at base; found in ant nests of the genus *Formica* → **Limestone Ant Fly** *Microdon mutabilis* p. 107

3a Reticular pattern fine, space between ridges not bigger than diameter of posterior spiracle; anterior spiracle not curved → 4
3b Reticular pattern coarse, space between ridges larger than diameter of posterior spiracle; anterior spiracle curved → **Chalk Ant Fly** *Microdon devius* p. 105

4a Anterior spiracle red–brown and almost three times longer than its maximum basal width; posterior spiracle high and narrow, almost conical; found in ant nests of the genus *Lasius* → **Heath Ant fly** *Microdon analis* p. 105
4b Anterior spiracle black–brown and at most twice as long as its maximum basal width; posterior spiracle short and wide; found in ant nests of the genus *Formica* → **Large Ant Fly** *Microdon major* p. 105

Key 2 Plumehorns *Volucella*

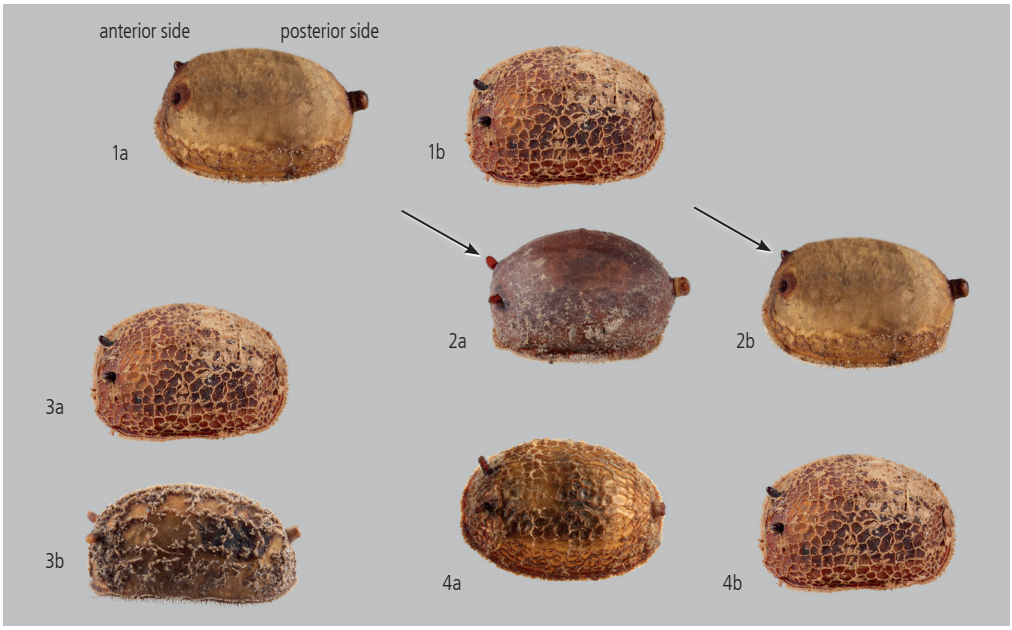
1a Fly does not look like a bumblebee → 2
1b Thorax and abdomen covered in long dense hairs; fly looks like a bumblebee (different colour morphs) → **Bumblebee Plumehorn** *Volucella bombylans* p. 371

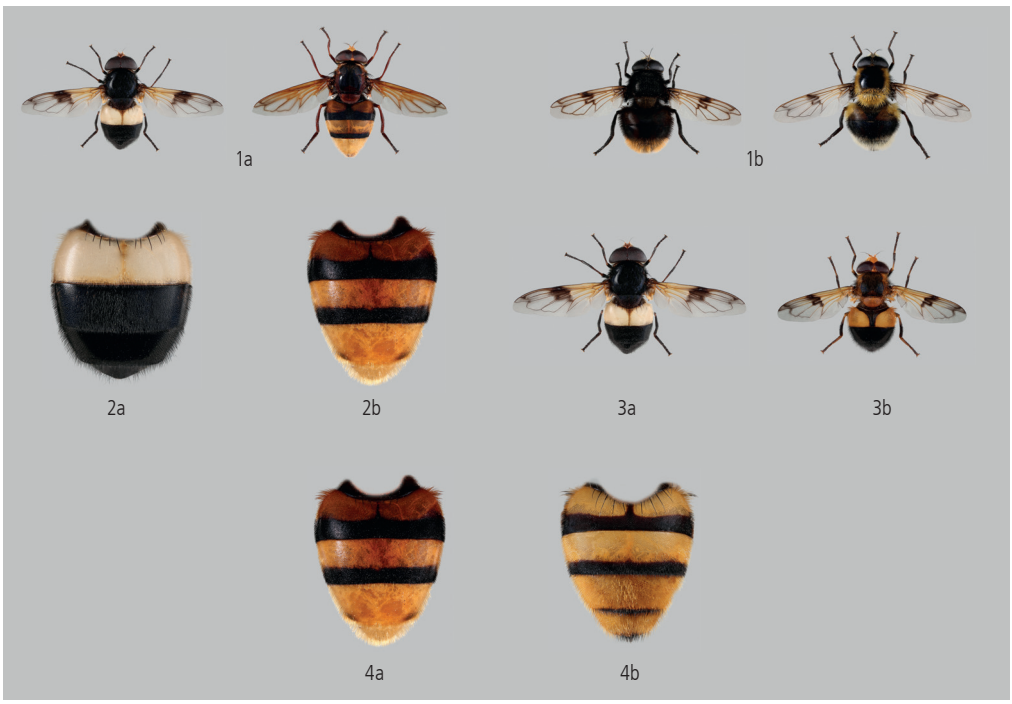
2a Tergites 3 and 4 black → 3
2b Tergites 3 and 4 with orange or yellow bands → 4

3a Tergite 2 ivory-white; side margins of scutum dark brown, sometimes dull orange; scutellum and at least posterior part of scutum with black hairs → **Pied Plumehorn** *Volucella pellucens* p. 373
3b Tergite 2 orange–yellow; side margins of scutum orange; scutum and scutellum with golden-yellow hairs → **Orange-belted Plumehorn** *Volucella inflata* p. 373

4a Tergite 2 with reddish pair of markings, tergite 3 with orange band, tergite 4 with yellow band; tip of abdomen yellow; sternite 2 black; scutum shiny → **Hornet Plumehorn** *Volucella zonaria* p. 371
4b Markings on abdomen uniformly yellow; tip of abdomen black; sternite 2 yellow; scutum dull → **Wasp Plumehorn** *Volucella inanis* p. 373


21a
21b


anterior side
posterior side
1a
1b
2a
2b
3a
3b
4a
4b


1a
1b
2a
2b
3a
3b
4a
4b

Key 3

1a Face yellow, often with a black central stripe → 2
1b Face black → 11

2a Very small flies, 4–7 mm; tergite 2 without large pair of whitish markings → 3
2b Large flies, 9–11 mm; tergite 2 with large pair of whitish markings
→ **Dark-saddled Hoary** *Leucozona laternaria* p. 147
(see also Key 4, 32b)

3a Eyes with uniformly distributed hairs, not with vertical stripes of hairs; scutellum black → 4
3b Eyes with vertical stripes of hairs; scutellum often with yellow posterior margin → 6

4a Male genitalia: larger, causing sternite 4 to be constricted and shorter than sternite 3. Female: tergite 4 hairs uniformly white → 5
4b Male genitalia: smaller; sternite 4 with straight hind margin and about as long as sternite 3. Female: posterior part of tergites 2–4 with black hairs → **Common Grass Skimmer** *Paragus haemorrhous* p. 213

5a Male genitalia: paramere boomerang shaped, inner side concave, without keel. Female: hairs on posterior part of tergites 2–4 erect, with a few semi-erect on hind margin → **Heathland Grass Skimmer** *Paragus tibialis* p. 213
5b Male genitalia: paramere triangular, inner side straight, with keel. Female: hairs on posterior part of tergites 2–4 adpressed → **Constricted Grass Skimmer** *Paragus constrictus* p. 213

6a Tergites 2 and 3 uniformly black or black with red markings → 7
6b Tergites 2 and 3 with yellow markings → **Yellow-banded Grass Skimmer** *Paragus quadrifasciatus* p. 217

7a Abdomen black or with only few red markings on middle of tergites and that do not reach tergite side margin → 8
7b Abdomen with extensive red markings, reaching side margin of at least tergite 3 → 9

8a Tergites 2–4 with silver dust bars underneath pale hairs (best seen by viewing from the side); abdomen usually black. Male genitalia: basolateral lobe with two teeth of equal size; paramere wide
→ **Grey-banded Grass Skimmer** *Paragus pecchiolii* p. 215
8b Tergites 2–4 without or with only faint dust bars; tergites 2 and 3 often with red spot. Male genitalia: lower tooth on basolateral lobe twice as big as upper one; paramere narrow → **Red-dotted Grass Skimmer** *Paragus albifrons* p. 215

9a Male genitalia: basolateral lobe with two teeth; outer upper border of hypandrium not extended into large triangular tooth. Female: tergite 7 usually with a shallow depression in middle; if absent, then central dark band on face occupies about one-quarter face width → 10
9b Male genitalia: basolateral lobe with one tooth; outer upper border of hypandrium extended into large triangular tooth. Female: tergite 7 without a shallow depression in middle; central dark band on face occupies about one-quarter face width → **Red Grass Skimmer** *Paragus testaceus* p. 215

10a Male genitalia: with a long, pointed protrusion between hypandrium and lingula. Female: tergite 7 with a shallow depression in middle → **Spiny Grass Skimmer** *Paragus finitimus* p. 217
10b Male genitalia: without a long, pointed protrusion between hypandrium and lingula. Female: tergite 7 without a shallow depression in middle → **Flaming Grass Skimmer** *Paragus flammeus* p. 217

11a Abdomen not with combination of being very slender and constricted, anterior half not slenderer than scutellum → 12
11b Abdomen very long and slender, anterior half even narrower than scutellum → **Common Dainty** *Baccha elongata* p. 211

12a Abdomen constricted at tergite 2; hind femur strongly swollen → 13
12b Abdomen not constricted; hind femur usually not strongly swollen → 27


1a
1b
2b
3a
3b
4a
4b
5a
5b
6a
6b
7a
7b
8a
8b
9a
9b
10a
10b
11a
11b
12a
12b

13a Wing: vein M1 and crossvein dm-m with rounded angle → 14
13b Wing: vein M1 and crossvein dm-m are bent at a sharp angle → 20

14a Sternite 1 present, wider than long; usually fore and mid tarsi more or less uniformly brown or yellow → 15
14b Sternite 1 mostly reduced or absent, if present longer than wide; fore and mid tarsi with basal three tarsomeres yellow, strongly contrasting with outer two black tarsomeres → **Variable Pufftail** *Sphegina sibirica* p. 319

15a Lower half of face white–yellow, although mouth edge may be black → 16
15b Face entirely black → 17

16a Humerus black, not contrasting with scutum; third antennal segment not very large; mouth edge usually black → **Common Pufftail** *Sphegina clunipes* p. 319
16b Humerus yellow, contrasting with black scutum; third antennal segment remarkably large; mouth edge usually yellow → **Elegant Pufftail** *Sphegina elegans* p. 319

17a Hind trochanter partly or entirely yellow → 18
17b Hind trochanter black → 19

18a Wing: costa and subcosta join at level of crossvein r-m. Male genitalia: surstylus fairly short with blunt tip → **Lesser Pufftail** *Sphegina verecunda* p. 321
18b Wing: costa and subcosta join before crossvein r-m. Male genitalia: surstylus elongated with pointed tip → **Red-horned Pufftail** *Sphegina nigra* p. 321

19a Katepisternum shiny; fore and mid legs largely yellow or yellow–brown; face below antennae hollowed out. Male genitalia: surstylus with very wide base abruptly narrowing to tip → **Shiny Pufftail** *Sphegina montana* p. 323
19b Katepisternum dusted; fore and mid legs black, except knees and sometimes basal third of tibiae yellow; abdomen relatively short and broad; face below antennae not hollowed out. Male genitalia: surstylus elongated, curved, with slightly wider base → **Stocky Pufftail** *Sphegina spheginea* p. 321

20a Wing: crossvein dm-m and apical part of vein M1 darkened → 21
20b Wing: crossvein dm-m and apical part of vein M1 not darkened → 24

21a Tergite 4 black; third antennal segment at least twice longer than wide → 22
21b Sides of tergite 4 with pair of small yellow markings; third antennal segment at most 1.5 times longer than wide → **Many-spotted Fen Fly** *Neoascia interrupta* p. 323

22a Tergite 2 usually with yellow markings; fore femur yellow or with faint brown ring → 23
22b Tergite 2 black; fore femur with a clear wide black ring → **Single-banded Fen Fly** *Neoascia unifasciata* p. 327

23a Yellow pair of markings on tergite 2 with more or less straight posterior margin; plates behind hind coxae just touch → **Smudge-veined Fen Fly** *Neoascia podagrica* p. 327
23b Yellow pair of markings on tergite 2 with oblique anterior and posterior margins, resembling an inverted V; plates behind hind coxae widely separated → **Butterbur Fen Fly** *Neoascia obliqua* p. 327

24a Tarsomeres 4 and 5 of fore tarsus yellow; third antennal segment at least 1.5 times longer than wide → 25
24b Tarsomeres 4 and 5 of fore tarsus black; third antennal segment barely longer than wide → **Short-horned Fen Fly** *Neoascia geniculata* p. 323

25a Tip of hind femur black or slightly dark orange; plates behind hind coxae connected; tergite 2 with yellow markings, sometimes missing in female. Female: abdomen constricted between tergite 1 and 2 → 26
25b Tip of hind femur bright yellow; plates behind hind coxae widely separated; tergite 2 black. Female: abdomen constricted halfway along tergite 2 → **Yellow-kneed Fen Fly** *Neoascia meticulosa* p. 325

13a

13b

14a

14b

15a

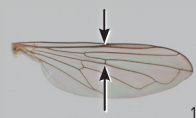
15b

16a

16b

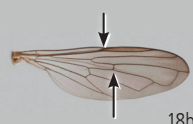
17a

17b



18a

19a



18b

19b

20a

20b

21a

21b

22a

22b

23a

23b

24a

24b

♀

25a

♀

25b

26a Tergites 2 and 3 uniformly black or black with yellow markings that do not reach side margins; mouth edge distinctly protruding. Male: tergite 8 with pale hairs. Female: tergite 2 gradually widening → **Black-kneed Fen Fly** *Neoascia tenur* p. 325
26b Tergites 2 and 3 with yellow bands that reach side margins over its full width; mouth edge slightly protruding. Male: tergite 8 with black hairs. Female: tergite 2 constricted halfway → **Broad-banded Fen Fly** *Neoascia annexa* p. 325

27a Face with facial tubercle and/or mouth edge protruding; face not entirely covered in long drooping shaggy hairs → 28
27b Face flat, sloping backwards, without facial tubercle and without protruding mouth edge; face entirely covered in long drooping shaggy hairs → **Key 3A**

28a Wing: vein M1 meets vein R_{4+5} only just before wing-tip → 29
28b Wing: vein M1 meets vein R_{4+5} well before wing-tip → 32

29a Abdomen partly orange → 30
29b Abdomen black → 31

30a Male: face with narrow black median stripe from mouth edge up to the facial tubercle; orange markings on tergite 3 usually reaching posterior margin. Female: black median stripe on tergite 2 widest near tergite posterior margin, but narrows abruptly just before posterior margin itself; groove on frons wider, reaching anterior ocellus → **Orange-belted Spineleg** *Myolepta dubia* p. 301
30b Male: face with strongly widening black shining median stripe from mouth edge to well over facial tubercle; if orange markings on tergite 3 present, not reaching posterior margin. Female: black median stripe on tergite 2 blends into a black transverse line along tergite posterior margin; groove on frons narrow, ending just before anterior ocellus → **Small-spotted Spineleg** *Myolepta potens* p. 301

31a Sides of thorax shiny; metasternum with hairs. Male: face with strongly widening black shining median stripe from mouth edge to well over facial tubercle → **Dark Spineleg** *Myolepta vara* p. 303
31b Sides of thorax dusted; metasternum bare. Male: face with short narrow black median stripe → **Obscure Spineleg** *Myolepta obscura* p. 303

32a Eyes with long hairs; face without facial tubercle → 33
32b Eyes with hairs or bare; if eyes hairs present, then facial tubercle also present → 35

33a Hind femur in middle at most 1.5 times wider than mid femur. Male genitalia: epandrium not elongated, about as long as wide. Female: posterior anepisternum with black hairs → 34
33b Hind femur distinctly thickened, in middle 2–3 times wider than mid femur. Male genitalia: epandrium elongated, almost 1.5 times longer than wide. Female: posterior anepisternum with white hairs → **Pine Haireye** *Psilota atra* p. 357

34a Male: hind coxa and lower part of katepisternum with white hairs. Genitalia: surstylus not remarkably long and slender. Female: hair fringe under scutellum black, with hairs shorter than hairs on tergite 3; hairs on posterior margin of tergite 4 two times longer than hairs on anterior part of tergite → **Anthracine Haireye** *Psilota anthracina* p. 357
34b Male: hind coxa and lower part of katepisternum with black hairs. Genitalia: surstylus remarkably long and slender. Female: hair fringe under scutellum white or mixed black and white, with hairs approximately as long as hairs on tergite 3; tergite 4 without long hairs on posterior margin → **Spindly Haireye** *Psilota exilistyla* p. 357

35a Face without orbital strip; if abdomen entirely black, often centre dull → 36
35b Face with orbital strip, a hairy strip on face along eye, from height of antenna to mouth edge; abdomen usually uniformly black → **Key 3B**


26a
26b
27a
27b
28a
28b
29a
29b
♀
30a
♀
30b
31a
32a
32b
33a
33b
34a
34b
35a
35b

36a Small stocky species; abdomen uniformly black, sometimes with blue or green sheen; abdomen usually dull in middle with shiny side margins; face with or without facial tubercle. Female: frons with transverse grooves → 37
36b Abdomen usually narrow; abdomen with red, yellow or blue–grey markings; if abdomen uniformly black, abdomen elongated and shiny; face with facial tubercle. Female: frons without transverse grooves → **Key 3C**

37a Tergites entirely metallic, with greenish sheen, centre not dull. Male: eyes separated → 38
37b Tergites metallic with black, blue or greenish sheen; if with greenish sheen, centre dull. Male: eyes meet on frons → 39

38a Fore tarsus and third antennal segment black; anterior anepisternum bare → **Green Shimmer** *Lejogaster metallina* p. 313
38b Fore tarsus and usually also mid tarsus with central tarsomeres orange; base of third antennal segment orange; anterior anepisternum with hairs in upper posterior corner → **Splendid Shimmer** *Lejogaster tarsata* p. 313

39a Wing: apical part of vein M1 perpendicular or slightly recurrent to vein R_{4+5}, giving cell r_{4+5} a rectangular appearance → 40
39b Wing: apical part of vein M1 not recurrent and ends at vein R_{4+5} in an acute or right angle, so cell r_{4+5} not rectangular → 46

40a Legs entirely black → 41
40b At least knees of legs orange → 42

41a Third antennal segment rounded. Male: eyes meet, with length of contact similar to ocellar triangle length. Female: posterior margin of tergites 4 and 5 without tubercle or notch → **Atlantic Mucksucker** *Orthonevra brevicornis* p. 315
41b Third antennal segment about twice as long as wide. Male: eyes barely meet. Female: middle of posterior margin of tergite 4 with a small tubercle; middle of posterior margin of tergite 5 with a small notch → **Slim-horned Mucksucker** *Orthonevra nobilis* p. 315

42a Eyes without dark horizontal band; third antennal segment at most three times as long as wide → 43
42b Eyes with a dark horizontal band; third antennal segment four times as long as wide → **Stripe-eyed Mucksucker** *Orthonevra elegans* p. 313

43a At least basal third of tibiae orange → 44
43b Only very base of tibiae orange → **Nordic Mucksucker** *Orthonevra erythrogona* p. 315

44a Wing: pterostigma uniformly yellowish; crossvein r-m not darkened; face narrow → 45
44b Wing: pterostigma bicoloured, yellow with dark-brown spot at base; crossvein r-m darkened; face wide → **Willow Mucksucker** *Orthonevra geniculata* p. 317

45a Male genitalia: surstylus wide and with a notch halfway, aedeagus with short projection. Female: side margins of sternite 8 convex; sternite 8 bare in middle → **Mire Mucksucker** *Orthonevra intermedia* p. 317
45b Male genitalia: surstylus narrow and without notch, aedeagus with long projection. Female: side margins of sternite 8 angled; middle of sternite 8 with hairs → **Stackelberg's Mucksucker** *Orthonevra stackelbergi* p. 317

46a Anterior part of scutum without two grey–white dusted longitudinal stripes; sternite 1 dull; body with green or black sheen → 47
46b Anterior part of scutum with two grey–white dusted longitudinal stripes; sternite 1 shiny; body with green sheen → **Stripe-backed Glimmer** *Riponnensia splendens* p. 311

47a Third antennal segment largely orange → 48
47b Third antennal segment entirely dark → 52


36a
36b
37a
37b
38a
38b
cell r4+5
39a
cell r4+5
39b
40a
40b
41a
41b
42a
42b
43a
43b
44a
44b
45a
45b
46b

48a Wing: basal veins yellow, contrasting with grey veins in apical half → 49
48b Wing: basal veins dark, not contrasting with rest of wing → 50

49a Proepimeron dusted; face at level of antennae distinctly wider than an eye. Female: body shiny black
→ **Yellow-winged Wrinklehead** *Chrysogaster cemiteriorum* p. 311
49b Proepimeron shiny; face at level of antennae about as wide as an eye. Female: body shiny blue
→ **Blue Wrinklehead** *Chrysogaster basalis* p. 311

50a Male: face at level of antennae about twice as wide as an eye; upper side scutellum with long hairs, hairs about as long as on scutum. Female: body shiny green; face at level of antennae wider than an eye → 51
50b Male: face at level of antennae about as wide as an eye; upper side of scutellum almost bare. Female: body shiny, dark purple; face at level of antennae narrower than an eye → **Dark-winged Wrinklehead** *Chrysogaster solstitialis* p. 309

51a Face wide. Male: facial tubercle distinct. Female: scutum bare → **Green Wrinklehead** *Chrysogaster virescens* p. 309
51b Face very wide. Male: facial tubercle indistinct. Female: scutum with short white hairs
→ **Broad-faced Wrinklehead** *Chrysogaster rondanii* p. 309

52a Eyes meet; fly is a male → 53
52b Eyes widely separated; fly is a female → 57

53a Male: tergite 8 with long hairs → 54
53b Male: tergite 8 with short hairs → **Naked Wrinklehead** *Melanogaster nuda* p. 303

54a Male: hairs on scutum black; hairs on hind tibia mostly black → 55
54b Male: hairs on scutum usually, at least in anterior part, partly pale; hairs on hind tibia mostly yellow → 56

55a Male: facial tubercle distinct; tip of surstylus bent inwards, pointing sideways → **Dark Wrinklehead** *Melanogaster aerosa* p. 305
55b Male: facial tubercle indistinct; tip of surstylus much less bent, more or less pointing forward
→ **Chalk Wrinklehead** *Melanogaster parumplicata* p. 307

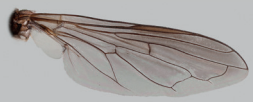
56a Male: hairs on scutum long; scutum dusted. Male genitalia: surstylus abruptly narrowed near tip
→ **Common Wrinklehead** *Melanogaster hirtella* p. 305
56b Male: hairs on scutum short; scutum centrally shiny. Male genitalia: surstylus parallel-sided, curved only at very tip
→ **Hidden Wrinklehead** *Melanogaster curvistylus* p. 307

57a Female: scutum with distinct hairs; abdomen upper surface with dull area, entire margins shiny → 58
57b Female: scutum almost bare; entire abdomen shiny black → **Naked Wrinklehead** *Melanogaster nuda* p. 303

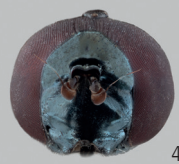
58a Female: scutum with short hairs; hairs on hind tibia mostly black → 59
58b Female: scutum with fairly long erect hairs; hairs on hind tibia mostly yellow
→ **Common Wrinklehead** *Melanogaster hirtella* p. 305
(the female of **Hidden Wrinklehead** *Melanogaster curvistylus* remains undescribed but probably ends up here as well)

59a Female: angle between face and mouth edge sharply defined → **Dark Wrinklehead** *Melanogaster aerosa* p. 305
59b Female: angle between face and mouth edge rounded → **Chalk Wrinklehead** *Melanogaster parumplicata* p. 307

48a



48b



49a

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50a

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51a

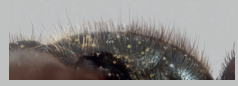
51b

53a

53b

55a

55b



56a

56b

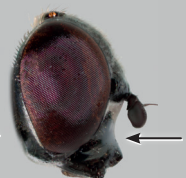
57a

57b

58a

58b

59a



59b

Key 3A

1a Tergites 2–4 about equal in size → 2
1b Tergites 2 and 3 large; tergite 4 minute, hardly visible from above, although very rarely in some females half tergite 3 length → **Mugwort Galleater** *Triglyphus primus* p. 273

2a Anterior anepisternum with hairs; hard to see without a microscope → 3
2b Anterior anepisternum bare; note, this is hard to see without a microscope → 5

3a Third antennal segment long, at least twice as long as wide. Female: tergite 2 with a pair of spots → 4
3b Third antennal segment short, about 1.5 times as long as wide. Female: abdomen uniformly black → **Obscure Psyllid-killer** *Trichopsomyia joratensis* p. 285

4a Wing: vein M1 perpendicular to R_{4+5}. Female: diameter of the markings on tergite 2 smaller than distance between inner extremities of markings → **Pale-footed Psyllid-killer** *Trichopsomyia flavitarsis* p. 285
4b Wing: vein M1 ends in an acute angle on R_{4+5}. Female: diameter of the markings on tergite 2 larger than distance between inner extremities of markings → **Large-spotted Psyllid-killer** *Trichopsomyia lucida* p. 285

5a Third antennal segment short, at most 1.5 times as long as wide → 6
5b Third antennal segment long, at least twice as long as wide → 19

6a Frons protrudes conically forward at level of antenna attachment. Male: mid coxa and hind trochanter without long spine. Female: frons with distinct dust markings → 7
6b Frons of normal shape, not protruding conically forward at level of antenna attachment. Male: mid coxa and hind trochanter with long spine. Female: frons bare or with very small dust markings → 15

7a Antennae attach above middle of head; abdomen not distinctly wide; abdomen usually either entirely black or with only one pair of pale markings → 8
7b Antennae attach at middle of head; abdomen wide; abdomen usually with two pairs of pale markings → **Four-spotted Pithead** *Pipiza quadrimaculata* p. 279

8a Hind femur not swollen; underside of hind femur without groove or ridges on tip → 9
8b Hind femur swollen; underside of hind femur with groove and ridges on tip → 10

9a Male: fore tarsus usually yellow; sides of face below antenna attachment point divergent. Female: tergite 5 about as long as wide → **Pale-footed Pithead** *Pipiza luteitarsis* p. 279
9b Male: fore tarsus usually dark; sides of face below antenna attachment point parallel. Female: tergite 5 approximately twice as wide as long → **Broad-faced Pithead** *Pipiza accola* p. 279

10a Fore tarsus dark, or with basal tarsomeres yellow, but outer tarsomere always dark → 11
10b Fore tarsus entirely yellow → **Poplar Pithead** *Pipiza festiva* p. 281

11a Hind femur swollen, but underside near tip without extra bulge; third antennal segment usually longer than wide → 12
11b Hind femur swollen, underside near tip with extra bulge; third antennal segment about as long as wide; thorax and abdomen with pale hairs → **Big-thighed Pithead** *Pipiza austriaca* p. 281

12a Wing without dark patch, or with dark patch with diffuse edges; third antennal segment 1–1.3 times longer than wide; hairs on face dark or pale → 13
12b Wing with distinct and sharply defined dark patch; third antennal segment 1.3–1.5 times longer than wide; abdomen usually black, sometimes with a pair of indistinct yellow markings on tergite 2; hairs on face pale → **Smudge-winged Pithead** *Pipiza lugubris* p. 283

13a Abdomen black or with a pair of markings on tergite 2 only. Male: face with black hairs. Female: tergite 4 anteriorly with band of black hairs; sternite 3 black (best assessed in living specimens) → 14
13b Abdomen with a pair of markings on tergite 2, almost always on tergite 3 as well. Male: face with pale hairs. Female: tergite 4 mostly with pale hairs; sternite 3 yellow (best assessed in living specimens) → **Square-spot Pithead** *Pipiza fasciata* p. 281