Collecting and preparing Sarcophagidae

Time of year and time of day. Sarcophagidae, particularly males, are most active during the warmest months of the year (from April to September) and during full on sunny weather. Productive collecting can be quickly halted by a passing cloud. Some studies and personal experience suggest that morning hours might be more productive for flesh flies, particularly in very hot weather.

Habitats. Flesh flies can be abundant in most habitats, including urban ones, but their diversity is greater in well-preserved woodland, grassland and coastal habitats. Species with a more specialised biology (e.g., as snail predators or scavengers) may be restricted to calcareous or chalk grasslands. Several Miltogramminae species are most easily collected in the sandy habitats used by their hosts.

Good collecting spots. Within the right habitat, good collecting spots for flesh flies include paths, woodland edges, small sunlit patches of forest floor, flowers (particularly umbellifers and composites), sunlit leaves of trees and shrubs, stones, small boulders, pebbly and sandy riverbeds and banks, dunes, and hilltops—provided these are surrounded by good natural habitat. Females will usually be much less abundant than males in most spots, except maybe on flowers.

Bait. Both males and females of several species, particularly of the subfamily Sarcophaginae, are attracted by rotting meat or faeces. Vertebrate carcasses (both small and large) and dead invertebrates are worth checking for flesh flies, but you may also bring your own bait in the field. Chicken liver and fish, for example, are known to work well—just make sure they are nice and ripe and carry them around in some good Tupperware to avoid annoying fellow collectors!

Collecting methods. Sarcophagidae can be collected with hand nets, sweep nets, Malaise traps, yellow pan traps and baited traps. With the right conditions and in the right habitats, individual collecting with a hand net can be very productive but will yield a majority of males. General sweeping of vegetation can yield additional species (particularly smaller ones) and a greater proportion of females; this method can also be resorted to when weather conditions (e.g., clouds or cool temperatures) reduce evident flesh fly activity. When a thorough inventory of an area is required, it is recommended to supplement direct collecting methods with indirect ones such as Malaise traps (with ethanol) and yellow pan traps (with a soap and water solution). These will almost certainly yield additional species and will provide a much more balanced sample in terms of sex ratio.

Rearing. Any rearing records of British Sarcophagidae would be of great interest to the Recording Scheme. Flesh flies can be reared from their larval feeding substrates or on artificial feeding substrates, such as chicken liver or minced meat, in the lab. Various species have been successfully reared from live earthworms, dead and live snails, other dead invertebrates or vertebrates, live acridid grasshoppers (genus *Blaesoxipha*), moth pupae, etc. Adults of several species of Sarcophagidae from across the three subfamilies have been successfully reared in the lab from first instar larvae obtained from wild-collected gravid females (including of some Miltogramminae), which will readily feed on minced meat or chopped up insects (see Richet 1988; *Entomologiste 44: 347–348*). Rearing of larvae to adult stage, whether on their natural substrate or on an artificial food source, is therefore relatively straightforward for several

species. Mature third instar larvae will pupariate within the feeding substrate or within the rearing container, where some slightly wet blotting paper should be placed for moisture.

Killing methods. Because many flesh flies are relatively large, most can be individually potted without using a pooter or aspirator. If you plan to pin them later, your flesh flies should be killed in ethyl acetate fumes. If no killing agent is available, flies can be placed individually in vials and placed in a freezer at home or in the lab. 50 mL self-standing plastic tubes are the ideal size for easy potting of individual flies from within the net. These can be modified as with a piece of sponge and abundant kitchen or toilet roll to avoid direct contact of the specimens with the small ethyl-soaked tissue at the bottom of the tube and ensure gradual release of the fumes. A well-prepared tube can last up to a whole day, but you should keep several charged tubes at a time for maximum productivity. NB: excessive amounts of ethyl acetate can lead to brittleness and damage to diagnostic features such as bristles and dusting. Specimens should not be left in the fumes for longer than 15–20 minutes before being transferred to a fumeless container, such as a tissue-lined tube or tupperware. Do not store too many dead flies per container, to avoid humidity and consequent damage to diagnostic features. In very dry weather conditions (uncommon in Britain), a soft leaf should be placed in each container to ensure that specimens do not dry out before pinning.

Preservation. As a general rule, you should aim to preserve your specimens in the same medium in which they were collected. If the flies were collected dry, they should be pinned shortly afterwards; if they were trapped in a liquid (ethanol or water and soap mixture), they should be preserved in ethanol. Air-drying of specimens from ethanol without use of a specialised laboratory method should be avoided, as it can lead to severe shrivelling and deformation of structures.

Pinning. Specimens should be pinned for long-term dry preservation, and pinning should take place a few hours after collection to avoid drying and consequent damage to specimens. When specimens are pinned too soon after death, *rigor mortis* will not allow the specimen to be prepared for an optimal visibility of diagnostic characters. Flies can be pinned at latest the following morning, but they should be preserved in a refrigerator overnight to avoid drying. There are two main options for pinning: direct pinning and double-mounting on a stage with a micropin. Direct pinning should be used for specimens over 7–8 mm long (pin sizes 0, 1 and 2) or over 1 cm (pin sizes 1 and 2); smaller specimens should be double-mounted on plastazote or poly strips using large micropins (sizes B to E). Double-mounting should be avoided for larger (>1 cm) specimens. Direct pinning should take place behind the thoracic suture and slightly to the right of the midline of the thorax. Micropinning can be done obliquely from an entry point just in front of the wing base (left side of thorax) or vertically, similar to direct pinning (with entry from the ventral or dorsal side according to personal preferences).

Setting fresh specimens. Once pinned, relatively little manipulation of specimens is required to enable a proper visibility of diagnostic characters, the most important being preparation of **male genitalia.** These can be prepared relatively quickly in fresh specimens, provided they are past the *rigor mortis* stage. The first step before setting the genitalia is to roughly unhinge them from their natural retracted position within the abdomen. With direct-pinned specimens, the genitalia can be unhinged with a fine pin or micropin (held with forceps or pushed into the end of a long matchstick or similar) while resting the pin across two fingers and securing its lower end with the thumb; the tip of the pin (or micropin) used to unhinge the genitalia should be inserted between the sides of tergite 5 (the last visible unmodified tergite) and the genital

capsule (syntergosternite 7+8 and epandrium—also known as the 1st and 2nd genital segments). This can be a slightly frustrating operation, and you may find that your specimen has slid up or down the pin in the process; it should not be an issue with fresh specimens, as you can push the specimen back to its original position on the pin before it dries. For double-mounted specimens, unhinging of the genital capsule should take place before staging and the much shorter (micro)pin will be more difficult to hold, so you may find it easier to pin it to a plastazote slab. Once the genitalia have been unhinged, the cerci (claspers) should be easy to hook out for final setting of the genitalia, which should be done with the help of fine micropins against a slab of plastazote. For this, direct-pinned specimens will have to be pinned into the plastazote at a very oblique angle almost parallel to its surface. A minimum of 2 micropins should be used to set the genitalia: one placed above the genital capsule to secure the latter and avoid pushing the abdomen too far up against the scutellum and wings, the other placed between the cerci (claspers) and the phallus, to ensure proper visibility of both structures. Set specimens should be left to dry for at least a week before the micropins are removed, as the genitalia can slowly return to their original position in incompletely dried specimens. The complexity of the setting will vary according to species, and species with smaller genitalia will be slightly more difficult to set. **Other characters.** Legs should be pulled away from the thorax, and tibiae and femora should be separated from each other to ensure proper visibility of leg and thoracic bristles. In females, mid legs should be pulled away from the thorax in a position perpendicular to the fly's main axis, to ensure visibility of the mid femoral organ, located on the posterior surface of the mid femur.

Relaxing dry specimens. Specimens that have been dry for some time (even up to several years) can be relaxed in a humidifying chamber for a few hours (but no longer than a day!) before attempting to perform the male genitalia setting techniques described above. This will be easier in some specimens compared to others, depending on age and other factors such as morphology of the genital capsule. Laurel leaves or menthol should be placed in the humidifying chamber to avoid the formation of mould. If a specimen cannot be relaxed, it is possible to carefully break off the genital capsule with very fine-tipped, hard forceps at the junction between syntergosternite 7+8 and the epandrium; however, if this operation is not carried out very carefully, damage to the phallus or other structures may incur.

Study of specimens in ethanol. Specimens in 70 or 75% ethanol are usually supple enough for male genitalia to be unhinged relatively easily. However, specimens in 96 or higher percentage ethanol are usually much more brittle and can easily break in undesired places when attempting to unhinge the genitalia. This can be solved by carefully breaking the musculature within the genital capsule with the tip of a pin and gently ripping the epandrium and other genital structures away from syntergosternite 7+8, while trying not to break them off completely. Females are often easier to identify in ethanol, since key features like the mid femoral organ and last abdominal sclerites are often better visible.