DISSECTION METHODS

In order for genitalia and other features to be scrutinised, it is usually necessary for the moth to be killed (or at least anaesthetised) and for it to be retained as a pinned specimen. The methodologies involved are dealt with by Heath and Emmet (1976) and most recently May and White (2006).

The aim of this section is to enable readers to develop their skills, starting with the 'easy' species and working up to those with only slight and comparative differences. We recommend working on larger species first to gain experience. Don't be ambitious - dealing with small moths is an art, but things do become easier with practice. It should also be emphasised that technique is, to some extent, down to individual preference and there may be scope for creative thinking in some areas. Indeed, we are aware that other workers have developed alternative techniques. However, our purpose is to outline the important practices and principles rather than attempt a definitive guide or an exhaustive list of alternatives. Although the taxonomic section of this book covers only macro-moths, these techniques apply also to micros, although in some cases with modifications.

Techniques may vary slightly according to species. For example, it is not necessary in many cases to carry out the full dissection procedure to examine features on the valvae of the males, as these can be seen by simply brushing the scales off this structure, often *in situ*. At the other extreme, in some cases very careful manipulation is necessary in order to see the diagnostic features, or these can look different according to their orientation or depending on whether a cover slip has been applied. It is important, and may be crucial, that the specimen is presented in the correct way for successful identification. It may still be necessary to refer the moth and slide preparation for a second opinion, especially in the case of suspected rarities.

Dissection – making a slide preparation

Equipment:

Microscope. For dissection purposes this should be binocular, giving magnifications of either 15x and 30x, or 20x and 40x, or have zoom magnifications ranging from (typically) 7x to 45x. Eyepieces with magnification of 10x are usually supplied, but 20x can be obtained. Lighting systems supplied are usually adequate, but can be supplemented with a fibre optic box. Compound microscopes reveal much finer detail when examining slides, but as they see the image 'upside down' are not suitable for dissecting. Modern microscopes can be fitted with a camera, which is a highly specialised and technical area, not covered here. A few other basic tools are needed. Some can be obtained from entomological suppliers or other retailers. In some cases they can be home-made. Here are some suggestions.

Fine forceps. Available from entomological suppliers.

Needles, brushes and probes. Used for manipulation. These can be either purchased or made using matches or cocktail sticks to create handles. Carefully saw a groove into one end of the stick and insert a fine pin. Glue this in place and bind it with cotton. Once dry, apply a coat of varnish. Create a number of these with a variety of pins, some straight and others bent or hooked at the tip. Brushes can be made in the same way, traditionally from a pin-feather of a Snipe or Woodcock. The shape and stiffness of these feathers make them ideal for brushing off body scales, etc. and they can be obtained from game merchants or gamekeepers. There is only one suitable feather per wing, situated just beyond the carpal joint. Failing these, experiment with other materials. The handle should not be too long, as this would magnify movements under the microscope.

Excavated glass blocks. Used for working on the dissection at the microscope and/or as staining baths.

Pipettes, glass tubes, glass slides and cover slips, syringe. A variety of these will prove useful.

Chemicals required:

Potassium Hydroxide (KOH), supplied as 10% solution
Euparal
Euparal Essence
Iso-propyl alcohol
1% Chlorazol Black (staining agent)
Purified or distilled water

Technique:

Step 1. Removal of the abdomen
Most examinations will involve dried specimens, in which case the abdomen should be carefully lifted until it detaches from the thorax. In some cases the hindwings may show signs of moving. Stop and carefully place a small drop of wood glue onto the adjacent part of the thorax and let this dry before attempting to detach the abdomen. Alternatively, slight downward pressure may then release the abdomen without bringing the hindwings with it. With soft, freshly-killed specimens, and where the moth is not being retained, use fine forceps to detach the abdomen or simply examine the intact moth if a full preparation is not required. Always remove the whole abdomen, since the genitalia (especially in females) may extend well forward and could otherwise be damaged, and other useful features could be present.

Step 2. Dissolving the fat and soft tissue within the abdomen
This is achieved by heating the abdomen in 10% aqueous Potassium Hydroxide (KOH) solution. KOH will work at room temperature but it can take many hours, making it more difficult to regulate. The aim is to heat without boiling so that no liquid leaps from the tube. KOH is caustic and contact with skin and eyes should be avoided. The Safety Data for this chemical is available from suppliers and on various websites. The same solution can be used to treat a number of moths before it loses its strength. An open glass tube is used, preferably one with a flat bottom. Tubes roughly 5 x 2.5 cm in size, no more than half full with the KOH, are ideal. Top up with water every now and again as evaporation will occur, causing the solution to become too concentrated.

In one method, water is brought to the boil in a saucepan and removed from the heat, and the tube stood in the hot water, which should come no more than halfway up the tube. Another method uses a light bulb fixed horizontally on a stand with a metal plate bolted on above it, upon which the tube sits. Since light bulb design has recently changed with the introduction of low energy bulbs and the phasing out of bulbs utilising the traditional tungsten filament, some experimentation may be necessary to achieve the desired result without overheating or overly prolonging the dissolving time. Take extra care, if using any kind of electrical heat source, that the solution cannot come into contact with the electrics.

The time taken will vary according to the solution temperature, which itself will vary according to the method, and as in all cooking, with the equipment used. However, 10-40 minutes is usually sufficient, depending on the size of the moth. Again, experimentation will be necessary. When ready, the abdomen will attain a translucent appearance and may tend to sink unless air bubbles prevent it from doing so. If the abdomen is left in solution too long, the genitalia and other sclerotised parts can begin to soften and distort, which may confound the determination, so it is better to undercook than to overcook.

Step 3. Initial preparation
Remove the now soft abdomen from the KOH solution with a hooked pin or pipette, and transfer to a dish or glass block containing water. It is best to use purified or distilled water as tap water can react with the acids in the body cavity and make some structures fragile. This is most likely to happen in hard water areas. Under the microscope, use a combination of small angled pins and a fine brush or pin-feather to remove as many scales as possible.

Lay the abdomen ventral side up. Using pins and brushes, apply gentle pressure and stroking movements to the abdomen to push out the dissolved fat and soft tissue. If the water becomes excessively cloudy as a result (especially with larger specimens), transfer to a dish containing clean water. Males have a pair of flattened structures (valvae) which open out at an angle (see figure I). In females there is no such structure and the ovipositor is the most immediately noticeable feature. You should soon be able to see whether you have a female or male, and the procedures for dealing with them differ. It is sometimes possible to identify the species at this stage.

Step 4 (males). Separating the genitalia from the abdomen

The male organs, including the valvae and aedeagus, are clustered together at one end of the abdomen, and these should now be obvious, the aedeagus lying horizontally *in situ* in the middle of the genitalia, usually above the juxta. The next step is to detach them from the abdomen. Hold the other (anterior) end of the abdomen firmly with the heel of an angled pin or forceps and, with a second angled pin or forceps, gently stroke towards the valvae. With luck the genitalia will float free of the body, but you may need to carefully tear the cuticle until the genitalia have become detached. If enough scales have been removed, you should see enough detail to avoid damaging anything of value while doing this.

Step 5 (males). Preparing the genitalia

It is recommended that the cuticle should be retained, in case it has features that may be useful in identification. Clean away as much soft tissue as possible. If the aedeagus is to be removed, do it at this stage. This may cause damage, so proceed with caution. With the valvae closed, hold down firmly with the heel of an angled pin or fine forceps around the base of the juxta, and use a second pin or fine forceps to pull the aedeagus away posteriorly. The aedeagus should pull clear of the outer membrane. Once it is free, any remaining membrane can be removed by cutting it off near the top of the juxta. If the aedeagus fails to pull free, this membrane can be gently torn at the point of constriction, using two pairs of fine forceps, and the aedeagus removed as described above. If stain is to be used, it is recommended that it is applied at this stage. Staining with Chlorazol Black enables better discrimination between important structures and those that can be discarded, such as membranes. Use only a small amount as too much can mask diagnostic features.

Sometimes it is also desirable to evert the vesica, the soft tube inside the aedeagus, to see the thorn-like structures known as cornuti, and in some cases it has diverticula, which can also be distinguishing features (see everting the vesica, below). In some families it is not possible to detach the aedeagus, or to flatten the valvae, or the parts may need to be presented from a certain angle.

Photography: the aedeagus can be photographed at this stage, before using a cover-slip in the slide-making stage, which can make the three-dimensional shape difficult to discern. Alternatively, a ring slide or vinyl prop (available from suppliers) can be used for the final preparation. These allow important features to be arranged in three dimensions.

Step 6 (males). Making a slide preparation

The valvae should be cleaned of scales and hairs, but stout spines should be left in place. Carefully open the valvae, removing any tissue that prevents them from doing so. Immerse these (and the aedeagus) in a water/alcohol mixture in several stages, for example 30%, 60% and finally 100% alcohol, for about 30 seconds. This helps the cleaning process and prepares the structure for the slide stage. It is important to go through the increasing strengths of alcohol. If the genitalia are transferred directly from water to 100% alcohol, distortion can occur. At each stage open the valvae, if necessary turning them upside down and with the heel of a pin press down on the back, against the glass surface (but be careful as they can become brittle and break). The idea is to use the alcohol's stiffening actions to encourage the valvae to remain fully open of their own accord.

At this stage, the genitalia can be transferred into Euparal Essence for several seconds. This is the solvent for Euparal, and it allows the preparation to clear and helps to remove air bubbles. Transfer to Euparal on a glass slide and if needed re-open the valvae. Arrange the aedeagus close by. Alcohol and Euparal will make things very brittle, so care is needed whilst manipulating the structure. Use Euparal Essence if needed to keep the Euparal liquid and to finish off the edges once the cover slip has been placed. Apply the cover slip at an angle to help prevent air bubbles. These usually disappear in Euparal. Label the slide, including a reference number to link it to the specimen, and cross-reference the specimen. The slide preparation should be kept horizontal and allowed to dry on a level surface for several months, otherwise the cover slip will slowly drift across the microscope slide.

Photography: valvae are normally displayed flat with ventral side upwards, but in Euparal small specimens may be able to partially spring back, disguising their shape. Consider making a temporary slide for photographic purposes: arrange the valvae on a glass slide with alcohol as a mountant and apply a cover slip, but keep feeding in alcohol as it will evaporate quickly. Once the shot has been taken, carefully remove the slip and apply Euparal to the specimen, as above.

Step 4 (females). Separating the genitalia from the abdomen

Since the female genitalia are more delicate, the next stage will typically involve removing part of the cuticle, rather than applying pressure, which would damage them. First you should identify the end bearing the ovipositor, near to which will be found a ventral opening called the ostium. This part should remain intact, but the segments anterior to this can usually be removed. Inside will be a bag-like structure called the bursa copulatrix (or corpus bursae) and from this to the ostium is a tube called the ductus bursae. Try not to damage either of these. Make a cut with a pin or fine scalpel at a junction between the two segments anterior to the one bearing the ostium. Place two angled pins inside the end of the abdomen away from the tip of the ovipositor and slit the skin open until you reach the initial cuts. Remove the cuticle and hopefully you'll be left with the ovipositor, the last segment bearing the ostium, along with the ductus bursae and bursa copulatrix (and appendix bursae if one is present in that species). Note that spermatophores (packets of stored sperm) may be present in the bursa copulatrix. These may cause clouding later but can, if necessary, be removed by making a small slit in the end of the bursa copulatrix. If stain is to be used, it is recommended that it is applied at this stage (see also Stage 5, males).

Photography: this is a good time for a photograph as alcohol can distort some delicate structures. The genitalia can be turned in the water in order to display important details. This can provide a useful reference for arranging the various structures on the final slide mount, which would normally show the genitalia with the ventral side upwards.

Step 5 (females). Preparing the genitalia

The genitalia should now be immersed in a water/alcohol mixture in several stages, for example 30%, 60% (for approximately 30 seconds) and finally 100% alcohol. This helps the cleaning process and prepares the body for the slide stage. As for males, the gradual increase in alcohol strength is important. In particular, the bursa copulatrix may collapse if the genitalia are transferred directly from water to 100% alcohol.

Transfer to a glass slide and proceed as under the second paragraph of Step 6 (males), ensuring that the ductus bursae retains its natural shape and does not become twisted. It should be noted that if a female has not mated, the bursa copulatrix may not be inflated and diagnostic features may not be visible as a result. Therefore, if a bred female is to be dissected it is a good idea to allow her to mate, if possible.

Everting the vesica

Hardwick (1950) described a technique of eversion of the male vesica of noctuid moths, which led to discoveries of vesical characters in noctuids and other large Lepidoptera. The vesica structures, including shapes, number and location of diverticula and cornuti, have proved to be very important in resolving taxonomically-difficult groups (reviewed and summarised by Mikkola, 2007).

This procedure is analogous to turning a sock or glove inside out. It is the most difficult procedure that must be undertaken, so must be practised on expendable specimens before attempting it in order to make a determination. After separation of the aedeagus from the rest of the genitalia, as much of the ductus ejaculatorius (a narrow tube that enters the aedeagus through a split membranous tube near the base) should be removed as possible, taking care not to remove or damage any part of the vesica.

Now hold the aedeagus firmly with a pair of fine forceps, and gently push the remaining part of the ductus and the vesica up inside the aedeagus with a blunt mounted needle or, very carefully, with the tip of a syringe needle. The aedeagus or vesica must not be punctured during this process or it will not inflate.

The syringe, fitted with a 30 gauge needle and filled with distilled or purified water, is then carefully inserted into the aedeagus through what is left of the ductus ejaculatorius. The aedeagus is then firmly, but carefully, clamped on to the hypodermic using a pair of fine forceps just above the entry point of the ductus ejaculatorius in order to seal it, and gentle pressure applied to the syringe.

The vesica should start to evert. When this happens, remove the syringe and transfer to a Petri dish previously filled with iso-propyl alcohol and repeat the procedure with the syringe filled with iso-propyl alcohol, this time fully inflating the vesica, increasing and maintaining full pressure until the entire contents of the syringe are used.

The vesica should now be fully everted and sufficiently hard so as not to collapse when mounted. If it fails to evert, usually due to a constriction in the vesica itself, or because some part of the internal armature (cornuti, etc.), is causing an obstruction, carefully tease out the vesica through the open, posterior end of the aedeagus using a pair of fine forceps. At this stage, if the vesica contains several cornuti etc., it may be advantageous to add a very small amount of liquid detergent to act as a lubricant. Before mounting in Euparal, Euparal Essence can be used to clear the preparation.

Examination of genitalia and other features *in situ*

In some cases, with males, it is possible to determine the species by simply extruding the valvae from the abdomen. In larger species this can, with practice, be achieved in the field without killing or seriously injuring the moth, by grasping it gently but firmly by the thorax and gently pinching the abdomen. The extruded valvae can then be examined with a hand-lens. It is often useful to first determine the sex of the moth, which may not be straightforward if the species in question is not sexually dimorphic. However, the tip of the abdomen of female moths is generally more rounded, and the tip of the ovipositor can often be seen, most easily with a hand-lens, as a single, elongated central structure (especially when scales have been lost after egg-laying). The abdomen of males is generally more blunt-ended and the scale tufts are more obvious. However, these differences are less obvious in some species. If a female is selected by mistake, this will immediately become apparent, as the ovipositor will be extruded when the abdomen is pinched.

Examination *in situ* is more easily performed on freshly-killed or anaesthetised specimens or on an abdomen softened with KOH (as described in the previous section). Place the moth on its back and press gently on the lower segments with a needle until the valvae open out. Many diagnostic features can be seen this way, although it may be necessary to brush away scales. If the moth is to be preserved, in order to minimise loss of scales from the wings and other parts of the body, pin it with a headless pin, turn it over and pin in a piece of plastazote (available from suppliers) and hold the wings in place using ordinary setting pins placed diagonally. Often, the valvae will stay open after they have been extruded allowing easy re-inspection. Other features such as the shape of the uncus and cucullus, and the projections on the eighth sternite of the *Epirrita* species, can be checked this way, but for subtle differences in shape, arrangement of spines, etc. it is necessary to perform a full dissection. In some cases, for example *Acronicta tridens* and *A. psi*, it is possible to check the feature in the field with a hand-lens by very gently pinching the end of the abdomen between finger and thumb.

On dried specimens, some features can be seen by gently brushing scales away from the tip of the abdomen. In general, the abdomen must first be removed (and it may disintegrate during brushing if fragile) and held with forceps, but the difference in the costal extensions on the valvae of *Xanthorhoe spadicearia* and *X. ferrugata*, for example, can with practice be seen by brushing with the specimen fully intact. If the moth has died with the valvae retracted, easily diagnosed features often cannot be seen, and it will be necessary to use KOH, which gives the option of proceeding to a full slide preparation, for future reference.

MORPHOLOGY

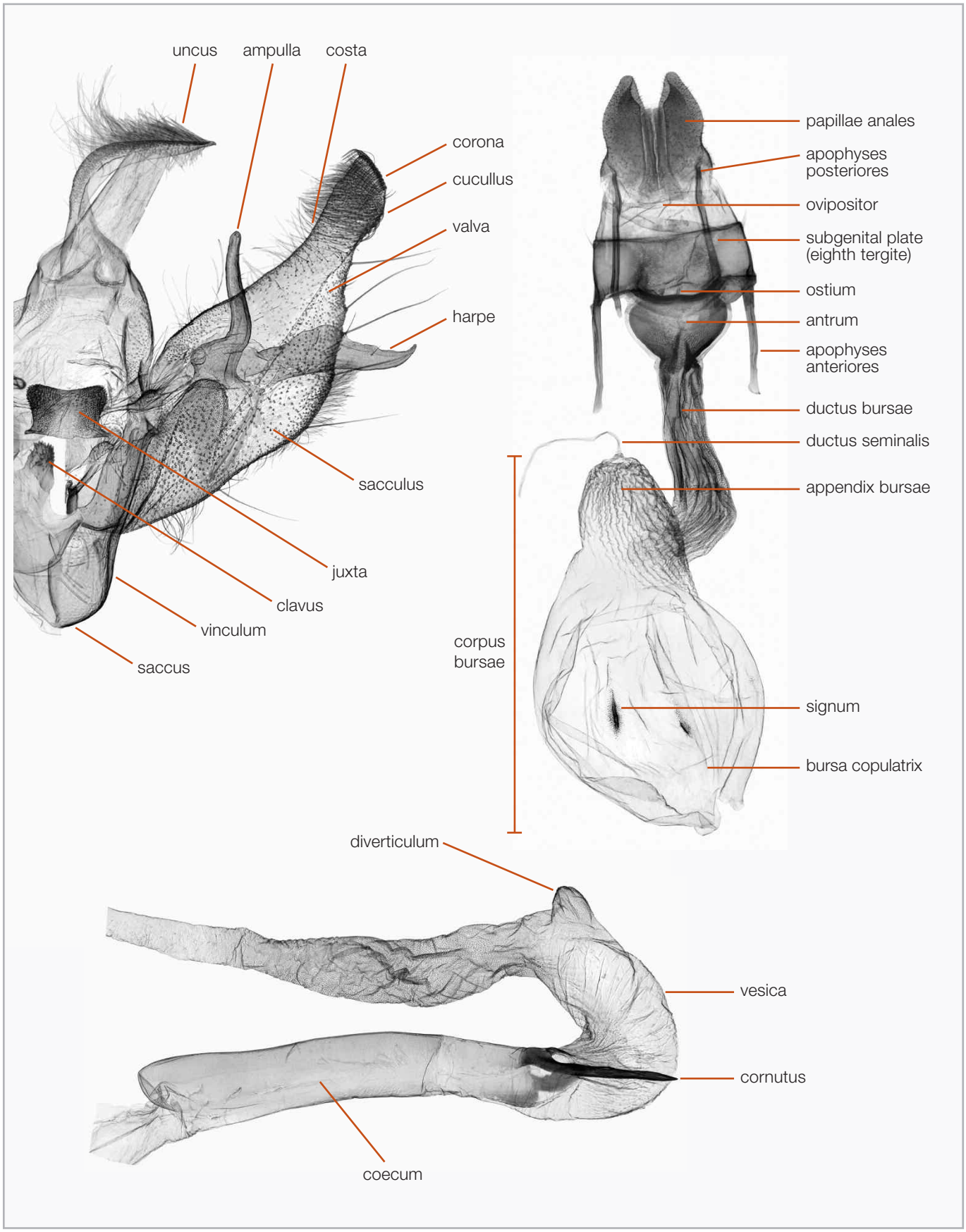

Figure I. Structural terms used to describe moth genitalia.
Male (top left), female (top right) and male everted aedeagus (bottom).

GLOSSARY OF TERMS

A cautionary note about anatomical terminology

Overleaf will be found a list of anatomical and descriptive terms used in this book, along with names of other major structures of the genitalia. The definitions have been drawn from a number of sources, including Torre-Bueno (1937 and later revisions) and Klots (1970). Where there may be ambiguity (see also below), further details are given. This is far from an exhaustive list of terms either for Lepidoptera or insects in general, and many others will be found in the aforementioned texts and elsewhere. Scoble (1992) describes the parts of the genitalia of Lepidoptera and their arrangement, and summarises our knowledge of the ways they are thought to have been derived from the terminal segments of the abdomen.

As mentioned in the introduction, and noted by Scoble (1992), a bewildering variety of anatomical terms has been used by a number of authors over many years, in an attempt to describe the various parts of moth genitalia, often inconsistently. The problem is especially acute in the males, and appears to be partly as a result of uncertainty or disagreement as to the evolutionary origins of these structures. In some cases, the same term has been used to describe several different, usually related structures, or parts of them, and one structure has had several names assigned to it by different authors. This is most notably the case for names given to the processes on the ventral surface of the valvae (clasping organs). In this book, we have avoided the use of the functionally descriptive, but confused, term ‘clasper’, which is often used to describe the whole clasping organ (i.e. the valva and cucullus) and equally often for a projection on the ventral surface of the valva. The same can be said of the term ‘harpe’, but its use to describe the whole structure is perhaps more a feature of the older literature.

In the case of *Cyclophora*, Hausmann (2004) uses the term ‘fibula’ to describe the thin process arising in the basal half of the valva in some species. One dictionary definition of fibula is a “clasp or buckle”, and therefore the term may be synonymous with harpe or clasper. In other cases, the processes on the ventral surface of the valvae are described as ‘extensions of the sacculus’. Here, the distinction from some uses of ‘harpe’ or ‘clasper’ may be unclear, but again where they are diagnostic we have generally followed the most recent authoritative works in describing these features. To further confuse the uninitiated, a long, narrow process under the valva is sometimes known as an ‘ampulla’, and in some instances the term ‘pollex’ has been used to describe a similarly-shaped structure. Confusion of terminology is not confined to the male valvae. In the females, some authors use ‘bursa copulatrix’ to include the ductus bursae whereas most keep them separate. The pointers used on the figures in this book should preclude confusion when using the text to identify a specimen, but the above discrepancies are worth bearing in mind if it is being used alongside other works or compared with them. Readers should refer to the glossary provided therein, or to the standard works listed above. Generalised diagrams showing the terminology for external features can be found in Skinner (2009).

GLOSSARY

Aedeagus tube-like organ of the male genitalia laying between the valves and functioning as a penis, often adorned with spines and useful in determining the species. It houses the vesica (sometimes referred to as the endophallus).

Allopatric referring to taxa occurring in geographically separate areas.

Ampulla in the male, a process arising from the sacculus, usually thin and tubular and on the costal side.

Anal angle in ventral view, the anterior extremity of the cucullus (in the male).

Anellus in the male, the membranous covering of the aedeagus.

Ante- before the middle, e.g. an antemedian line on the wing of a moth.

Anterior towards the front end of the body.

Antrum in the female, a chamber or cavity formed from part of the ostium in some species (see also ostium bursae).

Apex (pl. apices), apical, apically referring to the furthest point from the body or point of attachment.

Apophyses anteriores in the female, the pair of elongate processes arising from the eighth sternite.

Apophyses posteriores in the female, the pair of elongate processes arising from the ovipositor.

Appendix bursae in the female, a secondary swelling attached to the bursa copulatrix (which is then called the corpus bursae).

Basal, basally, basad closest to the body; towards the body or point of attachment.

Bifurcate partly divided into two, i.e. forked.

Bipectinate comb-like on both sides, e.g. antennae.

Biserrate saw-like on both sides, e.g. antennae.

Bursa copulatrix in the female, part of the bag-like structure connected to the ductus bursae, which is used to store sperm. If an appendix bursae is also present, this together with the bursa copulatrix constitute the corpus bursae. It is often adorned with spines, which may be distinguishing identification features.

Carina an elevated ridge or keel, not necessarily high or acute.

Cilium (pl. cilia) scale or scales resembling hairs, a row of which usually border the wings, or adorn the antennae or other organs.

Clasper(s) the valves in the male genitalia or parts of the armature thereof (usually on the median section or towards the base). In this book, use of the term has been avoided. It is also synonymous, in both meanings, with harpe.

Claviform stigma a mark in the central area of the forewing of noctuid moths, often club-like.

Clavus in the male, a process arising at the costal side of the sacculus. E.g. in *Mesapamea*.

Cline an ecotype or form exhibiting gradual differences over a geographical area.

Coecum in the male, a blind sac (part of the aedeagus).

Colliculum in the female, a small dorsal plate or narrow ring-like sclerite of the ductus bursae.

Cornutus (pl. cornuti) in the male, a spine arising from the aedeagus.

Corona in the male, a row of spines along the outer margin of the cucullus, extending across its inner face.

Corpus bursae in the female, the bag-like structure connected to the ductus bursae, used to store sperm. Comprises the bursae copulatrix and appendix bursae (which may be absent). It is often adorned with spines, which may be distinguishing features.

Costa, costal in male genitalia, referring to the uppermost (i.e. posterior) margin of the valva in ventral view. On the wing of a moth, the leading edge.

Cucullus in male genitalia, the tip of the valva, often necked, rounded and bearing spines.

Cuticle the outer skin of the body (also known as the integument).

Dentate toothed or strongly serrated.

Disc, discal referring to the central area, e.g. that of the wing of a moth.

Distal, distally, distad away from the body or point of attachment.

Diverticulum a blind side passage, forming a sac or swelling, e.g. in the vesica (as seen when everted) or bursa copulatrix.

Dorsum, dorsal, dorsally, dorsad referring to the back or upperside. On the wing of a moth, the dorsum is the back or trailing edge.

Ductus bursae in the female, the tube extending from the ostium to the bursa copulatrix.

Ductus ejaculatorius in the male, the single duct or tube through which the seminal fluid is ejected into the ostium of the female.

Ductus seminalis in the female, the tube connecting the bursa copulatrix with the oviductus communis (the median outlet of the female genital system).

Endophallus see vesica.

Excavate having a rounded depression as if dug out.

Fascia a band.

Fasciculate clustered or tufted.

Fibula Under *Cyclophora*, used to describe one of the processes of the valva in the males (possibly synonymous with harpe and clasper).

Gnathos in male genitalia, a hardened part of the vinculum near the uncus, which supports the anal tube.

Harpe in male genitalia, the hardened clasping organ on the inner face of the valva (see also clasper and valvae).

In situ (Latin) to examine the phenomenon exactly in the place where it occurs.

Integument see cuticle.

Juxta in male genitalia, a hardened plate-like structure between the valvae which supports the aedeagus.

Lamella ante-vaginalis in the female, a hardened plate partially surrounding the ostium placed anteriorly.

Lamella post-vaginalis in the female, a hardened plate partially surrounding the ostium placed posteriorly.

Medial, medially, median middle; the central area (medio-distal = away, more distant from, the middle).

Octavals projections on the posterior margin of the eighth sternite.

Orbicular stigma a round or oval spot in the discal cell in the forewing of some noctuid moths.

Ostial plate in the female, a hardened plate surrounding the ostium.

Ostium in female genitalia, the external opening.

Ostium bursae a chamber or cavity formed from part of the ostium (see also antrum).

Ovipositor in the female, the tubular or valved structure used to deposit the eggs, sometimes extendable beyond the apex of the abdomen.

Papillae anales in the female, a paired process at the apex of the ovipositor.

Pectinate comb-like (usually applied to antennae).

Phenotype an observable trait or characteristic or an organism.

Pollex in the male, a process on the valva, usually on the cucullus as an extension of the anal angle. Also sometimes used to describe a process arising from the median section of the valva.

Post- after, beyond, e.g. post-median line on the wing of a moth.

Posterior towards the hind end of the body.

Pre- before, e.g. pre-apical spine lies before the apex.

Produced drawn out, prolonged, extended from.

Proximal towards the body or point of attachment.

Quadrate square in shape.

Reniform stigma a kidney-shaped (or similar) mark at the end of the discal cell, usually referring to the forewing of Noctuid moths.

Sacculus in male genitalia, dominant part of the base of the valva, often adorned with spines.

Saccus in male genitalia, the lowest part of the vinculum.

Sclerite hardened part of the body forming a plate.

Sclerotised referring to a hardened part of the body.

Serrate saw-like; with notched edges like the teeth of a saw.

Seta (pl. setae) stiff hair or bristle.

Setose possessing setae.

Sexual dimorphism a systematic difference in form between individuals of different sex in the same species.

Signum (pl. signa) in the female, sclerotised spines and plates on the bursa copulatrix.

Simple unadorned (often applied to antennae).

Socius in the male, a paired extension of the vinculum.

Sternite(s) the ventral sclerotised plates of the abdominal segments.

Sub-basal near the base of.

Sub-elliptical roughly elliptical but with the broadest area towards one end, i.e. egg-shaped.

Sub-genital plate the plate beneath the genitalia (eighth tergite).

Sub-quadrate nearly or approximately square.

Sub-rectangular approximately rectangular.

Sub-terminal situated towards the end, e.g. sub-terminal band on the wing of a moth.

Sympatric referring to taxa occurring in the same geographical area.

Taxon (pl. taxa) a group of organisms adjudged to be a unit.

Tegumen the dorsal half of the large central transverse ring-like part of the male genitalia.

Tergite(s) the dorsal sclerotised plates of the abdominal segments.

Termen the outer edge of the wing of a moth, adorned with cilia.

Truncate with a squared-off ending.

Uncus in the male, the top part of the vinculum, sometimes forming a large hooked or curved structure.

Valva (plural valvae, or informally ‘valves’) the large pair of laterally extending clasping organs of the male genitalia (see also clasper and harpe), articulating with the vinculum.

Ventral, ventrally the underside.

Vesica in the male, the inner sac of the aedeagus, also known as the endophallus.

Vinculum in the male, the ventral half of the large central transverse ring-like part of the male genitalia.