2005 Workshop
Identification of Lepidoptera by Dissection

Overview

There has been a massive increase in recent years in the interesting hobby that we share - studying the lepidoptera and in particular, the moths. Inevitably when starting this interest, most people wish to simply identify the majority of the macromoths they encounter. In order to do this, it is perfectly reasonable to expect to use literature and photographs and be successful most of the time.

There are, however, many groups that defy identification as imagines based upon external features and to confirm to species level it is essential to assess the identification features contained within the genitalia structures of the adult moths. Examples of such groups of species are the Grey/Dark Dagger pair, the Marbled Minor group and the November Moth group.

When moving into the microlepidoptera there are even more species groups that require this treatment. Taking of specimens when dealing with micromoths is commonplace.

This one-day course will provide recorders with a basic knowledge in dealing with specimens and will contain a practical element that when used in conjunction with these notes, will allow attendees to begin dissections at home. Dissection is certainly a skill that requires regular practise and the more you do, the better results you will achieve, although perfect results are not always achieved even by experienced people.

This course will <u>not</u> cover relaxing, setting and storage of moths, subjects that will be covered in future courses. It is also not designed to be a forum for people to obtain specific determinations but if time allows we will try and accommodate this.

Killing Methods

When dissecting moths, there is obviously no doubt that you are killing a living thing. This is of course unfortunate but the fact is that if you want to confirm to species, you have to dissect some specimens. Ideally specimens should be obtained either from existing collections that are being disposed of or even from within spider's webs etc wherever possible as these are clearly already dead.

For live specimens, it is obviously important to kill the specimen as quickly and humanely as possible. There are many methods that can be used; some of these involve special techniques and chemicals/equipment, others do not.

Remember, not all moths have to be killed for the genitalia to be assessed (see below).

Freezing (includes storage)

One of the more common ways to kill specimens is the leave in the freezer for approximately an hour. Ideally, the specimens should be removed from the freezer after the hour and then allowed to defrost for perhaps ten minutes or so. At this stage they should be relaxed enough to set if required. If not being set, they can be dissected straight away but it is much easier to dissect long dried specimens. Most moths will dry out completely in a few days if left in a pot with air holes in. Items such as film canisters can be used for storage of dried specimens but if fresh they will go very mouldy very quickly if air holes are not punched in the top.

If specimens are left in the freezer for a long time, they will often be freeze dried and can be treated as dried specimens after defrosting. If this happens they will need to be relaxed if being set. To avoid freeze-drying you can add a moist piece of tissue between the top and the tube/pot. It is far easier to set a fresh specimen than one dried by air of freeze-drying. However, the freezer *is* an excellent way to store specimens for a long time as this avoids insect/pest damage.

Boiling water

One method which works well for most micros and slim-bodied macros, such as geometers is to kill them using boiling water. The principle is to fill a mug with boiling water (or use a hot water bath/Bunsen burner arrangement) and with the specimen in a sealed glass tube, place it into the water to a depth that ensures the specimen is surrounded by water. Within seconds the specimen will stop moving and be killed.

Mike Hull first made me aware of this technique. He found it useful when travelling abroad as it prevented the need for chemicals (see below) being transported overseas and all the problems that may be associated with that.

Sometimes, the heat can ripple the wings slight but this can be corrected when setting. Again the specimens emerge very relaxed for setting if appropriate.

Chemicals

The methods involving chemicals are all broadly similar. They involve either a killing bottle or a pot of some kind and the exposure of the specimen to noxious fumes of one kind or another. Each of the fumes has its individual qualities and can be basically summarized as follows:

Chopped laurel leaves - free (assuming you have a shrub nearby), maintain a moist atmosphere so specimen still relaxed, slow to kill.

Ethyl acetate - must be purchased, kills quickly and effectively, and dries specimens if left too long.

Ammonia - must be purchased, kills quickly and effectively, does not dry specimens if left too long, can cause discolouration to some species.

This list is not exhaustive but these are the main items currently used.

Other Methods

Pinching the thorax - some of the old collectors used to kill specimens instantly in the field by pinching the thorax between finger and thumb. May obviously damage a specimen and is perhaps a bit brutal for some/most.

Pricking with a needle laced with nicotine - another old method where a needle is dipped in a nicotine solution and allowed to dry. Large specimens apparently died rapidly when the thorax is pricked with this needle. Again not something many would be content with.

Rough setting and tentative identification

If at all possible any specimens should be set properly or even rough set to allow a tentative identification to be established. This really does make it much easier to get to the correct species rather than pawing through many gen plate images to find the group. You will certainly appreciate this if people give you specimens that have been rolling round in tubes for weeks and are left with no antennae/palps (vital features in the identification of many micros).

With many micros, this simply involves killing by one of the methods above, pinning (under the microscope on a small piece of plastazote) and then blowing the wings open from the rear so that the hindwings are visible.

Dissection

As will be shown below, it is not always essential to dissect the whole abdomen (in some of the males) but in females this is always required. The following sections have been broken down into the 'restricted dissection' and the 'full dissection'. The former can take 5 seconds and the latter over an hour!

The main thing to consider is that you should always plan to have enough time to spend on your dissections. It is so easy to rush and make a mess of them, especially with the smaller species. Try and get a production line going with one boiling while you dissect another.

Before going on, a word about safety. All of the chemicals referred to in this paper are potentially extremely hazardous or toxic, especially, but not exclusively, to young children. If you do not have the facility to lock them away somewhere then don't buy them until you have got this facility. In addition, you should always take great care to get as little (or none) of the substances on your skin as possible and work in a well-ventilated environment). You are advised to put the name of the chemical into a Google (or similar) search engine and view the available data for yourself if you are concerned about this.

Restricted Dissection Methods

There are two basic methods involved here. The first is squeezing the abdomen and this can be used on live specimens of male macromoths that can be released, still alive afterwards. Personally I have never tried this but with species such as the daggers and the minors it can be quite successful. This method can also be used on freshly killed specimens as part of the setting process. This can work in the field very well with some species.

After either stunning with CO2 or ethyl acetate (enough to knock out but not kill - the time depends upon the species) the abdomen can be squeezed between finger and thumb for larger specimens or between tweezers for smaller specimens until the valves are hanging out. The specific features can then be assessed either down the microscope or with a good hand lens.

For species such as daggers and minors, and the Gold-spot pair this can avoid the need for a full dissection saving time and also meaning that the specimen will look better in the collection (if this is a consideration for you**).** If pinning the moth and if a male that requires genitalia determination at a later date, give the abdomen a slight pinch while setting because if the critical area for separation lies within the uncus it will mean the specimen should be able to be identified *in situ* and removal of the abdomen not necessary. It is always a shame to have to remove the abdomen from a macro-moth in a collection.

The second method is brushing the scales off the tip of the abdomen that is a useful way to quickly identify some groups. Like most dissection methods, long dead and dry specimens are much easier to handle than freshly killed or live specimens. The groups this best applies to are the male pugs and the males of the November Moth complex. The basic principle is to either remove the abdomen (or possibly support the intact, upside down specimen with pins under the abdomen) and to brush away the

scales on the tip of the underside with a fine artist's paintbrush or woodcock/snipe 1st primary feather.
The male pugs have an often-distinctive plate in the final or penultimate segment that can be diagnostic for many species. Some (such as Green Pug and Sloe Pug) are not diagnostic and a full dissection should also be completed. The males of the November Moth group can be identified to species based upon the spines and shapes at the end of the abdomen. In some cases, this process can be undertaken in about 10-15 seconds once the method is mastered and if dealing with a distinctive species.

Full dissection

Once you have established that a full dissection is required, there are various stages in the process. The first point to make is that everything should be kept as clean as possible. Dust and dirt can make gen preps unattractive, and more importantly, potentially unusable for identification purposes. You are likely to be working with some potentially harmful chemicals and therefore it is recommended that the work be done in a well-ventilated room. Also, bear in mind that the following procedure is the one **I** use. You may well adapt this and others may use slightly different methods. *Don't be afraid to experiment and find your own way of doing things.*

1. Tools/Chemicals

The basic toolkit required for dissection is as follows:

- Needle-nosed forceps - one pair - approx £3
- Dissecting needles x 2 (make your own with a micro-pin pushed into the end of a long cook's match) - approx £0
- Snipe/Woodcock pin-feather glued into a dowel rod is very useful for removing scales from specimens – beg and borrow one from a local game bird butcher for free. (no jokes please). You can also use a fine sliver of plastazote glued to a rod.
- Excavated glass block/petri dish x 4 if possible (but you could get away with 2) - approx £2.50 each
- Glass block cover – can help reduce the loss of fluids by evaporation. approx. £1 each
- Low power stereo microscope best range of 10x-50x) - approx £80-£a lot
- Standard monocular turret microscope (useful occasionally for micro critical assessment but not essential for most specimens) - approx £50-£a lot
- KOH - approx £3 for 250ml (approx 10-20% solution on water)
- De-ionised water - the type used for irons/car batteries and can be bought from most supermarkets/car factors - approx £1 per litre bottle
- Isopropyl alcohol - approx £5 for 500ml (approx 300-500 dissections)
- Stain - approx £3 per small bottle (approx 100-200 dissections) - mercurochrome or chlorazal black the most popular. Beware of black, very easy to over stain with this.
- Mountant solvent - approx £5 per small bottle (approx 100-200 dissections) - Euparal Essence the most popular.
- Mountant - approx £6 per small bottle (approx 100-200 dissections) - Euparal the most popular as it is a permanent mount.
- Standard microscope slides - approx £5 for a pack of 50
- Microscope cover slips - approx £8 for a pack of 100 of the (highly recommended) round coverslips. Cheaper for the square coverslips but you use more mountant.
- Pipette - approx 50p - I use a separate one for each of the chemicals referred to above
- Glass rod - approx 50p

- Roll of toilet paper - stolen from bathroom! This can however, get a bit dusty. Good quality tissue can be better but slightly more expensive.

Total cost to set up is therefore approximately £50 assuming you have a microscope but this will do between 50-100 slides.

Anglian Lepidoptera Supplies do a full kit for £56.00 which includes all the above + Ethyl Acetate + postage (but not microscope!!!).

2. Removal of the abdomen

The first stage is to remove the abdomen from the specimen. This can be achieved easily on dried specimens by either using two sets of tweezers or a set of tweezers and a pin to twist the abdomen either up or down to snap it off that the thorax. The whole thing should come off intact and leave the wings and legs on the thorax.

Fresh specimens should preferably be left to dry for a few days/a week or two for larger specimens but can be dealt with when still 'wet'. The detachment of the abdomen can be tricky though and be careful that the whole thing comes away. The main reason for ensuring the whole abdomen is detached intact is that in some species, the female genitalia in particular extend virtually to the thorax within the abdomen.

3. Softening the abdomen/removal of fat

The bodies of moths (including the genitalia) like all insects are encased within a hard exoskeleton called chitin. This needs to be softened so that the structures can be extracted and manipulated for identification and, if appropriate, mounting on a microscopic slide for storage and future reference.

The standard product used for this is a 10% concentration of potassium hydroxide (KOH). *This is a caustic substance and care should be taken not to get any on your hands or the remaining parts of the specimen if this is going to be retained*. If you do get any on your skin, don't panic but immediately wash off in plenty of water.

Specimens can either be softened in a cold solution (usually takes a few days for larger specimens) or in a warmed solution. Either way, place enough KOH in a test tube or glass pot to cover the specimens and then place the specimens in the solution.

There are several ways to maintain the constant heat required to soften the abdomen but care should be taken not to overheat as the solution can spit with obvious dangerous consequences. The method used by many is to have a water bath at just below boiling point with a peg or piece of wire holding the tube inside. Another is to use the heat from a 40w standard bulb (*do not use a bulb with a higher wattage as this will produce too high a temperature and the KOH can spit if too hot*).

An example of my own boiling equipment is shown below.

It is also possible to use a method suggested by Malcolm Storey as follows. Use a small glass jam jar (one of those tiny sample pots) and a small Heinz beans tin. You can stand 4 small specimen tubes (containing micro abdomens, in KOH) in the jam jar and the diameter is such that they can't fall over. Then pour boiling water into the beans tin until it flows into the jam jar. Leave until cold and they're done. If necessary repeat with more hot water.
The purpose of this process is to make the specimen soft and also to clear the bulk of the fat from the specimen (in particular from the bursa of female specimens). The specimen can take anything from 10 minutes (can be as little as five minutes with tiny specimens, especially if the KOH is already warm) to about an hour to soften and clear properly depending upon the size of the abdomen. Once the specimen is 'soft and squishy' it is usually ready to be dissected.

4. Initial assessment

The initial assessment of the specimen is designed to confirm whether you are dealing with a male or a female specimen as the methods used are somewhat different. This step can be omitted if the sex of the specimen is obvious (e.g. some species have different morphology for males and females).
Take the specimen from the KOH solution with either pin or needle-nosed forceps and place into a watch glass or petri dish with enough de-ionised water to cover the sample. Ensure that the specimen is maintained under the water as much as possible to reduce air bubbles. Squeeze the middle of the abdomen gently to partly push out the abdomen tip and the shapes of the male or female genitalia will be obvious. The females have a twin ovipositor tip and the males have claspers held together. This should look roughly like one of the images below.

Typical female (left) and male (right) abdomens after softening on KOH

5. Dissection

This is the part that everyone dreads but is in truth quite a straightforward process as long as it is done with care and patience. When you get to some of the micros they can be quite tricky and some species require certain parts of the abdomen to be retained to assist the identification but on the whole it is achievable by anyone with a steady hand. Before you start an unfamiliar species, always have a look at the published information to see what the plate looks like and what parts, if any, of the abdominal skin should usually be retained. This will give you an idea how to do the

dissection and what to expect. Some recorders argue that we should always retain the abdominal skin and place alongside the genitalia on the slide. The reason for this is that features for identification may become apparent in the future which we don't presently use.

With the abdomen in the water,

Females - The female genitalia basically comprise the ovipositor, the lamellae, the ostium (entrance hole to the ductus bursae), the ductus bursae (long or short tube from the ovipositor to the bursar) and the bursa copulatrix (large or small bag at the end of the ductus bursae and often has small or large darker markings (signum/signa) or can be clear of markings. Care should be taken to ensure that the bursa copulatrix is not ripped off as it is quite delicate and often is essential to the identifcation of the specimen. The procedure I adopt is to use the fine forceps (to tear) and a dissecting needle (to hold) to rip the abdomen up one side then the other and then across the top at approximately the 7th or 8th segment. The whole thing will then look like the photo on the left. Then *carefully* tear the remaining skin around the lamellae so that all that is left is the actual genitalia parts. Be warned that in some groups (tortrix moths for one) the last procedure cannot be done effectively and the skin should be left on at segment 7 or 8 and the scales cleaned off.

Some other groups (e.g. Coleophoras) have the bursa copulatrix so far up the abdomen toward the thorax that great care must be exercised when doing the detachment of the abdomen and also the initial stages of the dissection. Again, sometimes it can be appropriate to leave a large proportion of the female micro genitalia structures whole – i.e. don't remove any parts from the abdomen. This is obviously dependent on how see-through the skin is and how successful the removal of the scales has been but in most cases it allows good viewing of the structure. To be successful with this though, no or very little stain is used.

The final stage is to remove any remaining tissues not directly required. Normally at this stage I grasp the ovipositor with the fine forceps (and holding the other stronger parts of the plate with a dissecting pin) pull the ovipositor as far as it will come out (without straining it too far) to allow the shape and length to be displayed in the final plate. Sometimes staining (see below) highlights additional bits of fat etc that need to be removed. You may sometimes at this stage see cloudiness or large air bubbles within the bursa copulatrix. Either can be removed by putting a small hole in the bag with a pin and then using the fine forceps to gently push out the offending substances. Fat will not normally disappear during the remaining processes but air bubbles (even quite large ones) usually disappear during the slide mountant curing process.

Males - there is little doubt that most male specimens provide far better features for identification on the whole than female specimens. This is not always the case though.

The process for males is broadly the same as that described above for females. There are usually just two main structures that need to be retained and these are the main plate and the aedeagus. Sometimes it is appropriate to retain the abdominal skins (in

groups such as the November moths and the pugs) and you should check the literature to confirm if the specimen you have requires this treatment. Many of the smaller species of micros, such as the Eriocrania, have the male structures as an integral part of the abdominal skin. Separation of the male genitalia should not be attempted in these groups. Again, a check of the literature will help you decide what to do. Some continental books show male structures, such as the genus Scrobipalpa, opened up. This extremely delicate process is achieved by splitting the structure at its narrowest point. This is very difficult and it is probably best to leave the structure entire in most cases. The MBGBI volumes show the structures in one piece.

Once again, the procedure adopted is to use the fine forceps (to tear) and a dissecting needle (to hold) to rip the abdomen all the way up one side then the other and then *carefully* tear the remaining skin around the main plate (which contains the aedeagus) so that all that is left is the actual genitalia parts. If appropriate retain the part of the abdomen which has the plate. At this stage, there is sometimes a bit of skin left at the vinculum (bottom of the plate where the valves join) that needs to be torn to allow the valves to be hinged apart and opened. It should now look a bit like this.

Then clean off any scales with a pin or the forceps and the plate is then ready for final preparation. The difference between an uncleared (above) and a cleared plate (below) with the aedeagus removed is obvious. In most cases the aedeagus should be removed by grasping it with forceps from the back and pulling it out. Take care not to pull too hard as the whole structure can be destroyed if you do. Sometimes you have to detach some of the skin around it before it will come out. In some cases the aedeagus does not contain features useful in identification and can therefore be left in situ. This is appropriate for some of the smaller species as they are just too difficult (for me anyway!) to extract without ruining the whole thing. Again check the literature to see what is the best thing to do.

You may sometimes at this stage see air bubbles within the structures. They can usually be removed by using a pin or forceps to gently push out the offending substances. Air bubbles (even quite large ones) usually disappear during the slide mountant curing process.

6. Staining

This stage is not essential but can help highlight features that may not be seen otherwise. Care should be taken though as staining can also hide the features if the stain is applied for too long. Personally I only tend to stain female specimens (and male pug abdominal plates - see above) but some recorders stain all specimens and others don't stain any. You will learn to use your judgment on what should and should not be stained but get used to this with common species and not (like I did once) with

a new garden species that then became unidentifiable as I overstained it - this was the first specimen I had tried to use a stain with.
Assuming you are using a water based stain (recommended) the procedure is to shake the bottle, and then using a pipette place enough stain in the glass block to cover the specimen completely. Then transfer the specimen from the water to the stain for an appropriate period. Personally I try 10-second bursts and then transfer back to water, giving the specimen a few taps with the forceps to remove excess stain in the water. Have a look to see if it's how you want it to look, and back to the stain for another 10 seconds if a deeper colour is desired. This way you build the stain gradually and reduce the possibility of overstaining.
If the specimen is over cooked in the KOH it will bleach the genitalia somewhat, staining will bring back the features.

7. Assessment

At this stage it is often best to attempt an identifcation as the plate can be moved around and check at different angles to confirm the features required. These can sometimes be masked when the slide is completed at it effectively becomes two-dimensional. You can look at the plate either in the water or you can make a temporary slide with a slide, a coverslip and a few drops of water from the watch glass, placing the plate in position. This can also often help in forcing air bubbles and remaining fat out.

8. Isopropyl Alcohol (alcohol)

The next stage of the process is to harden and dry out the plate. This is an essential stage as it removes the water from the plate. Water left in the plate can cause milkiness in the final slide and ruin it. Using a glass block cover can reduce the loss of liquids through evaporation.
Take the plate from the water using either forceps or a pin and drop into the glass block containing enough alcohol to completely cover it. You should then make the plate as flat as possible whether it is a male or a female. Females can just be pressed down with a pin or forceps and the parts arranged in the best way to show off the relevant features. With males the claspers should usually (but not always - check the species - some such as Bryotropha species for example are best viewed closed from the side) be held apart in the open position until the plate hardens. This is often easier with the plate lying face down rather than face up. Hold for about half a minute or so and the plate should then hold itself in place. Be careful with the specimen now as it becomes relatively fragile but this is a good time to gently remove any remaining scales from the plate.

9. Place in the solvent (euparal essence)

The next stage is very simple. Again using the appropriate solvent for the mountant you use (in my case euparal - solvent = euparal essence) pipette enough into the watch glass to fully cover the specimen and then place the specimen into the solvent for enough time to allow the whole specimen to be completely soaked. This process is called 'wetting'.

10. Final stages

While the specimen is soaking in the solvent take a blank slide and wipe it with a tissue dipped in the alcohol. Polish it off with a clean lint free cloth to ensure there is no grease or 'bits' on the surface. Place enough mountant onto the centre of the slide

relative to the size of the specimen and the slide cover. This can be as much as 3 drops (run off the glass rod) for large macros and as little as half a drop for micros.
Then take the specimen from the solvent and arrange it within the mountant as best you can to see the features required. Then take a slide coverslip (make sure it is clean) between finger and thumb (on the edges not on the surfaces – or use tweezers) and at an angle slide the bottom of it towards the mountant. You will see when it hits the mountant and when it does use a pin to gently lever down the slide cover onto the mountant. Do this carefully like a hinge so that the likelihood of trapped air bubbles is reduced. The slide coverslip will then work its way down and the mountant will work its way to the edges. If appropriate you can press the coverslip down gently with a pin or forceps, to ensure the mountant spreads properly or push it around slightly so that the parts are displayed in the best way. This can also help push bubbles out of the slide. If you do get some bubbles in the slide then don't panic, as they should eventually disappear. If they are very large or your specimen rolls around as the coverslip drops (this can happen) then you can always lift the coverslip with a pin, put the specimen back in the solvent and after cleaning the coverslip and slide with solvent, start again. Sometimes if the mountant does not work its way all the way to the sides, you can help this by putting closed forceps into the mountant solvent and then opening them next to the slide cover to allow the blob of solvent to drop onto the slide. Alternatively if genitalia not in the required position, try manipulating the cover slip, this can work sometimes.
Occasionally with larger specimens, the coverslip will lift slightly overnight and leave the mountant not quite at the edge. If this does happen (I normally check them the next day) then you can get a blob of mountant on a pin and drop this in the appropriate place next to the coverslip. This will then normally run in and fill the gaps. If a very large specimen, the use of a ring slide may be required.
The next stage is to make a label for the slide or otherwise mark it in some way to identify it later. Ideally, use the same label for the slide and the specimen if the specimen is to be retained. Alternatively you can use a handwritten gummed label or a glass pen to mark the slide. Do this immediately as it is very frustrating trying to remember which specimen the plate came from if you don't! If you are dissecting a specimen which is the only specimen of that species that day then a normal numbering method by date is fine. However, it may be that you have more than one specimen from one day and in this case a number or letter sequence on the slide and specimen to link them both together can be useful. One example of this may be to use a number sequence 01020304, where the first (01) relates to the first, second, third, etc prep. done that day and the rest is the date on which it was done. The example being the first prep done on the 2nd March 2004.
Depending upon the type of mountant used, they can take up to 6 months to fully cure and personally I store all of my slides horizontally in a standard slide case, which in turn is placed upright (like a book) on a shelf. This prevents them from getting dusty and allows them to cure over time. I store mine in Bradley & Fletcher code number order so they are easier to find. Make sure you mark them male and female so you can find them more easily for reference.
Then move onto the next one!

Mountant	Solvent	Alcohol stage required?
Euparal	Euparal essence	Yes
Numount	Xylene	Yes
Aqueous mountant	Water	No
Alcoholic Mountant	Isopropyl alcohol	Yes
Canada Balsam	Turpentine	Yes
DPX	Xylene	Yes

Ready-reckoner for compatible chemicals

Identification

O.K. so you have got your slide ready for identification and want to know what to look for. It really helps if you know the names of the parts of the genitalia and can then converse with others about what you mean. The following are typical male and female genitalia designed to help understand the main structures. Any good textbook (such as the MBGBI series, Goater etc) will have references to these parts and some will have the structures named. If you ever need further help or are not sure of what a part is called, then please feel free to contact me and I will try and help.

Publications which provide possible sources of diagrams for Moth Genitalia are provided below.

This is not an exhaustive list but will hopefully provide you with a good list of future Christmas Day and Birthday presents! Some of them (in fact most of them) are not cheap. Another option is to contact either your local moth recorder or someone else nearby that currently undertakes dissections.

MBGBI Series of books (particularly good for the micros) - Volumes 3 and 4(i+ii) are particularly good but there are also some in Volume 2. Volumes 9 and 10 also have some of the critical species but not Common /Lesser Common Rustic.
British and Irish Pug Moths - A.M.Riley and G.Prior – good and covers all the UK pugs
British Pyralid Moths - a Guide to their Identification - B.Goater – excellent book on this group with some critical genitalia drawings
Tortricidae of Europe. volume 1 and 2 - J.Razowski – apparently excellent and contains all but a few UK species, all with genitalia diagrams.
Pierce/Pierce and Metcalfe books – these are very comprehensive and include many species not shown elsewhere but the illustrations are variable to say the least. This series will be digitised by Pisces shortly.
Microlepidoptera of Europe Series by Apollo Books – good but expensive and all not yet completed.
Geometrid Moths of Europe Series by Apollo Books – good but expensive and all not yet completed.
Noctuidae Europaeae Series by Apollo Books – good but expensive and all not yet completed.

Shane Farrell
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