

Processing freshwater ostracods from archaeological deposits, with a key to the valves of the major British genera

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Summary

Techniques for the extraction, preservation and examination of freshwater ostracods from Holocene deposits are reviewed, and a key to the valves of British Holocene and modern genera provided.

Introduction

Ostracods are a diverse group of small, bivalved crustaceans, which occur in almost all aquatic environments, including both permanent and ephemeral lentic and lotic waters. The animals themselves are essentially shrimp-like, but with a bivalved, calcareous shell (carapace). Many species occur in fresh waters, almost 100 of which are known from modern Britain and British Pleistocene and Holocene deposits (Griffiths and Evans, in press).

The analysis of ostracod valves from ancient freshwater sediments has become increasingly important in palaeolimnology and other branches of palaeoecology. Ostracod valves preserve well in a wide variety of depositional environments, and may also occur in very large numbers. As a result, small sediment samples may yield a useful range of species and their various ontogenic stages. These assemblages can provide quite specific information on the nature of the environment from which they derive.

Although many archaeozoologists are aware of freshwater ostracods, very few attempts have been made to apply ostracod analysis within environmental archaeology. There are a few published accounts of ostracods recovered from archaeological sites; however, formal ostracod analyses are unusual— notable exceptions being the work of Robinson (1986) and Bradbury *et al.* (1990). Despite this,

ostracods are recognized as valuable indicators of palaeohydrology, palaeoclimate, and other parameters (see De Deckker and Forester 1988; Carbonel *et al.* 1988; Delorme 1989).

This article gives some details of methods whereby ostracod valves can be extracted from archaeological sediments, and information on the handling and storage of specimens. An identification key to the genera of British Holocene freshwater taxa is provided.

Methods

1. Sources of material

Ostracods may be expected to occur in almost any non-acidic, water-lain deposit. As ostracod shells are calcareous, optimal preservation occurs in calcareous deposits such as tufas and marls, but they also occur in other fine sediments. Preservation is poor in peats and other acidic, humic sediments, from which subfossil ostracods may be entirely absent (Kempf 1971).

In river valleys and other non-lacustrine environments, ostracod valves are usually collected during bulk sediment sampling (e.g. monolith sampling) from sections exposed during excavation, perhaps primarily for Mollusca, thin section studies or other types of sedimentological analysis. In other environ-

ments, and particularly in the case of lake deposits, sampling usually involves taking sediment cores with a Russian or Piston corer, or similar devices.

Once obtained, ostracod-bearing sediments should be wrapped carefully, protected from desiccation, and labelled. Cores can be protected by extrusion into plastic piping, and then wrapped in cling-film and silver foil. Sediments should be stored until required, deep-freezing being the best method.

Small numbers of ostracods may also be collected during the routine sieving of sediments for molluscs, vertebrate remains, etc. In this case, many smaller species may be lost unless a proportion of each sample is sieved through the finer mesh sizes. Ostracods thus extracted should be mounted directly onto micropalaeontological slides and stored dry, or (less desirably) preserved dry in labelled specimen tubes. Ostracod shells must never be stored in formalin which, unless buffered, causes dissolution of the calcareous shell matrix.

2. Sampling resolution and sample size

Sampling resolution may be dictated by a number of factors, including both the nature of the sediments under examination, and the

time and resources available for specimen processing and identification. The range of other taxa being investigated during palaeoenvironmental analysis may also constrain sampling strategy and sample size. The number of ostracod valves within a given amount of sediment may vary by several orders of magnitude; lacustrine calcareous muds and tufas may include hundreds of valves per gram of sediment, but in some deposits valve densities may be less than 1 valve per gram. Where ostracod densities are high, it is usually possible to sample distinct horizons of a sediment sequence at regular intervals. This sampling is usually based on depth-related lithostratigraphic intervals, and undertaken in a consistent manner in regularly spaced horizons (for example, with sampling at 1 cm intervals, by examining one in every five samples, or taking alternate 5 cm sections). Sampling across lithostratigraphic boundaries should be avoided wherever possible. As resources permit, an entire sequence may be sampled, or just key horizons at strategic points within a sequence.

Equitability of samples may be achieved either volumetrically ($x \text{ cm}^3$ of sediment/sample) or by weight (x grams of sediment/sample). Where weights are used, they should be based on the weight of the sample following air- or oven-drying to constant weight. Processing samples by standard weight is preferred

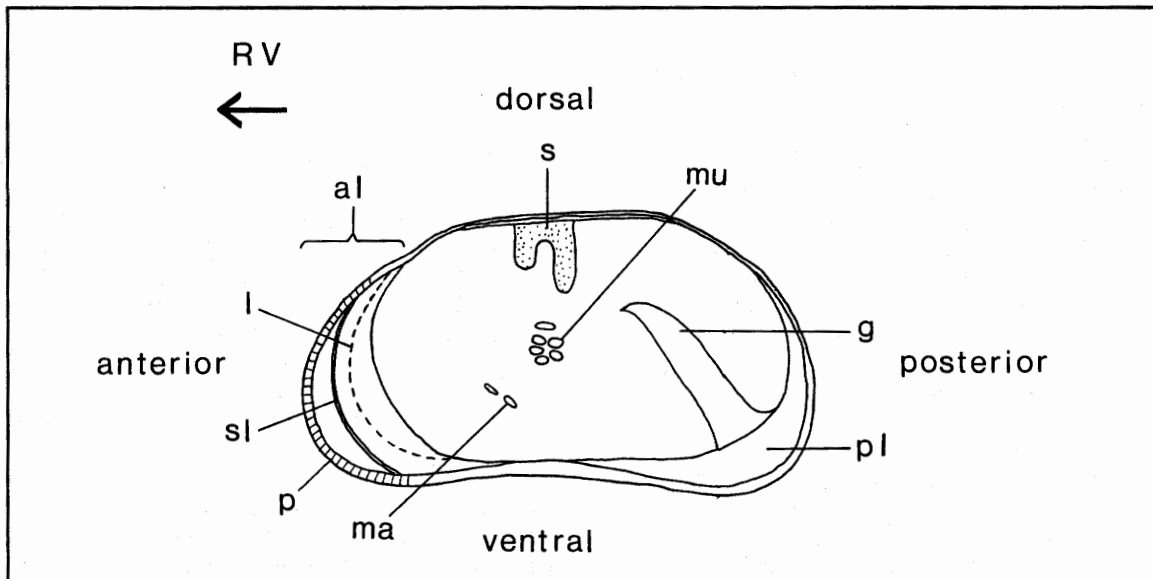


Figure 24. Inner view of the right valve (RV) of a composite freshwater ostracod, showing diagnostic characters used in the key: al = anterior inner lamella; g = genital impression; l = list; ma = mandibular scars; mu = muscle scars; p = pore; pl = posterior inner lamella; s = sulci; sl = selvage.

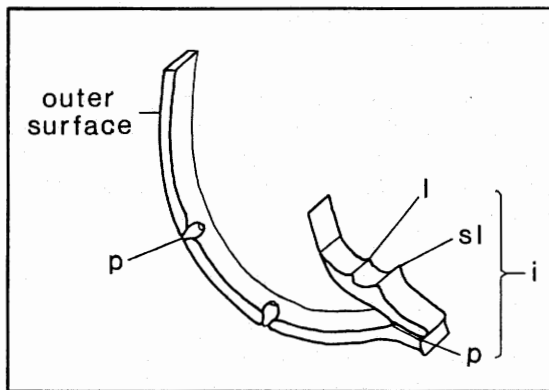


Figure 25. Cross-section through the antero-ventral margin of an ostracod valve: *i* = inner lamella; *l* = list; *sl* = selvage; *p* = pores (based on Van Morkhoven 1962).

where ostracod extraction is coupled with sampling for Mollusca. Twenty grams of sediment is usually sufficient from most calcareous depositional environments, although a certain amount of trial and error is often required to determine sample size. Lake marls, in which ostracod numbers may be very high, may require the adoption of a sub-sampling regime. Although rather different sampling strategies are possible from one sequence to another, within-sequence consistency is important.

3. Processing

Techniques for the extraction of ostracods are essentially similar to those used in the

extraction of terrestrial Mollusca (Evans 1972). Most sediments require disaggregation in dilute (approximately 5% v/v) hydrogen peroxide. Digested residues should be washed through a 125 μm sieve and then oven-dried. This process may need to be repeated several times, particularly when the sample contains large amounts of organic matter. Should valves prove difficult to remove from such sediments, extraction under water, or even under dilute alcohol, may be required. Dry extractions are best undertaken in a small sorting tray coated with blackboard paint (which increases contrast and reduces static). Wet extractions are best undertaken in square 'Petri' dishes. Where sediments are compacted, the reaction rate of the digestion can be increased by applying heat at the start of the digestion process, or by either increasing the concentration of the peroxide solution or the number of digestions. In such circumstances, care must be taken to avoid damaging the valves, and repeated checking is required. Other methods are used occasionally; some workers repeatedly freeze and thaw samples until the sediments disaggregate, or utilise flotation techniques (e.g. De Deckker 1979). We have found the performance of flotation techniques to be both unreliable and unpredictable.

Valves rarely require additional cleaning, although sometimes this may be necessary to allow the examination of the muscle scars (Figs. 24, 26). Cleaning should be undertaken with extreme caution and many agents, e.g. laboratory detergents, may cause valves to

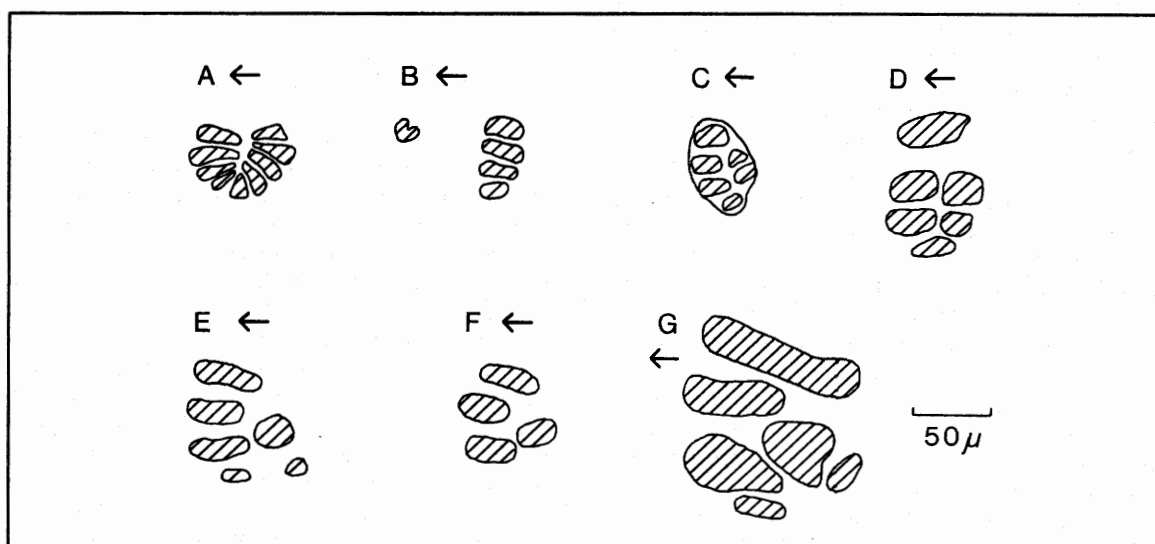


Figure 26. Major muscle scar field types, seen from ext. LV (redrawn from Ghetti and McKenzie 1981).

disintegrate. Gently brushing with a fine sable hair paintbrush and deionised water is recommended.

Many ostracod valves are damaged by microbial, chemical and physical processes, and in practice it is often difficult or impossible to identify valves that have been badly damaged. Identifications should be limited to largely intact valves and carapaces, although valve fragments representing <50% of the original may be retained for referral if necessary. Ostracods are best extracted by individually hand-picking valves from sediments under a dissecting microscope, with a fine, damp sable paintbrush. The extracted valves are then cleaned (if required) and transferred to micropalaeontological slides, previously washed with an aqueous solution of gum tragacanth (Macarthy's, Romford, Essex). Gum tragacanth dries quickly, but is water soluble and will mount damp ostracod valves firmly, but releases them readily if required. Micropalaeontological slides are sealed with a normal microscope slide, are easily stored, and will protect the specimens for many years, with minimum curatorial effort and negligible expense.

4. Sub-sampling

Although it is theoretically preferable to extract all ostracod valves from a given sediment sample, their abundance may make this impracticable. Ostracod valve numbers may also be increased considerably by the presence of juvenile moult-stages. Examination of juveniles provides additional taphonomic information, as a full range of juvenile moult-stages strongly indicates *in situ* preservation (see Whatley 1988). Unfortunately the identification of juveniles is often difficult, so many ostracod workers do not examine juvenile moult-stages at all. Adults are recognisable by the greater degree of calcification of the valves, by their size as compared with that of the juvenile moults, and sometimes by the presence of impressions of the testes or ovaries on the inner wall of the shell (Fig. 24). The size of the sample examined is largely a matter of personal preference; we have achieved good specific representation from samples of 500 valves. Species-acquisition curves, and other methods of determining optimal sample size are unreliable, as these may differ considerably from one horizon to the next so that separate

determinations would be required for each part of the sequence.

A standard sub-sampling regime has been proposed by Danielopol and Casale (1989) in which the ostracod sample is spread evenly upon a gridded, numbered micropalaeontological tray. In our variant of this procedure, the tray is divided into 32 equally-sized, numbered squares, and 500 ostracods are extracted from the numbered squares as dictated by a train of random numbers (generated by a random-number generator). In addition, the number of valves (where an intact carapace = 2 valves) is counted from ten squares, again following the sequence of random numbers. This results in sample size of 500 valves, and ostracod counts from ten squares of the tray. The mathematical distribution of ostracods in the tray is then described following Elliott (1977). This allows the estimation of the total number of ostracods in the original sample.

5. Data presentation

From the extrapolated values obtained, ostracod data may be presented as either tables of raw numbers, or as diagrams of percentage abundance or absolute numbers. These can be plotted in exactly the same way as molluscan diagrams. In addition, various techniques may be used to characterize ostracod assemblages, e.g. diversity indices such as Brillouin's Index (Magurran 1988), clustering and ordination techniques, e.g. TWINSpan or DECORANA (Gauch 1988) or taxocene analysis (Griffiths and Evans 1992).

6. Identification of ostracod material

There are very few works that deal with the identification of subfossil freshwater ostracods other than Absolon (1973), which deals with central European species. The valves of British marine and brackish-water faunas are illustrated in detail by Athersuch *et al.* (1989). Modern British freshwater species are illustrated by Henderson (1990), although the taxonomic keys are primarily based upon soft-part anatomies. Ecological information may be obtained from Klie (1938), Nüchterlein (1969) or Henderson (1990), and a good introduction to the taxonomic literature is provided by Kempf (1980a-d; 1991). The collection of modern reference material is also a valuable aid to identification, and allows the development of a first-hand appreciation of

natural ostracod communities (e.g. Griffiths and Evans 1991).

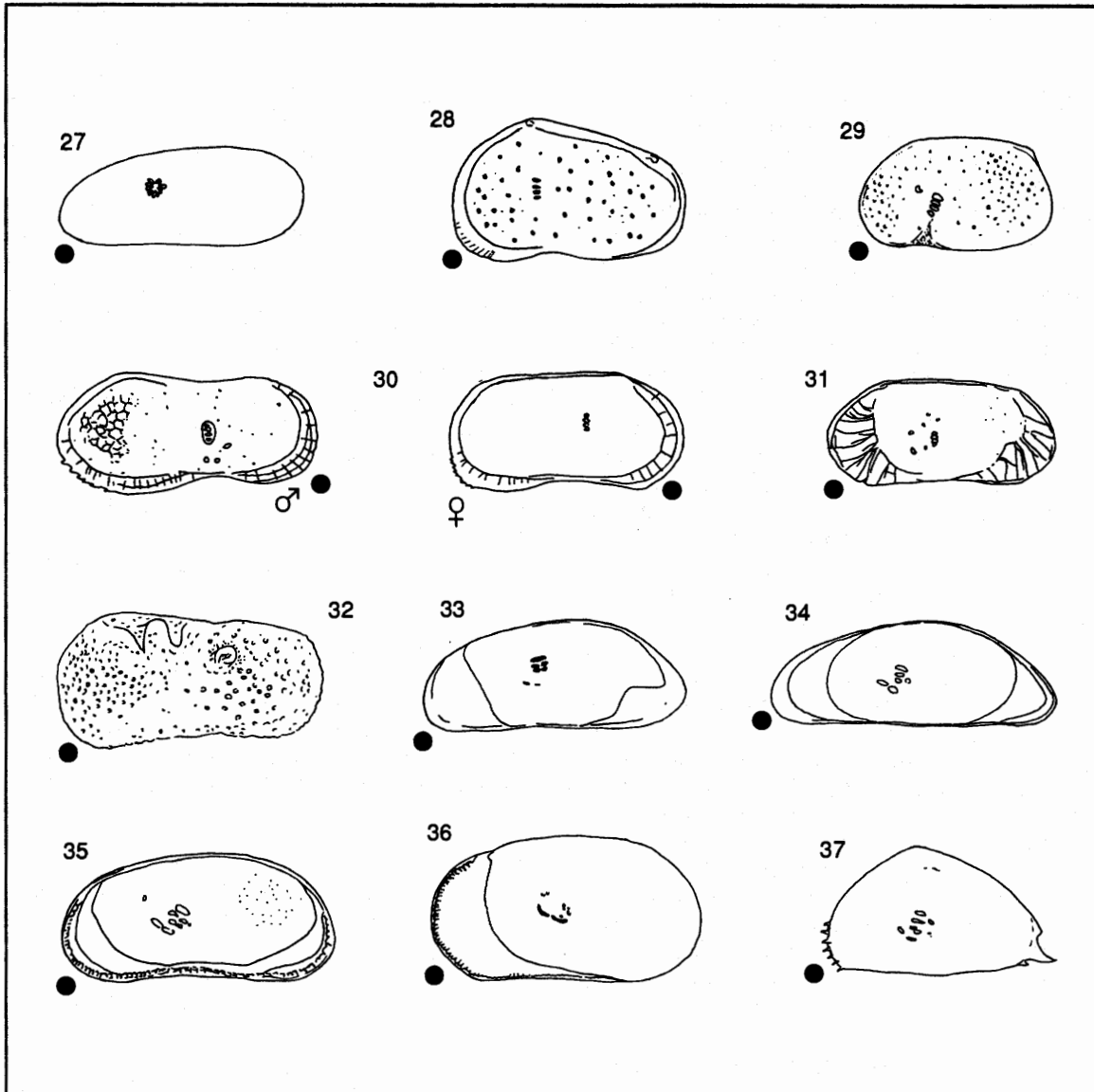
Ostracod valves can be orientated by examining the location of the muscle scars, which usually lie anterior to the mid-point of the valve. Valves should be examined under a high-power dissecting microscope, with particular attention being paid to the overall shape and structure of the shell (Fig. 24), the structure of the inner lamellae (Fig. 25), and the pattern of the muscle scars (Fig. 26). Where the muscle scars are not visible, improved resolution may be achieved by gently brushing the valve with dilute food colouring, or by placing the valve onto a microscope slide in a drop of glycerine, and then viewing in transmitted light. An excellent introduction to ostracod morphology is provided by Van Morkhoven (1962).

The key presented here will allow the assignation of most adult specimens to genera, although other faunal works should also be consulted. Identification to genus is often sufficient for preliminary environmental reconstruction (Griffiths and Evans 1992). The generic nomenclature used here conforms to current European usage (e.g. Meisch *et al.* 1990; Griffiths and Evans, in press). The group *Candona s.l.* is taxonomically difficult, and although it is usually agreed that *Candona s.l.* is composed of several genera, reference must be made to specialist faunal works. Absolon (1978) illustrates the species of *Candona s.l.* known from the German Quaternary.

Valve key to the genera of Holocene and modern British freshwater ostracod taxa

In the key and figures the following abbreviations have been used: RV—right valve, LV—left valve, ext.—exterior, int.—interior.

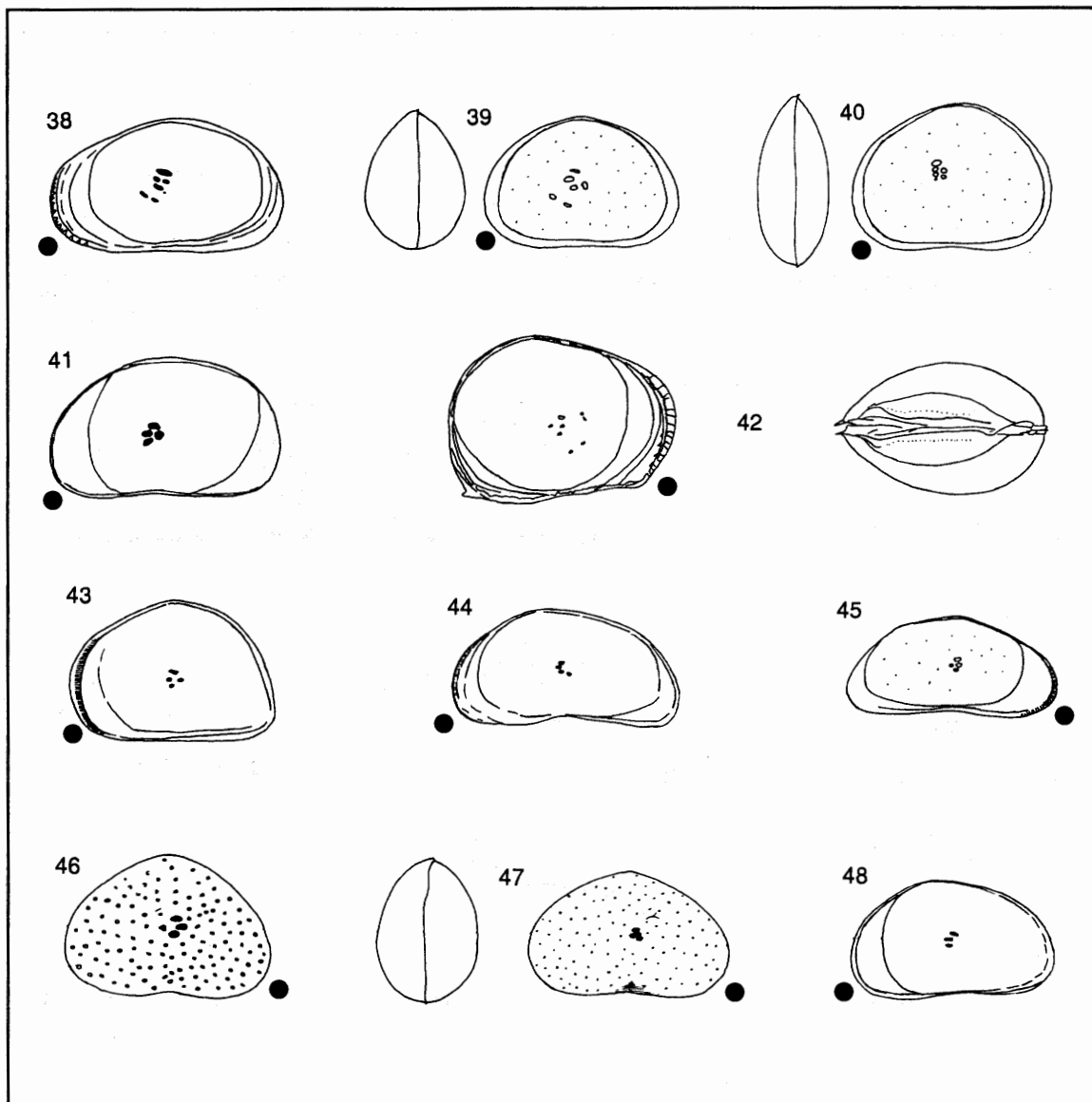
01. Valves swollen, elongate, narrower anteriorly than posteriorly. Muscle scars in a rosette with (usually) nine components (Figs. 26A, 27) *Darwinula*
— Valves and scars not like this 02
02. Muscle scars as in Fig. 26B; four scars arranged in a vertical row, often with a 'V-shaped' scar in front 03
— Muscle scar pattern of various types but never as above 06
03. Valves clavate, robust, often noded, but without reticulate surface ornamentation. Internal surface of valves with large, conspicuous pores (Fig. 28) *Cytherissa*
— Valves not as above, and with punctate or reticulate surface ornamentation 04
04. Valves swollen, especially posteriorly, sub-oval in lateral view, with regularly punctate external surface. Never beaked in dorsal view (Fig. 29) *Metacypris*
— Valves and carapaces not sub-oval or swollen. Males 'slipper-shaped', females sub-trapezoidal (Fig. 30). Surface with reticulate or punctate external ornamentation, sometimes also noded. Anterior end often beaked in dorsal view 05
05. Anterior pore canals short and straight (Fig. 30) *Limnocythere*
— Anterior pore canals long and branched (Fig. 31) *Paralimnocythere*
06. Valves >0.8 mm, conspicuously punctate. Lateral outline sub-rectangular (Fig. 32). Dorsal inward folds (sulci) present (Fig. 24). Valves plain, spined or with denticulate margins. No anterior rostrum *Ilyocypris*
— Valves without sulci, and otherwise not as above 07
07. Valves elongate with a 'Z-shaped' posterior inner lamella (Fig. 33) *Stenocyprina*
— Lacking this structure 08
08. Shell in lateral view sub-lanceolate (Fig. 34) *Dolerocypris*
— Valves not sub-lanceolate 09
09. Valves large (>1.8 mm), sub-rectangular (not sub-triangular) and with large, conspicuous muscle scars (Fig. 35) *Herpetocypris*
— Valves not as above 10
10. Valves flat, 1.1-1.4 mm long. Anterior inner lamella dorsally notched, very wide, and extending ventrally for the first two-thirds of the valve (Fig. 36) *Isocypris*
— Valves without these structures 11
11. Valves 2.0-2.6 mm long, globular, with very conspicuous selvage on anterior inner lamella. Either two large backwardly-pointing lateral spines, or tiny spines on the anterior margin, and a postero-ventral spur (most noticeable on RV; Fig. 37) *Cypris*
— Without conspicuous anterior selvage, or if present valves not large and globular, and lacking these external structures 12



Figures 27-37. Figure 27. ext. LV Darwinula; Figure 28. int. RV Cytherissa; Figure 29. ext. LV Metacypris; Figure 30. int. LV Limnocythere; Figure 31. ext. LV Paralimnocythere; Figure 32. ext. LV Ilyocypris; Figure 33. int. RV Stenocypris; Figure 34. int. RV Dolerocypris; Figure 35. int. RV Herpetocypris; Figure 36. int. RV Isocypris; Figure 37. ext. LV Cypris. ●—marks anterior end.

- 12. Valves >1.2 mm. Anterior inner edge of right valve with a row of tiny denticles (Fig. 38) *Heterocypris*
— These denticles not present 13
- 13. Muscle scars as in Figs. 26D or 26E .. 14
— Muscle scars not like this 16
- 14. Muscle scars resembling a 'paw print' of four or five elements with a rounded or slightly elongate scar above (Fig. 26D) .. 30
— Three slightly elongate scars anteriorly and

- in a vertical row, one smaller one below, and (usually) two smaller scars behind (Fig. 26E) 15
- 15. Valves swollen (Fig. 39) *Cyclocypris*
— Valves relatively flat (Fig. 40) *Cypria*
- 16. Dorsum curving gently downwards anteriorly, but posteriorly broadly rounded. Valve with wide anterior inner lamella. Not globose as in *Cyclocypris* (Fig. 41) ... *Scottia*
— Valves not like this 17



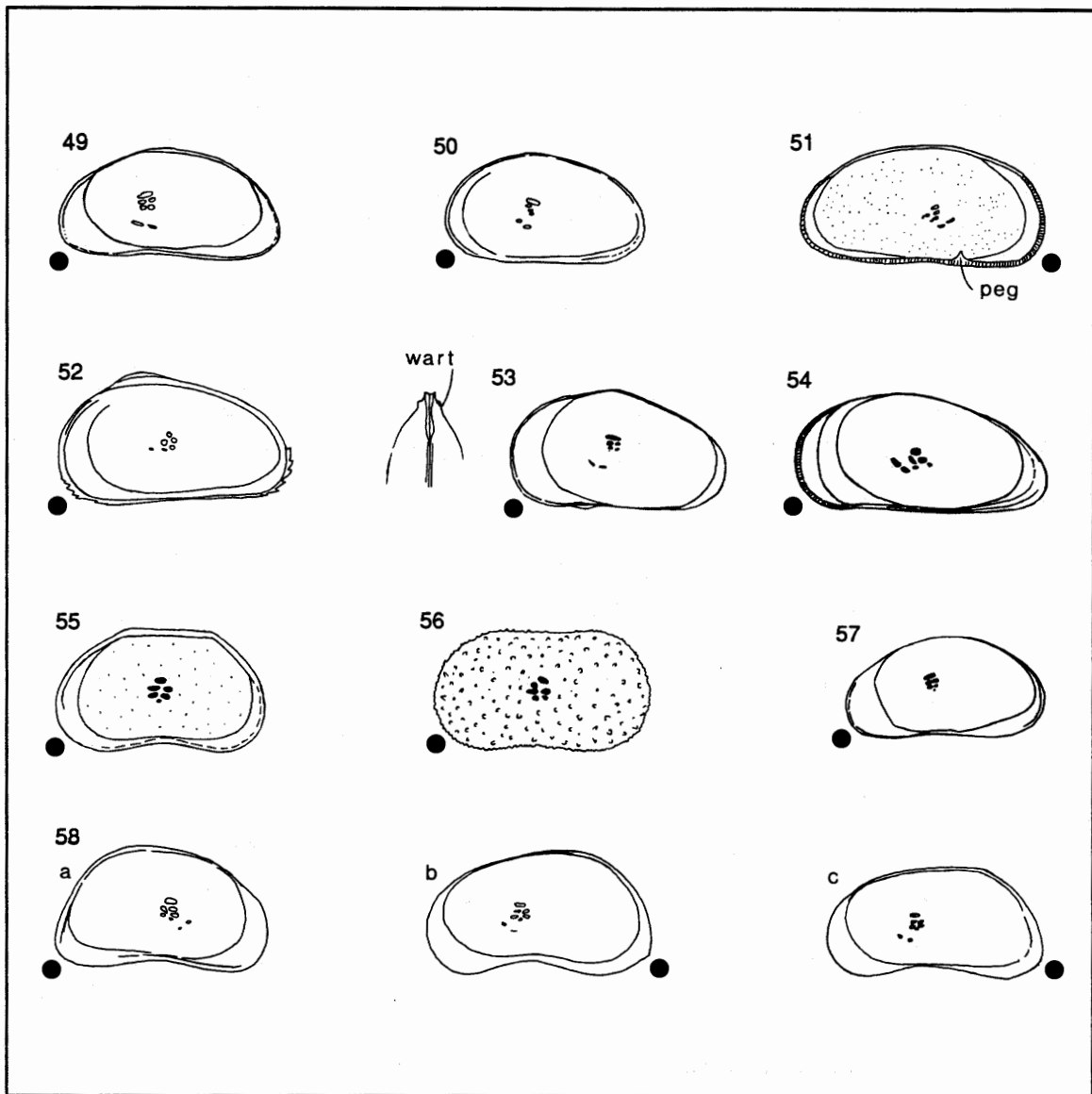
Figures 38-48. Figure 38. int RV Heterocypris; Figure 39. dorsum and int. RV Cyclocypris; Figure 40. dorsum and int. RV Cypria; Figure 41. int. RV Scottia; Figure 42. ventrum and int. LV Notodromas; Figure 43. int. RV Cyprois; Figure 44. int. RV Potamocypris; Figure 45. int. LV Cavernocypris; Figure 46. ext. RV Sarscypridopsis; Figure 47. dorsum and ext. RV Cypridopsis; Figure 48. int. RV Plesiocypridopsis. ●—marks anterior end.

17. Valves swollen, >1.0 mm, conspicuously ventrally flattened (Fig. 42) *Notodromas*
 — Valves not ventrally flattened 18

18. Valves with unusual, irregularly sub-ovoid outline, pore canals of anterior margin very conspicuous, in dorsal view valves not markedly swollen (Fig. 43) *Cyprois*
 — Valves not like this, and lacking highly conspicuous anterior pore canals 19

19. Valves <0.85 mm, muscle scars simple (Fig. 26F). Outline sub-ovoid or crescentic 20
 — Valves >0.95 mm. One large elongate scar above, scars below irregular and not tightly clustered 24

20. Valves crescentic, with curved or angled dorsal margin (Fig. 44) *Potamocypris*
 — Valves sub-ovoid or elongatedly sub-ovoid 21



Figures 49-58. Figure 49. int. RV *Psychrodromus*; Figure 50. int. RV *Strandesia* and *Bradleystrandesia*; Figure 51. int. LV *Tonnacypris*; Figure 52. int. RV *Prionocypris*; Figure 53. dorsum and int. RV *Eucypris*; Figure 54. int. RV *Trajancypris*; Figure 55. int. RV *Nannocandona*; Figure 56. ext. LV *Paracandona*; Figure 57. int. RV *Candonopsis*; Figure 58. (a) int. RV (b, c) int. LV *Candona* s.l. ●—marks anterior end.

21. Valves elongate, with a wide posterior inner lamella (Fig. 45) *Cavernocypris*
 — Valves sub-ovoid or sub-triangular, with a narrow posterior inner lamella 22
22. Valves heavily punctate, with a markedly peaked dorsal margin; not conspicuously swollen (Fig. 46) *Sarscypridopsis*
 — Valves smooth or punctate, not dorsally peaked 23

23. Valves sub-ovoid, very swollen in dorsal view, and with narrow anterior inner lamella (Fig. 47) *Cypridopsis*
 — Valves not distinctly swollen, elongatedly sub-ovoid, with a wide anterior inner lamella (Fig. 48) *Pleisciocypridopsis*
24. Valves robust, height/length ratio <0.5, elongatedly sub-ovoid with a wide anterior inner lamella (Fig. 49) *Psychrodromus*

- Valves sub-ovoid, or roundedly sub-triangular, height/length ratio usually >0.5 25
25. Valve surfaces finely punctate or tuberculate, but no other ornamentation of any type. Anterior inner lamella narrow 26
— Anterior inner lamella always wide. Valves smooth or sometimes with denticulate margins, or a row of small 'warts' on the anterior external surface 27
26. Valves sub-ovoid, exterior surface modestly pitted (Fig. 50) *Strandesia*
— Valves elongatedly sub-ovoid, exterior surface modestly tuberculate *Bradleystrandesia*
27. Anteroventral inner lamella of LV with a blunt peg (Fig. 51) *Tonnacypris*
— Peg absent 28
28. Ventral margins denticulate (Fig. 52) *Prionocypris*
— Denticles absent 29
29. Valves with a row of warts on the exterior anterior surface (Fig. 53) *Eucypris* s.s.
— Valves lacking these warts, shell outline sub-clavate (Fig. 54) *Trajancypris*
(If <1.1 mm, *Eucypris pigra*)
30. Valves punctate or tuberculate 31
— Valves without surface ornamentation 32
31. Shells <0.60 mm long, dorsal margin straight, ventral margin concave, slight antero-posterior asymmetry (Fig. 55) *Nannocandona*
— Adult shells c. 0.75 mm long, anterior and posterior margins equally rounded, the dorsal margin somewhat concave. Surface of valves with conspicuous tuberculate ornamentation (Fig. 56) *Paracandona*
32. Valves sub-reniform and with a wide, almost straight anterior inner lamella. Uppermost muscle scar markedly elongate (Fig. 57) *Candonopsis*
— Anterior inner lamella not like this, muscle scars arranged in a 'paw print' with the uppermost scar round or sub-ovoid. Most species 1.0-1.8 mm long, but sexually dimorphic and of a variety of shapes, although never sub-ovoid or markedly swollen in dorsal view. Always lacking surface ornamentation. (Fig. 58 a-c) . . . *Candona* s.l.

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