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Fluid Preservation Course
7th – 10th April 2008
Shirehall Study Centre, Norwich

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Course Summary

The Fluid Preservation course, or indeed Master class, given its scope and series of extensive workshop practicals, is an essential training experience for natural history curators, conservators and anyone with responsibility for fluid collections. Many a natural history collection can lay claim to a dark suppurating secret locked away at the back of an unsuitable cupboard! For this reason alone, this course is worthwhile hosting, given the success stories of our 'before and after' projects, which were practiced on and preened over for the duration of the four day course.

The level of expertise required for attendance can range from the novice to the experienced practitioner, as the course provides a 'top-up', if you will, to those who actively conserve, and a fully comprehensive introduction for those whose experience does not extend beyond topping-up or being alert to the warning signs of a collection in need of conservation.

The format of the course provides the opportunity to put the lecture series into practice by supervised workshop sessions. The material provided by the Norwich Castle Museum, Natural History Department gave plenty of scope for encountering the many problems of caring for fluid collections. This provided a far-reaching application to the theoretical.

Lectures followed by question and answer sessions were interspersed by lab workshops where each attendee could choose a number of projects to work on throughout the week. Given that many of the conservation measures needed for fluid preservation are time consuming, the four-day duration was time enough to see a relatively long-term project to fruition, and in most cases with excellent results.

Fluid preservation is not just simple 'topping-up'; neither is it simply reapplying sealant to leaky jars. It is the application of chemistry for the long-term preservation of biological material and the practical measures required to store such media effectively. Firstly you must understand the histological and historical context of fluid preservation (many preserved specimens are ancient!) to calculate their worth: do they come with documentation and scientific data, is the specimen of scientific and/or of historic value, to what extent has the specimen deteriorated. In effect: to conserve or not to conserve?

Knowledge of the practical reparative methods is essential as it can be the case that poorly sealed or unsuitable jars are the culprit for escaping vapours and fluids, and chemical reactions. Therefore, knowledge of the materials available is essential. Learning the brave skill of glass cutting, drilling and grinding is invaluable when replacing cracked lids or even old Bakelite lids, as is the sealant process.

Mounting specimens within jars can be a fiddly process, especially with such groups as the Coelenterates (sea anemones, jellyfish and hydroids). Methods of attachment along with the appropriate type of thread, adhesive, backing plate or suspension material were demonstrated and made available. The first experiment involved using Celloidin glue to attach snail shells to glass and then immersing in 70% industrial methylated spirit (IMS). This proved to be an ongoing obsession for the group (whose shell would be the first to bob to the surface with shameful failure!)

Fig 1. Beulah Garner with jellyfish in hand.

Assessing what fluid a specimen has been formerly preserved in is essential (in the absence of any preservation data) both to the safety of the conservator and to the future preservation and maintenance of the specimen. Various methods were employed and tested, including making our own floatation devise: an effective 'Heath Robinson' alternative to gravity detectors. From this action can be taken as to what preservative the specimen should then be stored in and the appropriate labelling employed – this is another subject in itself.

Each of our practical projects was supervised step-by-step. Most of the specimens required re-hydrating, which can be a lengthy process and involves the use of TEEPOL or Decon 90, either in cold preparation, a heat bath or local injection of fluid. The next step is to 'fix' the specimen with formalin, even if it has been fixed previously. If specimens were to be stored in IMS then a 'ladder' process of reintroduction of the specimen into various dilutes of IMS was followed to minimise osmotic shock. Any chemical must be assessed for its compatibility to the specimen and to the storage medium, for example, if a backing plate is made of plastic then IMS is not suitable. Various other tests can be carried out post immersion. There may be some discolouration to the preservative, which could be pigment or lipid leaching from the specimen. Lipid leaching can lead to deterioration of the specimen, and so pH must be taken to ascertain contamination levels (Fig 2).

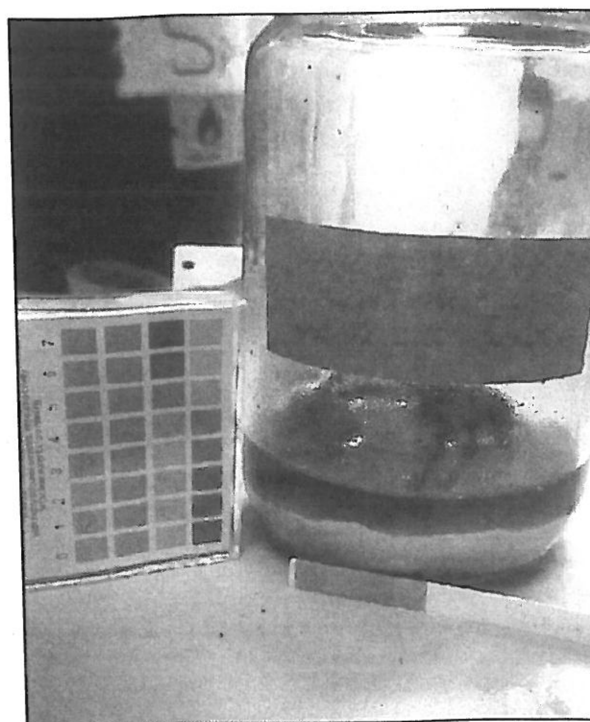

Fig 2. Testing the pH of preservation fluid.

Each specimen is unique and requires an individual assessment and resulting treatment. It is not guesswork, it is a lengthy process. It is however, incredibly worthwhile given the results we achieved as a group under Simon Moore's excellent instruction over the week.

The outcomes of the course are the acquisition of a number of new skills and recommendations for best practice. It is important for natural historians/curators to have an understanding of the time involved in assessing and conserving fluid collections, enabling them to work effectively with in-house departments and to schedule an efficient programme of maintenance. A heightened awareness of the financial and time constraints of ensuring a dedicated programme of future conservation practice is beneficial to all hierarchies within the museum environment. The methods available to the natural science practitioner for trouble-shooting, problem solving, reparative and pre-emptive work, resulting from this course, should ensure a high standard of collection care for the future of your fluid collections.

Monday 7th April 2008

After an unscheduled fire alarm (we like to keep people on their toes!), the course started off with a general talk about fluid preservation and its history. Simon Moore (himself well-preserved, despite [or perhaps because] of the amount of noxious chemicals he's dealt with over the years!) described fluid preservation as 'an open field', where the technology is constantly being pushed forward. However, much of the basics of fluid preservation are still the same as they were 100 years ago.

Biological specimens were historically stored in glass 'battery' jars, with a polished side for viewing through. Today the problem is that a lot fewer people know how to look after such specimens. Another common problem is that many spirit collections are in a variety of jars and containers, fluids and lids. Specimens often get dried out because the seals are not terribly good. Fungal decay can even occur if the industrial methylated spirit (IMS) is under 30% or formalin (40% saturated formaldehyde gas in water) is under 2%. Normal fixing strength for formalin is 10%. Formalin is aqueous, so it would take a very warm store for it to evaporate away. Glycerol can sometimes be used in order to save a collection from drying out.

Dried-out specimens-
To re-hydrate or leave? That is the question! Arthropods in particular don't re-hydrate well. But it is advisable to re-hydrate soft specimens. It is recommended that DNA samples be taken before re-hydration, as the process can 'scramble' the DNA (Fig 3).

Re-hydration fluid can be used (TEEPOL or Decon 90), heated up to about 40°C with a top on it so that it doesn't evaporate. After re-hydration, re-fixing must occur with an internal fixative (e.g. injecting formalin). Air bubbles can be removed from a specimen by placing it in water and using a mild vacuum pump – this stops the specimen from floating (Fig 4).

You can tell much older jars from newer because the older have 'pontil' scars on the bottom (where they were hand-blown). Newer jars tend to have been ground flat.

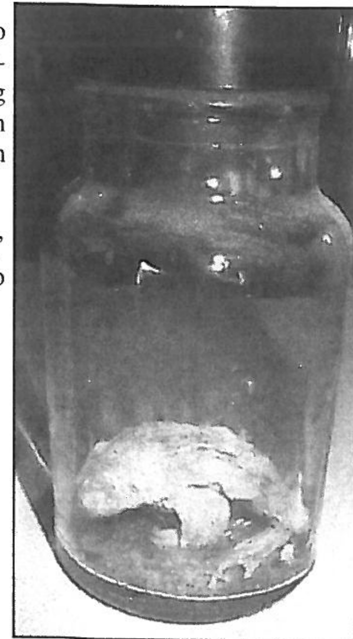

Fig 3. Dried out frog specimen.

Fig 4. The rehydration process in action.

Other problems-
Temperature fluctuation problems arise when formalin is used as a preservative (rather than a fixative). Formalin can solidify when the temperature fluctuates between 5°C to 25°C. Ideally formalin preserved specimens should be kept at a constant temperature of between 10°C and 15°C.

These solids can be dissolved away by putting the specimen in IMS. Crystals in the solids may have already broken the specimen, so glass needles can be used to fix the specimen. Glue called celloidin (a solution of

pyroxylin in alcohol) is also used to cement the specimens in IMS to glass. Gelatin is celloidin's equivalent when specimens are preserved in formalin.

Formalin is still used as a fixative today, despite its carcinogenic properties! Other alternatives to formalin have been sought out over the years, but formalin is still generally considered the most effective. Formalin essentially works by coagulating proteins into a stable condition. Fresh specimens can produce cloudy suspensions of insoluble salts in formalin – especially when incorrectly buffered. De-ionized water, plus a phosphate buffer will prevent this from happening (the pH should be about 7).

Dilute IMS is best made up to 24 hours in advance as tiny bubbles form in it when added to water. Many specimens change colour when stored in IMS but not in formalin.

Tuesday 8th April 2008

Narcotization, historical sealants and pelagic mounting-

Certain animals close up or retract, so they need to be anaesthetised before fixing. Sea anemones have a two-stage narcotizing barrier. Stage one: they respond to physical stimuli, and, Stage two: they respond to chemical stimuli. Fixation must occur before autolysis (self-digestion) occurs. They also open and close according to the tide (circadian rhythms). Powdered menthol is often used in narcotization.

Historical sealants-

When working on 18th and 19th Century jars you may be required to use the same sealant as was originally used. Recognition is essential in these cases. Greases are vital on ground glass jars, but some silicone-based greases may solidify in time. Simon prefers 'paraffin soft white', which doesn't seem to solidify. To make an effective seal, the jar top must be ground down flat (this also gives a key for the sealant to work on). If the seal is yellowish and flaking, it is probably gelatine. Gelatine seals that look white in colour are no good (they are unstable). Silicone seal has a rubbery feel to it, and can be very tricky to remove if it was properly put on in the first place. However, you can buy silicone-reversing agent these days.

Pelagic mounting-

Animals that swim or float should not be destined to sit in a heap at the bottom of a jar – especially if they are going on display. They need to be correctly mounted on discs of acetate sheet, suspended by monofilament. Older jars tend to have suspensory glass loops or knobs under the lid. Glass floats (a bit like Christmas tree baubles) can also be used. Larger, more solid specimens such as vertebrates can be mounted on a back-plate (Fig 5).

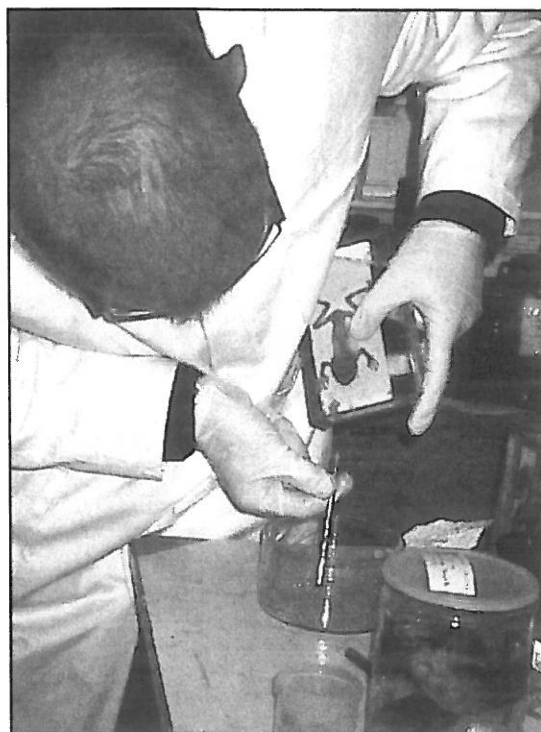

Fig 5. Neil Mahrer with frog transparency (neil is the one on the left).

Sometimes copper wire was used to hang specimens. This will stain the fluid blue/green and the specimens green (the fluid can be replaced, but the staining to the specimen is irreversible). Glass rods or cradles are good for holding specimens in place as glass is inert and can be easily heated and bent to suit the needs of the specimen.

Wednesday 9th April 2008

Preservatives-

Preservative fluids may suit a variety of different taxa. Opresol has been found to be effective for many invertebrate groups as a PFP (post-fixation preservative). However, as with most newer preservatives, the

long-term preserving stability has yet to stand the test of time. Other preservatives can react in the long-term with plastic jars (crazing them). Alcoholic preservatives will soften plastics with time. If in doubt – use glass jars. Also, some preservatives are not capable of maintaining preservation for densely muscled specimens.

Absolute alcohol (99%) is 74 over proof. Dowicil (made in the 1960s) reacted with protein to produce formaldehyde. Its powdered form was easy to carry, but some people got confused about amounts and strengths, and it didn't work – as a result it is no longer made!

A tri-part colour preservation technique was developed in the 1920s. Despite keeping the specimen colour quite well, unfortunately it does quickly yellow the fluid.

Transparencies are stored in a variety of fluids including glycerol.

Contaminants-

This occurs due to specimen reaction, seepage and things like copper wire, etc. Yellow alcohol can be cleaned with charcoal and filtering, but heavily lipid contaminated fluid will remain in solution. It is much easier and less time consuming just to change the alcohol. Latex-based injection dyes eventually break down and stain the specimen plus the fluid pink.

Fungi start to appear in 30% IMS. The following can adversely affect/completely alter DNA: fungal contamination, re-hydration, formalin (when used as a preservative rather than a fixative), and ethyl acetate. Black fungal staining of specimens is irreversible. It is very important to record the fixative and preservative on the label.

Thursday 10th April 2008

Transparencies, Containers, labels and tubes in jars-

Once prepared, transparencies should require only minimum attention since their preservatives are viscous and dense fluids. They are stained with calcium specific dyes to show skeletal elements. This method is useful for showing unusual bone deformities.

When diluting dense fluids such as glycerol with water, you will experience mixing lines called 'Schleren Optics' (you also get this when ice melts into whisky – although I was scolded for using this analogy as apparently one should always drink Scotch neat!).

Benzoate transparencies can have problems with crystallization – the specimens should be soaked in absolute alcohol (no water) in order to clear this.

Older 'Copenhagen' jar lids tend to degenerate in time or become too rigid. However, they are easy to use and good in the short-term (up to 20 years or so).

Ground glass jars by Dixon, etc. (Fig 6) are very expensive – but they are the best. Le Parfait-type jars are good, but you need to replace the rubber gasket often (which is a lot of work, especially in a large collection).

Screw-on lids are preferred in North America. Newer plastic lids should outlive the old Bakelite lids. Bakelite tends to be slightly patterned, which helps in telling the difference between modern plastic and old lids.

Visijars (plastic jars) have a finite lifespan (as they become crazed after a while). Also they sometimes reflect the light more strongly than glass, which can obscure the specimens on display.

Labels (and what to do with them!)-

Ensure fastness of ink and robustness of paper. Exterior labels should be for display purposes only. Vital data should always be inside the jar. Old, historic or frail labels should be stored in an archive, and the data from them transferred to a new label. The problem with external labels is that they detach themselves over time, they can obscure the level of the fluid in the jar, and they can become stained and wrinkled. Glycol-based preservatives make labels transparent.

Internal labels are best – 'goatskin' parchment (which isn't real goatskin) is best (Wiggins Teape Paper Ltd.). Handwrite these labels using India ink. A diamond scriber can also be used to write on the glass what preservatives were used, etc.

Tubes in jars-

Tubes in jars are used for small things (especially arthropods). Simon prefers them upside-down to minimise the risk of their drying out if the jar lid becomes compromised or the shelf is too deep to see the jar. Others prefer the tubes with the lid on top to stop specimens from coming out of the tubes and therefore detached from their labels.

Pith or cork are no good for stoppers as they stain the preservatives and eventually crumble away. Rubber quickly perishes and stains the fluid. Nylon stoppers are probably the best as cotton wool gets trapped in antennae and legs. Pierce the nylon stopper with a fine needle to stop pressure from inside the tube pushing them off.

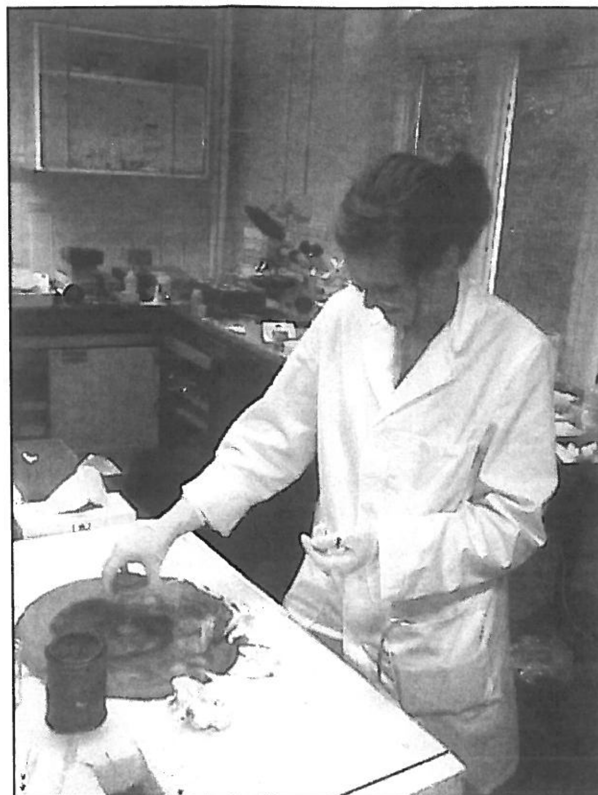

Fig 6. Melissa Gunter grinding a glass jar rim.

Storage areas-

Wooden shelves are good as spills evaporate off them easily. Metal shelves tend to rust very easily. Good ventilation is essential. A list of the fluids used in the store to be kept near the entrance is also a good idea. Very tall, narrow jars need additional support of struts and bars.

Posting fluid preserved specimens-

Formalin specimens – use tissue or wadding soaked in de-ionised water (with no actual fluid to slosh around). Put the wrapped specimen in a poly-bag, then heat-seal it, then pack it for the post.

For alcohol specimens – use tissue/wadding soaked in IMS and then the same procedure as above.

Another method might be to use either agar or tert-Butanol, which solidifies around the specimen below 20°C.

Some foreign museums send specimens back in osmotically different fluids because of laws in their country. Find out if this is the case before sending specimens and either come to a compromise or don't send them the specimen!

Comments

"I found the course very interesting and extremely instructive, given I didn't know much about museum conservation in general. I only wish I had dealt with more entomological specimens. This could be something to consider for the next courses; perhaps participants could give a general list of the groups of specimens they will find useful to work on during the practice."
Alessandro Giusti
Assistant Curator (Lepidoptera), Natural History Museum

"General thoughts on the course were very positive. There were inevitably periods of waiting around during the practical work but I don't see how that could be avoided so no beef there. The facilities were tight but never actually led to annoying queuing, so again no problem."
Neil Mahrer
Conservator, Jersey Heritage

"Overall I thought it went well, technical side - mounting specimens and preparing containers particularly useful has haven't done much of that before."
Tony Parker
Assistant Curator (Vertebrates), National Museums Liverpool

"Name badges! More demonstration from Simon, like case studies and examples of various practices, so we could then take our specimens and follow his lead. It was a really good week. Resources were excellent."
Beulah Garner
Trainee Natural History Curator, Horniman Museum and Gardens

"From my point of view and coming from a totally different background/discipline I can honestly say that I found it most informative and extremely interesting. The main reason for my being there ... is the Damien Hirst work that has recently come into our collection, but as the course unfolded it was clear that there were other works in our collection that the principles and techniques would apply to which was a real bonus. I suppose you could say in hindsight that participants do not have to be from a museum 'wet specimen' background; which would open it up to other interested institutes/individuals."
Keith Morrison
National Galleries of Scotland

"I much enjoyed the course and found it extremely useful. Regarding the organisation; I liked the proximity of the lab to the room where we had the talks and the mixture of talks and practical work and the way we could pop through easily to check up on specimen progress."
Cathy Caudwell
College of Life Sciences, University of Dundee

"I thoroughly enjoyed the course and am really glad I got to attend. Having had no background knowledge of fluid preservation before I attended, I came away more knowledgeable and with the skills and confidence to apply these course techniques to some of our specimens. I particularly enjoyed the lab based practical where we could work through the problem specimens and a real highlight was realising that dried-out or neglected specimens could be re-hydrated and restored to good quality. The only suggestion I have is possibly to allow everyone to acquire a problem specimen that needs to be mounted. Only so everyone can have a go at mounting with the microfilament or using celloidin to repair it etc. but apart from that it was great."
Amy Romanes
Natural Sciences Curatorial Assistant, National Museums Scotland

"I thought the course was very good and run very well. Simon's papers, while extremely helpful and full of useful information, in no way compared with the hands-on experience and in-person instruction you receive during the course. As we all found, each specimen is completely different with separate and unique issues to assess and deal with. It was a good course and I was glad I was able to attend."
Melissa Gunter
Conservation Intern, Museum Victoria, Melbourne, Australia

Norwich Fluid Preservation Course 2008. Top row (from left to right): Melanie Rolfe, Cathy Caudwell, Amy Romanes, Keith Morrison, Melissa Gunter, Tony Parker, Beulah Garner. Bottom row (from left to right): Neil Mahrer, David Waterhouse, Alessandro Giusti. (Photography by Simon Moore)