Biology Curators Group Newsletter

Title: Liquid Preservation - How Little We Know

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3 Filling the jars with water - even more disastrous for the specimens.

Alternatively they can use:

4 Leuco-basic fuchsin impregnated papers which go pink with formalin and other aldehydes (including curators' hands) which may be fine but can be messy, time consuming and the curator is still inhaling fumes from the discarded papers.

5 Use an LCD readout specific gravity meter - a small amount fluid is sucked into the meter using a rubber bulb and a precise readout of the fluid's specific gravity is obtained - fine for alcohols but it will not distinguish between low grade alcohols and formalin; the meter is expensive and slow to use.

6 The Simon Moore method (below). Although this also does not distinguish between low-grade alcohols (of which there should be none in your collection!) and formalin it has the advantage of being much faster, cheaper (home-made), much safer (no sniffing) and it's accurate!!

You will need: a dropping bottle with reservoir and mapping pins of assorted colours with heads small enough to fit into your dropping bottle reservoir.

1 Make up a range of those preservative solutions for which you will be testing.

2 Remove heads of red, yellow and blue pins using pliers (these colours are not obligatory!).

3 Test flotation of pin heads in solutions and replace pins (point first) into pin heads to weight them.

4 Trim off pins to various lengths so that some will float, some will sink in the various solutions: eg yellow has no pin, red has half a pin, blue has pin right through.

5 When each pin has been trimmed to correct weight, push the remainder of the pin into the head.

6 Put weighted pin heads into bottle's reservoir.

7 Test - suck up fluid into reservoir, give a shake to get rid of any adherent air bubbles, note the distribution of floaters and sinkers:

8 Yellow will float below 55% alcohol and in 10% formalin, it sinks in 60-80% alcohol; red will float in 30% alcohol and 10% formalin, it sinks in 50% and 70% alcohol; blue floats only in formol-glycerin.

9 A simple method of just distinguishing between 70% alcohol and formalin will only require one red ball - floats in formalin, sinks in alcohol (if strength greater than 55%).

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A SHORT NOTE ON PRESERVATIVES THE IDENTIFICATION PROBLEM - A POSSIBLE SOLUTION

During the course of a one year, externally-funded conservation project in the Hunterian Museum (Zoology Section) in Glasgow, work was undertaken to address a backlog relating to various parts of the collections, including the wet material. Some 2,000 jars were dealt with in the available time.

The main problem encountered in this project with regard to preservative was one of identification. Many curators rely on smell, but this was obviously not to be recommended where some of the jars contain formalin or unidentified, possibly toxic, fixatives or preservatives. There is a published method using a strip test to distinguish between formalin and alcohol, but it was found to be difficult, time consuming and expensive to make the strips up. The method used in this project to tell preservatives apart was more or less discovered by chance while labelling the jars.

It was found that a strip of Goatskin Parchment label (8mm x 20mm), when dropped flat on the surface of preservative behaved in different ways:

* Alcohol (down to about 30%) will soak through the paper immediately and the label sinks after a short time.
* Formalin (even at low concentration) repels the paper and the label will float on the meniscus for a long time, sometimes curling up at the edges.
* Phenoxetol is neutral, being mostly water, and the label sits flat on the surface for a time until the fluid slowly soaks through.

Although this method has not been rigorously tested, it never failed in use, and sometimes identified alcohol when the nose could not. It also identified the common situation where the preservative is mostly alcohol but with a little formalin residue from the fixing process (this mixture frequently fools the strip test method). With practice, it was also possible to recognise some other preservative types, eg alcohol with glycerine. At the very least, the method readily identified formalin which the nose should never be allowed near!

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LIQUID PRESERVATION - HOW LITTLE WE KNOW

There is a wealth of information in specialist books and journals on the liquid preservation of biological material, but very little of this concerns plants. Following the reorganisation of the science departments at the Natural History Museum in 1990 a newly-formed Curation Programme undertook the task of monitoring and improving methods of specimen conservation. In my role as Curator of Algae I had to decide the fate of the largest liquid-preserved collection in the department as well as manage other disparate holdings, such as pressed herbarium specimens,

microscope slides and rocks housed in packets and boxes.

Recently, I began the search for published information on fixatives and preservatives specifically tailored to suit a range of botanical material. To my surprise I discovered that very little research has been carried out on the liquid preservation of plants and that the long-term effects of preservatives on gross and fine structure are not well documented.

Why liquid-preserve in the first place? Some plant groups, such as succulents, do not lend themselves readily to the squashing and drying which is used for the preparation of traditional herbarium specimens. Similarly, the flowers and fruits of some groups, such as orchids, are difficult to dissect out when dried and, therefore, have been preserved routinely in alcohol.

Illustrators prefer to draw plants fresh or liquid-preserved, rather than dried, and where an accurate measurement is essential it is easier to obtain dimensions from wet-stored material than from specimens glued flat to a herbarium sheet. Micro-algae and diatoms are usually liquid-preserved in the field and the samples taken back to the laboratory for processing onto microscope slides and identification, unless the researcher has the opportunity to examine the fresh material immediately after collection.

It is likely that most curators of higher plant wet stacks use 60% or 70% industrial methylated spirit (IMS, aka alcohol) as preservative, whilst algal material will be in 3-5% formaldehyde (8-10% formalin). Health and safety issues have highlighted the possible dangers of exposure to formalin and this, combined with its unpleasant smell, has encouraged many curators to transfer their holdings to alcohol. This may be better for the safe handling of material but what of the effects on the specimens? The collection has to be the important issue here, not the convenience of the curator. It is quite possible to plan a facility and practice for the safe handling of formalin-preserved material which will also satisfy Health and Safety requirements. In your wet stacks you could be using alcohol, formalin, the two combined or mixed in various proportions with other substances.

What to use for the successful, long-term preservation of a wide range of botanical material should be the objective of the conservator or curator. Should different materials be preserved in fluids specially devised to suit their particular requirements or will one universal preservative suit all? To change the fluid, or, if the collection has been neglected, to leave it dry or top it up? Do we know what actual mix and strength of preservative was used for an old, inherited collection? All these questions are of vital importance but when we look for answers we find a deplorable lack of hard facts.

For a range of, apparently, tried and tested fluids for fixing and preserving biological material there are some published accounts, eg Horie (1989); Wagstaffe & Fidler (1955 and 1968); and for botanical material only, Bridson & Forman (1992). Unfortunately most recipes are not accompanied by a reference to any previous experimentation or laboratory testing, and are handed down like "tablets of stone". Most of the assessments of long-term effect are based on zoological and pathological material and are no real basis for use with botanical specimens, where the tissues are so different.

This article seeks to promote an awareness of the problem amongst curators and asks them to search out references, unpublished data or any information that will improve the maintenance of botanical wet stacks. If, from your own or a colleague's experience, you know of date-lined, documented collections I would be grateful for data on their condition so that I can start a do's and don'ts of botanical liquid preservation for publication at a later date.

Meanwhile, a few "in-house" experiments are planned which will monitor the effects of different preservatives, methods and storage conditions in both the long and short term.

One positive contribution to our knowledge of fluids in the preservation of botanical material is that of Page (1979). He carried out experiments on conifer specimens hoping to find a method that would prepare them for eventual dry or herbarium storage. His chemical pre-treatment involved the use of ethyl alcohol and glycerol but immersion was temporary and the material was not stored wet.

There are other treatments where fixation and preservation are adapted to the specific needs of electron microscopy or cytology and these topics are not covered here. Finally, the question of appropriate containers and storage is almost as important as the preservative used and there is still no real solution for the large, older collections housed in less than perfect conditions. The resources needed to re-bottle and re-house these specimens are enormous and the final decision should be the right one based on adequate research. Here at the Natural History Museum a long-term study of containers has persuaded the Zoology Department to abandon all but ground-glass stoppered jars for alcohol-based storage. However, in the Botany Department we have had good results with the use of the so-called "Copenhagen" or "Danish" museum jars supplied by Grathwol. Our formalin-based preservatives does little damage to the plastic snap-top lids, unlike the alcohol used in zoology, which makes the plastic brittle and liable to split.

Comments on, or answers to, the questions raised above are sought by the author who will endeavour to make the findings available to all those who are interested.

In conclusion

Both published and unpublished data, including casual observation, on botanical material in liquid preservation needs to be collated and evaluated.

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