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Adhesives for fluid-preserved specimens

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Abstract
Fluid preserved specimens, whether stored in formalin or alcohol-based preservatives, often require adhesion to backing plates, especially if used for display or education. Techniques for adhering delicate specimens (or parts of), labels and flags, also applications as gap-fills are mentioned together with reversibility and health and safety issues.

Introduction
Most of us have to store, maintain and sometimes display dissections or mounts of whole biological specimens. When internal labels or bits of specimen become detached through physical abuse, many tend to hide these specimens away where they become forgotten and/or neglected.
As Art Museums and Galleries also start to acquire fluid-preserved biological specimens their untrained staff start to panic, unsurprisingly, when such works of art start to deteriorate over time.

Adhesives
Most of us tend to store biological specimens in 70-80% Industrial Methylated Spirit (IMS), having hopefully fixed the fresh specimens in 10% formalin to stabilise proteins, amino acids and other related tissues.

For alcohol preservatives, celloidin must be used as an adhesive, for formalin-preserved material, prepared gelatine is used. Celloidin goes under many different names:

- **Necoloidine** – an 8% solution of pyroxylin (dry nitrocellulose flakes which are moistened with IMS to prevent instability (explosive/combustion) problems). This gels in 70-80% alcohol to produce a hard, semi-opaque solid which has an adhesion strength of about 100g for a 0.5mm layer increasing to c. 5Kg for a 2cm thickness preserved in IMS. It is/was used as a larger specimen embedding medium for microtomy.

- **Pyroxylin** – alcohol-moistened flakes of nitrocellulose. These are dissolved in a compound solvent - a 50:50 mixture of absolute ethanol and di-ethyl ether to produce Necoloidine.

- **Pro-celloidin/Collodion** – is a more European term for pyroxylin powder. Again it is alcohol-moistened and tends to flock together forming easier to handle soft lumps.

For formalin-preserved specimens you must use prepared gelatine and which also acts as a reversible (warm water soluble) jar sealant. This adhesive is prepared from leaf gelatine only so that the molecular strands are long enough to form good adhesion. Powdered gelatine may be fine for foodstuffs but not for adhesion:

- The gelatine leaves (11g for a small sample of adhesive) are hydrated in cold water (hot will melt them!) until very limp but not starting to dissolve. This varies from about 2 minutes to overnight depending on leaf thickness.

- Once soft, the leaves are heated, using a hot-water bath or *bain-marie* to prevent charring, until molten – do not overheat or this will break down the molecular strands of the gelatine.

- 2.5ml of glycerol are mixed into the warm mixture followed by 1.5ml of glacial acetic acid (using a fume extractor!).

- The mix is poured out onto a glass sheet or tray and once cool, it solidifies into a soft, rubbery and slightly sticky consistency.

- It shored be stored in an air-tight container.

- The glycerol prevents drying out and the glacial acetic acid acts both as a fungicide and a glass-binding agent (essential when used as a battery jar sealant).

    - Leaf gelatine – 11g
    - Glycerol – 2.5ml
    - Glacial acetic acid – 1.5ml

Techniques

IMS-preservation:
This will only work for specimens store in IMS in 80% strength or below to 60% (lower percentages will cause gradual specimen deterioration). Absolute IMS will dissolve celloidin, the water content of the preservative grade brings about the gelling and subsequent tack strength (Figs 1 and 2):

- Remove parts of specimen to be joined from the IMS preservative.

- Once touch dry or even slightly moist, pipette some solvent (ether-alcohol mix, see above) onto the surfaces to be joined.

- Apply celloidin or pipette around the base of a specimen (or label) to be mounted and join surfaces together. Support the joined surfaces so that they don't break apart.

- After 5 minutes, the celloidin will stop flowing if tilted, and will ripple. After a further 5 minutes, immerse into preservative strength IMS to gel (about 10 minutes to half and hour. For larger amounts it can take overnight).

- Reversibility can be brought about by applying solvent or peeling off the gelatinous layer but if this might compromise the specimen, always use the solvent.

- Bear in mind that these substances are highly flammable and that the ether (solvent grade) can still cause drowsiness.

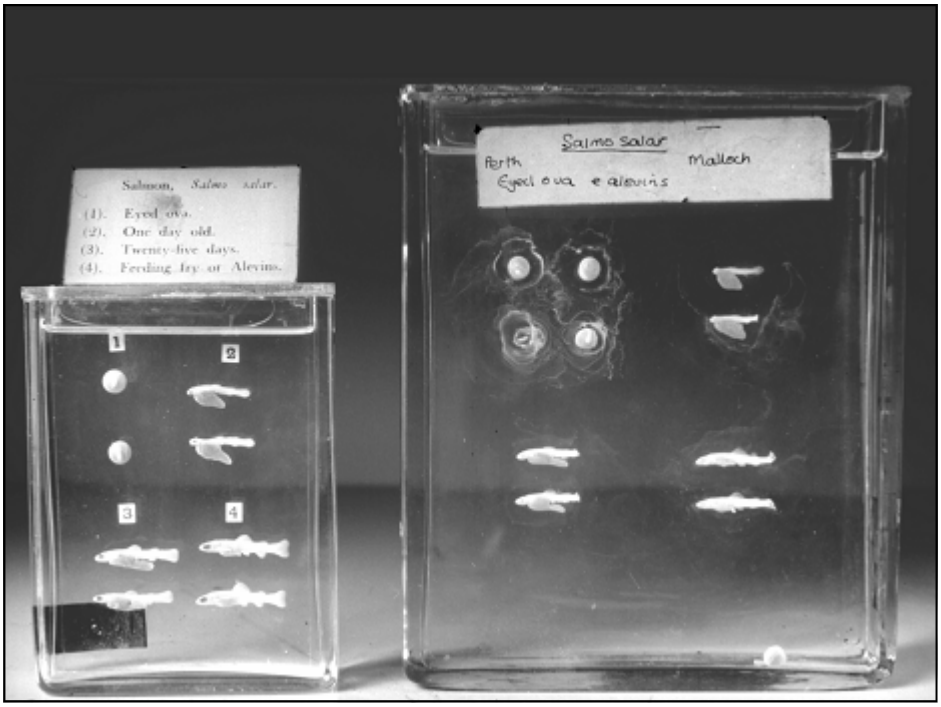

Fig 1. Celloidin mounting of salmon ova and alevins – the right-hand set of ova was breathed upon during gelling, hence the opacity and the dropped ovum.

Fig 2. Re-attaching a fallen tentacle from a jellyfish is straightforward enough; gap-filling a tear to the bell using a watch-glass to emulate natural curvature, is slightly more challenging.

Formalin preservation:
This is also used for attaching specimens in plastic (perspex) jars or 'visijars', bearing in mind that alcohol will severely soften plastics!

- Melt the gelatine in a hot-water jacketed beaker – 2 beakers of slightly different volume are fine.
- Apply to both (touch dry) specimen surfaces and join before it cools.
- Once cool, submerge in formalin – it will gel.
- Try not to use too much gelatine at a time, if the beaker contents cool and require re-heating, the molecular strands of the gelatine start to break up and will result in adhesion loss.
- Bear in mind that this join will only last for 30-40 years. Hydrolysis from the water in the preservative will slowly undo the gelatine molecular fibres.

Fig 3. For fallen limbs of arthropods, needles of glass form a useful and inert armature to support enough weight for either adhesive to remain effective.

Conclusions

These adhesives have been tested over a 40 year period and although the gradual failure of gelatine has been noted over that period, the celloidin technique seems to last even longer.

Further Reading

Moore, S J. 1980. Problems with Museum Jars solved. *Biology Curators' Group Newsletter* 2 (9): 382-389.

Moore S J. 1999 Fluid Preservation. In *Care & Conservation of Natural History Collections* (eds Carter & Walker). Butterworth-Heinemann, London.