Wax On, Wax Off:
A Guide to Fossil Vertebrate Micropreparation

Scott K. Madsen
Utah Geological Survey
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Cover Illustration

View of an in-prep *Sphenodont* skull from the Jurassic age Morrison Formation, Utah; the skull is slightly larger than a thumbnail.

INTRODUCTION

In 1996, Rich Cifelli edited "Techniques for Recovery and Preparation of Microvertebrate Fossils" (see References) which included a section I wrote called "Some Techniques and Procedures for Microvertebrate Preparation" dealing with manual preparation. That paper contains a lot of "how to" detail regarding specific materials, techniques, and sources for equipment and materials that are still relevant. However, over ten years later, I am still frequently asked for advice on buying tools for microprep, how to set up a work station, and what techniques to use. This happens often enough that I decided it's time for an update. This Guide is an introduction to microprep that emphasizes what I consider to be the most important components of microvertebrate preparation. Most of my suggestions are presented more or less in the sequence that I follow when I approach a project. Please refer to Madsen's (1996a, b) papers for more specific techniques and applications.

This Guide is aimed mostly at beginners who are already comfortable with the basics of preparation. If you have spent time using non-pneumatic hand tools, so much the better because Micro-jacks and the like are seldom a part of microprep. Microprep is about adjusting and adapting to a different scale; think small, *very* small. Also, the subject of microprep inevitably leads to more philosophical musings about preparation, conservation, and other non-technical aspects of the work. These things are just as important as technical skill with a needle, so, if you're new to this field, please bear with me. Beginners take note: my recipes for success are by no means the only way and I encourage you to seek out as many alternative sources of information and techniques as possible; some are referenced in the text and others follow in References and Resources.

WHAT IS MICROPREP?

Micropreparation (herein referred to as "microprep") is any manual preparation of vertebrate fossils on a scale that requires the use of a microscope. Perhaps it is by simple virtue of the scale that microprep can be so compelling and impressive to the uninitiated. This is especially true of those not familiar with the concept that the tiniest of creatures coexisted with the largest dinosaurs on the planet. During my 20 year tenure at Dinosaur

National Monument, I "performed" live microprep through the use of a micro-video camera attached to my Nikon SMZ-U microscope while I worked on various Mesozoic mammals, frogs, lizards, and other tiny creatures. One of my favorite tricks was to focus on the tip of a single denticle of a baby theropod tooth and zoom back until the public could see the whole tooth mounted on a pin head. At first a single denticle would fill the whole monitor, a clear but incomprehensible image, and as the rest of the tooth slowly appeared people would recognize what they were seeing and gasp in astonishment; you could actually hear a wave of recognition pass through the crowd! Here was a dinosaur tooth the size of a sand grain and they couldn't have been more filled with wonder if they were looking at the largest dinosaur. This was showmanship and the public loved it, but the real value was that it never failed to draw them into the world of the very small. It was a hook to capture the public's fascination and then teach them about extinct ecosystems and the art of fossil preparation.

"Wax on, wax off" - the Micropreparator mindset

No, the title to this paper does not refer to Carbowax, paraffin, or beeswax. It refers to the degree of focus, control, and discipline required to practice microprep and how that can have a meditative, almost Zen-like quality to it. This was illustrated to me one day in 2007 when one of my interns, Tom Nelsen, spent many hours learning the joys of carbide sharpening (the first skill I teach students, in part because it weeds out those who lack patience and persistence). After hours of trial and error, I heard him muttering "wax on, wax off, wax on, wax off…" If you're not familiar with this phrase, check out the first *Karate Kid* movie and you'll better understand Tom's mantra; he was acknowledging the degree of focus, concentration, and repetition required to master the most basic skill of micropreparation.

People have often remarked to me that they think microprep must be much harder than work on macrofossils. I think it is just different. The preparation of either very tiny or very large bones (or, as George Engelmann once referred to them, "the ridiculous to the sublime") requires its own set of techniques, tools, and materials. To do either of them with consistent high-quality results takes equal parts of skill and experience. But one

thing that sets microprep apart from the rest of the crowd is that it tends to be very unforgiving of even the slightest lapse of attention or slip of the needle. We can get away with little dings and scrapes in a large sauropod femur, but when the femur is only a centimeter long and one millimeter wide, a small mistake can obliterate the specimen. In this regard, it can be very intimidating, especially for a beginner. A little bit of fear and trepidation is good until you get the hang of it; just don't let that fear paralyze you.

Having said that, realize that some folks just aren't cut out for microprep. It requires long hours at the scope sitting virtually stone-still except for the barely perceptible flex of a finger and thumb and, in the course of a day, progress may be impossible to detect with the naked eye. Not everyone has the kind of mindset it takes to pursue this work. If that's not you, that's okay; there are plenty of big bones to work on.

There is a gray area that should be mentioned at this time: the instances when one may use a microscope (and other concepts mentioned here) applied to macro-fossils. Perhaps I am a perfectionist, but I happen to be one of those types who is haunted by those stray bits of distracting matrix left on otherwise clean femurs or in the deep recesses of iguanodont braincases. They just bug me and I have to get rid of them even if it takes a microscope and a carbide needle to do it. The same is true with removing overly thick (and unnecessary) glue coatings from modest sized bones or working out the fine details of the intricate tongue and groove structures of bone sutures on larger skulls. What may be considered overkill prep to some often means the difference between being able to readily distinguish between mere cracks and a real suture. To me it is also the difference between a good prep job and a great prep job. I want people to ooh and ah when they see my work and you should want that, too.

WHAT IS THE GOAL?

Before any work begins you should ask what the goal of the preparation is. Determine, as early as possible, how the specimen will ultimately be "presented." This decision should involve consultation with researchers or others and may depend much on what can safely be done (your call); considerations include whether or not the specimen is to be

cast, photographed with SEM, or used for other purposes the researcher has in mind (their call). Researchers can be pretty demanding sometimes so be prepared to stand your ground when they ask the impossible or be ready to hand the specimen over to someone who is better qualified.

Questions to ask: Can the specimen be entirely freed of matrix and is this desirable? Is the specimen best prepared on one side (in relief)? Which side is "best" for display or research purposes? How are you going to hold the specimen as prep proceeds (will you have to leave or even create a "handle" and where should it be)? Will you need to use Carbowax or other materials for a temporary matrix and what are the consequences? What is the best/safest way to store the specimen? I hate to have to add this too, but my experience has been that you'll need to consider the best way to protect the specimen from the abuse of those people less careful than you; I love preparing a specimen the first time but I dislike repairing them!

TOOLS, EQUIPMENT, AND WORKSPACE

I have seen some pretty amazing field prep done on microfossils by experienced hands with just a hand lens and a piece of carbide. However, at some point, no matter how good you are, you can only be as good as your equipment allows you to be (take note researchers and those who hold the purse strings). If you have a junky microscope and poor lighting, your work results are going to be poor. Trying to learn how to do microprep with substandard tools can be brutally frustrating and I have seen some potentially good preparators give it up because of this obstacle. What is worse is that many don't even know their tools are substandard. Most of these folks have simply never been exposed to a good microscope, light-source, or carbide.

I approach every microfossil with the attitude that I've got one shot at getting it right. To be handicapped by lousy equipment in a professional lab is simply an absurd concept. It makes no sense to invest a fortune (relatively speaking) in collecting specimens only to neglect the lab work that follows. Most hand tools are relatively cheap (Foredom grinders, carbide rod, Carbowax, etc); a good microscope and light source are not cheap

but they are essential for a lot of our work. To paraphrase Harvard preparator Bill Amaral (1989), your prep microscope should be at least as good as that of the researcher's microscope (maybe even better).

When I enter a lab to make recommendations for a microprep setup, the first thing I do is go to the work table and check out the scope, light-source, type of needles being used, and the sharpener for those tools; pretty much everything else is secondary to these essential tools. When I look through the scope and see a dim hazy image and a blunt hypodermic needle sitting nearby I recoil.

On the other hand, to be quite frank, I am all too often dismayed when I walk into labs and witness the sorry condition of equipment and tools that many people use. I'm referring to air tools lying in the sand tables, gunked up glue bottles lying on their sides with more glue on the outside of the container than in them, etc. Take pride in your work and take care of your tools! If you do, maybe your employer will be more likely to buy you better ones!

For more information and illustrations regarding tools, equipment, and setting up a microprep workspace, see Amaral (1989), the links in References section, and my setup (Fig. 1).

Figure 1. My work setup at Dinosaur National Monument. Notice the following equipment: sturdy table, comfy adjustable chair, adjustable boom stand (a), fiber optic light (b), selection of sharpened carbide needles (c), and moveable work surface (d). The air line is not readily visible in the wires coiled on the boom stand. Also notice the relaxed shoulders and neck, a result of correct work surface and eye-piece heights.

Tables

First and foremost, you need a solid vibration-free work surface. The same is true for whatever surface your scope and light-source are mounted on, whether it is the same table or the wall. Stability is everything. I prefer a heavy table with one or more sturdy shelves in front of me and to the sides: these provide a variety of surfaces for holding everything from light sources to vials as well as attachments for air lines and power cords. A rail mounted flush with the sides and back of the table is a good idea for helping to capture bits of bone or teeth that fly away during prep – and I can guarantee they will. I find there is no such thing as too many electric outlets as well. Velcro is a great way of keeping electric cords and air lines out of the way.

Chairs

Investing in a good chair will improve your comfort, stamina, and precision. It does not have to be an expensive chair, but it should be very adjustable. You should be able to position yourself relative to the microscope eye-pieces so that you don't have to strain your body to see. Adjustable arms can be very useful for bracing and stability. I have been in too many labs where the chairs people are using are junkers compared to those used by the secretary or computer jock in the next room. Ergonomic furniture is not just a luxury; it can improve stamina and the quality of your work and help prevent injuries.

Air lines

I use pneumatic tools for microprep on very rare occasions (usually I have been sorry that I did). But an air line is incredibly useful for blowing away matrix from the specimen. This little addition is worth its cost for the trouble it will save you keeping the specimen free of debris. Without one you are stuck having to hold a squeeze bulb all the time or blow with your lips (each is inconvenient and/or poses risk to the specimen). Arrange for compressed air at the workstation that aims a *gentle* and narrow air stream at the work. I mount my line to one of the light pipes with Velcro so it's pointed right where I am working. Be sure to use a valve that allows just a *little* air to run through the line (have you ever seen those videos of an escaped fire hose? Been there, done that!). It is also important to know when to turn the air *off* – be aware of loose pieces that may fly away forever. Make sure the air line has a good desiccator – water buildup in the lines has caused at least one disaster for me. Amy Davidson (1998) shows a schematic of a chip-blowing (preparation) needle; with slight modifications, this concept can be used to build a stationary blower.

Microscopes (and accessories)

I am sure there are many great microscopes out there, but all I can say is that I miss my old Nikon SMZ-U that I had at Dinosaur National Monument. It had fabulous optics (with a stereo zoom range from 0.75x to 7.5x), a wide field of view and a flat image at all magnifications (i.e., no curvature of field aberrations); these are all important qualities for a good microprep scope. The more whistles and bells you can afford the better. This

means a full complement of accessory lenses, including 0.5x – 2x objective lenses, and 10x, 15x, and 20x eyepieces.

People are always asking me how to upgrade a scope's power. The most important thing to remember is that changing objective lenses to a greater power reduces the focal length (working distance for your fingers) between the lens and the specimen, so you might want to think first about getting more powerful eyepieces. A really great scope with all the extras (like my SMZ-U) will cost up to $20,000. A serviceable scope for microprep can be bought for a fraction of that, but my sense is that with optics you get what you pay for. Unfortunately, this primary piece of hardware is also the one most often lacking in labs, usually because the cost is beyond that of many program budgets. One solution is to encourage a researcher who is serious about studying microfossils and has skill in grant writing to include a good lab scope in the next grant proposal.

Don't forget to buy a protective lens for the objective lens so you don't ding the glass with matrix or debris when you are prepping or sharpening carbide. I get rid of the rubber eye-guards on the eyepieces because they just make it harder to get my eyes "centered" properly, though many people find them necessary. I'm very diligent about keeping my scope clean, especially the eyepieces, and cover it every night or when not in use. Be extremely careful with how you clean glass lenses (especially the objective); they can cost a fortune!

Get a heavy boom stand with long arms and universal mounts for the scope body so you can turn and twist it to any angle, and consider bolting the stand to the table so it doesn't tip over from the weight of your scope (and all the accessories mounted to it). This can be a real hazard; I learned the hard way when my scope crashed onto a specimen, crushing it and almost taking out the objective lens, too. Bolting a stand down is preferable to weighting it with sandbags; bags tend to leak and they take up space better used for tools.

For microprep in the middle of a large block, invest in a mobile floor stand (like you see in hospital operating rooms) with a long articulating boom arm. These are worth their weight in gold (some are about as valuable; prices range from about $650 to $3,000) and prevent the hassles and risks associated with finding a place to put your scope and light source on the block. Fiber optics also provide the option for extra long light pipes so you can mount the light source on the boom stand and run the light pipe the length of the articulating arm.

There are some amazing micro-video cameras available today that can send high resolution images directly from the microscope to a computer or monitor for live action views, video and still image capture, or storage and manipulation of data. They are also just plain fun to use and are a great aid in teaching prep, educating the public, and documenting your work. The use of this technology requires special configurations of the microscope body that are typically found only on more expensive microscopes.

Light sources

Forget about the old illuminators; they can produce a lot of heat and the light quality is poor. For microprep you will need a variable intensity fiber-optic unit. Look for twinned, long and supple light pipes with light-focusing lenses. I generally prefer a ring light that fits around the outer rim of the objective lens because it doesn't require constant adjustment. The idea is to get the light where you need it at all times. This is especially true at high magnification when you are trying to get that last bit of rock out of a hard-to-reach place.

Hand Tools

Use the best tools you can get your hands on! I got rid of hypodermic needles, sewing needles, dental tools, and Exacto-knives years ago. Dental tools can be very useful in the field, but for microprep the only use I've found for them is the occasional time I have to reach around a corner (usually this involves the inside of a skull) that can't be reached with a straight carbide needle.

Carbide

Carbide is about the only metal on the market that has the strength to hold an edge well and will not "spring" when you apply or release pressure. Use high quality diamond sharpeners to get the proper edge for the job and keep them sharp! This takes some discipline but is worth the trouble. And it is truly amazing how sharp a point you can make with carbide (Figs. 2 and 3).

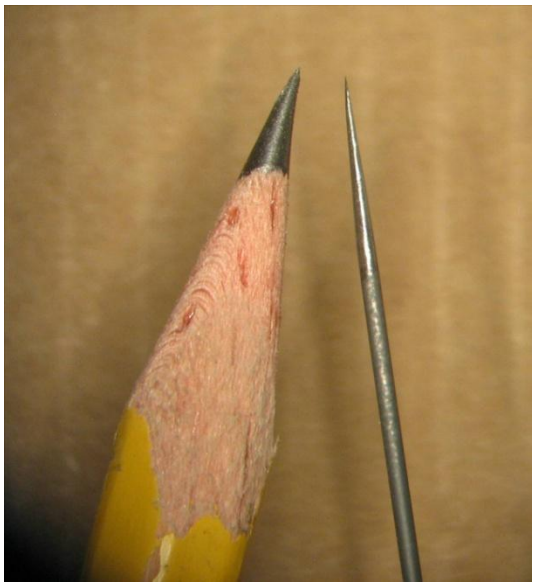

Figure 2. Magnified view of sharp point of carbide needle. Pencil for scale.

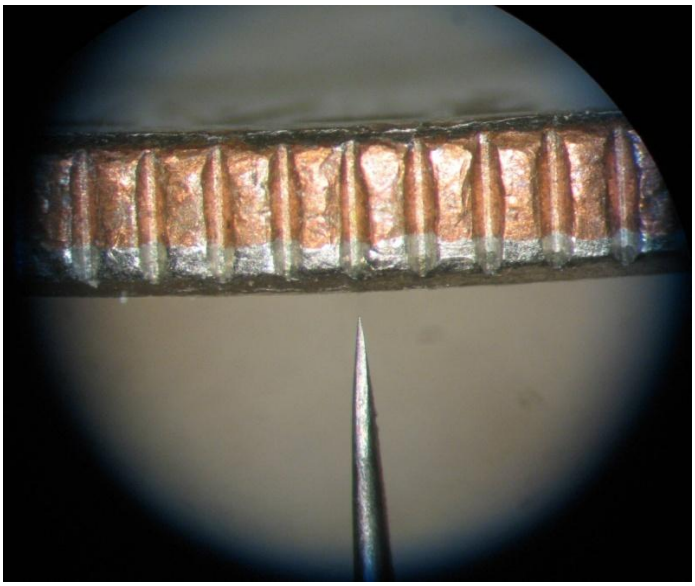

Figure 3. Magnified view of beveled edge of carbide needle. Edge of quarter for scale.

Carbide is available from several vendors, but I get mine from MSC Industrial Supply. It is called "carbide rod, C-2 solid carbide", and comes in many thicknesses (some vendors use inches and others use millimeters for measure so it can get confusing at times). Be aware there is "bad" carbide out there; if you see little cracks and voids appearing in it as you sharpen it under the scope you should probably try another piece. I consider about 1mm to be the best thickness for most microprep work; any thinner and it tends to break too easily and much thicker of a rod is overkill and takes forever to grind to shape. I break it to a reasonable length in my fingers and sharpen it under the scope using a diamond wheel mounted to a grinder with a variable speed foot control (this is about the only use I have for the grinder). The best diamond wheel I have found for this purpose is

also from MSC (3/8" x 3/8", 150 grit, diamond grinding pin, item# 03506128, $43.00). These wheels seem to cut faster and more smoothly than any other I have found and they last a long time.

Sharpening carbide might sound easy but actually requires a fair bit of patience and technique. I prefer to brace the grinder handpiece on a pad at a slight angle to the table and increase the magnification until the abrasive wheel fills the field of view. Hold the carbide rod at about a 20° – 30° angle to the top of the head and rotate it in your finger and thumb; I move the carbide in and out along the surface to even out the wear on the head. Gentle pressure is most effective so resist the urge to apply pressure. With 1mm carbide I generally start trimming at least ¼ inch from the tip to create a nice gradual taper. For a super-fine point you will have to drop the carbide to a very low angle relative to the diamond wheel. To bevel or shape the final tip, increase the magnification until the tip of the point fills the field of view and adjust your tools to allow even more precise grinding.

I'll say it again. Keep tools sharp! Dull tools just make bigger holes when you screw up and tempt you to use pressure instead of precision. I start each morning with a sharpening session. I typically have from 6 – 12 carbide needles on hand of various gauges, sharpened on both ends, and with a variety of different-shaped tips to handle changing rock conditions. I store them with one point down in a high-density foam pad and sort them with the sharpest ones closest to me so I don't have to hunt for what I need.

I seldom use pin vice holders for carbide in microprep except when I'm removing a lot of rock with a thicker-gauge point. I get the highest control of carbide and best "feel" using just my fingers. You might want to try this. If you use a pin vice I'd recommend one with a narrow tapered end, as the wide ones can pose a hazard to the specimen when you are working up close in confined spaces as well as block your view.

Forceps

Another tool I always have close at hand is my Dumont needle-nosed forceps. I've had the same pair for over 30 years, in part because I use them for picking things up, *not* digging around in cracks. They are lightly sprung and great for picking up the tiniest of bones and teeth. The finer the tip the tinier the object you can pick up without either crushing it or launching it across the room. I am not a fan of vacuum tweezers as they are cumbersome and tend to have tips too large for most micro-work.

Your personal kit

If you are serious about this business, get your own personal tools. This may sound harsh, but I recommend having your own set of tools and don't let other people mess with them. The first job I ever had was working in a restaurant and I can still remember watching the head chef come in every morning and opening his own briefcase full of fancy knives; I now understand why. Most tools develop personalities (the diamond sharpening wheel is one of them) that you get to know well and that other folks tend to screw up. The most important tools in my kit are the carbide needles and diamond sharpener. I came in one morning and found someone sharpening an Estwing hoe-pick with my diamond bit – I could have killed him! Hide your tools if you have to. I know a lot of labs where people have to share everything but I don't encourage this practice.

CONSERVATION PHILOSOPHY

The modern preparator's obsession with the subject of "glue" is a good thing. The discussions can get tedious, repetitive, passionate, and personal beyond reason, but they have resulted in a much more reasoned and disciplined approach to our work. In fact, it's unfortunate that more preparators don't obsess about it. Thinking more about what we use and how we use it is good for the specimens, and that is the bottom line.

This leads me to the next topic that should be learned and learned well: the appropriate use and application of various adhesives and consolidants. This skill is both the hardest and most critical thing you will need to learn. While I have my personal recommendations, my first advice is to avoid being dogmatic in your approach to

materials (and be skeptical of those who are). Have an open mind; be well informed, creative, and ready to think outside the box. Use materials and techniques because they make *sense*, not because they happen to be lying around the lab or the last person used them.

Most of us are neither chemists nor conservation materials experts and must therefore rely on studies done by these specialists for much of our information on the properties of these materials. I am a firm advocate of rigorous testing and the need for comparative studies of *all* of the adhesives and consolidants that have been used in preparation. But I also believe that important evidence indicating the effectiveness and longevity of these products rests on the shelves of museum collections around the world and we should be paying more attention to that. I am also wary of generalized statements regarding what is "best." For instance, of the typical solvent-based systems in use today, I've found that it is very hard to compare and contrast Acryloid B-72, Butvar (B-76), or PVA (B-15) because there is an infinite range of ways to mix them in multiple solvents.

I am my own worst critic and when I look at my own past work my eyes are drawn to the flaws. Learn from your mistakes and especially from those of others so you can avoid making them in the first place. Whenever I look at museum collections I look for flaws in preparation technique and materials, particularly adhesive and "filler" failures, and ask questions about the materials that were used. Look for clues like loose pieces and peeling of adhesive materials from bone and rock. These features usually indicate shrinkage of the material that can be catastrophic for microfossils. Note what has worked well for others and steal all the ideas you can. Seek out the most reliable materials with proven long term track records. Buying the cheap stuff off the shelf at the local hardware store is usually not the best way to go. The online *PREPLIST* is the best place to find the most up to date information and explore options (see References and Resources section).

Regardless of what you use, I recommend that the user of any substance pay attention to instructions, labels, Material Safety Data Sheets, and product brochures that come with the substance. No chemical that we use for prep is made specifically for our use and

contents can be changed without warning; this includes adhesives advocated by the conservation community, as well as those they disapprove of. For example, the formula for Duco cement used to include cellulose nitrate, but was later changed to polyvinyl butyral, essentially Butvar. Cellulose nitrate is fine for home use, but has poor qualities for fossil preparation compared to polyvinyl butyral. I still don't use Duco off the shelf for fossil preparation because I can make a similar and more versatile product myself with raw materials, but I want to emphasize the fact that it's hard to know exactly what you are using all the time. Get in the habit of checking labels and keep records of what materials you use.

There is a growing body of conservation literature that covers the subject of adhesives well, and I'd encourage anyone in the prep business to read these reports and perform your own experiments. Pay close attention to the materials you are using and how they behave in different circumstances; this knowledge base will give you the ability to adapt and make the best choices.

Consolidants and Adhesives: Solvent-based systems vs. Cyanoacrylates

Because of the difference in size and other physical characteristics, your approach to the application of adhesives and consolidants on microfossils is often completely different than on large bones. For instance, at the microfossil scale great strength is not as necessary as when you are working on a 200-pound sauropod femur, but the setting time, viscosity, and amount of adhesive required to do the job are critical factors; a single drop of glue might be enough to completely envelope a Mesozoic mammal jaw!

The rule of thumb when using adhesives and consolidants on microfossils is to treat every specimen as if it will be photographed using an SEM (scanning electron microscope). This means the treatment has to be not only invisible to the naked eye, but also must not coat a surface in any way (this includes glue, wax, Carbowax, and everything else). Anything that does not soak in will appear to the SEM as a solid – these machines can't be fooled! As Bill Amaral says, "get the glue *into* the specimen, not *on* it." This can be

achieved in two ways; applying an adhesive and physically removing excess material, or using an adhesive that soaks in thoroughly enough to leave no surface trace.

Let's take a look two of the most commonly used types of adhesives and consolidants and think about how you can best use their properties to your advantage.

Solvent-based adhesive systems (Acryloid, Butvar, and PVA) are first dissolved in a solvent (usually acetone and/or alcohol) and then applied to a specimen; curing (hardening) occurs as the solvent evaporates leaving the polymer (glue) behind as a solid. Curing time may be very slow and bond strength varies a lot depending on numerous factors. In general, for the consolidation of shattered bone or porous rock on the microfossil scale I want maximum control of penetration and setting properties.

Based on previous studies (Madsen, 1996b) and experience, I prefer using Butvar B-76 mixed with various ratios of acetone and alcohol rather than PVA or Acryloid B-72. The key here is the ability to adjust the volatility and viscosity of the solvent system so that the polymer can disperse into a substrate and do its work before the solvent evaporates, and Butvar B-76 seems to be best suited for this. In general, a higher acetone concentration will yield a thinner and faster setting mix, while a higher concentration of alcohol will yield a thicker and slower setting mix. There isn't a cookbook to tell you how to find the correct ratios of solvent to use in a given situation, but this determination gets easier with experience.

Cyanoacrylates (CAs) are another class of adhesives that have been in use in paleontology for at least 30 years. Known to the public as "super" glues of very high strength, they are available in a variety of formulations that offer different viscosities and setting times. CAs don't cure through the evaporation of a solvent but cure through a chemical reaction in the presence of a weak base, in this case, a fossil. Due to their particular properties, I have found cyanoacrylates adhesives to be a very useful alternative to solvent based adhesives when working on microfossils.

There has been much critical discussion of the use of cyanoacrylates in fossil preparation (Elder et al., 1998); some are valid and others may be premature or unfounded. But it has been my experience in 30 years of using CAs for microprep that I have never experienced a single bond failure or damage to a specimen due to shrinking of the material or causes other than human error. That's a pretty good record.

CAs are said to be more "brittle" than other adhesives we commonly use (Butvar, PVA, Acryloid), but brittleness is a relative term. My observation is that CAs in small quantities (when used as adhesives or film) are actually quite flexible, even after many years. This is easy to see under a scope when you poke it with a sharp needle or pry up a film – it bends. This quality can sometimes be used to your advantage. For instance, a very thin layer of CA may be applied to a bone surface, allowed to set and then peeled off the surface removing the last vestiges of stubborn matrix. Of course, this trick can also be done using other adhesives in place of CAs, but often times CAs simply do this job more efficiently. Again, experience should guide you in making the best choice in a given situation.

In addition, and of greatest importance for microprep, CAs have remarkable penetrating properties. Ethyl CAs (like PaleoBond's Penetrant Stabilizer) wick into tiny cracks like nothing else does. This is an extremely useful property when working on tiny bones and even more so if you are repairing a mammal tooth that is less than 1mm wide. CAs require far less fluid to do the job than epoxies, Acryloids, and other polymers, and don't rely on evaporation of solvents to form a bond. This allows me to set bone and tooth fragments correctly and achieve the tightest fit possible. For example, when using other adhesives, if you are trying to place a cusp back on a broken tooth, the cusp to be attached will need to "float" on a blob of resin and be pressed into place. This requires mechanical force, excess resin flowing out from the join, a less than perfect fit, and the need for further cleanup; I don't like any of these properties. CAs, correctly applied, can eliminate these problems.

It is my experience that CAs are harder to "reverse" (dissolve) than most other adhesives, so when I use them I do so with great discretion. The best strategy is to know exactly what you are going to do and how to do it so you get the job done right the first time. This often means *rehearsing your moves before you commit* with the application of the adhesive.

TECHNIQUES AND TIPS

How to position your body and equipment

Hold the tool loosely in the fingers. Relax your body, especially the hands, shoulders, and neck and don't forget to breath. Let the tool do the work. If you're holding the specimen, hold it gently – don't pinch or squeeze too hard. Save your energy; when you meet resistance try to find the soft spots in the matrix and avoid using excessive pressure. Work around tough sand grains as forcing them will usually just break the carbide tip. Make sure all cracks are secured (as Bill Amaral says, "if it moves, glue it").

Everyone has their own way of holding a tool, but my needle is always between my right thumb and index finger. For high-precision work I rest my right thumb on my left thumb and use it as a brace and "stop" that prevents pushing the needle too far (Fig. 4). Use the sandbag to focus the specimen; a gentle squeeze or relaxation of the left hand will change the height of the bag (and thus the focus). Be aware of where all of your fingers are in relation to the specimen so as to avoid touching delicate bone and breaking it.

Figure 4. Hand configuration where position of thumbs act as a brace and stop.

It's often advisable to place a high-density foam pad (about 1 foot square) under your work to cushion falls in case something escapes your grasp. I use a white pad so I can better find objects that get away. Use foam that won't easily crush and disturb the focus of the work under the scope. I often place all of my work (pad, sand bag, and all) on top of a thin board so that if I need to clear the workspace for another project I can move the whole setup and not have to rearrange everything later (see Fig. 1).

For sandbags, I prefer to use sand in cloth sample bags, doubled so they do not leak. Others prefer BB's in place of sand (it is cleaner) but I don't like the texture of it. Have lots of filled bags on hand of different sizes to assist in positioning specimens; they're also handy as hand rests and for elevating your hands above the specimen.

Removing matrix

In general the idea is to remove the matrix *without ever actually touching the specimen with the tools,* although in reality it seldom works out this way. Tease, pop, slide, flake, lift off the matrix. Depending on the situation this may mean either working towards the

specimen or away from it. Ideally, you should only have to make actual contact with the specimen if you are forced to shave matrix or consolidants off.

The fabric of matrix usually has weaknesses (as in the grain of wood) often associated with the original bedding; take advantage of this characteristic. I call the process of removing gross quantities of matrix "planing" (as in carpentry). When planing off matrix go in at a low angle to the specimen or rock surface and use a beveled-edge needle so as to remove as thin a layer as possible (especially when working close to the specimen). This will help reduce the chances of producing "discovery" marks. Look for color changes that often come in the form of "halos" around bone. Use your fingers to sense resistance in the matrix and when you do, stop pushing so as to not break the carbide point (and the resulting wax on, wax off sharpening mantra) or force off more matrix than you desired. If work slows a lot, try planing in a different direction or rotate the tool in the fingers 90-180 degrees and try again. Take advantage of thin or sharp edges of rock; they are usually more vulnerable and can usually be trimmed off faster. Always look for weaknesses to take advantage of, but avoid pushing or prying out chunks of rock; they may be attached to bone below so it is often better to stabilize them in place and plane them down.

Clean your work area frequently so that if you accidently lose a tiny fragment of bone on your table you'll have less debris to sift through during recovery. It's also a good idea to dump all your waste rock in a special container in case you need to sort through it later for a missing piece.

Know when to quit and call your project "done". Often a point is reached where you could continue to remove more matrix but to do so would substantially weaken a specimen. This sometimes requires a tradeoff of revealing less information for the sake of specimen integrity. The person preparing the specimen usually has the best intuitive feel for when this point is reached.

Using adhesives

A number of specialized techniques for the application of adhesives in microprep have been described and well-illustrated elsewhere and will not be re-described here in full. However, the critical need for the would-be micropreparator to learn the art of manipulating truly small quantities of adhesives with precision should be re-emphasized. When objects to be prepared are dwarfed by pinheads, even the smallest of manufacturer's applicators will prove to be too big and sloppy for your use.

Of all the means I've learned for creating and applying small adhesive drops (known as "microdots" around my lab), the use of single paper fibers has proven the most effective. This can be achieved in two ways. Paper or cardboard can be saturated with adhesive (the least viscous CAs lend themselves to this technique particularly well) and a sharp needle stroked across this surface picking up a tiny bundle of fibers. Contrary to what you might think, this tiny saturated bundle is *not* what you want to apply; rather you are hoping to see an isolated fiber sticking out of the bundle with a single minute drop on its tip – that's the gold! Alternatively, you can tear off a ragged little shred of tissue paper, roll it in your fingers to make a handle and swipe the other end through a small drop of adhesive; as with the needle technique, you hope to end up with a microdot of adhesive isolated at the end of a fiber. Either technique takes a lot of finesse and luck to get right, and sometimes many attempts to find that right drop where you need it, but it beats the alternatives. Finding a good source of paper of the right grade helps; personally, I'm a fan of the yellow legal pad cardboard backing.

My general rule of thumb, especially when using CAs, is less is best. Never forget how far and fast these substances can travel along tiny cracks or wick through porous material.

Finally, learn to use your hardeners as softeners. Consolidants like Acryloid, Butvar, and PVA are typically dissolved with acetone and/or alcohol. On contact with matrix they will usually soften it briefly before they stabilize the mass. This can be a maddening property (especially if you're fighting gravity) but can sometimes be used to your advantage by softening stubborn matrix. Timing is everything when using this technique

and you will have to act fast to remove the treated matrix before it becomes even harder than it was originally.

Polyethylene Glycol and Cyclododecane

Polyethylene Glycol (PEG), commonly referred to as Carbowax, is a water-soluble synthetic wax that has been used for decades by preparators as a temporary filler or support, among other uses. I have found this material extremely useful for microprep, and my favorite applications for its use are also described and illustrated in Madsen (1996a); the reader is encouraged to experiment with these and other techniques. In addition, another material called cyclododecane (CDD) now provides an alternative to PEG (Arenstein et al., 2004). CDD is also a wax-like substance, but unlike PEG (which requires some amount of water for removal), CDD sublimates (changes directly from a solid to gas) and therefore requires no mechanical or chemical forces to get it off your specimen. The potential advantages of this property are obvious. It should be noted that the toxicity of CDD fumes are still unknown and its use should be confined to a fume hood or well ventilated work area.

Mounting and storing microfossils

I generally don't consider my prep job done until the specimen is safely stored; in the case of microfossils this often means they are mounted on pins and stored in vials. Avoid using glue to mount teeth and other small specimens on pins. Many people do a poor job of mounting them (tending to mount them crooked or smear glue on the specimen), but more importantly, someone *always* seems to want to remove them later. Getting the specimen off the pin usually requires mechanical force (which may break the specimen) and/or solvents (either smearing glue or weakening bonds in repaired specimens). Use some form of non-tacky wax for small mount jobs, especially if you're mounting them on pin heads. Microcrystalline synthetic waxes of the type that are used for "paste-up" jobs in the printing industry are good for this use. I've used wax for this purpose for many years and have seldom had any problems with specimens falling off, including when they were mailed. Another benefit of wax is that if the specimen is knocked against the glass when it's returned to the vial (when most damage occurs), it's

much more likely to survive the blow if the mounting medium has some "give" to it. In addition, using wax instead of glue allows you all the time you need to orient the specimen correctly or make later adjustments. A small groove or depression impressed into the wax is usually all it takes to "capture" the specimen.

Learn to set the teeth or jaw fragment on the pin in the correct orientation. This usually means that if the cork is resting on a flat surface the cusps are pointed straight at the viewer looking through a scope. Pins need to be inserted straight into the cork and centered to reduce the chance of the specimen hitting the glass. If rotating the pin or cork within the vial may cause contact of specimen to glass consider using a larger vial.

Lastly, I try to avoid the use of cotton for microfossil storage. Cotton has its uses for cushioning more robust specimens, but all those fibers love to cling to tiny projections like teeth and cusps. I've wasted a lot of time picking tiny fragments of teeth and bones out of cotton balls in my career, time that would have been better spent on the front end of the process by creating proper storage. There are also many excellent alternatives to vial storage and you should explore these options.

SUMMARY

Micropreparation deals with the mechanical preparation of vertebrate fossils on a scale that requires the use of a microscope. As such, it requires specialized tools and skills, as well as a temperament that can deal with long hours of sedentary work and intense focus. The quality of work is also limited by the quality of tools; a high quality stereo-zoom microscope is the most important piece of equipment, but a comfortable and well planned work space is critical for efficiency. Custom ground carbide needles should be used for most work and these need to be fashioned with great precision. The micropreparator should have a broad knowledge of the properties of adhesives and consolidants and apply these materials with a clear purpose and strategy that insures the longevity and scientific utility of the specimen; a similar approach should be applied to determine the best means of storing and handling microfossils after preparation.

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REFERENCES AND RESOURCES

There is a rapidly growing body of preparation and fossil conservation literature. Much of it is available on-line and I intentionally tried to limit the references in this paper to those currently in electronic formats, but some of the best information is still only found in hard copy. The links below are a good place to start exploring the literature.

Amaral, W. W. 1989, Microscopic preparation, *in* Leiggi, P. and May, P. (eds.), Vertebrate paleontological techniques, Vol. 1: Cambridge University Press, Cambridge, p. 129-140.

Arenstein, R., P., Davidson and **Kronthal.** 2004. An investigation of cyclododecane for molding fossil specimens. http://vertpaleo.info/documents/Arenstein_et_al_2004.pdf

Davidson, A. 1998. A foot-controlled, chip blowing needle for micropreparation of fossil vertebrates. http://www.vertpaleo.org/education/documents/Davidson_1998.pdf

Elder, A., **Wenz**, and **Madsen**. 1998. Understanding cyanoacrylate adhesives and consolidants and their use in vertebrate paleontology. *In* J. Martin, J. Hogansen, and R. Benton (eds.), Proceedings for the fifth conference on fossil resources, 5:141-143. http://www.vertpaleo.org/education/documents/Elder_et_al_1998.pdf

Madsen S. K. 1996a. Some techniques and procedures for microvertebrate preparation. Oklahoma Geological Survey, Special Publication 96-4: 25-36. *www.snomnh.ou.edu/pdf/reprints/unused/96-6.pdf*

Madsen, S. 1996b. Testing properties of preservatives in the preparation lab. The link below from ReBecca Hunt's www.dinochick.com provides verbatim text of a platform talk from: Journal of Vertebrate Paleontology 16(3) Sept. 1996 Abstracts of papers. http://74.125.95.132/search?q=cache:_o9hf-9zu5cJ:www.dinochick.com/SKM%2520Prep%2520session%2520AMNH%25201996.doc+scott+madsen+preparator&cd=4&hl=en&ct=clnk&gl=us

PREPLIST may be found at: http://www.vertpaleo.org/education/preplist.cfm

The Paleontology Portal is an excellent on-line source of preparation and conservation information with links to many other useful sources: http://collections.paleo.amnh.org/

Also please check out Matt Brown's excellent web page www.fossilprep.org.