explosion, but I've had to clean up boil overs on a number of occasions.

2) Once the reaction subsides and the solution turns a golden yellow color, it is just a matter of washing the diatoms. There are a couple of ways to do this:

a) centrifuge the diatoms, then throw out the liquid, add wa ter, centrifuge, throw out the liquid, etc.

b) fill the large beaker with water and let the diatoms settle to the bottom (1-2 hrs), carefully pour off the fluid, and add more water, let the diatoms settle, etc. The diatoms will appear as a milky white residue in the bottom of the con tainer. The sample may have to be concentrated to really see this). If any more information is needed please feel free to contact me.

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Preparing Powders For Electron Diffraction Studies

Questions often come up concerning the best way to prepare powders for electron diffraction analysis in the TEM. This is actually fairly simple:

1) Take an oxide powder, grind it up with mortar and pestle and take a carbon coated grid and swipe it across the fines. Two glass slides can also use to grind up the powder.
2) A number of materials can be evaporated onto a carbon coated grid or onto a cleaved NaCl sample and then float that off on water onto a grid.
3) A molybdenum wire can be smoked in air to provide crys tals. If the wire is left in the smoke long enough, there will probably be enough crystal to cover a grid. Heat the wire across the terminals of an evaporator or heat with a torch to generate the white smoke. Thus MoO3 sample will pro- vide a good rotation calibration sample. Magnesium can also be burned to produce MgO crystals.

If there aren't sufficient particles in a single diffraction pattern to provide diffraction rings, take multiple exposures from different areas on the same piece of film.

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A Tip on Purifying Glutaraldehyde

Being an old timer, I have been around since before the advent of vials of glutaraldehyde. In the 1970s, the Merck index described a method for purifying glutaraldehyde using activated charcoal.

Basically, glutaraldehyde begins to polymerize when stored at room temperature. Since best fixation is achieved with the monomeric form, this purification protocol was designed to remove the more complex forms.

Storage under nitrogen in a sealed vial slows the polymerization, and lower concentrations degrade more slowly. Storing at 4°C is advised.

Method:
1) Mix the 25% glutaraldehyde with 1g of activated charcoal per 100 mL of liquid.
2) Allow to stand for 30 minutes, then vacuum filter through a Whatman #1 filter.
3) Repeat three times.
4) Store at 4° C

Repeat once a year.

I have used this method for more than 30 years and consistently obtained very satisfactory fixation. It is messy, but worth the effort.

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