Capstone Research Paper:
The Victorian Art of Arranging Diatoms

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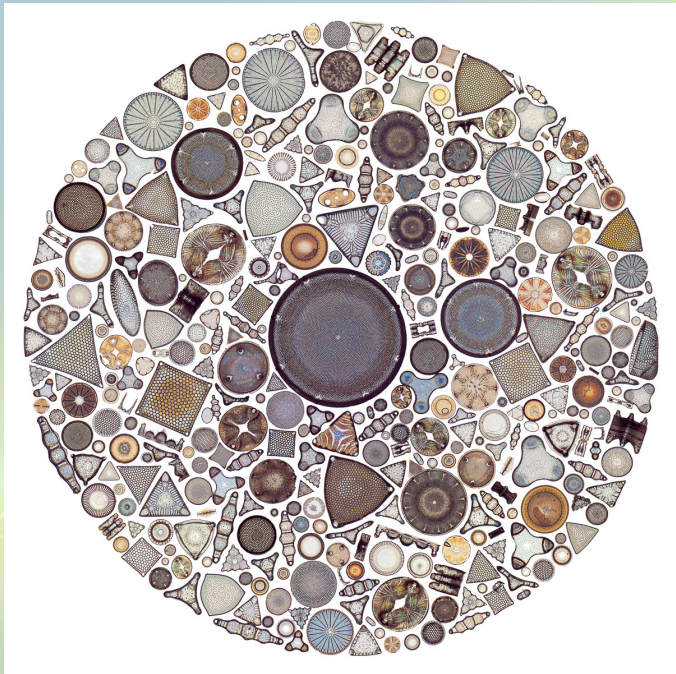

Beats, S. (Photographer). (2015, June). [digital image].

Retrieved from http://www.microscopy-uk.

org.uk/mag/artjun15/sb-Diatom-Arranging.pdf

Abstract

This paper serves as a guideline for my capstone project. Notable diatom arrangers of the past and present, and how to collect, clean, arrange, mount, and illuminate diatoms in order to create a photograph of a diatom arrangement are all topics discussed in this paper. The art of arranging diatoms was very popular in the Victorian period, but it has declined in popularity. There aren't many practicing diatom arrangers alive today. A very existent fear is that this beautiful form of art will be lost in history if people lose interest to the point where there are no practicing diatom arrangers left in the world. My goal of this assignment is to learn all aspects of arranging diatoms. This will involve collecting and cleaning diatoms in order to arrange and photograph them. I expect this project to be full of obstacles, since I have never arranged diatoms before. I will have to be flexible and knowledgeable of other alternate processes in order to achieve the end goal of creating a diatom arrangement. I have researched not just one, but many methods of cleaning and preparing slides of diatoms. There are many articles describing different diatom arrangers' processes. This will allow me to have backup plans if my original plan doesn't work out.

Keywords: diatom, diatom arrangement

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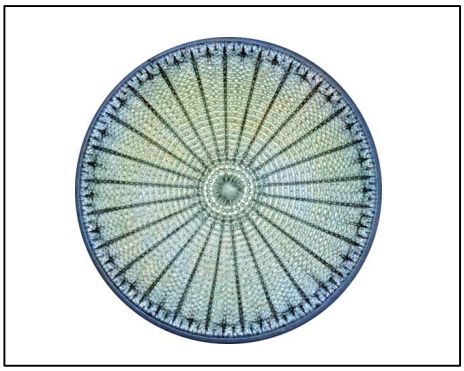
Diatoms are incredibly beautiful microorganisms that many people don't know about, let alone are able to observe in person. Diatoms are, by definition, "any of numerous microscopic unicellular, marine, or freshwater algae of the phylum Chrysophyta, having cell walls containing silica" (The Definition).



Grave, E. (Photographer). *Diatom (highly magnified)* [digital image]. Retrieved from https://www.britannica.com/science/diatom

There are over 100,000 species of diatoms (Miller), and they range from five to one thousand microns long (Diatoms). They are "one of the most common types of phytoplankton" (Nagy, 2011, p.xi). Diatoms live in very diverse climates. They are present in marine waters, oceans, fresh water (streams and lakes), hypersaline or brackish water, high and low temperatures, and a variety of pH levels (Nagy). Diatoms are almost perfectly symmetrical. They were one of the earliest subjects to be viewed under a microscope. Due to their availability their first application was "to test microscope objective resolving power" (Lynk). Today, they are used to track climate change (Lynk). This paper discusses historic and modern diatom arrangers, and how to collect,

clean, arrange, mount, and illuminate diatoms in order to create a photograph of a diatom arrangement.

Diatom Arrangers of the Past and Present

In the Victorian era (1830's – 1900) (Lynk), microscopists mostly collected diatoms, as well as foraminifera and radiolaria (Franchini). "All of the professional mounters during the Victorian era prepared and offered numerous diatom slides of material obtained from locations around the world" (Lynk). In 1849 G. Shadbolt created the first diatom arrangement. Some notable diatomists from the Victorian era are Charles Baker, and Johann Dietich Möller. Klaus Dieter Kemp and Steven Beats are modern diatom arrangers (Bracegirdle).

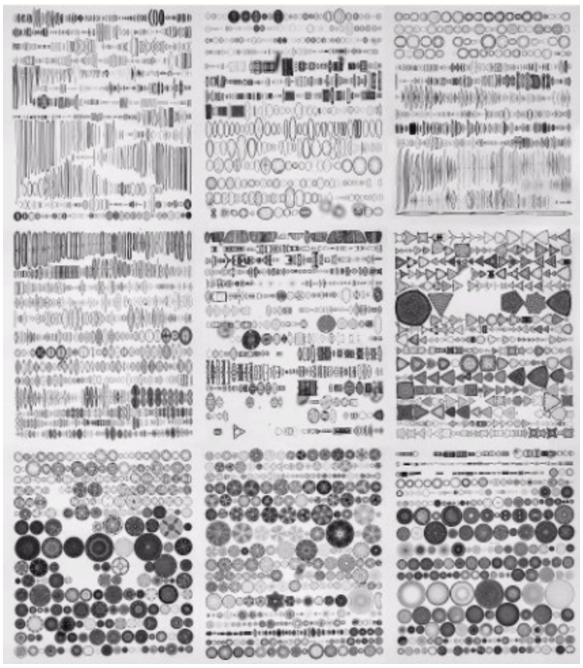
Charles Baker (1814-1893)

By 1851 Charles Baker had created a well-established company. His company sold microscopes, slides, and other scientific instruments. By 1864 he was selling slides of diatom arrangements. Möller created some of these slides (Bracegirdle).

Johann Dietrich Möller (1844-1907)

Johann Dietrich Möller first began making diatom arrangements in 1867, with the help of his brothers. Throughout his lifetime he created slides of diatom arrangements containing differing amounts of diatoms, which were then sold to customers. These include: a 20 arranged diatom species test slide (2,162 created and sold), a 100 arranged diatom species slide (1,009 created and sold), and a 400 arranged diatom species slide (597 created and sold). Möller also created a special 1,715-diatom species slide for Brazil's emperor. He made "two slides with the [diatoms'] names photographed onto the slide" (Bracegirdle, 1998). These two slides contained 80 and 335 species of arranged diatoms. His biggest arrangement is 6 millimeters by 6.7

millimeters and contains 4,026 specimens arranged into 133 rows. This slide is known as the *Universum Diatomacearum Möllerianum*.



Schoors, J. (Photographer). (2015, March). [digital image]. Retrieved from http://www.microscopy-uk.org.uk/mag/indexmag.html?http://www.microscopy.uk.org.uk/mag/ artmar15/vip-diatom-slide.html

"It took [Möller] forty days just to place the diatoms" (Bracegirdle, 1998). After the slide was finished in 1890, Henri Ferdinand Van Heurck purchased this slide. Möller is also famous for the indexes of diatoms that he included when a person bought his slides. These indexes of diatoms allowed him to win many awards. Möller was a pioneer of diatom arranging. Unfortunately, he never wrote down his process, which means that when he passed away in 1907 he took his methods of arranging diatoms with him to his grave (Bracegirdle).

Klaus Dieter Kemp (1937-)

Klaus Dieter Kemp was highly influenced by Möller. At the age of nine Kemp emigrated from East Germany to England. He worked at Flatters and Garnett Limited from the age of fourteen to twenty-eight. His duties included preserving specimens, especially diatoms. Gordon McKechnie taught him how to mount slides. Since, he had been selling his completed slides to the company for six years Wilfred Garnett gave Kemp the J. A. Long diatom material. From 1968 to 1974 Kemp "worked at Salford University and then [from 1974-1993 he] joined Harris Biological Supplies as production manager" (Bracegirdle, 1998). He left that company in 1993 in order to be self-employed. Four years later he was well known for his diatom and butterfly scale arrangements (Bracegirdle). Today he is well known across the world. Kemp "reinvented the art form [of arranging diatoms] to a requirement that made it a teachable skill. He also devised an inexpensive *micromanipulator* which can be constructed with simple machine tools" (Nagy, 2011, p. 11). Ever since I saw the video *The Diatomist* by Matthew Killip (see references), I have had a fascination with diatom arrangements. Just like how Möller influenced Kemp, I got my inspiration from seeing Kemp's diatom arrangements.

Steve Beats

In June of 2015 Steve Beats published an article (see references) on tips and tricks that he learned while attempting diatom arrangements for the first time. He has been interested in microscopy, as a hobby, for about forty-five years. Beats sold his slides in order to have extra money to purchase microscopy gear. After he saw a step-by-step article on arranging diatoms (see Hummelink under references) Beats sold his diatom arrangement slides with the hope that he'd be able to make some of his own. He accomplished his goal. It took him 130 hours to place 400 diatoms. "Two years and 2000 hours of effort later I'm still learning, still practicing, and

still making mistakes. But it's a truly satisfying (and ongoing) experience that I'd strongly encourage others to try" (Beats, 2015).

Collecting Diatoms

Diatoms are relatively easy to find and collect. "Diatoms are often visible to the naked eyes as a golden coating growing on vessels, and they commonly from brown films on aquarium glass or rocks" (Nagy, 2011, p. xi). Mud or silt contain "contaminants that are difficult to separate" (Nagy, 2011, p. 3), so it's best to obtain debris-free samples not in contact with them. Diatoms can be collected in freshwater, brackish water, marine water, and fossil diatom sites all over the world (Nagy).

Collecting Diatoms in Freshwater

There are many methods to collect diatoms in freshwater. One way to do this is to gather "samples of stalked multicellular green algae (or aquatic weeds) that grow on the bottom of the stream of lake" (Nagy, 2011, p. 3).

Blair. (Photographer). (2015). [digital image]. Retrieved from https://www.study

blue.com/notes/note/
n/bio-101-study-
guide-2015-16-blair/
deck/15679050

Place the algae into a zip-lock plastic bag and shake, pummel, or agitate the bag. This will cause "the water in the bag [to] rapidly become a cloudy golden-brown or olive-drab color as the diatoms are released from the weed and become suspended in the solution" (Nagy, 2011, p. 3). Pour the water into a vertical glass cylinder through a coarse sieve. The sieve will stop the green algae and allow the diatoms to easily flow through and be contained in the bottom of the glass cylinder. Allow the diatoms to settle for a few hours or overnight. Repeat this process four to five times in order to gather all of the diatoms. The previous process doesn't cause any damage to the diatoms. Other places to find diatoms are on "the undersides of lily pads, or damp concrete in the shadow of a bridge", because "diatoms tend to prefer cold water that is not in direct sunlight" (Nagy, 2011, p. 4). Another method of collecting diatoms in freshwater is to find rushes and strip off diatoms that grow in sheaths along this plants' stems. "Other species may

(2015, April 10). [digital
image]. Retrieved from

https://thomascountyag.

com/2015/04/page/3/

grow as a shiny or glossy coating on rocks or on wood exposed to splashing water, appearing like a mucous growth with the characteristic color" (Nagy, 2011, p. 4). Some diatoms "grow in microscopic, essentially colorless tubes, or on the ends of tubes, and the whole colony appears like a dirty off-white cotton mop attached to rocks in" a stream (Nagy, 2011, p. 4).

Leekie, J. (Photographer). (2014).

Didymo is distinctive – and rather

disgusting [digital image].

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dfo-mpo.gc.ca/sep-pmvs/sci-icp/

streamtalk/14-02/14-02-eng.html

The scientific name for this type of plant is *Didymosphenia geminate,* but it has been "renamed by the public as 'Rock Snot' " (Nagy, 2011, p. 4). Didymo is a highly invasive species. It's fatal to plants and insects, if it expands enough to blanket the bottom of a stream. Fly fishermen have transported it all over the world by way of the felt soles on their boots. Diatoms can also be seen "as a fine brown or golden-brown dust" amid "pebbles on the bottom of slow moving waters" (Nagy, 2011, p. 5). These are individual diatoms. A turkey baster can also be used to collect sediment, which also contains single diatoms, off the top of a flowing stream. Like the first

method, put the water in a cylinder so that the diatoms settle at the bottom over a period of time (Nagy). There are a number of ways to gather diatoms in freshwater, and depending on the methods used, different types of diatoms can be collected.

Collecting Diatoms in Brackish and Marine Waters

Like freshwater, oceans also contain single and colonial diatoms, but it is more challenging to collect diatoms in the ocean compared to freshwater. Go to an area with rocks that has still water. Use a turkey baster to collect single diatoms off the top of the water, similar to the sediment method used to collect freshwater diatoms. Buoys, anchor lines, and sea debris (driftwood and plastic) also contain green algae that can undergo the same process as the first method of collecting freshwater diatoms listed above. Another way to collect diatoms from marine and brackish waters is to first find a seashell. Brush the seashell with a the bristles of a toothbrush, while rinsing the shell with water. Once the toothbrush turns brown, then this is an indication of the presence of exotic diatoms. Diatoms can also be collected by "tow[ing] a very fine funnel-shaped net behind a boat at low speeds" (Nagy, 2011, p. 6). Not everyone has access to a boat, so this option is only available to a limited group. It's significantly more difficult to collect diatoms in brackish and marine waters.

Collecting Fossil Diatoms

Finding fossil diatoms are an even greater challenge. Nagy explains how fossil diatoms are formed:

> There are sites around the world where diatoms fell as sediment out of marine or freshwater bodies of water over time, and formed deep concentrations on the bottom. Over time the organic material decomposed and the diatom frustules were pressed together, resulting in diatomaceous earth, or diatomite. (Nagy, 2011, p. 6).

The most famous place to gather diatomaceous earth is Oamaru, New Zealand. Some other famous freshwater fossil diatom locations include Terrebonne and Klamath Falls, Oregon, and the Toome Bridge in Ireland. Diatomists from the Victorian era often gathered collected fossil diatoms from Klamath Falls. Notable marine deposits include: Lampor, California; Dunkirk, Maine; Szent Peter, Hungary; Mors, Jutland, Denmark, France, and Russia (Nagy). Diatomaceous earth can be purchased, as well as collected and cleaned freshwater, and brackish and marine water diatom samples (see Dailey under references). If I don't find adequate samples or run out of samples in the winter, then that is another option.

Cleaning Diatoms

Cleaning diatom samples is an important step in order to have a good, clean diatom arrangement. "The goal of cleaning diatom samples is to have as a final product a sparkling white suspension of diatom" parts "in distilled water, free of diatom fragments and containing sediments, so that a single diatom valve can be examined accurately" (Nagy, 2011, p. 7). There are many ways to clean diatoms. The three main ways are sulfuric acid, peroxide, and incineration methods. Another method that I found uses bleach. The methods I am going to try are ranked in order: 1.) Bleach, 2.) Incineration, 3.) Peroxide, and 4.) Sulfuric Acid. These methods are ranked in this way due to ease of use and safety level. If the first method doesn't work or is inadequate then I will go to the second method, and so on, until I find the best method to clean diatoms. If I am not allowed or don't have the materials to do a certain method, then I may resort to buying already cleaned samples. I am only going to go into detail about the bleach method; information about the other methods can be found on the references page.

Bleach Method

This method is the easiest and safest method. Household bleach is "readily available, does not require heating, is inexpensive to use, and does not necessitate the use of accessory equipment (i.e., fume hoods)" (Carr, Hergenrader, Troelstrup, 1986, p.153). Step one is to clean off any preservatives that might be on the diatom samples. First, place diatoms in a small beaker and add a few drops of distilled water. Next, allow the diatoms to settle at the bottom. Afterwards, carefully decant the water to get rid of as much water as possible. After step one is complete, mix one part bleach with one parts distilled water and diatoms in the beaker. Cover the beaker with a watch glass in order to keep unwanted particles from contaminating the sample. Let the beaker sit for one to two hours and agitate it throughout the waiting process. Decant and rinse the sample at least six times with distilled water. Allow the diatoms to settle to the bottom of the beaker between each rinse cycle. After the last rinsing, drain the extra water and move the diatoms to a storage container. It is unclear as to whether this method will be able to clean marine diatoms, because it has never been tested. This method may also cause the loss of very small plankton, because they don't easily settle. They tested the sulfuric acid and peroxide method in order to compare results to the bleach method.

> When large amounts of organic matter were present in our samples, the nitric acid cleaning procedure [also known as the sulfuric acid method] had to be repeated. The hydrogen peroxide method was unpredictable; either no reaction occurred, or sometimes violent eruptive bubbling caused loss of samples from their containers. (Carr, Hergenrader, Troelstrup, 1986, p.155)

According to Nagy, other processes, including the bleach method, may be less dangerous than the sulfuric acid method, but he claims that the other methods are also less effective than the sulfuric acid method.

Arranging Diatoms

Arranging diatoms is going to be one of my biggest challenges, but tips and methods that I have found will be helpful. A two-millimeter glass capillary tube can be melted in order to make a glass needle. “Other types of needles have been used in the past, but pulled glass from a capillary tube has stood the test of time, since it is both flexible and easily replaceable” (Nagy, 2011, p. 12). Another important tool is the micromanipulator. This device allows the diatom arranger to lift, lower, and position the glass needle in the exact position he wants it in order to place the diatom on the slide. It is advised to use an achromatic 10x objective. This resolving power “is a compromise between acquiring the magnification needed to see the diatoms, and having sufficient working space between the front of the objective and the microscope slide [in order] to lift diatoms and move them” (Nagy, 2011, p. 12). If I can’t arrange diatoms, then another idea is to individually photograph diatoms and combine them all into one photograph (Michael Peres) of a “modern diatom arrangement”.

Static Electricity

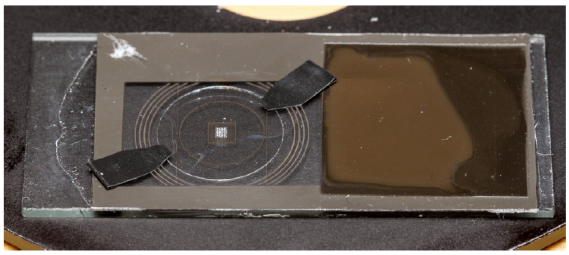
Picking up diatoms from one slide and placing them on another is can be hard due to static electricity. Static helps to secure diatoms onto a slide and allows diatoms to stick to a glass needle. The downside of static electricity is that it can cause diatoms to stick or “be repelled (pinged) away from the needle” (Beats, 2015, p. 5). Rubbing paper fibers on the glass needle or using an anti-static gun will help to reduce static. Beats recommends the Milty Zerostat 3. This product doubled his diatom arranging speed. A newly made glass needle will create too much static electricity, so it’s best to wear it down before attempting to arrange diatoms. “Rubbing” a diatom off a needle onto the glass will not work. Instead allow the diatom to hang off the needle a little bit, so when you go to place it on a slide the diatom and not the needle touches the slide.

The diatom may take a couple of seconds to fall off the needle and onto the slide, but be patient because “rubbing” it will make things worse. Static electricity plays a big role in arranging diatoms (Beats).

Microfilm

Microfilm is clear film with a microscopic design printed on it. “Anything that was white (i.e. the background) in the original is clear on the film. Anything that was black comes out dark brown and opaque (i.e. the lines of the design)” (Beats, 2015, p. 13). Microfilm can be used as a guide in order to get a clean design when placing diatoms on a slide. The design must be made up of lines and mirrored, because the “diatoms will be viewed through the other side of the coverslip after mounting” (Beats, 2015, p. 13). Beats explains in detail how to mount the coverslip to the slide:

> The film roll has to be cut to put the template at one end of the frame and an equal sized dark area (for the keeper) at the other end. The insulation tape is used to hold one end of the film in place prior to applying a couple of small drops of balsam. The film is pressed down to touch the balsam where surface tension and air pressure take over to pull the film onto the glass and hold it flat. […] The film should be stuck with the emulsion side up (shiniest side down), so the imagery will be in direct contact with the underside of the arrangement coverslip. This ensures it is not too out of focus when you are focused on diatoms sitting on the surface of the arrangement coverslip. (Beats, 2015, p. 14)



Beats, S. (Photographer). (2015, June 13).

Retrieved from http://www.microscopy-uk.

org.uk/mag/artjun15/sb-Diatom-Arranging.pdf

The adhesive used to mount the microfilm to the slide is half Canada balsam and half Xylene. "(It never dries). Twenty-four to forty-eight hours on a hotplate at about eighty-five degrees Celsius will dry it enough for careful use but it needs another three to four weeks in a warm place to become 'robustly dry'. (Beats, 2015, p. 14)

Slide Preparation

There isn't just one way to make a slide; the types of materials and the amounts can differ for each slide and subject. "Microscope slides on diatoms are made by placing a sample between a microscope slide and thin coverslip of glass, typically with a 'mountant' between the two which stabilizes the subject matter and causes the coverslip to adhere to the glass slide" (Nagy, 2011, p. 9). A high refractive index mountant is needed in order to see the diatoms. There are diatom mounters that we know of from the Victorian period, but they were never written down so exactly how the mounts were made will never be known.

Types of Mounters

There are many mounters to choose from. Styrax is one example. Dr. Henri Van Heurck created it in 1885. He used "resinous gum from a shrub, *Styrax liquidambor*" (Nagy, 2011, p. 10). There is an American tree called *Styrax officinalis*. Styrax is not sold commercially, but it can be made. Kemp swears by Styrax, saying that it "is the most stable mountant now available" (Nagy, 2011, p. 10). Naphrax and Zrax are two ready-to-use mountants that can be bought online. Naphrax is leftover after Napthalene and formaldehyde condenses. This is "not an archival material", because eventually little pockets of air, or bubbles, may form (Nagy, 2011, p.

10). Dr. Bill Dailey, an organic chemistry professor at the University of Pennsylvania, created Zrax. It "has similarities to Hyrax and […] promises to be more stable over time than Naphrax" (Nagy, 2011, p. 10). Beats came up with a modified version of Debe's Fixative. This product has "good strength while minimizing visible artifacts" (Beats, 2015, p. 9). His version is made up of "25 ml glacial acetic acid, 12 g isopropyl alcohol, 6 g of isobutanol, [and] 0.3 g [of] high quality, white bovine gelatin (pH 5.0 – fine powder)" (Beats, 2015, p. 9). The difference between Beats' version and the original Debe's Fixative is that Beats used "much less gelatin and a slightly different ratio of alcohols to acids" (Beats, 2015, p. 9). The process to make Beats' altered version of Debe's Fixative is as follows:

> Form the gelatin powder into a small pile and mix with a few drops of distilled water until it is the consistency of thick wallpaper paste. Use a minimum amount of water but ensure the "paste" is clear with no white lumps of powder remaining. Drop this moistened gelatin into the acetic acid in a screw top bottle and shake until all the gelatin dissolves (this may need up to an hour of intermittent, vigorous shaking). Do not heat the mixture as this will subtly change the molecular structure of the gelatin and can reduce its strength.
>
> Mix the alcohols in a separate container. When the gelatin is dissolved, add the alcohols to the acetic acid a few drops at a time, mixing well between drops. Use a separate pipette for dropping the alcohol and do not get any of the acid/gelatin mix into the pure alcohol or gelatin may precipitate out.
>
> When all the alcohol is mixed in, filter the finished Debe's through a 0.2um Whatman filter into smaller storage vials. You can use filter paper and funnel, but cover the top with silver foil to reduce the evaporation of the alcohol while filtering. The recipe makes

about 50ml of Debe's (a lifetime of arrangements). Store in a cool, dark place, but not the refrigerator.

So far, this mixture has proved to be very stable and it doesn't seem to suffer solidification or appearance of specks of gelatin over time like the stronger formulation can. On top of that, it sticks diatoms very firmly with virtually no detectable soaking in when the film is applied thinly

enough.

The Debe's is applied to a clean coverslip by placing a drop in the centre of the slip using a small glass rod or similar. The fixative will spread to the edge and form a thin flat film on it's own. If the Debe's doesn't spread evenly then the coverslip is not clean enough. I now clean my coverslips using Pirahna mix (dangerous) followed by 5 minutes in an ultrasonic bath of isopropyl alcohol and then heat dry immediately before applying the Debe's.

Coating thickness is controlled by the size of the drop and by the size of the cover slip to some extent. If a drop spreads well but doesn't reach the edge, then you get the thinnest possible coating, but it will only just hold onto the diatoms. Only use the thinnest coating for very delicate frustules or very small arrangements. Normally you want the Debe's to spread right to the edge to get a thicker strong coating that holds frustules very firmly. (Beats, 2015, p. 10)

Depending on the cost and whether I have access to the materials needed to make Beats' "Modified Debe's Fixative" it seems like an easy enough process to try. Plus, it makes a lifetime supply. If I don't end up making this then I will buy some of Dr. Bill Dailey's Zrax mountant.

Illumination Techniques

There are many types of illumination techniques that are produced with a microscope, but some are better than others for photographing diatoms. Phase contrast is the least effect illumination technique, because "this technique creates either light or dark-colored artifacts […] surrounding objects of interest" (Nagy, 2011, p. 15). I think that the illumination techniques brightfield and darkfield are good techniques. Many of the diatom arrangements you see on Google use one of these techniques. "Darkfield imaging may be produced with a simple stop in the light path to the condenser on lower powers or with digital image manipulation by inverting the values of a brightfield image" (Beats, 2015, p. 9). Inverting a brightfield image to turn it into darkfield prevents every spec of dirt or dust from showing up, but the colors aren't as vibrant. White and black values may also be negatively affected if you do this, so based on some experimentation with photographs of diatom arrangements, this process only works for certain arrangements. Using crossed polarizing filters with a darkfield image may also be able to boost the vibrancy of colors. Another type of illumination is as follows:

> Oblique illumination involves adjusting the light path through the condenser so that only a sector of the glass is used. This can cause an interesting three-dimensional effect that looks somewhat similar to the images produced by Differential Interference Contrast after Normanski. (Nagy, 2011, p. 15)

"Hoffman Modular Contrast may be regarded as a type of oblique illumination and cannot be used with objectives of a higher degree of correction than Planochromats, thus limiting the resolving power of the optical system" (Nagy, 2011, p. 15). Nagy believes that interference illumination techniques produce the best photographs of diatom

arrangements. “Normanski Differential Interference Contrast may be adjusted to provide a pseudo-darkfield image, or adjusted so that any of the interference bands may serve as a brilliant color background to one’s image” (Nagy, 2011, p. 16). Focus stacking with s shallow depth of field can also be used to improve the overall quality of the photograph. Five to ten shots would be needed. Illumination is going to be another challenge for me, because I’m only really comfortable with brightfield. I have done darkfield though too, but I’m not as comfortable with it compared to how comfortable I am with brightfield.

Conclusion

Notable diatom arrangers of the past and present, and how to collect, clean, arrange, mount, and illuminate diatoms in order to create a photograph of a diatom arrangement are all topics discussed in this paper. This project will be a huge learning process for me, because I have photographed diatoms before, but they were still alive. I need to be flexible and be willing to try new things if one method doesn’t work out. In order to have a good photograph at the end I need to be able to have properly collected and cleaned samples, the right material, and proper illumination. Because of this I will not move onto the next process until I have each process down. I’m not sure how I want to display the finished product, but like Beats “I hope [to encourage] someone to try diatom arranging for themselves. [It pains me that] the art will die out if people don’t join in” (Beats, 2015, p.16).

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Mentor Choices

1. Professor Ted Kinsman
2. Professor Michael Peres
3. Professor Dan Hughes