Five things that will damage your pH probes

Since pH is one of the most important variables to control in hydroponic culture almost all hydroponic growers have and use pH probes. There are however several things that can go wrong with these probes due to the very nature of the sensor and the way in which other substances interact with it. Today we will learn about some of the worst things that you can do to your pH probes and how you can potentially avoid these issues.

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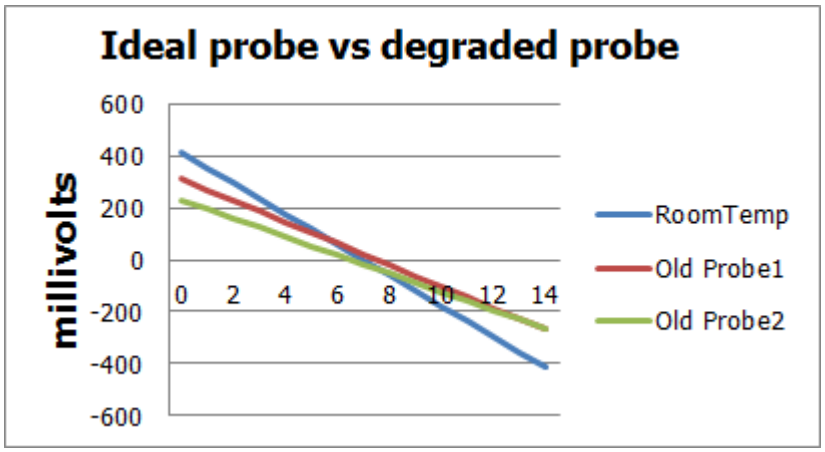

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1. Let them dry. These probes are made from glass and the readings depend on the potential difference between the inside and outside surfaces of the sensor. These are determined mainly by pH since hydronium (H3O+) ions interact strongly with the glass surface. For measurements to be accurate the surface needs to be in equilibrium with the media that is being measured. If you let the electrode dry then the hydration of the surface will be lost and the equilibrium state will be much harder to achieve (a dry probe should be placed in a KCl solution for at least 4 hours before being used). Any junctions within the probe might also dry which will require further stabilization before the probe can be

used . Dry pH probes are therefore a big no no.

2. Keep them in water. Although keeping pH probes in water is better than letting them dry this has a similar effect in that it alters the composition of the glass with time. Since the solution around the probe is much more diluted, with time ions in the glass will have no problem migrating away from the probe, creating defects within the glass that will mess with your sensor's calibration. Ideally you will want to store your pH probes in a concentrated KCl solution (usually around 150-300g/L) which will prevent any of these migration effects and will ensure that your probe remains stable in the longer term. If you buy KCl you can use distilled water to prepare your own pH probe storage solution.

3. Measure very basic solutions. Since pH probes are made of glass and glass is mainly made of silicates this means that basic solutions will tend to react with your pH probe. When the pH goes above 10 a pH probe will start to dissolve in solution, completely altering the surface and making the sensor lose calibration very quickly. In general avoid measuring the pH of any solution above 10 so that this effect can be kept to a minimum.

4. Measuring solutions with chemicals that react with glass. Besides basic solutions – where hydroxide ions dissolve glass – there are a variety of substances that can affect the performance of pH probes by reacting with the glass. This includes solutions containing silicate species and solutions containing fluoride ions. If the solution has ions that can react with glass then the pH probe's lifetime will be diminished and much more frequent calibration will be required. Try to avoid long term measurements of solutions containing large amounts of these ions and beware that weekly calibration might be necessary.

5. Not cleaning the probe. When measuring solutions such as hydroponic nutrient solutions the pH probe is usually

subjected to an environment filled with potentially microorganism contaminants. If the probe is not properly cleaned then microbes can form a biofilm over the glass that will seriously affect the accuracy of pH readings. A probe can be cleaned with a bleach or hydrogen peroxide solution to remove these contaminants but the probe will then need to be recalibrated as the film will have effectively changed the glass surface to some extent.

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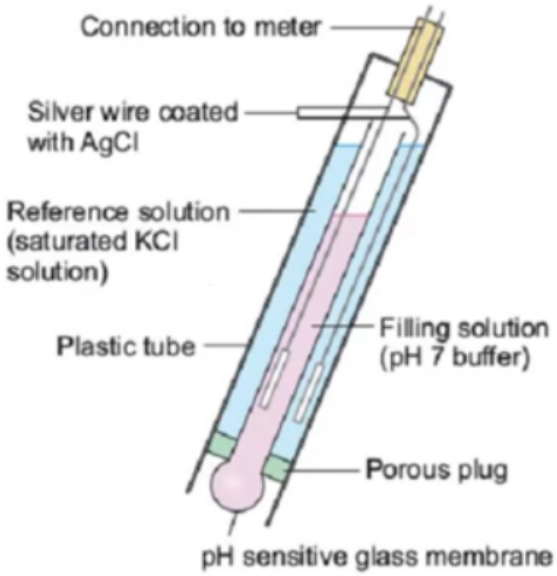

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Having pH probes that give accurate reading for a long time is not a difficult task if you take proper care of your sensors. Storing them adequately, ensuring they are not exposed to harmful conditions and cleaning them ensures that they will last for a much longer time. IF you keep track of your pH sensor calibrations you might notice changes in the calibration slope – as in the first image in this post – which indicates a loss of sensor sensibility (the slope becomes less pronounced). You can use a sensor until around 20% of the sensor’s sensibility is lost, time after which you’ll need to buy a new probe.

There are also several sensors that can be used for long term

continuous measurements – which are made in a much more robust manner – we will talk about industrial quality and in-line sensors in a future post.

Vapor pressure deficit (VPD) in hydroponics

If you have read books or articles about greenhouse environmental control you have probably heard about Vapor Pressure Deficit, also known as VPD. This is an important variable to measure as it helps us understand the conditions our plants are facing, gauge their water use and even predict whether we will be getting better or worse yields. Today I am going to talk about vapor pressure deficit in hydroponics, what this variable means, what it takes to control it and why it is so important to understand and even change this value to obtain better results.

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Table 3. References VPD (kPa) for greenhouse production, recommended by Argus Ltd, (2009)

T(°C)	rH (%)													
	35	40	45	50	55	60	65	70	75	80	85	90	95	100
15	1.11	1.02	0.94	0.85	0.77	0.68	0.60	0.51	0.43	0.34	0.26	0.17	0.09	0
16	1.18	1.09	1.00	0.91	0.82	0.73	0.64	0.55	0.45	0.36	0.27	0.18	0.09	0
17	1.26	1.16	1.06	0.97	0.87	0.77	0.68	0.58	0.48	0.39	0.29	0.19	0.10	0
18	1.34	1.24	1.13	1.03	0.93	0.83	0.72	0.62	0.52	0.41	0.31	0.21	0.10	0
19	1.43	1.32	1.21	1.10	0.99	0.88	0.77	0.66	0.55	0.44	0.33	0.22	0.11	0
20	1.52	1.40	1.29	1.17	1.05	0.93	0.82	0.70	0.58	0.47	0.35	0.23	0.12	0
21	1.62	1.49	1.37	1.24	1.12	0.99	0.87	0.75	0.62	0.50	0.37	0.25	0.12	0
22	1.72	1.59	1.45	1.32	1.19	1.06	0.92	0.79	0.66	0.53	0.40	0.26	0.13	0
23	1.82	1.68	1.54	1.40	1.26	1.12	0.98	0.84	0.70	0.56	0.42	0.28	0.14	0
24	1.94	1.79	1.64	1.49	1.34	1.19	1.04	0.89	0.75	0.60	0.45	0.30	0.15	0
25	2.06	1.90	1.74	1.58	1.42	1.27	1.11	0.95	0.79	0.63	0.47	0.32	0.16	0
26	2.18	2.02	1.85	1.68	1.51	1.34	1.18	1.01	0.84	0.67	0.50	0.34	0.17	0
27	2.32	2.14	1.96	1.78	1.60	1.43	1.25	1.07	0.89	0.71	0.53	0.36	0.18	0
28	2.46	2.27	2.08	1.89	1.70	1.51	1.32	1.13	0.94	0.76	0.57	0.38	0.19	0
29	2.60	2.40	2.20	2.00	1.80	1.60	1.40	1.20	1.00	0.80	0.60	0.40	0.20	0
30	2.76	2.54	2.33	2.12	1.91	1.70	1.48	1.27	1.06	0.85	0.64	0.42	0.21	0
31	2.92	2.69	2.47	2.24	2.02	1.80	1.57	1.35	1.12	0.90	0.67	0.45	0.22	0
32	3.09	2.85	2.61	2.38	2.14	1.90	1.66	1.43	1.19	0.95	0.71	0.48	0.24	0
33	3.27	3.02	2.76	2.51	2.26	2.01	1.76	1.51	1.26	1.01	0.75	0.50	0.25	0
34	3.46	3.19	2.92	2.66	2.39	2.13	1.86	1.59	1.33	1.06	0.80	0.53	0.27	0
35	3.65	3.37	3.09	2.81	2.53	2.25	1.97	1.69	1.40	1.12	0.84	0.56	0.28	0

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Vapor pressure deficit – measured in kPa – basically measures how much water vapor pressure we would need to put into a room with a certain humidity and temperature to get it to the point where relative humidity would be 100%. The larger the VPD the more water you need to put into the air to get it to saturate while the lower the VPD the closer the air is to full saturation. Since air holds more water with increasing temperature this means that at a fixed relative humidity the VPD is directly proportional to the room's temperature. This simply means that the hotter the room, the higher the VPD and the colder the room, the smaller the VPD if humidity remains constant.

The problem with a very low VPD – room close to 100% humidity – is two fold. First, it's difficult for any organism to evaporate water and second, it's easy for water to condense on any surface it temperature drops just a bit. For humans this basically means having to wear a t-shirt soaked with your own sweat but for plants this means both an inability to cool their surfaces and an inability to transport nutrients to their leaves. A low VPD generates a lot of stress because it makes plants unable to properly transport water.

A high VPD is equally problematic as it means that the plants need to transpire a lot. If air can hold a lot of additional water vapor this means that plants will lose more water through their stomata and this permanent loss puts pressure on the roots to transport more and more water. If root mass is not large enough or water availability is not high enough then plants will face important problems and will simply tend to wilt as the air takes away more water than what the plant can effectively transport through its tissues. You can actually often create models using VPD to predict a crop's water usage (see image below).

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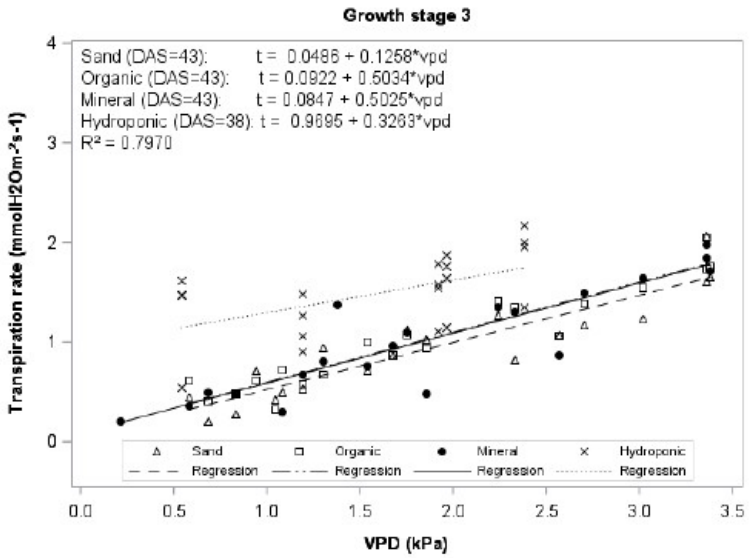

Figure 7: Transpiration rate in Panicum (*Panicum maximum* cv. *tanzania*) under VPD with different substrates in growth stage three.

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The first graph in this post (which I took from this study on tomatoes) shows the optimum VPD – in green – as a function of humidity and temperature for greenhouse production of tomatoes. In general a range between 0.5 and 1.1 seems to work best but the window under which these conditions are possible becomes narrower as temperature increases. Ideally we would want to be somewhere around 20-25°C where we should sustain humidity values between 65-70%. This would give us a VPD value between 0.7-0.8 which is around what is commonly held to be most beneficial for greenhouse crops under normal conditions.

However optimal VPD can also change depending on lighting conditions and other sources of supplementation. For example the optimal VPD during the day is usually higher than the optimal VPD during the night. In general it's better to have a drop in VPD during the night relative to the VPD that is maintained during the day. Declines in canopy carbon dioxide exchange rates can be correlated with increases in the VPD during this time (see here for a study about this on soy bean). If you're supplementing carbon dioxide – which puts further transpiration stress on the plants – the optimal VPD is also likely to be lower than if you didn't use any

supplementation at all (you can see a practical application of this here).

Changing the VPD can be a challenge but under closed environments it is much easier to do. You can reduce the humidity using a dehumidifier to increase your VPD and you can use a humidifier to increase your VPD. Ideally you will want to use an AC unit to keep your temperature at exactly the value you want it to be and you can then use a humidifier/dehumidifier to control the exact point where you want your VPD to be by controlling the value of your relative humidity at the fixed temperature provided by the AC unit.

Maximizing yields per area in hydroponics

Since the 1940's hydroponics – which I use to talk about a broad variety of soilless growing methods – have promised to deliver better plant yields than soil culture with less water usage and higher fertilizer efficiency. However there are many different types of soilless cultures that vary in their initial cost, media used, irrigation method used and potential for yield. Today I want to talk about the decisions that need to be made if you want to maximize yields in a hydroponic crop and the compromises that you may have to make in order to get there.

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There are mainly two ways in which yields can be increased in crops. The first is to increase the amount of production you can achieve per plant and the second is to increase the amount of plants you can have per area. Maximizing crop production requires using methods that allow you to reach the best compromise between these two, maximize the product of plants per area with production per plant. This often means not having the largest amount of plants you could possibly grow per square meter and not having the largest possible yields you could have per plant.

Assuming that plants are getting adequate lighting and carbon dioxide there are two things that can be done to maximize the amount of yield per plant. The first is to ensure that plants can get optimum contact with nutrient solution as often as possible. This means that nutrient solution should always be saturated with oxygen and that irrigation should happen as often as possible. This ideally means that the media should not allow for any waterlogging but should allow the solution to flow freely and constantly. The second is that the nutrient solution should contain adequate amounts of all nutrients – all within the plant's sufficiency ranges – with adequate temperature, pH and EC values. The optimum nutrient ratios in solution depend on the plant being grown and they can play a

substantial role in getting better yields per plant, especially in flowering crops. Here are some scientific articles with some experiments about some of the above (1, 2, 3, 4, 5, 6).

A typical problem when maximizing yields per plant is that this immediately means larger energy expenditure. It often means close to constant irrigation systems with highly efficient oxygen pumps. It also means potentially more expensive media – such as expanded clays or rockwool – with closed systems where solutions need to be closely monitored. Systems of this sort are more vulnerable to power outages and they are much less forgiving with grower mistakes. Plants are much more dependent on the ideal conditions being created around them and deviations from these conditions can eliminate any potential advantages that were obtained when going for this system class.

Our next area of yield maximization is to increase the number of plants per area. To do this we basically need to increase two things: light and ventilation. The main limiting factor in having more plants is the light that they can receive so either changing to systems where light can be better distributed – such as growing towers – or using more lights can alleviate this problem. Some growers have even used high power LED strips between plants to fix this issue. Since plants also absorb carbon dioxide around their leaves we also need to ensure we have stronger ventilation to ensure none of our plants are getting starved. Increasing plant density also increases the propensity of our plants to catch and transmit diseases so environmental manipulations like lower humidity are often coupled with increases in density to decrease these risks. See these articles for more on yields, light and density (1, 2, 3, 4).

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Increasing plants per area automatically decreases yields after one point but it is often the case that you can get larger final yields per area by compromising some yield per plant in the process. Even if plants yield 10% less this might be worth it if you can include 2 more plants for every 10 within your hydroponic crop. The key to maximizing yields per area is to find how far you can push this before getting substantial issues that may dramatically decrease plant yields.

It is worth noting that steps taken to maximize yields are also often steps taken in making the crop more susceptible to problems. While lower yielding setups, like for example run to waste setups with sparse plant density, are often easy to manage and very forgiving, more technical setups like closed loop constant irrigation systems at high plant densities can be much better yielding but much more prone to problems, requiring much closer monitoring and attention. This is why many growers might get better yields with setups with lower yielding potential, because their mistakes are punished much less harshly under these conditions.

A few basics of leaf tissue analysis in hydroponic crops

Adequate nutritional control is difficult. Although there are several tools to control your plant's chemical environment – such as pH, EC and ORP – in the end the main interest we have is to control the composition of plant tissue and how this composition affects plant development and yields. One of our sharpest tools to achieve this is leaf tissue analysis which allows us to look at plant composition levels and figure out if anything is wrong with our plants. Today I want to talk about this powerful tool, why it is not so simple to use, how to use it and why it can be so important in helping you figure out what's wrong with your crops.

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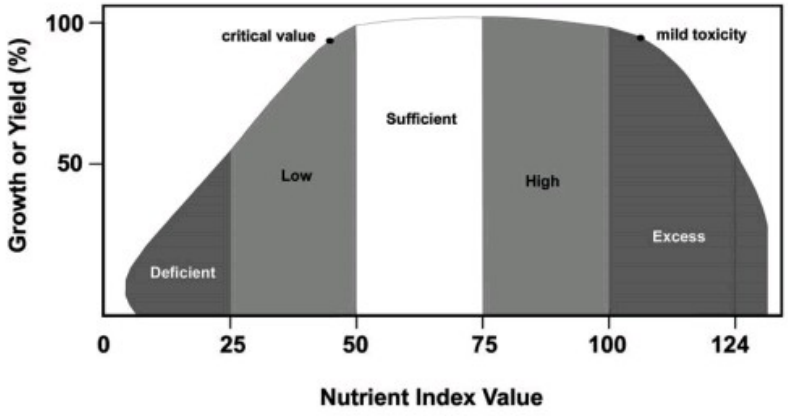

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The general model for nutrients and crop yields explains that plants will absorb nutrients till a point of maximum yield. After this point increasing nutrients will not increase or decrease yields substantially for a while but after a given point toxicity will prevail and plant yields will decrease due

to nutrient toxicities and potentially osmotic pressure issues. This model is simplistic as it leads to an overall linear understanding of plant nutrients which is why growers often find leaf tissue analysis puzzling and confusing.

In leaf tissue analysis we most commonly obtain a sample from the plant's most recent mature leaves. This tissue is analyzed by a lab and we obtain a chart where the percentage composition of the plant tissue for the different elements is given. We can then look at reference values for healthy plants and if any of our nutrients are outside this range then there is certainly something wrong with our crop's nutrition. Sometimes the lab will also give you some reference values but bear in mind that this aren't necessarily healthy plants but the average of what the lab gets for the plant specie you are growing. You either want an academic/government reference for healthy sufficiency ranges or you want to grow healthy plants yourself and take a reference sample to use for your future crops.

The tricky part is to interpret the tissue analysis. For example let's suppose that your tissue analysis comes up with low phosphorous. The immediate intuitive response that we get from the general model of nutrient sufficiency is that we should increase P in solution to get the P up within the leaves. However nutrient relationships are non-linear and in many cases what you have isn't a general lack of enough nutrient in solution but a problem getting that nutrient up to the leaves. In the case of P for example it might range from having excess chloride to having a nutrient solution that is too cold. I haven't seen a single case in hydroponics where low P in leaf tissue has actually been due to low P in the nutrient solution.

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Most Recent Mature Leaf — All Growth Stages

Macronutrients					
N	P	K	Ca	Mg	S
3.5–5.0%	0.30–0.65%	3.5–4.5 %	1.0–3.0%	0.35–1.0%	0.2–1.0%

Micronutrients					
Fe	Mn	Zn	Cu	B	Mo
50–300 ppm	25–200 ppm	18–80 ppm	5–35 ppm	30–75 ppm	0.1–1.0 ppm

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It is important then to realize that a problem within leaves is not necessarily a problem with concentration for that specific nutrient being too low or high in solution – in my experience it rarely is – but more so a problem with the balance of nutrients in solution or the environment that is causing a nutrient absorption issue. There are all sorts of antagonic and synergistic relationships between the different nutrients and the environment that will make this hard to interpret in many cases. To know what might be the cause first you will want to address all environmental issues that are known to cause toxicities/deficiencies and then look into addressing nutrient issues relative to the solution. You will want to pay a lot of attention to ratios instead of absolute concentrations.

You can have a perfectly good nutrient solution and the absorption problem might be related with something like transplant stress, root pathogens, incorrect carbon dioxide supplementation, light issues, temperature/humidity problems, etc. Growers tend to focus on the nutrient solution as the potential source and cure to all plant problems but the key is often in the environment and crop management more than within the actual nutrient solution. Even when the cause is the nutrient solution growers often misdiagnose the problem and increase or decrease nutrient concentrations, more often than not making the problem worse.

Due to the above it is not surprising that few hydroponic growers find tissue analysis very useful. While in soil crops

tissue analysis is usually used to manage fertilization and soil amendments in hydroponics the environment and solution are so controlled that the problems become much more difficult to diagnose and the solutions are often not what you would consider intuitive. It certainly requires a lot of reading and experience to properly interpret leaf tissue analysis and tackle the causal factors that are causing issues in hydroponic crops. However with enough experience or guidance leaf tissue analysis can be a great tool to know what your plant is taking, what it’s not and how these issues can be fixed.

Managing a Run To Waste (RTW) hydroponic crop from a nutritional perspective

Today it’s very common to create hydroponic crops using techniques where nutrient solution is not recycled. This type of crop, commonly called drain-to-waste (DTW) or run-to-waste (RTW) offers the advantage of having a very cheap setup – since no recirculation is used – with the big disadvantage that nutrient control becomes harder as there is no constant feedback of how the plants are affecting the nutrient solution. Today I want to talk about the main differences between a RTW crop and a recirculating crop and how nutrient management needs to be done in order to be effective in RTW setups.

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One of the most important difference between both crop types is the substrate. In a RTW crop you want the substrate to have a much higher capacity for water retention since you want to irrigate less frequently and ensure the plants are in contact with nutrient solution as much as possible without having aeration problems and a lot of run-off. Having a lot of run-off means wasting more nutrients in a RTW setup so you want to have a media that can minimize this. This means having a media like peat moss or coco coir where water retention can be very substantial.

The problem with the above is of course salt accumulation within the media. If you irrigate the plants with a full strength nutrient solution and you irrigate when the level of moisture descends then this is both because the plants have absorbed water and the solution has evaporated to some extent. This means that the next time you irrigate your total amount of salts will be the amount from your current irrigation plus the amount accumulated in the media. This can quickly turn into a very problematic situation where the plants are subjected to extremely high conductivity levels.

This is why run-off monitoring is key in RTW setups. You usually want to water your plants enough to allow for some run-off – usually 10-20% of the plant container's volume – so that you can perform measurements of pH and EC over that run-off. This is why it's so important to have the plants over

trays where run-off can be collected as measuring the run-off is very important to ensure that your plants are receiving adequate nutrition. Measuring the run-off of every plant is impractical so collecting the solution from many plants in a single tray and then measuring that output is a lot easier. Alternatively – if you cannot place the plants on trays – you can use a suction lysemeter to take out solution from a few plants after watering to monitor conditions around their root zone.

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In a RTW setup your run-off will always tend to be more concentrated than your input – this is normal – but you want to have conductivities in your run-off no more than 30% greater than your input concentration. If this is the case you should do plain water irrigation until your run-off has at least the same conductivity as your originally desired input. When you water with plain water also make sure you adjust the pH of the water to the value you desire. One of the lead causes of bad results in RTW setups is to have salinity build-ups that cause nutrient lock out simply due to a general lack of run-off monitoring. In general if watering using conductivities close to or above 2 mS/cm plain waterings should be done once for around each 1 or 2 nutrient solution feedings.

The pH is also very important. Depending on your media your pH can change substantially between your input and your run-off but in general you want your output pH to be as close to the desired pH as possible. You can compensate a bit by changing the pH of your input solution – for example if your run-off pH drops you can increase the pH of your input solution – but never increase your input pH above 6.5 or below 5.5. Some

media like peat can acidify solutions a lot with time, in these cases it’s very important to pretreat the media to avoid these problems with output pH. A strongly buffered input solution can also help in these cases. *Before starting your crop always test the run-off pH/EC of the media without plants to ensure you can make any needed treatments before you actually start your crop.*

The key to successful RTW setups from a nutritional perspective is run-off monitoring. Once you start monitoring your outputs you will see how your plants respond to your input solution and you’ll be able to better control the plants’ root zone environment. Of course these issues are all eliminated by recirculating setups since in that case the nutrient solution returns to the tank and there is a constant feedback of how the plants are affecting the solution. This can make recirculating setups much better at giving higher yields.

Using coco coir in hydroponics

Side by side with peat moss, coco coir is one of the most commonly used media in hydroponic culture. Its excellent root propagation and aeration properties, coupled with its adequate water retention, make it an ideal medium for hydroponic culture. Nonetheless, there are several issues that can arise when using coco coir, particularly due to its chemistry and variability. Today we are going to talk about using coco coir in hydroponics, what the main problems with coco can be and how these problems can be avoided.

TABLE 11.7 Levels of pH and EC in Coir

pH	EC ($mS\,cm^{-1}$)	Reference
4.9–6.4	0.17–2.32	Noguera et al. (2003b)
5.6–6.9	0.13–1.26	Evans et al. (1996)
4.8–6.8	0.32–0.97	Meerow (1994)
5.5–5.7	0.80–1.90	Handreck (1993)
5.0–5.7	0.12–1.51	Prasad (1997b)
4.9–6.6	0.32–0.41	Smith (1995)
6.0–6.7	0.2–0.4	Kipp et al. (2000) (coir dust)
5.9–6.1	0.2–0.9	Kipp et al. (2000) (coir chips)

Values of Noguera et al. (2003b), Evans et al. (1996), Meerow (1994) are based on saturated media extract, Smith (1995) on 1:5 water extract, and the others on 1:1.5 water extract. EC values have been converted to 1:1.5 water extract (see text).

EC and pH values for different coco coir sources

Coco coir is basically ground up dried palm tree husks. Although it is organic, it is much more fibrous than peat moss and for this reason, it does not suffer from some of the pH and decomposition issues commonly found with peat. Although coco is biodegradable, its decomposition can take more than 20 years, reason why it is a suitable media for hydroponics. It can even be used several times within a hydroponic crop in order to save production costs, as long as plant material is removed and the media is properly treated between crops.

Since coco coir comes from large plants grown across a variety of different conditions, the actual chemical makeup of the coco can change very substantially. The table above shows the pH and EC of different coco coir sources. As you can see, we have everything from an EC of 0.1 mS/cm to an EC of 0.9 msS/cm, with pH values that cover anything from 4.9 to 6.8. This is mainly due to the big variations in the ions contained within the coco and how these ions interact with the plant material.

Coco coir also has a high cation exchange capacity, meaning that it can retain large amounts of ions. These are only taken out if they are replaced by others with stronger affinity for the media or when strong interactions with chelating agents are possible. This is generally why coco is treated with calcium nitrate solutions, to remove many of these ions from the media structure and allow the media to be as neutral as possible when used in hydroponic culture. However, many coco

producers do not treat the media at all – or simply wash it with plain water – leaving a lot of potassium and sodium within the coco that needs to be accounted for. A lot of micro nutrients that are tightly bonded to cation exchange sites are often also often present inside the coir.

TABLE 11.9 Chemical Properties of Coir ($CaCl_2$/DTPA Extractable Macronutrients) ($mg L^{-1}$)

P	K	Ca	Mg	Na	Reference
–	183–222	100–172	36–58	85–92	Kipp et al. (2000) (coir dust)
–	47–98	56–60	31–79	30–78	Kipp et al. (2000) (coir chips)
8–17	304–720	6–15	8–28	110–114	Handreck (1993)
	69–128				Prasad (1997b)

TABLE 11.11 Chemical Properties of Coir ($CaCl_2$/DTPA Extractable Micronutrients) ($\mu g L^{-1}$)

Fe	Mn	B	Zn	Cu	Reference
	1100–1500	120–180	700–1300	170–2200	Handreck (1993)
79–157	814–1540	66–77	429–527	0–6	Kipp et al. (2000) coir dust
45–140	484–561	66–154	364–552	240–448	Kipp et al. (2000) coir chips
4100–7700	900–5000	200–400	500–1100	100–300	Prasad and Maher*

*Prasad and Maher unpublished data.

Some of the chemical properties of different coco coir sources

If you want to ensure your coco is as neutral as possible in terms of nutrients, you can extract it with a 1 g/L solution of calcium nitrate and then with 2g/L of tetrasodium EDTA. This will extract both macronutrients that are exchangeable for Calcium, and micro nutrients that can be extracted when using EDTA. The EDTA step is important, as coco can hold a large amount of micro nutrients within it, that can be exchanged and used by the plant. If you want your nutrients to all come from solution you will need to remove these contributions from the media. After this, you will then want to run plain water to remove any excess Ca and EDTA and then run your full strength nutrient solution for a few days. This will strip the coco from excess ions and equilibrate the cation exchange sites with your nutrient solution's composition.

Note that these steps aren't necessary to grow successfully with coco, but they can give the grower more control over the nutrients received by the plants. You can alternatively run

nutrient solution through the coco and then perform an analysis of the output, so that you can compensate for the nutrients that are given by the coco through the growth cycle. This of course means that you need to spend money doing solution analysis through the crop’s life to ensure that you’re adequately compensating for the coco’s contributions through the entire growing period.

When properly treated, coco can be a very good media for growing hydroponic crops. The larger aeration, better chemical stability and fibrous structure makes it better for root growth than most peat moss sources. Yields for several plants are also often larger or just as good in coco when compared with peat moss. The lack of important decomposition during growth cycles is also a big advantage over peat, as important drops in pH due to media decomposition can be avoided and the media can be more readily recycled.

Measuring ion concentrations in hydroponics using electronic tongues

One of the biggest problems in hydroponic research is the measuring of individual ion levels in hydroponic solutions. Right now there is no commercial solution for the accurate tracking of individual ions in hydroponic solutions and this makes it impossible to track ions in real-time to measure how nutrient absorption reacts to different environmental and chemical conditions. The only way to currently do this is to carry out more expensive and cumbersome ICPE analysis that provides a snap shot of a solution’s composition in time.

However there is a solution that might be coming up within the next few years which is the use of electronic tongues to measure the concentration of a large variety of ions in solution.

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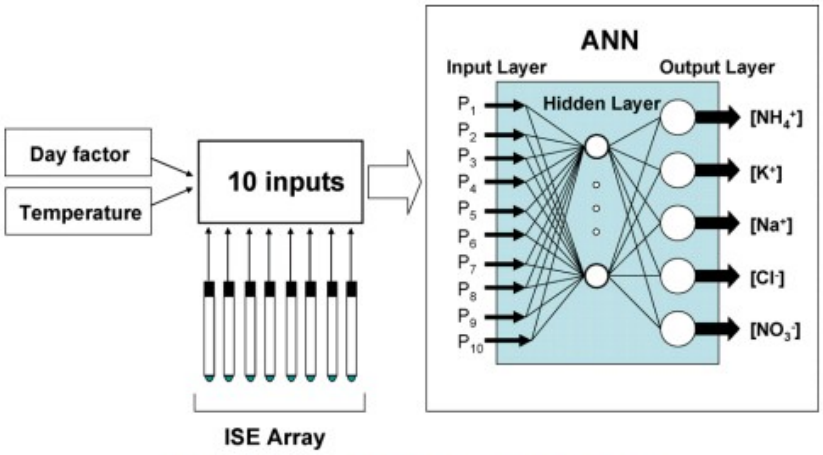

Fig. 2. Schematic representation of the electronic tongue approach.

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Many of you may be thinking, what about Ion Selective Electrodes (ISEs) ? These electrodes are designed to measure the concentration of individual ions in solution and they are perfect when you’re trying to measure a single ion against an unchanging background. The issue with ISE is that they work via the interaction of ions with molecules that have a especially strong interaction with them (what we can an ionophore) but the interaction can also be strong with other ions, generating interference. For example a K+ ion selective electrode usually uses an ionophore like vancomycin but this ionophore also has strong interactions with NH4+ (ammonium) ions. Since the concentration of ammonium also changes with time in hydroponics this means that your reading will be changed not only by how K+ concentration varies in solution but also by how NH4+ concentration changes.

In reality interference is not generated by a single ion but by a good portion of the ions present in a hydroponic

solution. This means that it is practically impossible to use an ISE in an accurate manner in hydroponics because you will always be getting changing interference from the other ions in solution. In the experiments I have done attempting to track nutrients using ISE this problem has always been so bad that the results become practically useless, regardless of how you calibrate the electrodes (since the concentration of the ions that interfere changes relative to the ion you want to monitor).

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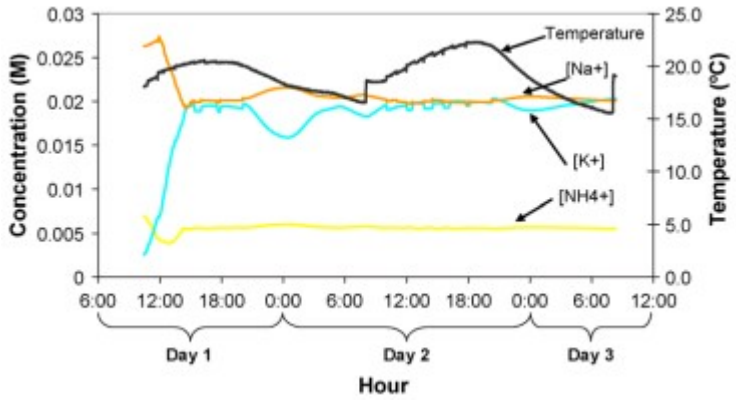

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Electronic tongues are an intelligent idea to circumvent these problems. The idea is to use many ISE for different ions – with many for the same ion and many generic ionophores that have poor selectivity – and then to use statistical modeling tools – mainly neural networks – to come up with ways to figure out the noise/signal/interference and get accurate measurements for ion concentrations regardless of what the actual readings of the electrodes are. The neural network is trained with data from solutions with varying concentrations of all the ions being monitored and this allows the creation of a robust prediction engine that can be used to get actual ion concentrations. M. del Valle's group in Barcelona has done some of the pioneering work in this area, the images in this post have been taken from some of their research papers on the subject (for example this one and this one).

Through this research they have been able to come up with ISE arrays that – using the neural network models – can measure concentrations in real-time for nitrate, chloride, sodium, potassium, ammonium, calcium, magnesium and phosphate. This means that you can effectively monitor how plants absorb different ions, not only allowing you to carry out experiments surrounding nutrient absorption but also allowing you to know which ions are getting depleted so that you can replace them. This brings a totally new dimension into hydroponic culture that simply isn't accessible right now.

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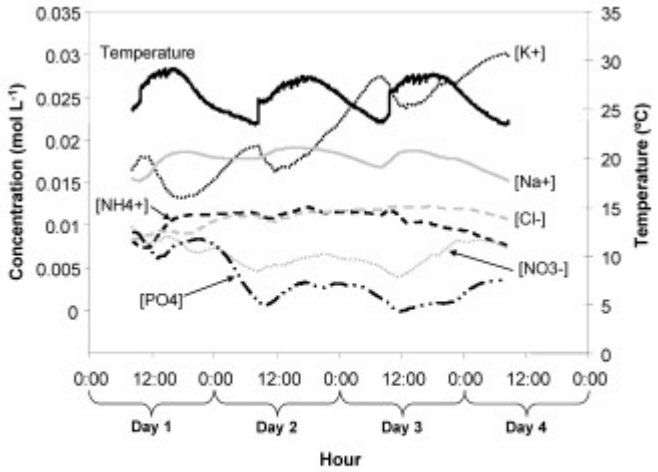

Figure 3. Representation of the concentration values predicted by the electronic tongue during the second application, in summertime, for the considered ions, ammonium, potassium, sodium, chloride, nitrate, and phosphate, in the nutrient solution during 3 days of continuous monitoring.

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It might take a significant time for these sensors to reach commercial applications – mainly due to the expensive calibration that is needed due to the variability in fabrication – so it might be years before we see something like this available to the general public. However if you have a commercial hydroponic setup that is large enough you definitely can follow this research to make your own ISE array and build an electronic tongue with them. This will give you access to a ton of information that is inaccessible to all of your competition.

Using Peat Moss in Hydroponic Culture

There are several different types of media available for hydroponic culture and from these peat moss is one of the most popular due to its low cost and high availability in some countries. This media is made up of decaying mosses and is used mainly in drop irrigation systems of both a recirculating and non-recirculating nature. However the organic nature of the media provides several important challenges to the hydroponic grower which – when not controlled – can lead to important problems associated with nutrient availability, inhibiting plant growth. Today we are going to talk about the characteristics of peat moss as well as how we can amend this media to make it suitable for hydroponic cultivation.

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TABLE 11.2 The von Post Scale for Assessing Degree of Decomposition of Peat (von Post, 1922)

Degree of decomposition (H)	Quality of water exuded	Proportion of peat exuded
1	Clear, colourless	None
2	Almost clear, yellow brown	None
3	Slightly turbid, brown	None
4	Turbid brown	None
5	Very turbid, contains a little peat in suspension	Very little
6	Muddy, much peat in suspension	One-third
7	Very muddy	One-half
8	Thick mud, little free water	Two-thirds
9	No free water	Almost all
10	No free water	All

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Peat’s main characteristic is its organic nature. Since it is made up of decaying organic matter this means that the chemical nature of the media will change depending on the degree of decomposition of the media and also depending on the

particular moss species that were used to produce the peat moss. You can know the degree of decomposition of a peat moss sample by using a simple procedure. Place a handful of wet peat in your hand and then squeeze it, the result – how the exuded water looks and whether peat is squeezed between your fingers – will tell you all about your peat. The von Post scale – developed in the 1920s – will then allow you to tell how decomposed your media is in a scale from H1 to H10.

Highly decomposed peat will tend to remain more chemically stable as the organic decomposition process has already been carried out. For this reason you want to buy what is commonly known as “black peat” (H7-H10) where microbial activity has already dialed down and the peat moss more closely approaches what we would call an “inert media”. This however does not mean that Peat moss is chemically inert at this point as it does contain as a significant amount of substances that can affect your nutrient solution.

One main characteristic of peat is that it’s acidic. This means that the pH of untreated peat will usually be between 3 and 4.5, too low for use in hydroponic applications. Peat is generally amended with calcium carbonate (lime) to make its pH go up and remain there but this process can be ineffective if the peat can still decompose very significantly (if you buy peat with decomposition < H7). This also contributes high amounts of Ca into the media which might lead to nutritional problems if Ca is also applied normally in solution. To alleviate these issues peat is also sometimes treated with lime/dolomite mixtures so that the counter-ions are both Mg and Ca. Alternatively – but more expensively – this problem can be solved by using phosphate buffer solutions that are run through the peat for a significant period of time. A potassium monobasic/dibasic phosphate buffer at a pH of 6.5 with a 100 mM concentration can buffer the peat moss. For this the buffer needs to be applied until the run-off pH out of the peat comes out unchanged. Then tap water should be applied to remove the

K/P from the media. Note that this will only work for black peat that's already gone through most of the decomposition process as lighter peats will simply decompose further and acidify the media again.

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TABLE 11.3 Cation Exchange Capacities of Different Peat Types (Puustjarvi and Robertson, 1975)

	Cation exchange capacity (CEC)	
Species or peat type	cmol kg^{-1}	meq L^{-1}
Undecomposed sphagnum moss peat	130	80
Sphagnum sedge peat	110	60
Sedge peat	80	40
Highly decomposed black peat	160	240

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However if all you can get is already treated peat moss then you should run nutrient solution through your peat for a while before putting your plants in to ensure that the peat's cation exchange capacity has already balanced with your nutrient solution's composition, this will also help remove nutrients applied to the peat that deviate the nutrient concentrations from what we want within the media. Peat can have a significant cation exchange capacity as showed in the table above – even more so for black peat – so a commercial source of peat may exchange a significant amount of nutrients with your solution. Peat is also not very good at retaining anions so the media will be unable to supply any N or P which will be leached very easily from the media. This inability to retain anions basically means that they will only be available when the plant is watered, reason why you should take care to correctly monitor moisture in your media to maximize your productivity.

For hydroponics it is therefore best to find untreated black peat and treat it yourself. If this is not possible then try to find unfertilized black peat – which has had only lime but no other nutrients added to it – and then use that. A great

characteristic of peat moss for hydroponics is that its nutritional content is low – allowing great control over the nutrients added through the composition of the nutrient solution – but this advantage is eliminated when the peat moss is filled up with fertilizers by companies that produce it for non-hydroponic purposes.

If you're using black peat also make sure to check how the peat behaves when watered, if the peat compacts too much you might want to add some perlite to your peat to increase the aeration of the media and prevent excessive compacting from happening. Add perlite until you get the desired balance between aeration and moisture retention. This is not necessary with all black peat sources but it can often be required.

Automated media moisture monitoring in hydroponic crops

> Irrigation control is one of the most important things to control in a hydroponic crop. Irrigate too frequently with a media that has high water retention and your plants will start to wilt as their roots die due to lack of oxygen and reductive conditions, water too sparingly and your plants will not grow as much as they could and maybe even die from the drought conditions you're imposing on them. On today's post we will discuss the topic of irrigation, more importantly how to know when to water your crops and how to control this process using sensor based approaches instead of just using look-and-feel to determine when to water your plants.

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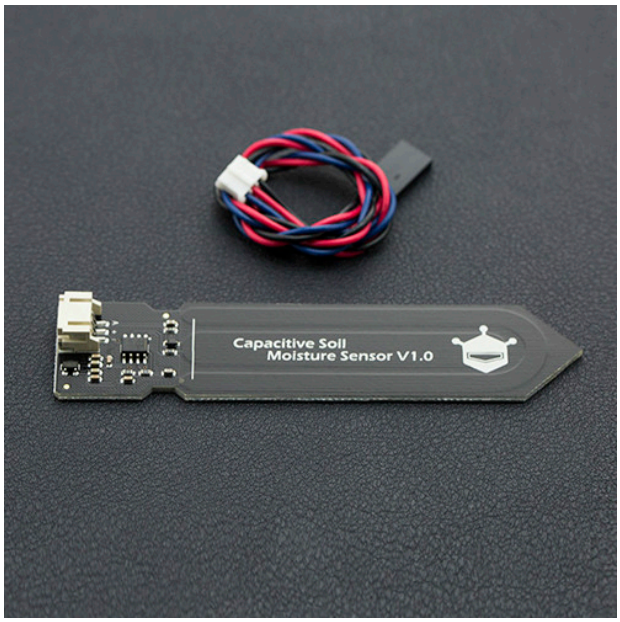

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Plant roots need to have access to water and nutrients. This means that the root zone needs to be saturated with nutrient-rich water as often as possible while avoiding oxygen depletion and salt accumulation. This means that irrigation needs to be controlled to ensure that plants get as much as possible, as often as possible, without going into any excess that would be detrimental to growth. Sadly there is no solution that is true for all crop setups and gauging irrigation frequency requires a close monitoring of what is going on within the crop.

To really know when to irrigate crops you should have a way to properly monitor moisture levels. This can be achieved through several methods, for example with tensiometers or with simple weighting of the plants, but many of these methods are often not cheap or practical for routine practice. Manual inspection of plants can also be misleading since top level moisture perception is subjective and can often lead to very suboptimal results.

In today’s world the best way to monitor moisture without

having to pay a high cost is to use simple capacitive moisture measuring sensors. These sensors are corrosion resistant and independent of salt concentrations in solution and therefore provide you with a good measure of moisture within your root zones without having to worry about the conductivity of the nutrient solutions. My favorites right now are this small capacitive sensor for smaller media containers and the chirp for larger containers. If you don't want the chirp features and just want sensor readings you can also get this simpler I2C sensor for larger containers. Both of these sensors are cheap and can be installed in crops with many plants.

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If you want to go with the simplest possible setup the chirp provides auditory signals when plants need to be watered, although this is not the ideal way to setup the sensors. Ideally you would want to connect these sensors to an arduino so that you can process the data. The arduino mega is particularly well suited for this task as you can connect up to 16 analogue input sensors to it, however you can use less analogue inputs with a normal arduino. Both sensors provide sample code for measuring values from an arduino, you can then output them to an LCD screen or save

them within a computer. You can even connect the arduino's digital outputs to a relay so that you can automatically trigger your irrigation system when a custom set moisture level is reached.

Obviously you do not have to place a sensor within each plant. Just monitoring around 10-20% of your crop will give you enough information to know exactly how moisture levels behave within your crop and when you should ideally water them. This will eliminate all the guessing from your watering and will allow you to water your media perfectly while completely accounting for how long it takes for water to leave your plants. This means you no longer would need to just guess when to water, but your watering will be perfectly tailored to what your media allows and what your plants need.

Hydrobuddy v1.60: A new update with important changes

During this past few weeks I have been working on modernizing Hydrobuddy in order to get it to compile with the latest versions of Lazarus and the Free Pascal Compiler (FPC) so that other people can more easily build the software from source. Today I want to talk about the latest release for Hydrobuddy (v1.6) that comes with some important changes that take the software a step forward and seek to make usage and building of the program much easier. If you're interested in downloading the source or binaries for the new version of Hydrobuddy please visit its official page here.

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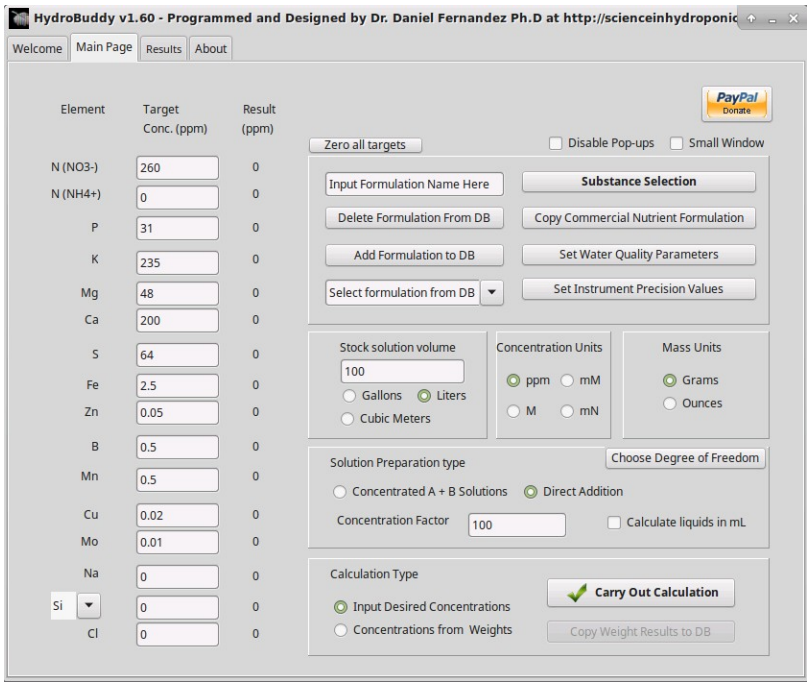

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I have decided to greatly simplify the program in order to remove sections and features that were complicated and really not used very often. This helped eliminate libraries that were previously required, some of which are no longer compatible with the latest version of the Lazarus IDE. I have therefore removed the program's ability to automatically update on startup and have also completely eliminated the data log section of the program. The elimination of the automatic updating makes the program much easier to compile as it eliminates some complicated requirements that were significantly difficult to install for those unfamiliar with the Lazarus RAD environment.

In addition to these changes I have also eliminated the Windows and Linux installers since these two made the overall setup and building process more complicated while they provided little additional benefit. The elimination of the installer means that the program can now be installed by simply extracting a zip file – how it was installed in the very beginning – something that makes it suitable for portable applications while before there might have been permission issues when attempting to run the installers on Windows/Linux.

In addition to the above I have also created separate versions of the program databases for Linux and Windows since these files are not compatible between operating systems and trying to build on Linux/MacOS – with the databases present that were Windows files – caused issues when testing the program. There are now suitable conditional headers that use the appropriate table files depending on the operating system being used.

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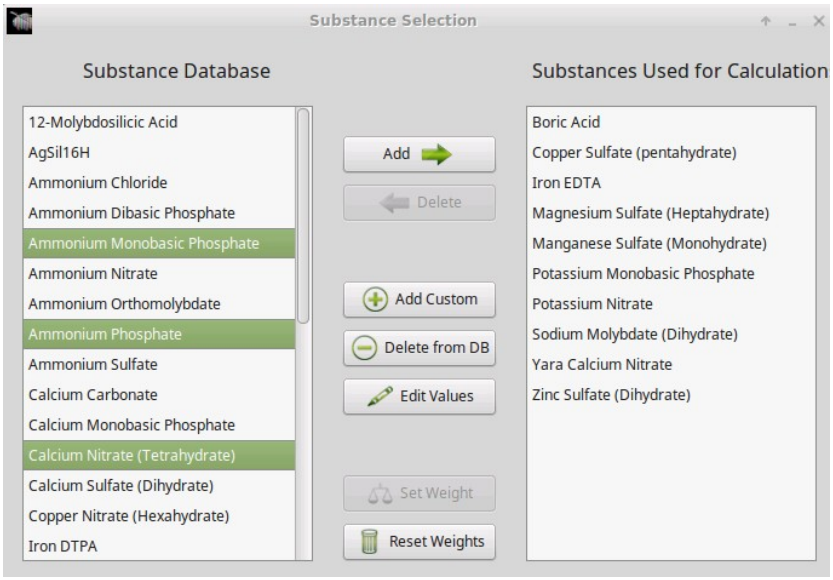

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I have also implemented a few additional features that improve some practical aspects when using the program. The substances used form now allows for multiple selects within the two substance columns so that you can perform multiple substance additions and deletions at the same time. This becomes very useful when you're changing your substance selections all the time since it allows you to easily add/delete multiple substances at the same time. In addition to this I have also implemented a "Zero all targets" button in the home page which basically sets all the ppm targets to zero. This can be very useful when you want to target particular single nutrients or you want to write targets from scratch.

Hydrobuddy's source is also now available via a [github]

repository to enhance the level of contributions from other programmers. This means that others can now checkout the source, modify it and contribute their own code changes to the program so that we can implement additional features or functionality.