# What is the effect of amino acids in hydroponics?

It is very common for hydroponic nutrient manufacturers to add amino acids to their products. They often mention significant benefits that range from strengthening plants to greatly increasing yields or product quality but they rarely mention any peer reviewed evidence studying these effects. Today we are going to look at the use of amino acid applications in hydroponic culture and the effects that amino acids have been shown to have when used in a variety of different crop types. We will see some of the benefits and the problems that they have shown to cause as well and we'll discuss whether it is actually worth it to apply them in a hydroponic nutrient solution.

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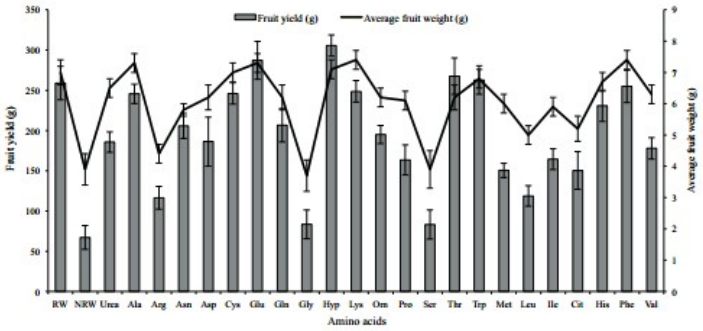


**Fig. 3.** Effect of twenty two amino acids on the yield of strawberry plants grown in non-renewed nutrient solution in closed hydroponic system. RW: renewed, NRW: non-renewed, amino acids are presented as their three letters abbreviation.

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Amino acids – which I am going to use here to refer to L-alpha amino acids – are basically organic molecules that are used as the basic block for protein construction in all life forms. Plants are able to synthesize all the amino acids they need internally while in the case of animals many of these amino acids need to come from other animal or vegetable sources. However since amino acids can be added to nutrient solutions and plants can absorb them (see here) it is interesting to

wonder what the effects they might have.

There are two ways in which amino acids can affect a hydroponic crop. They may be absorbed and used directly by the plant or they may create a chelate with a metal ion and affect that metal's absorption. It is very difficult to separate both effects – except when specific metal absorption studies are carried out – so the effect on yields is generally a combination of these two. The specific amino acids used and their proportion are also critical to these effects as both plant absorption and the stability of metal chelates depend on the exact structure of the amino acids in solution.

There is significant evidence that amino acid applications reduce nitrate assimilation (see here, here and here) this is not surprising given that amino acids compete with nitrate in the nitrogen cycle and may be more readily assimilated by plants. This seems to be especially the case if nitrate concentrations are low and the plants are N deprived. The effect is most important for glutamine, not surprising as glutamate synthesis is basically the mechanism used for ammonium incorporation by plants.

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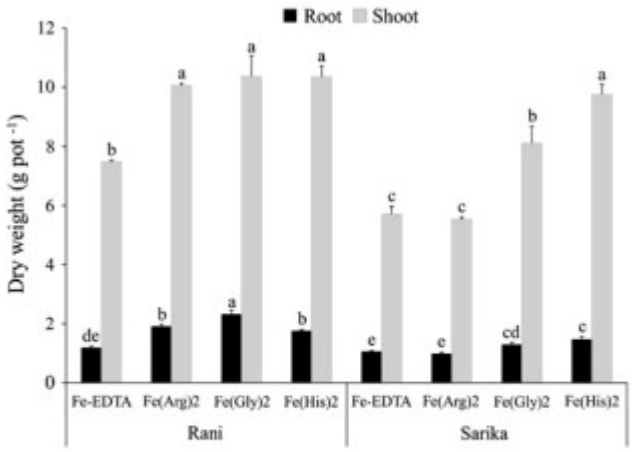
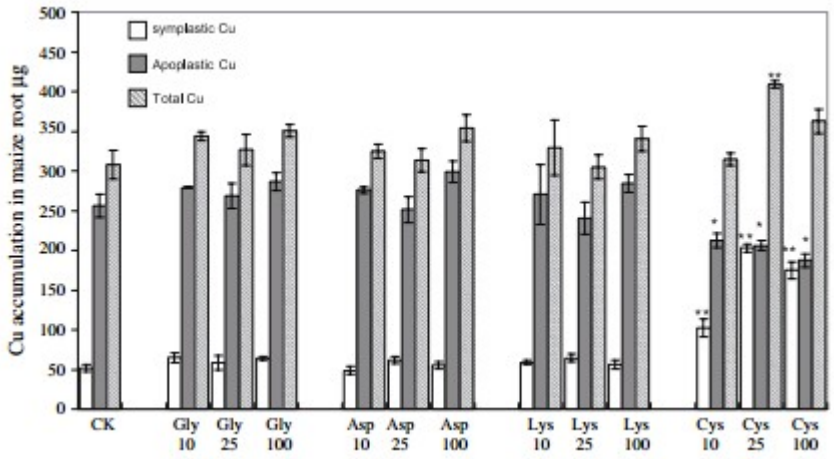



**Fig. 1** Root and shoot dry matter weights of two tomato cultivars grown in nutrient solution containing Fe-EDTA, Fe-arginine [$Fe(Arg)_2$], Fe-glycine [$Fe(Gly)_2$], and Fe-histidine [$Fe(His)_2$]. *Error bars* represent standard error ($n = 3$). *Bars* having different letters in root or shoot are significantly different at the 5% level by LSD

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There is also evidence that amino acids can help plants under stress conditions. For example strawberries in autotoxic conditions – meaning that they have made a nutrient solution toxic after a lot of recirculation – benefited greatly from an amino acid cocktail application (here) and Canola plants have shown to have increased yields under saline conditions with proline applications (here). Plants under heavy metal stress can also benefit from the presence of amino acid, for example rice seedling have shown to benefit from amino acid applications under cadmium stress (here).

There are also limited studies in the use of amino acids as metal chelates in hydroponics. A 2012 study (here) compared different Fe chelates with Fe EDTA and showed that some of these chelates work better than the traditional EDTA chelate in Fe absorption. Fe glycine showed the best absorption across roots and shoots plus the best yields in tomatoes (second image in this post). This shows that Fe glycine may be a good candidate for the replacement of Fe EDTA in hydroponic solutions.  Another study (here) also compared different Cu containing amino acid chelates and found that cysteine may be effectively used for Cu fertilization and phytoremediation.

Is it worth it to apply amino acids in hydroponics? This may depend on the exact conditions the plants are facing. While amino acids have proved beneficial for the assimilation of

specific nutrients – like Fe and Cu – or the alleviation of some stress conditions (salinity, autotoxicity), there isn't any strong evidence suggesting wide range beneficial effects under normal plant growing conditions, especially if these are close to ideal. In normal hydroponic solutions introducing large amounts of amino acids may even have significant negative effects due to their effect on ion absorption and N metabolism. Further evidence is required before general recommendations for exogenous amino acid applications can be made.

This doesn't mean that amino acids might not be beneficial under normal conditions, just that we have no evidence yet showing which amino acid profiles might work best for which plants and under what concentrations and we do know that there can be potentially harmful effects if these parameters are not studied carefully.

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# Calibrating your digital humidity sensors

On a recent post I talked about vapor pressure deficit and its importance in hydroponic culture. To adequately control VPD it's necessary to accurately measure relative humidity and in order to do so it's necessary to have adequately calibrated humidity sensors. Since most of today's humidity sensors are digital this becomes even more important as these sensors can get damaged very easily, especially if the dew point is reached at any given point in time. Today I am going to talk about humidity sensor calibration, how it can be easily carried out and why you should do it in order to ensure that your humidity sensors are being accurate enough for your

cultivation needs.

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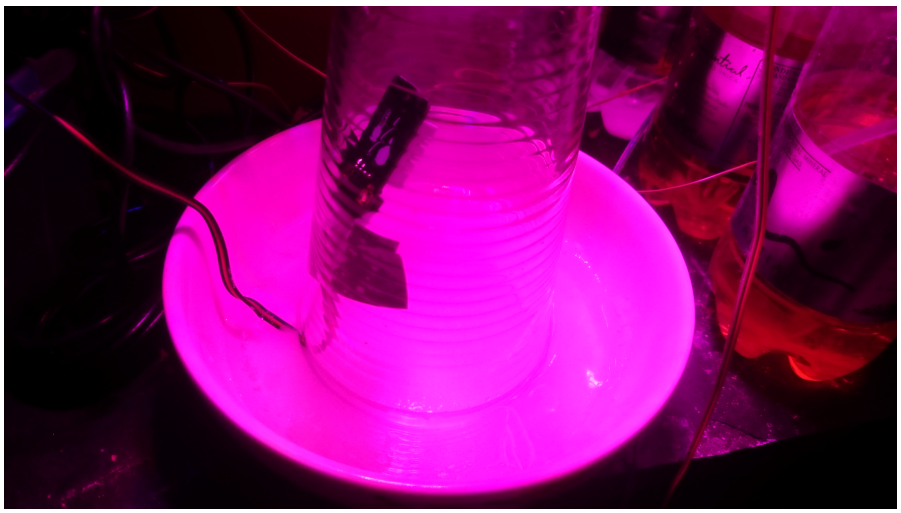

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Most modern digital humidity sensors are based on conductive polymers whose resistance changes with the amount of water in the air. If the polymer is in equilibrium with water vapor in the air then this change will be proportional to relative humidity. Sensors like those from the SHTX and DHTX series work using this principle. However if the polymer gets wet – water falls on the sensor or the dew point is reached – or if it faces very low humidity conditions for a long time then the humidity sensor will stop working correctly and it will need to be reconditioned and calibrated.

Reconditioning of these sensors is usually carried out by exposing the sensor to higher temperature dry conditions and then exposing the sensor to a controlled higher humidity lower temperature environment. These are some typical instructions for humidity sensor reconditioning. Once this process is carried out the sensor is now ready to be calibrated. Depending on the sensor you're using you might be able to change some calibration parameters to adjust the sensor to changes in its response or you might just use the calibration procedure to check the sensor's accuracy and discard it if it

isn’t behaving properly.

Calibration of digital humidity sensors can be carried out by putting them in the atmosphere composition generated over a saturated solution of a given salt. This table shows the expected relative humidity values at different temperatures for different salts. Basically you want to use a glass container where you can prepare a solution that has so much salt that there are undissolved crystals within it and then place your sensor in a closed environment above this solution (without touching it!). You can achieve this by drilling a hole at the top of a container with a lid to place the sensor (like it’s showed here), alternatively you can stick the sensor with electrical tape inside a glass and then place it upside down in a small amount of solution. This last process – first image in this post – completely eliminates any issues caused by potential holes and the atmosphere reaches equilibrium a bit faster. Another potential option is to create a paste with water and salt and place this past with the sensor inside a zip lock bag.

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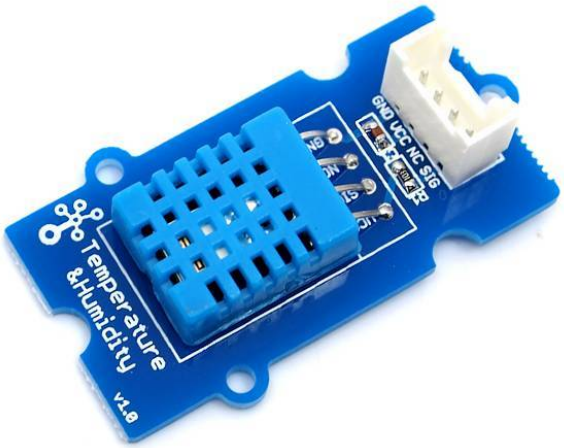


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For starters you can perform a single measurement with a saturated sodium chloride solution – which should give you a

humidity of around 75%. This is a good way to check if the sensor is working properly without the need to buy any additional materials. If you want you can then get some additional salts, like potassium chloride, magnesium nitrate and potassium nitrate, which should give you several different calibration points to draw an appropriate calibration curve to gauge how your sensor is working across the entire humidity range. Ideally you would want to have two salts with equilibrium points above 50% and two below 50% relative humidity.

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# Probes for constant immersion in hydroponic nutrient solutions

If you have a hydroponic crop then you probably have to measure and monitor the pH and EC of your nutrient solutions. This means taking probes out of storage, ensuring they are calibrated and then carrying out measurements. This process can be very inconvenient, reason why growers might prefer to carry it out less often, even if this means they will have a lot less data. However there are several solutions that can enable constant monitoring of hydroponic nutrient solutions without the need to constantly take out, calibrate and then store away probes. Today we will talk about why regular probes are not suited for this and what types of probes are needed if you want to do this.

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Usually low quality EC/pH pens cannot be kept within nutrient solutions because they are not built to withstand constant contact with nutrient solutions. This is both due to the electrode composition – the actual glass or metal electrodes not being robust enough – and the actual junctions and other components not withstanding the nutrient solution as well. Although hydroponic nutrient solutions are not particularly harsh environments – with a slightly acidic pH and moderate ionic strengths – probes for constant monitoring of nutrient solutions must be designed with constant immersion in mind.

For constant monitoring of pH in nutrient solution tanks you want a proper submersible electrode assembly like this one. These electrodes are usually mounted on PVC fixtures and can be easily mounted on tanks to provide constant readings for the nutrient solution. The electrode comes with a standard BNC connector meaning that it is compatible with a wide variety of pH controllers. If you don't want to mount it on the tank but you just want the electrode to be like a normal probe but constantly submerged then you can use something like this industrial probe which comes with a pH controller as well that can be used with any other probes you purchased and interfaces with an arduino or raspberry pi to get and store readings. For

probes like this last one I usually wrap the entire outside body of the probe in electrical tape to give further strength to the probe/cable junction.

For conductivity readings you will want to go with electrode-less EC probes (like these ones) which over PVC mountings as well with the advantage that they do not suffer from polarization issues – like normal EC pens use – so they lose calibration much more rarely and can give much more accurate readings across a wide range of different solution types and conductivity values.

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For the grower who wants it all there are also probes like the Mark I-A probe which is a tank-mounted probe assembly that does EC, pH and ORP readings, all in one single fixture. This is incredibly practical since it is able to implement all the readings you need in one single fixture. The problem of course is that calibration of any reading requires you to remove all three sensors so this can be a bit inconvenient when you want to ensure that any of the readings are indeed accurate.

Of course submersible robust probes are more expensive but they are much more convenient. They get damaged much less frequently, require much less maintenance, provide constant readings and need to be calibrated only a few times a year. For example the industrial EC and pH probes I use in my home

hydroponic setup have only required calibration once a year, even then the loss in calibration was only around 0.2 units for the pH sensor and 0.3 mS/cm for the EC one so I probably could have continued using the probes without calibrating them for 2 years without having to face any dramatic consequences. If you spend 300-400 USD on high quality robust probes you will probably have them for much longer, with far more accurate results along the way.

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# 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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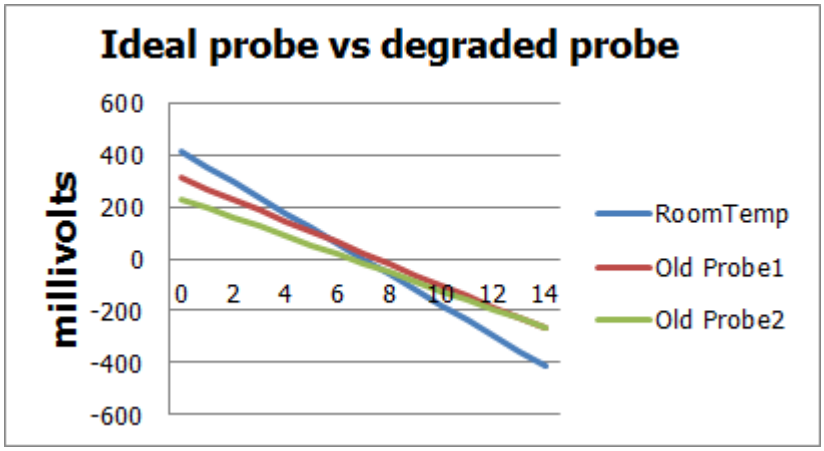

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**1. Let them dry.** These probes are made from glass and the readings depend on the potential difference between the inside and outside surfaces of the sensor. These are determined mainly by pH since hydronium (H3O+) ions interact strongly with the 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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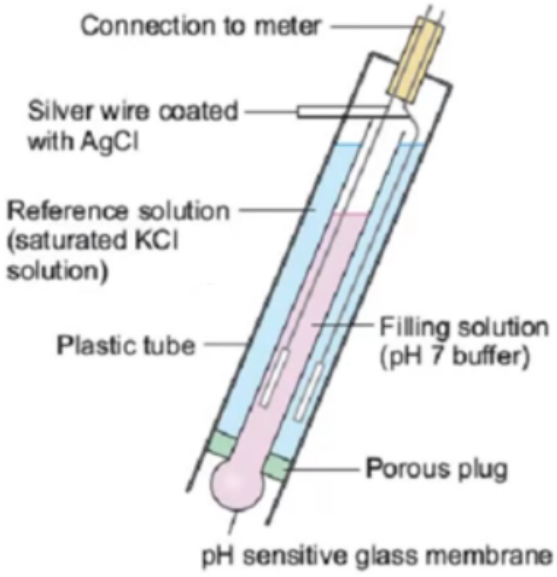


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Having pH probes that give accurate reading for a long time is not a difficult task if you take proper care of your sensors. Storing them adequately, ensuring they are not exposed to harmful conditions and cleaning them ensures that they will last 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.

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# 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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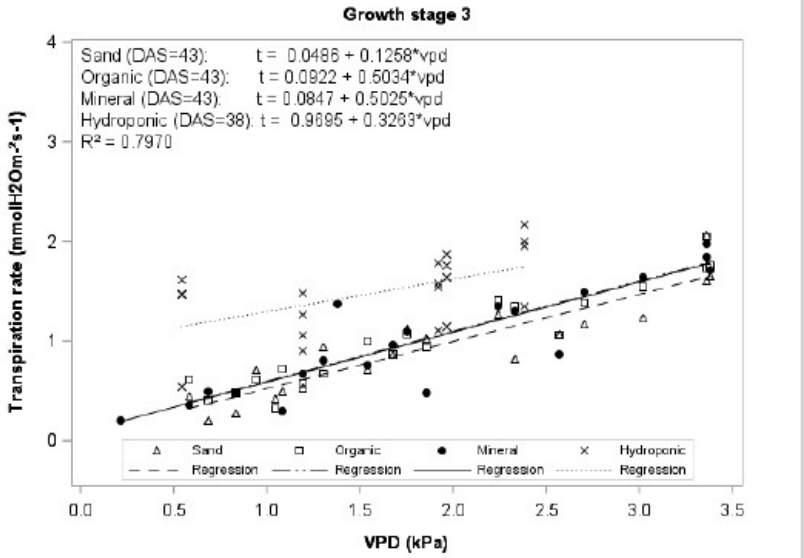


**Figure 7:** Transpiration rate in Panicum (*Panicum maximum* cv. *tanzania*) under VPD with different substrates in growth stage three.

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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.

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# 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.

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# 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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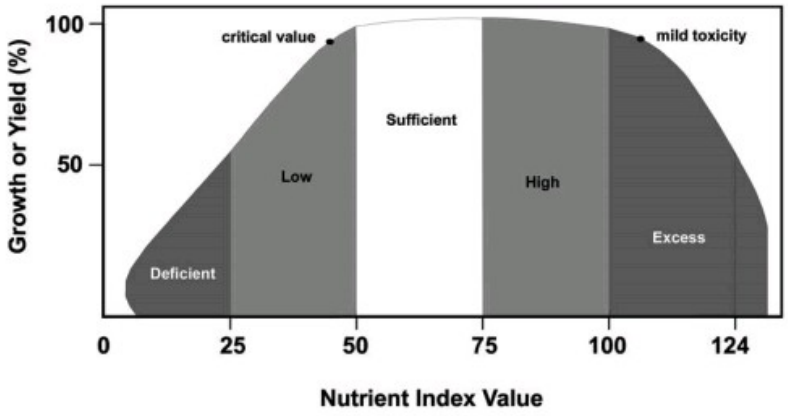

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The general model for nutrients and crop yields explains that plants will absorb nutrients till a point of maximum yield. After this point increasing nutrients will not increase or decrease yields substantially for a while but after a given point toxicity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

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# 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.

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# 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.

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# 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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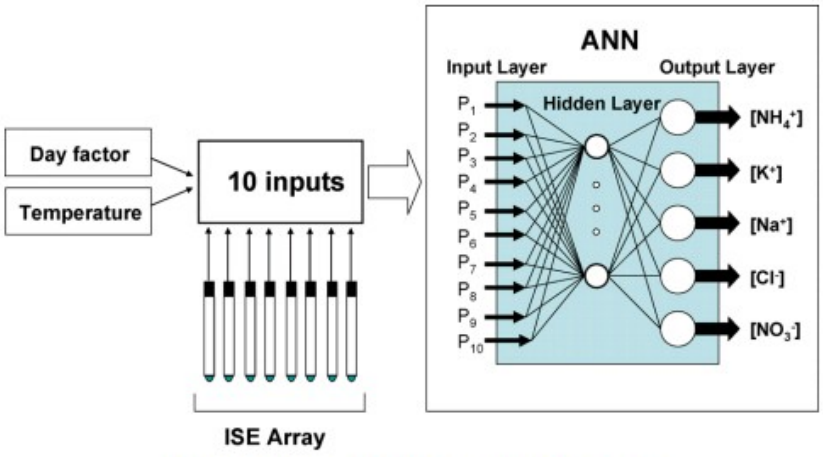


Fig. 2. Schematic representation of the electronic tongue approach.

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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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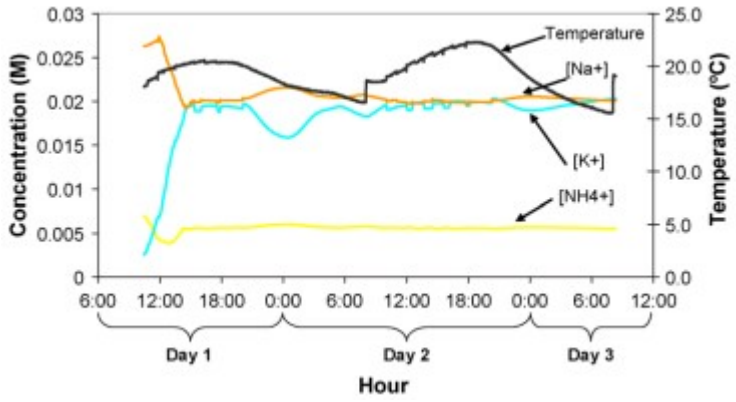


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Electronic tongues are an intelligent idea to circumvent these problems. The idea is to use many ISE for different ions – with many for the same ion and many generic ionophores that have poor selectivity – and then to use statistical modeling tools – 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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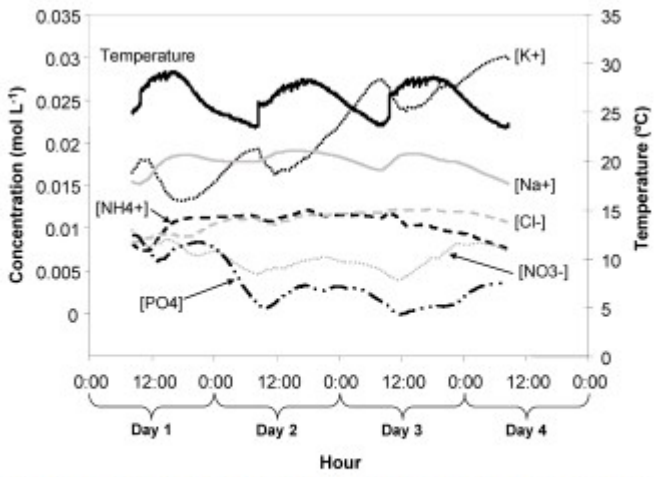


**Figure 3.** Representation of the concentration values predicted by the electronic tongue during the second application, in summertime, for the considered ions, ammonium, potassium, sodium, chloride, nitrate, and phosphate, in the nutrient solution during 3 days of continuous monitoring.

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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.