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Intracellular pH Response to Weak Acid Stress in Individual Vegetative Bacillus subtilis Cells

Rachna Pandey,^a^ Norbert O. E. Vischer,^a^ Jan P. P. M. Smelt,^a^ Johan W. A. van Beilen,^a^ Alexander Ter Beek,^a^ Winnok H. De Vos,^b,c^ Stanley Brul,^a^ Erik M. M. Manders^d^

Molecular Biology and Microbial Food Safety, SILS, University of Amsterdam, Amsterdam, The Netherlands^a^; Department of Veterinary Sciences, Laboratory of Cell Biology and Histology, Antwerp University, Antwerp, Belgium^b^; Department Molecular Biotechnology, Cell Systems and Imaging Group, Ghent University, Ghent, Belgium^c^; Van Leeuwenhoek Centre for Advance Microscopy, SILS, University of Amsterdam, Amsterdam, The Netherlands^d^

**ABSTRACT**

Intracellular pH (pHi) critically affects bacterial cell physiology. Hence, a variety of food preservation strategies are aimed at perturbing pHi homeostasis. Unfortunately, accurate pH quantification with existing methods is suboptimal, since measurements are averages across populations of cells, not taking into account interindividual heterogeneity. Yet, physiological heterogeneity in isogenic populations is well known to be responsible for differences in growth and division kinetics of cells in response to external stressors. To assess in this context the behavior of intracellular acidity, we have developed a robust method to quantify pHi at single-cell levels in Bacillus subtilis. Bacilli spoil food, cause disease, and are well known for their ability to form highly stress-resistant spores. Using an improved version of the genetically encoded ratiometric pHluorin (IpHluorin), we have quantified pHi in individual B. subtilis cells, cultured at an external pH of 6.4, in the absence or presence of weak acid stresses. In the presence of 3 mM potassium sorbate, a decrease in pHi, and an increase in the generation time of growing cells were observed. Similar effects were observed when cells were stressed with 25 mM potassium acetate. Time-resolved analysis of individual bacteria in growing colonies shows that after a transient pH decrease, long-term pH evolution is highly cell dependent. The heterogeneity at the single-cell level shows the existence of subpopulations that might be more resistant and contribute to population survival. Our approach contributes to an understanding of pHi regulation in individual bacteria and may help scrutinizing effects of existing and novel food preservation strategies.

**IMPORTANCE**

This study shows how the physiological response to commonly used weak organic acid food preservatives, such as sorbic and acetic acids, can be measured at the single-cell level. These data are key to coupling often-observed single-cell heterogeneous growth behavior upon the addition of weak organic acid food preservatives. Generally, these data are gathered in the form of plate counting of samples incubated with the acids. Here, we visualize the underlying heterogeneity in cellular pH homeostasis, opening up avenues for mechanistic analyses of the heterogeneity in the weak acid stress response. Thus, microbial risk assessment can become more robust, widening the scope of use of these well-known weak organic acid food preservatives.

Microbes have evolved to maintain a narrow range of optimal intracellular pH (pHi) values. For instance, under optimal growth conditions, Bacillus subtilis maintains its cytoplasmic pH at neutral or slightly higher values, with the exact range depending somewhat on the measurement tool used (compare data from reference 1 with our data). pHi affects many biological processes, such as enzyme activity, reaction rates, protein stability, and the structure of different molecules, such as nucleic acids. Thus, the pHi of bacteria is very important to ensure optimal growth, and conversely, perturbing the physiological pHi is a strategy that is often exploited by the food industry for preservation purposes. Weak acids, such as sorbic, acetic, lactic, and benzoic acids, are naturally occurring preservatives that are commercially used in the food industry. These molecules are long known to inhibit the outgrowth of both bacterial and fungal cells (2), thereby allowing for the extension of the shelf-life of food products. Sorbic acid and its salts inhibit the growth of various bacteria, including spore formers, at various stages of their life cycle, including spore germination, outgrowth, and vegetative cell division (3). The widely accepted theory of weak acid preservative action suggests inhibition of growth through lowering of the pHi. According to the theory, undissociated acid molecules pass, depending on their lipophility, more or less readily through the plasma membrane by diffusion. In the cytoplasm (pHi >7.5), the acid molecules dissociate into charged anions and protons. These cannot pass across the lipid membrane and hence accumulate in the cytoplasm, lowering the pHi of the cell. The acidification of the cytoplasm, in turn, inhibits metabolism. A recent study by van Beilen et al. (4) shows that sorbic acid has an ability to act as a classical uncoupler, transporting protons over the membrane, whereas acetic acid,
which is less lipophilic, does so to a much lesser extent. This is corroborated by the fact that sorbic acid has a greater effect on the membrane potential, while acetic acid only carries bulk volume protons across the membrane until steady state is reached. Studies by Holyoak et al. (5) and Bracey et al. (6) showed that in Saccharomyces cerevisiae, the inhibitory action of weak acid preservatives evokes an energetically expensive stress response. This response in S. cerevisiae is based on a membrane-localized efflux system that removes both the accumulated anions as well as the excess protons from inside the cell (7, 8). The attempts to restore homeostasis, however, require significant amounts of ATP, resulting in a drop in available energy pools for growth and other essential metabolic functions. In summary, weak acids inhibit the growth of microbes in a number of ways, including through membrane perturbation, inhibition of essential metabolic reactions (3, 6), and stress on pH homeostasis (31), as well as the accumulation of toxic anions (6, 9).

Direct measurement of the pH may be used as a proxy for cellular metabolism and thereby provide rapid insight into survival strategies at the single-cell level. The pH of the cells can be measured by various methods, such as 31P nuclear magnetic resonance (NMR), fluorescent dyes (most noticeably 5(and 6)-carboxyfluorescein diacetate succinimidyl ester) and the distribution of radiolabeled membrane-permeable weak acids (10–14). The advantage of these methods is that they do not require genetic modification. In the case of fluorescent dyes, single-cell measurements are possible (15). The disadvantage of using weak organic acid dyes is that they may themselves alter the pH. The disadvantage of the 31P NMR and radiolabeled compounds is that they require extensive cell handling and high cell density, which also disturb the cell’s physiology. Fluorescent proteins make an attractive and noninvasive alternative for measuring the pH of the bacterial cell, although obviously genetic modification is a prerequisite. pHluorin, a ratiometric and pH-sensitive green fluorescent protein (GFP) variant (16), allows direct, fast, and localized pH measurements. It has been successfully used in S. cerevisiae (17, 18) and more recently in B. subtilis (19–21) to probe cellular responses to various growth conditions, glucose pulses, respiratory chain inhibitors, and a few other treatments. A specific advantage of a fluorescence microscopy-based method is that it can provide information with (sub)cellular resolution (22, 23). This allows for the capturing of interindividual phenotypic heterogeneity that arises from factors, such as differential growth kinetics and stochastic effects, at the level of gene expression and protein activity. Taking advantage of this added value, we analyzed the effect of sorbic and acetic acid on the perturbation of the pH of Bacillus subtilis vegetative cells using an improved IpHluorin reporter.

**MATERIALS AND METHODS**

**Growth conditions.** To monitor the pH of exponentially growing B. subtilis cells for an extended period of time, the B. subtilis PptsG-IpHluorin (trpC amyE3’ spcr PptsG-IpHluorin amyE5’) construct was used (20). This construct consists of the pHluorin gene (16), which was inserted after the first 24 bp of comGA adjacent to the promoter PptsG, resulting in IpHluorin. This promoter drives the expression of the gene encoding the glucose-specific phosphotransferase system II. Thus, we were able to obtain expression of IpHluorin in vegetative cells growing on a glucose-containing medium. The B. subtilis 168 laboratory wild-type strain PB2 and B. subtilis PptsG-IpHluorin were grown exponentially in Luria broth (LB) at 37°C, under continuous agitation at 200 rpm. The exponentially growing cells were reincultured in a minimal defined medium with 80 mM MOPS [3-(N-morpholino)propanesulfonic acid] (24), buffered to pH 7.4 (here referred to as MOPS medium). The MOPS medium contained spectinomycin (50 µg/ml) when appropriate, and cells were grown until exponential phase at 37°C, under continuous agitation at 200 rpm. The optical density at 600nm (OD600) was measured in time to determine if cells were in the exponential phase. Cells in the early exponential-growth phase (OD600 ~0.2) were used for time-lapse microscopy experiments (see below). In stress experiments, 3 mM sorbic acid (KS) and 25 mM acetic acid (KAc) at pH 6.4 were used to test for their effect on the growth and pH of exponentially growing bacteria.

**Phototoxicity measurements.** Phototoxicity is a detrimental phenomenon in live-cell imaging, which occurs upon repeated exposure of fluorescently labeled cells to intense light. In order to test for possible phototoxicity, exponentially growing B. subtilis PB2 and B. subtilis PptsG-IpHluorin cells (grown in MOPS medium under live-imaging conditions, see below) were repetitively exposed to excitation light of two different wavelengths (390 nm and 470 nm) with exposure times of 100 ms and 30 ms, respectively, for a period of 5 h, with intervals of 5 min and 10 min. The exposure time was chosen in such a way that the bleaching in each channel has the same rate. The generation time of the cells was calculated with a home-written script for ImageJ (http://imagej.nih.gov/ij/) (25), Multichannel-SporeTracker. The total number of cells assessed for B. subtilis PB2 cells grown in the absence of fluorescent light was 107 and was between 77 and 164 for B. subtilis PptsG-IpHluorin cells cultured in the absence and presence of fluorescent light. The effect of phototoxicity on the cells was regarded as negligible when the generation times of vegetative cells did not differ significantly (t test, P > 0.05).

**Fluorescence time-lapse microscopy (live imaging).** In order to ensure the unbiased growth of aerobic bacteria, a closed air-containing chamber that has been described previously (25) was used for time-lapse fluorescence microscopy. In this chamber, cells were sandwiched between the glass coverslip and a thin (160 µm) 1% agarose-medium pad, molded in a Gene Frame, to ensure their immobilization in the presence of sufficient culture medium and enough oxygen for undisturbed growth. The pad was loaded with 1 µl of exponentially growing vegetative cells (OD600 ~2). Time-lapse series were made by making use of a temperature-controlled boxed incubation system for live imaging set at 37°C. The specimens were observed with a 100X/1.3 plane apochromatic oil objective mounted on a Zeiss wide-field fluorescence microscope (Axiovert-200; Zeiss, Jena, Germany) controlled by the MetaMorph 6.1 software. For ratiometric imaging, light from a Xenon arc lamp was filtered by a monochromator (OptoScan; Cairn Research Ltd., United Kingdom) and tuned to 390 nm or 470 nm, each with a bandwidth of 30 nm. The microscope was equipped with a standard GFP filter cube (Chroma) with 510-nm long-pass (LP) emission filter. Images were acquired with a Cool-Snap HQ charge-coupled-device (CCD) camera (Roper Scientific). For phase-contrast imaging, a red filter (610 nm LP; Schott AG, Germany) was placed in the light path to protect the cells from phototoxicity. For control experiments, the time-lapse series of phase-contrast and fluorescence images were recorded at a sampling frequency of 1 frame per 10 min (see Results for the final choice of 10-min intervals) for 5 h, and for stress experiments, the cells were imaged for 10 h (also 1 frame per 10 min). Two biological replicates and 15 to 30 technical replicates (recorded fields of view on one slide) were recorded in parallel per experiment. In every field of view (technical replicate), 2 to 8 vegetative cells were identified and followed over time. This resulted in the analysis of approximately 30 to 60 vegetative cells from the start of each imaging experiment per biological replicate.

**pH measurements in a microcolony and in single cells within a microcolony.** For pH measurements, two image analyses tools for ImageJ were used. Multichannel-SporeTracker (https://sils.fnwi.uva.nl/bcb/objectj/examples/sporetracker/SporeTracker.htm) was developed for pH measurements at the microcolony level. This program runs in combination with ObjectJ (https://sils.fnwi.uva.nl/bcb/objectj/), a plugin for ImageJ. ObjectJ supports graphical vector objects that nondestruc-
tively mark images on a transparent layer. It provides back-and-forth navigation between results and images. The results table supports statistics, sorting, color-coding, qualifying, and macro access.

Multichannel-SporeTracker allows accurate measurements of the intensity of IpHluorin in the cell, calculates the ratio \( \frac{E_{390}}{E_{470}} \) of IpHluorin, and deduces the pHi and the generation time of the vegetative cells growing into a microcolony at any desired time frame (Fig. 1). The generation time is calculated from the area \( \log_2 \) of the growing cell population. The pHi measurements are based on the ratio of the fluorescence emission at 510 nm after excitation at 390 nm and 470 nm, respectively \( \left( \frac{E_{390}}{E_{470}} \right) \). To calculate the pHi, fluorescence images were first aligned with the corresponding phase-contrast images. Before measuring the fluorescence intensity of the cells, temporal intensity fluctuations were buffered by subtracting the mode (most frequent) value per frame throughout the time-lapse image stack. Then, the fluorescence intensities of IpHluorin-expressing cells were measured in both fluorescence channels within cellular regions of interest, and the \( \frac{E_{390}}{E_{470}} \) ratio was calculated. By correlating the ratio with a calibration curve (mentioned below), the pHi of the cell was determined.

For pHi measurements at the single-cell level, a custom-made script for IJ/Fiji ColiMetrics.ijm was used (https://www.uantwerpen.be/cell-group/). The algorithm segments individual bacteria and tracks them over time. It also converts the two-channel fluorescence image (excitation at 390 nm and 470 nm) into a color-coded image representing the pHi for every individual cell in a microcolony (i.e., pH maps). pH maps are hue, saturation, value (HSV) images, in which the hue represents the ratio of both fluorescence channels, converted into a pH value according to a sigmoidal fit of the calibration curve, and the value is the average intensity of both channels (expansion of a macro described before; see reference 26). For single-cell analysis, individual bacteria were tracked over time up to the point of cell division. \( \frac{E_{390}}{E_{470}} \) ratios were measured per cell and plotted as a function of time.

**Calibration of pHi.** *B. subtilis* PptsG-IpHluorin cells were grown to exponential phase in MOPS medium to pH 7.4 containing spectinomycin (50 \( \mu \)g/ml). At an OD\(_{600}\) of 0.4, the cells were centrifuged \((1,073 \times g \text{ for } 10 \text{ min})\) and resuspended in phosphate-citrate buffers \((0.1 \text{ M citrate and } 0.2 \text{ M } K_2\text{HPO}_4)\), with pH values ranging from 5.5 to 8.5, as described previously by us (20). The cells were then permeabilized with the potassium

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**FIG 1** Multichannel-SporeTracker output for pHi measurements in growing *B. subtilis* cells. Shown here are collective plots of 4 individual cells measured every 5 min for 5 h. Bottom to top: \( \log_2 \) (surface area occupied by cells); fluorescence intensities measured at 510 nm when excited at 390 nm (FluorA) and 470 nm (FluorB), respectively; and the ratio of the excitation wavelength \( (390 \text{ nm and } 470 \text{ nm}) \) of fluorescence intensities (2nd panel from the top) and pHi (top). Note that here 4 cells are shown from the batch that starts at pHi of \( >8 \); in cells from other batches pHi was lower \( (\sim 7.5) \) at the onset of imaging (see the text and Fig. 5 lineage tracking for comparison).
ionophore valinomycin (1 μM) and the protonophore nigericin (1 μM) (13). This treatment allows for the equilibration of the pHi with the externally set pH. Subsequently, cells in the phosphate-citrate buffer of different pH (5.5 to 8.5) were transferred to agarose pads with the ionophores and immobilized on agarose pads containing both compounds at set pH values ranging from 5.5 to 8.5. The cell fluorescence emission intensities were measured, and the ratio (E390/E470) was plotted against pHi. At least 200 cells were measured per data point. Error bars indicate the standard deviations. The figure gives a comparison between the observations (○), including error bars (95% confidence limits of the observed values), and a fitted (●) curve generated according to the Henderson-Hasselbalch equation. See the text for further details of the statistical analysis.

The data were fitted to a slightly modified Henderson-Hasselbalch equation. It describes the relationship between the ratio of the intensity of emission at 510 nm after excitation at 390 nm and 470 nm (E390/E470) and pHi. The relationship between the ratio of the emission intensity at 510 nm after excitation at 390 nm and 470 nm (E390/E470) and pHi.

\[
\frac{E_{390}}{E_{470}} = \frac{[H^+]^{b}}{[H^+]^{a}}
\]

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The data were fitted to a slightly modified Henderson-Hasselbalch equation. It describes the relationship between the ratio of the intensity of emission at 510 nm after excitation at 390 nm and 470 nm (E390/E470) and pHi. Here, this was calculated as \( R = \frac{[10^{-\frac{pH}{pK_a}}]}{[10^{-\frac{pH}{pK_a} - 1}] + 1} \times b + a \). pKa equals the \( -\log_{10} \) of the dissociation constant, and b and a are parameters without physiological meaning, but they are used to enable a quantitative fit of the R values. The fitting procedure was conducted as follows: in total, 876 observations were available; 22 observations almost equally distributed across the different pH values were either negative or far beyond the set (3 standard deviations) to which they belonged. After elimination of these outliers, the average of each local clusters was taken. In total, 62 average values were obtained. To test the robustness of the model, 20 average values were selected randomly and left out. The 42 remaining values were fitted according to our modified Henderson-Hasselbalch equation. The estimates of the parameters, including confidence intervals, are shown in Tables 1 and 2. For nonlinear regression models, the correlation coefficients between the parameters can be considered acceptably low (27). The average residual sum of squares (RSS) was calculated for each pH value that was studied. No significant correlation was found between average RSS and pH. The average sum of squares was 0.00968.

### RESULTS

**Long-term ratiometric imaging of IpHluorin-expressing cells is not phototoxic.** A typical fluorochrome can only withstand a limited number of excitation cycles. Excessive illumination eventually leads to irreversible loss of fluorescence (photobleaching) and the production of free radicals that can damage cellular components compromising cell viability (phototoxicity). The combined effect, i.e., photodamage, restricts long-term fluorescence live-cell imaging. Photodamage can be mitigated by the parsimonious use of illumination light but cannot be eliminated completely (28). To assess whether our imaging conditions allowed for monitoring bacterial cells without excessive photodamage, we compared the generation time with and without fluorescence illumination. In addition, we assessed the effect on cell growth of IpHluorin expression.

The generation time of wild-type *B. subtilis* PB2 cells grown in the absence of excitation light (93 ± 13 min) was similar to the generation time of the IpHluorin-expressing cells grown in the absence of excitation light (92 ± 17 min) (Fig. 3). Therefore, we concluded that IpHluorin expression is not harmful to the cells.

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**FIG 2** Calibration curve of *B. subtilis* PptsG-IpHluorin, which describes the relationship between the ratio of the emission intensity at 510 nm after excitation at 390 nm and 470 nm (E390/E470) and pHi. The *B. subtilis* PptsG-IpHluorin cells were permeabilized using 1 μM nigericin and 1 μM valinomycin and immobilized on agarose pads containing both compounds at set pH values ranging from 5.5 to 8.5. The cell fluorescence emission intensities were measured, and the ratio (E390/E470) was plotted against pHi. At least 200 cells were measured per data point. Error bars indicate the standard deviations. The figure gives a comparison between the observations (○), including error bars (95% confidence limits of the observed values), and a fitted (●) curve generated according to the Henderson-Hasselbalch equation. See the text for further details of the statistical analysis.

**FIG 3** Effect of fluorescent light (excitation at 390 nm and 470 nm and emission at 510 nm) on *B. subtilis* PptsG-IpHluorin. Movies of *B. subtilis* PB2 cells grown in the absence of fluorescent light and *B. subtilis* PptsG-IpHluorin cells in the absence and presence of fluorescent excitation light (390 nm and 470 nm) with a time interval of either 5 min or 10 min were made during 5 h. Generation time was analyzed by Multichannel-SporeTracker. The total number of cells assessed for *B. subtilis* PB2 cells grown in the absence of fluorescent light was 107; for *B. subtilis* PptsG-IpHluorin cells in the absence of fluorescent light, it was 164; and for regularly illuminated PptsG-IpHluorin cells, it was 77 (for the specimens inspected every 10 min) and 92 (for those illuminated every 5 min). No cell lysis was observed while analyzing either of the incubations.

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**TABLE 1** Parameter estimates of the slightly modified Henderson-Hasselbalch model (see “Calibration of pH,” in Materials and Methods) describing the relationship between pHi and the ratio of the IpHluorin fluorescence emission intensity upon excitation at 390/470 nm.

<table>
<thead>
<tr>
<th>Parameter</th>
<th>Estimate (mean)</th>
<th>SE (mean)</th>
<th>Lower bound</th>
<th>Upper bound</th>
</tr>
</thead>
<tbody>
<tr>
<td>pK&lt;sub&gt;a&lt;/sub&gt;</td>
<td>7.112</td>
<td>0.055</td>
<td>7.001</td>
<td>7.223</td>
</tr>
<tr>
<td>b</td>
<td>1.625</td>
<td>0.048</td>
<td>1.528</td>
<td>1.722</td>
</tr>
<tr>
<td>a</td>
<td>0.703</td>
<td>0.040</td>
<td>0.621</td>
<td>0.785</td>
</tr>
</tbody>
</table>

**TABLE 2** Correlations of parameter estimates reported in Table 1.<sup>a</sup>

<table>
<thead>
<tr>
<th>Parameter</th>
<th>pK&lt;sub&gt;a&lt;/sub&gt;</th>
<th>b</th>
<th>a</th>
</tr>
</thead>
<tbody>
<tr>
<td>pK&lt;sub&gt;a&lt;/sub&gt;</td>
<td>1.000</td>
<td>-0.047</td>
<td>0.625</td>
</tr>
<tr>
<td>b</td>
<td>-0.047</td>
<td>1.000</td>
<td>-0.681</td>
</tr>
<tr>
<td>a</td>
<td>0.625</td>
<td>-0.681</td>
<td>1.000</td>
</tr>
</tbody>
</table>

<sup>a</sup> The average RSS was 0.00968.
under the tested conditions. Next, cells were exposed to sequential pulses of light of two different wavelengths (390 nm and 470 nm for 100 and 30 ms, respectively) with 5-min or 10-min intervals between fluorescence measurements. Figure 3 also shows the effect of repeated exposure of cells to 390 nm and 470 nm excitation on the generation time of the B. subtilis PptsG-IpHluorin-expressing cells. Both conditions, i.e., 5-min and 10-min intervals, resulted in a slight increase in generation time (116 ± 3 min and 114 ± 21 min, respectively) without causing cell death (no cell lysis was observed while analyzing either of the incubations microscopically). We conclude that our settings are suitable for up to 5 h of pH live imaging and went for the longer 10-min interval in further experiments.

**Sorbic and acetic acid impact on pH and growth of B. subtilis cells.** Sorbic and acetic acid have detrimental effects on bacterial cells. The two acids have a similar pKa of 4.76, but sorbic acid is lipophilic, whereas acetic acid is hydrophilic in nature. Here, the effect of exposure at pH 6.4 to 3 mM potassium sorbate and 25 mM potassium acetate was studied in vegetative B. subtilis cells at the single-cell level. Figure 4 shows the effect of sorbic and acetic acids on the pH and generation time of B. subtilis PptsG-IpHluorin vegetative cells growing in microcolonies. In sorbic acid-stressed cells, the average internal pH of the microcolonies decreased from 7.2 to 6.8 (Fig. 4A and Table 3), and the generation time increased significantly (Fig. 4A and Table 3). In 25 mM potassium acetate-stressed cells, a similar trend in internal pH and increase in generation time were observed (Fig. 4A and Table 3). Figure 4B gives the correlation graph between the average internal pH of the microcolonies and generation time for control cells, as well as cells stressed with the two acids. PptsG-IpHluorin-expressing cells are typically lowered 50% in their growth rate by the addition of 3 mM potassium sorbate or 25 mM potassium acetate to liquid cultures while monitoring population-level pH (4). Here, growth rate inhibition by these concentrations of both weak organic acids is increased, and the observed standard deviation for the population is high. This might reflect some light sensitization by the weak organic acid stresses. However, we have also shown previously that in the MOPS-buffered defined medium used here, there is an ~30% increase in generation time of wild-type B. subtilis cells compared to liquid culture conditions (25). Hence, although we cannot exclude that under our live-imaging conditions, weak acid stress response may sensitize the PptsG-IpHluorin-expressing cells to light, it will not be the major response seen.

In conclusion, acid-stressed cells and control populations displayed significant differences in both pH, and growth rate (P < 0.001). Figure 4C shows selected time points from movies (see Videos S1 to S3 and Table S1 in the supplemental material) of B. subtilis PptsG-IpHluorin vegetative cells in the presence and absence of 3 mM potassium sorbate and 25 mM potassium acetate, which are color-coded by their pH. Noticeably, as was observed previously in a study by van Beilen et al. (see Fig. 1b in reference 4), the pH of control cells started in some cells at values above 8, indicating a somewhat stalled metabolic activity at the onset of imaging. In this regard, we noted a clear batch variation between the two biological repeats shown. Growth rate and average colony pH calculations with Multichannel-SporeTracker were always performed from the time point at which a clearly detectable surface increase, i.e., growth, had resumed. pH was then generally ~7.5, values seen previously by van Beilen et al. in liquid populations (4, 20).

**Lineage tracing of individual cells in microcolonies reveals pH heterogeneity.** As noted in Materials and Methods, single B. subtilis cells grow and divide to form microcolonies. We observed that within a developing microcolony, the E<sub>390</sub>/E<sub>470</sub> fluorescence ratio of individual B. subtilis cells differs. This shows that there is heterogeneity in pH between individual B. subtilis cells in a microcolony at a given time point of culture. Figure 5 shows for control conditions a typical example of tracking individual cells, growing from a single cell up to a microcolony. It became clear that under those conditions, after a transient drop in pH presumably due to the increased levels of acetate made by the growing bacteria themselves (see, e.g., reference 4), individual bacteria are likely able to mount to various extents a response that allows them to finally...
again raise their internal pH, albeit to various degrees. Such variation in pH, is also observed for the cells that underwent intentional weak organic acid stress. This is true in particular for those exposed to sorbic acid (Fig. 4A and B). Strikingly, under conditions of 3 mM potassium sorbate stress, microcolonies emerged with pH values well within the range of those from control cells as well as values that were well below. While pH is clearly a major determinant of growth rate, it is definitely not the only one, certainly not under sorbic acid stress.

DISCUSSION

Here, we deployed a derivative of green fluorescent protein (GFP), IpHluorin, to probe at the single-cell level the pH of B. subtilis cells. The use of this genetically encoded reporter holds various advantages, such as inherent labeling, strong signal-to-noise ratio, and concentration independence. A potential disadvantage is its requirement for molecular oxygen, precluding its use in anaerobic species, such as Clostridium species.

Using B. subtilis cells stably expressing IpHluorin, we have established a robust microscopy-based assay for simultaneously measuring pH and generation time. We first tuned the imaging conditions so as to minimize phototoxicity. Subsequently, we established a calibration curve showing a strong correlation of the fluorescence ratio with the externally adjusted pH ranging from 5.5 to 8.5. Once optimized, we benchmarked our assay using two well-known weak acid preservatives, namely, sorbic and acetic acid. Also, to analyze the microscopy images, we developed a semiautomated image analysis tool based on the previously published SporeTracker (25), called the Multichannel-SporeTracker. This tool calculates the internal pH and the generation time of exponentially growing B. subtilis PptsG-IpHluorin-expressing vegetative cells. It allows us to monitor individual cells and subpopulations and deconvolute the population-level information at the single-cell level.

The analysis of the effect of sorbic acid and acetic acid on vegetative cells showed that at low concentrations of sorbic acid, the generation time increases with decreasing pH. Similar results were obtained from the analysis of acetic acid-treated cells, albeit at higher acid concentrations, and a wider distribution of generation times is seen. Thus, at the selected concentrations, the two acids reduce the pH, and the growth rate to a similar extent. This result corroborates the notion that sorbic acid is the more effective preservative of the two. van Beilen et al. showed that sorbic acid is unable to recover pH during acid stress. These observations reflect the notion that sorbate acts as a classical uncoupler, which shuttles protons over the membrane, whereas acetate is believed to do this to a much lesser extent (4). The data of the study by van Beilen et al. show that sorbic acid has an effect on the membrane potential, while acetic acid carries only bulk volume protons across the membrane until steady state is reached, leaving ΔΨ relatively unaffected. In line with this, and in accordance with data previously obtained in yeast (reviewed by Orij et al. [17]), van Beilen et al. (4) showed that at the population level, growth rate and pH behavior in B. subtilis can be correlated. We now also demonstrate a similar correlation at the single-cell level, demonstrating that pH can be assessed as a good indicator of individual bacterial health. From this knowledge of pH, one could infer at the individual cell level the activity of metabolic pathways that are key to cellular energy conversion. Such data may be used by food microbiologists to feed contemporary models that aim at quantitatively predicting microbial food stability. This study suggests that heterogeneity at the individual cell level is prominent, with important implications for weak organic acid-based food preservation strategies. We can now, through lineage tracing, also start to verify mechanistically whether under long-term weak organic acid stress conditions, subpopulations of Bacillus cells arise that might be more able to restore their pH, hence explaining their better survival in foods and outgrowth potential to new (micro)colonies that can spoil foods. It may also be applied to the analysis of other potential food spoilage organisms, e.g., Zygosaccharomyces bailii (29).
In conclusion, our microscopy-based single-cell analysis technique effectively allows for gauging pH$_i$ and relating it to generation time. In doing so, the method can provide further mechanistic insight into the principles of existing and novel food preservation strategies. The analysis can be extended to the ratio-metric assessment of the dynamics of the pH$_i$ of spores during preservation strategies. It will allow one to point out the phase where weak acids have a maximum effect and also could provide key information about the timing of weak organic acid action on individual germinating and outgrowing spores. This information can be coupled to risk management of the unwanted growth of bacteria in food and hence help the food industry combat food spoilage. An attractive platform for implementing such an assay would be a microfluidics-based lab-on-chip system (30). Such a platform allows for rapid measurement of pH$_i$ values under dynamically changing conditions (change of medium types or supplements) coupled to monitoring of the dynamics of spore germination and outgrowth. This type of experiment should provide ways to deconvolute the population data with respect to the effects of different sequences of stresses on the germination and (out)growth efficiency of B. subtilis spores.

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REFERENCES


Erratum for Pandey et al., “Intracellular pH Response to Weak Acid Stress in Individual Vegetative *Bacillus subtilis* Cells”

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Rachna Pandey,a Norbert O. E. Vischer,a Jan P. P. M. Smelt,a Johan W. A. van Beilen,a Alexander Ter Beek,a Winnok H. De Vos,b,c Stanley Brul,a Erik M. M. Mandersd

Molecular Biology and Microbial Food Safety, SILS, University of Amsterdam, Amsterdam, The Netherlandsa; Department of Veterinary Sciences, Laboratory of Cell Biology and Histology, Antwerp University, Antwerp, Belgiumb; Department Molecular Biotechnology, Cell Systems and Imaging Group, Ghent University, Ghent, Belgiumc; Van Leeuwenhoek Centre for Advance Microscopy, SILS, University of Amsterdam, Amsterdam, The Netherlandsd

Volume 82, no. 21, p. 6463–6471, 2016, https://doi.org/10.1128/AEM.02063-16. Page 6470, Acknowledgments: The first paragraph should include the sentence “Gertien Smits is thanked for her ground-laying work, as well as the many stimulating discussions on the measurement of intracellular pH and its role in cell growth.” Page 6470: References 16 through 18 should read as follows.