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Calculation of ATP production rates using the Seahorse XF Analyzer

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Abstract

Oxidative phosphorylation and glycolysis are the dominant ATP-generating pathways in mammalian metabolism. The balance between these two pathways is often shifted to execute cell-specific functions in response to stimuli that promote activation, proliferation, or differentiation. However, measurement of these metabolic switches has remained mostly qualitative, making it difficult to discriminate between healthy, physiological changes in energy transduction or compensatory responses due to metabolic dysfunction. We therefore present a broadly applicable method to calculate ATP production rates from oxidative phosphorylation and glycolysis using Seahorse XF Analyzer data and empirical conversion factors. We quantify the bioenergetic changes observed during macrophage polarization as well as cancer cell adaptation to in vitro culture conditions. Additionally, we detect substantive changes in ATP utilization upon neuronal depolarization and T cell receptor activation that are not evident from steady-state ATP measurements. This method generates a single readout that allows the direct comparison of ATP produced from oxidative phosphorylation and glycolysis in live cells. Additionally, the manuscript provides a framework for tailoring the calculations to specific cell systems or experimental conditions.

Keywords ATP; ECAR; glycolysis; oxidative phosphorylation; Seahorse XF Analyzer

Subject Categories Immunology; Metabolism; Methods & Resources

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Introduction

ATP is critical for cellular energy transduction. Essential cellular functions such as maintenance of ion homeostasis, macromolecule biosynthesis, motility, and autophagy involve reactions coupled to ATP hydrolysis to support these otherwise energetically unfavorable processes. The two dominant ATP-producing pathways in mammalian cells are glycolysis and oxidative phosphorylation (Chandel, 2015). Glycolysis involves the cytoplasmic breakdown of one molecule glucose into two molecules of pyruvate, generating two molecules of ATP in the process. The fermentation of pyruvate to lactate and subsequent lactate efflux enables continuous glycolytic turnover. Pyruvate and other carbon-rich energy substrates such as fatty acids and amino acids can also be taken up by mitochondria and completely oxidized during oxidative phosphorylation. The chemical energy in these substrates is converted to potential energy by the mitochondrial respiratory chain, and this reservoir of potential energy is used to drive ATP synthesis (Rich & Maréchal, 2010; Nicholls & Ferguson, 2013).

It is now clear that redistributing the balance between oxidative phosphorylation and glycolysis can characterize – and in some cases determine – cell function and fate (Lunt & Vander Heiden, 2011; Sullivan et al., 2018; DeBerardinis & Chandel, 2020; Shin et al., 2020). Glycolysis and offshoot pathways play essential roles in generating precursors for macromolecule biosynthesis (Lunt & Vander Heiden, 2011), supporting one-carbon metabolism (Ducker & Rabinowitz, 2017), and establishing cytoplasmic reducing power (Lewis et al., 2014). Biological processes such as stem cell pluripotency (Gu et al., 2016), oncogenesis (DeBerardinis & Chandel, 2020), growth factor signaling (Pavlova & Thompson, 2016), and immune cell activation (van den Bossche et al., 2017) are therefore frequently associated with a preferential shift towards glycolysis. Conversely, terminal differentiation is often accompanied by an increased capacity for oxidative phosphorylation and the complete oxidation of
glucose and fatty acids (Moyes et al., 1997; Wilson-Fritch et al., 2003; Chen et al., 2008; Divakaruni et al., 2014; Hazim et al., 2022).

Despite the importance of cellular ATP production rates and the distribution between glycolysis and oxidative phosphorylation, common measurement approaches are often limiting. Snapshot measurements of cellular ATP content are routinely used to characterize changes in energy metabolism, but these are generally informative only in extreme cases of energetic dysfunction such as ischemia (Brand & Nicholls, 2011). During physiological cell activation, cells can readily increase the rate of ATP production to match the increased rate of consumption without appreciably changing steady-state ATP levels. A far more useful metric is the ATP:ADP ratio, reflecting the free energy change associated with ATP hydrolysis into ADP and inorganic phosphate. However, biochemical or mass spectrometry-based approaches to measure ATP:ADP are limiting. They are destructive measurements conducted on cell lysates, and therefore unable to measure real-time changes. They are also prone to experimental noise that masks subtle but meaningful differences.

Kinetic measurements of energy status are therefore extraordinarily specialized, requiring transfection of a genetically engineered ATP-ADP sensor and an expert understanding of the caveats involved when using molecular probes (Tantama & Yellen, 2014; Koveal et al., 2020). Importantly, all these approaches lack direct information about whether the measured ATP is derived from oxidative phosphorylation or glycolysis.

Fluorescence-based, multi-well technologies such as Seahorse XF Analysis can characterize these bioenergetic shifts, as simultaneous measurements of oxygen consumption rates (OCR) and extracellular acidification rates (ECAR) can gauge the relative balance between oxidative phosphorylation and glycolysis (Pelletier et al., 2014; Divakaruni & Jastroch, 2022; i.e. OCR:ECAR ratio). However, precise quantification of the total rate of ATP production and the balance between these two pathways is not straightforward. Direct comparison is precluded by differences in unit scale between OCR (pmol O2/min) and ECAR (mpH/min). Quantitative comparison between these two pathways is not straightforward. Direct comparison is precluded by differences in unit scale between OCR (pmol O2/min) and ECAR (mpH/min). These measurements very often correlate with oxidative phosphorylation (OCR) and glycolysis (ECAR), but do not directly reflect the quantity of ATP produced via each pathway (Divakaruni et al., 2014). A strategy to convert OCR and ECAR readings into rates of ATP production from oxidative phosphorylation and glycolysis involved three principal steps (Mookerjee et al., 2015; Mookerjee et al., 2017):

i. Transforming ECAR (mpH/min) into a proton production rate (pmol H+/min) so rates of medium acidification could be directly compared against rates of oxygen consumption (pmol O2/min) or lactate efflux (pmol lactate/min) with similar units.

a. mpH/min → pmol H+/min

ii. Accounting for sources of acidification not associated with glycolysis and fermentation so changes in H+ production quantitatively reflect lactate efflux.

a. pmol H+/min → pmol H+ lactate/min

iii. Converting rates of oxygen consumption and lactate efflux into ATP production from oxidative phosphorylation and glycolysis using established stoichiometry.

a. pmol O2/min → pmol ATP OxPhos/min
b. pmol H+ lactate/min → pmol ATP Glyco/min
c. pmol ATP/min = pmol ATP OxPhos/min + pmol ATP Glyco/min

The work presented uses a 96-well Seahorse XF Analyzer platform. Although some values cannot be directly translated to 24-well instruments due to differences in volume of the measurement microchamber, the experimental strategy to calculate these values is universal across all Seahorse XF platforms.

**Transforming ECAR into a proton production rate**

It was first necessary to convert ECAR into values of pmol H+/min so rates of medium acidification could be easily compared against...
OCR (in pmol O₂/min) and parallel lactate efflux assays (in pmol lactate/min). This unit conversion required calculating the buffering power of the experimental medium. Buffering power is defined here as the pH change in the measurement microchamber elicited by a given amount of H⁺ (Fig 1A). Importantly, sequential additional of H₂SO₄ showed the change in pH was directly proportional to the added H⁺ over a range greater than 0.5 pH units (Fig 1B). Predictably, as the concentration of HEPES in the medium increased, there

![Figure 1](https://www.embopress.org/doi/10.15252/embr.202256380)

**Figure 1.** Extracellular acidification is directly proportional to lactate efflux in the absence of respiration.

A. Sample trace of H₂SO₄ injections to determine buffer capacity of Seahorse experimental medium. Error bars are obscured by the symbol (n = 16 technical replicates from a single experiment).

B. Changes in pH from sequential additions of 62.5 nmol H⁺ via the injector ports. The experimental medium is DMEM supplemented with 8 mM glucose, 2 mM glutamine, 2 mM pyruvate, and the indicated concentration of HEPES (pH 7.4). The microchamber volume of the 96-well XF Analyzer is 2.28 μl. Step-by-step calculations are available in Dataset EV1 (n = 6 experimental replicates).

C. The H⁺ production rate is given for A549 cells treated acutely (20 min prior to assay) with varying concentrations of 2-deoxyglucose (2DG) or 2DG with iodoacetic acid (IAA) to block glycolysis. The experimental medium contains 5 mM glucose with 5 mM HEPES, 200 nM rotenone (complex I inhibitor), 1 μM antimycin (complex III inhibitor), and 2 μM oligomycin (complex V inhibitor to block 'reverse' hydrolysis of ATP upon respiratory chain inhibition) (n = 6 biological replicates).

D. Sample trace of standard curve used to measure lactate collected from experimental medium (n = 3 technical replicates).

E. Plot of lactate efflux rates measured as in (D) against H⁺ production rates measured in the Seahorse XF Analyzer. Experimental conditions and titration of the rate with glycolytic inhibitors are as in (C). Parenteral values given in the key are the fitted slopes for each individual cell type (n = 6 biological replicates).

F. Aggregate lactate:H⁺ ratios, calculated as in (E) for 17 different cell types, yielding an average value of 1.53 ± 0.12 (mean ± standard deviation).

All data are mean ± S.E.M. unless otherwise specified.

Source data are available online for this figure.
was a smaller drop in pH elicited from the added H⁺. The results demonstrate that extracellular acidification measurements in mpH/min are readily converted to values of pmol H⁺/min by applying a scalar factor accounting for the buffering power of the medium and the volume of the measurement microchamber. Sample calculations and step-by-step instructions for transforming ECAR into values of pmol H⁺/min are provided in the Datasets EV1 and EV2.

**Accounting for sources of acidification other than lactate efflux**

It was then necessary to determine whether measurements of H⁺ efflux could indeed accurately measure lactate efflux. In principle, there could be several reactions that could cause a meaningful net pH change in the experimental medium in addition to the incomplete oxidation of uncharged glucose into anionic lactate (Fig EV1A). Other glycolysis-linked reactions can result in medium acidification, such as the efflux of anionic pyruvate or CO₂ evolution during the oxidative pentose phosphate pathway (Fig EV1B). Additionally, several mitochondrial dehydrogenases will generate CO₂ during the complete oxidation of energy substrates and oxidative phosphorylation. These include the 2-oxoadid dehydrogenase family (pyruvate dehydrogenase, α-ketoglutarate dehydrogenase, and the branched chain keto acid dehydrogenase) and isocitrate dehydrogenase (Fig EV1C). Furthermore, specific cell types may release organic acids under certain conditions, such as hepatocyte efflux of ketone bodies during robust fatty acid oxidation or neuronal release of glutamate during prolonged depolarization.

Rather than attempt to account for every possible acidifying reaction, we began by testing the hypothesis that respiratory acidification (CO₂ evolution from mitochondrial dehydrogenases linked to O₂ consumption) would be the only acidifying process with a large enough flux to affect the measurements on a scale comparable with lactate efflux in the basal state. If this hypothesis is correct, then the following testable predictions should hold. First, if respiratory acidification is indeed the only measurable source of non-glycolytic acidification, any remaining acid production should be attributable to glycolysis when respiration is inhibited. Similarily, in the absence of glycolysis, respiration should be directly proportional to the remaining acidification rate.

**In the absence of respiration, glycolysis is the exclusive source of measurable extracellular acidification**

We first measured whether lactate efflux was directly proportional to H⁺ production after chemical inhibition of the respiratory chain. In the presence of electron transport chain inhibitors, cellular H⁺ production was gradually reduced with increasing concentrations of glycolytic inhibitors in A549 cells (Fig 1C). We then measured the lactate content in the spent experimental medium from these samples using an enzymatic lactate assay (Fig 1D) to determine the relationship between lactate efflux and acid production in the absence of respiration (Fig 1E). As a negative control, no measurable acidification was observed in cultured pancreatic β-cells (Zhao et al., 2001), which do not express appreciable levels of lactate dehydrogenase or monocarboxylate transporter-1. Using 17 different cell types comprised of primary and immortalized cultures, this analysis yielded an average lactate:H⁺ ratio of 1.53 ± 0.12 (Fig 1F; Appendix Table S1).

Unexpectedly, this value deviated from our expectations and was substantially different from previous efforts to correlate H⁺ production and lactate efflux rates (Mookerjee et al., 2015). In principle, the lactate:H⁺ ratio should never be greater than 1, since lactate efflux should never exceed the total H⁺ efflux given their 1:1 stoichiometry. In fact, the theoretical lactate:H⁺ should be < 1 due to acidifying reactions associated with glycolysis but distinct from lactate efflux (Fig EV1B). We reasoned that this discrepancy could be due to the incomplete coverage of the measurement well by the fluorometric sensor of the Seahorse XF cartridge: although the enzymatic lactate measurements used medium collected from the entire well, outer portions of the well are likely undetected by the fluorometric sensor spot(s) during the acidification measurements (Gerencser et al., 2009; Fig EV2A).

To test this, we scraped cells from the measurement well from either the center of the three risers directly under the sensor (creating a “donut” shape of cells in the well) or on the rim of the plate outside the three risers (leaving cells in the center untouched; Fig EV2B). As predicted, both OCR and ECAR were significantly reduced when cells in the middle of the well were removed, but losing cells along the outer rim did not significantly affect the measurements (Fig EV2C). To ensure the results were not due to changes in cell number, the lactate:H⁺ ratio was calculated for these conditions. Consistent with the sensor not seeing the outer edges of the well, lactate:H⁺ was lowered close to 1 when cells were removed from the outer rim, and increased beyond 8 when cells were removed from center of the plate (Fig EV2D). Rather than a systemic error with the buffer capacity or enzymatic measurements, the result demonstrates that incomplete sensor coverage of the measurement well is a main driver of why lactate:H⁺ values deviate from theoretical expectations.

**In the absence of glycolysis, respiration is the exclusive source of measurable medium acidification**

Having shown that medium acidification is tightly correlated with glycolysis in the absence of respiration in Fig 1, we then tested whether medium acidification would be tightly correlated to respiration in the absence of glycolysis. We therefore measured acidification rates in HepG2 cells offered glucose-free medium containing pyruvate, glutamine, and the glycolytic inhibitors 2-deoxyglucose and iodoacetate. The rates of respiration and acidification were titrated by adjusting flux through the electron transport chain (with oligomycin, FCCP, and rotenone with antimycin A) and blocking substrate uptake and oxidation (with varying concentrations of the mitochondrial pyruvate carrier inhibitor UK5099 and the glutaminase inhibitor BPTES; Fig 2A and B).

We observed a proportional relationship between respiration and medium acidification in the absence of glycolysis (Fig 2C). Furthermore, we again extended this analysis to a panel of 21 different primary and immortalized cell preparations to yield an average H⁺:O₂ ratio of 0.38 ± 0.05 (Fig 2D; Appendix Table S2). However, similarly to the values obtained for lactate:H⁺, the composite H⁺:O₂ again deviated sharply from expectations and previous attempts to correct ECAR for respiratory acidification. The value obtained was much lower than the theoretical H⁺:O₂ of complete oxidation of either pyruvate (0.80) or glutamine (0.67; Mookerjee et al., 2015).

As such, we conducted a series of experiments to better understand the factors that shape the H⁺:O₂ value and whether experimental perturbations would change respiratory acidification in expected ways. We first examined whether oxidation of different substrates would perturb H⁺:O₂ values, as complete oxidation of...
various substrates yields varying H\textsuperscript{+}/O\textsubscript{2} ratios. Indeed, the H\textsuperscript{+}/O\textsubscript{2} ratio resulting from pure glutamine oxidation was fractionally lower than pure pyruvate oxidation (Fig EV3A–C).

We then further examined whether choice of respiratory substrates could influence H\textsuperscript{+}/O\textsubscript{2} ratios in isolated mitochondria, a simplified system where the experimental conditions are highly
controlled and metabolic reactions can be well defined. We tested acidification from uncoupled respiration in rat heart mitochondria offered (i) pyruvate with malate (P/M), (ii) P/M and inhibitors of multiple enzymes creating a truncated TCA cycle (P/M truncated; Mookerjee et al., 2015), and (iii) succinate with rotenone (S/R). The results qualitatively exhibited the expected behavior: the \( H^+/O_2 \) ratio for mitochondria oxidizing P/M increased in the presence of inhibitors which blocked downstream TCA cycle reactions (e.g. the alkalinizing succinyl CoA synthetase reaction). Moreover, succinate oxidation in the presence of rotenone, which will support oxygen consumption without generating CO\(_2\), shows robust respiratory rates with negligible changes in pH (Fig EV3D and E).

However, even in this reductionist system, the measured \( H^+/O_2 \) fell far below expected values (theoretical \( H^+/O_2 \) of P/M truncated = 1). We therefore used the XF24 system to further ensure that the result was not a peculiarity of an individual instrument platform. Using the same workflow as before, and accounting for the larger microchamber volume of the 24-well platform, nearly identical results were observed where respiratory acidification in isolated mitochondria fell short of theoretical predictions (Fig EV3F).

Lastly, in addition to showing that respiratory substrates could affect the \( H^+/O_2 \) ratio, we wanted to determine whether forced efflux of organic acids other than lactate could also affect the measurements in predictable ways. For a proof-of-concept example, we measured whether NMDA-induced efflux of glutamate would alter the \( H^+/O_2 \) in primary cortical neurons. Neurons were offered exclusively \( \beta \)-hydroxybutyrate with glycolytic inhibitors, so lactate efflux or sources of glycolytic acid production could not interfere with the measurements. As expected, NMDA-induced efflux of glutamate in primary cortical neurons caused a pronounced spike in the acidification rate (Fig EV3G) and the \( H^+/O_2 \) ratio (Fig EV3H) that was sensitive to the NMDA-receptor antagonist MK801 (Divakaruni et al., 2017). The result was not attributable to glycolysis, as lactate efflux was negligible and unchanged across conditions (Fig EV3I). Taken together, the demonstration that \( H^+/O_2 \) ratios respond in expected ways to changes in substrate oxidation and non-glycolytic acid efflux provided additional confidence in the accuracy of the measured values.

**Converting rates of oxygen consumption and lactate efflux into ATP produced from oxidative phosphorylation and glycolysis**

The results from Figs 1 and 2 demonstrate that rates of \( H^+ \) production can, in principle, quantify lactate efflux after correcting for respiratory acidification and accounting for the sensor coverage of the plate well. We therefore transformed \( H^+ \) production rates into lactate efflux rates in equation (1) as follows:

\[
\text{pmol lactate/min} = 1.53 \times (H^+\text{Total} - 0.38 \times \text{OCR}_{\text{mito}}). \tag{1}
\]

where 1.53 = average lactate: \( H^+ \) value obtained in Fig 1; \( H^+\text{Total} = H^+ \) production rates read by the instrument; 0.38 = average \( H^+/O_2 \) value obtained in Fig 2; \( \text{OCR}_{\text{mito}} \) = basal mitochondrial respiration.

To validate whether these average values obtained under constrained conditions could be broadly applied across different cell types under typical assay conditions (i.e. complete assay medium with both glycolysis and oxidation phosphorylation functional), we compared lactate efflux calculated using the XF Analyzer against an enzymatic assay in A549 adenocarcinoma cells, C2C12 myoblasts, and murine BMDMs (Fig 3A). Prior to adjusting the measurements as in equation (1), the \( H^+ \) production rate underestimated the rate of lactate efflux (comparing leftmost bars in each graph with rightmost). But as expected, Seahorse XF measurements accounting for respiratory acidification and sensor coverage resulted in lactate efflux values (middle bars) that matched enzymatic measurements. Taken together with Fig 1, the results suggest the consensus values and approximations used here should be applicable across a broad range of cell types under normal assay conditions.

The ability to quantify lactate efflux with rates of \( H^+ \) production allows the transformation of ECAR into a rate of ATP produced by glycolysis. All necessary parameters are obtained during standard respirometry profiling (Fig 3B and C), and sample calculations with representative data sets are available in Datasets EV2–EV4.

The rate of mitochondrial ATP production can be estimated by applying a P:O ratio (moles of ADP phosphorylated per mole of oxygen consumed) to the respiration that drives ATP synthesis (Brand, 2005), which is determined using sensitivity to the ATP synthase inhibitor oligomycin. As detailed elsewhere, oligomycin-sensitive respiration will slightly underestimate ATP-linked respiratory activity (due to overestimating proton leak), though this is a small, \( \sim 10\% \) overestimate which can be adjusted (0.1*OCR\(_{\text{Leak}}\) in equation 2; Affourtit & Brand, 2009). Moreover, assumptions must be made for the selection of P:O ratio based on which substrate is being oxidized. Here, we use a value of P:O = 2.73 (P:O\(_2\) = 5.45), the maximum value for complex I-driven respiration (Watt et al., 2010) and within the range of values for complete glucose oxidation (Mookerjee et al., 2017). The rate of ATP from oxidative phosphorylation can therefore be estimated as in equation (2):

\[
\text{ATP}_{\text{OxPhos}} = 5.45^* (\text{OCR}_{\text{ATP}} + 0.1^* \text{OCR}_{\text{Leak}}). \tag{2}
\]

where 5.45 = P:O\(_2\) (moles of ADP phosphorylated per O\(_2\) consumed); OCR\(_{\text{ATP}}\) = ATP-linked respiration (respiration sensitive to oligomycin); OCR\(_{\text{Leak}}\) = Respiration associated with proton leak (respiration insensitive to oligomycin).

The rate of glycolytic ATP production can be estimated with 1:1 stoichiometry to the lactate efflux rate (equation 1), and the total ATP production rate is given by the sum of equations (1) and (2). See Datasets EV2–EV4 for sample calculations.

\[
\text{ATP}_{\text{Glyco}} = \text{Equation 1}. \tag{3}
\]

\[
\text{ATP}_{\text{Total}} = \text{ATP}_{\text{OxPhos}} + \text{ATP}_{\text{Glyco}}. \tag{4}
\]

Notably, this calculation will slightly underestimate rates of glycolytic ATP, as it fails to account for the ATP generated during glycolysis when glucose-derived pyruvate is funneled into mitochondria for complete oxidation (Mookerjee et al., 2017). Accounting for this would require knowing (or assuming) the fractional contribution of glucose oxidation to oxidative phosphorylation relative to other substrates. To test whether this omission would appreciably affect experimental conclusions, we measured rates of ATP production in A549 cells offered complete medium or a simple salts medium supplemented with only glucose. Offering cells only glucose in simple salts allowed calculation of this additional glycolytic ATP by applying known stoichiometry and assuming respiration is exclusively driven by glucose oxidation with no
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Figure 3. Transformation of ECAR into lactate efflux allows calculation of ATP production rates.

A. The H⁺ production rate from the XF Analyzer (left bar in each graph), the calculated lactate efflux rate from the XF Analyzer (center bar), and the lactate efflux rate measured from an enzymatic assay (right bar) are given for A549 cells, C2C12 cells, and primary bone marrow-derived macrophages (BMDMs). Cells are offered 8 mM glucose, 2 mM pyruvate, and 2 mM glutamine in experimental medium supplemented with 5 mM HEPES. The shaded component of the H⁺ production rates for each cell type is the calculated contribution from respiratory acidification (n ≥ 4 biological replicates).

B. The oxygen consumption rate from a representative experiment with A549 cells is presented. Assay medium is as in (A). The parameters needed to calculate the ATP production rate are indicated within the figure (n = 6 technical replicates).

C. The H⁺ production rate from the kinetic trace in (B) is presented. Only the initial rates are shown, as these are the only measurements required to calculate the ATP production rate (n = 6 technical replicates).

D. ATP production rates calculated for A549 cells in DMEM supplemented with 8 mM glucose, 2 mM glutamine, and 2 mM pyruvate and Krebs–Henseleit buffer supplemented with only 20 mM glucose (n = 4 biological replicates).

E. Maximal respiratory rates measured in response to oligomycin and FCCP for treatments as in (D) (n = 4 technical replicates).

All data are mean ± S.E.M. Statistical analysis for 3A was analyzed by one-way, repeated measures ANOVA followed by Dunnett’s post hoc multiple comparisons tests. Individual pairwise comparisons for 3D and 3E were analyzed by a two-tailed Student’s t-test. *P < 0.05; **P < 0.01; ***P < 0.001.

Source data are available online for this figure.

contribution from endogenous substrate stores. However, rates of both oxidative phosphorylation and uncoupler-stimulated respiration were substantially limited when cells were offered medium containing solely glucose (Fig 3D and E). Additionally, rates of glycolytic ATP production between groups were only marginally altered despite accounting for the cytoplasmic ATP production linked to mitochondrial glucose oxidation (Fig 3D). We therefore chose not to account for this component, favoring – on balance – an approach where cells are offered complete DMEM to measure rates of oxidative phosphorylation and uncoupled respiration that are not restricted by substrate limitation.

Lastly, prior to applying the method to various models of cell activation, we measured how altered rates of organic acid efflux in response to activation stimuli may affect the measurements. We chose adrenergic activation of brown adipocytes as a model system, as adipocyte lipolysis is known to cause release of fatty acids into the experimental medium (Thompson et al, 2010). Fig EV4A–C show that stimulation of brown adipocytes with norepinephrine results in the hallmark increase in uncoupled respiration, as well as an increased rate of H⁺ production. To determine whether the contribution of organic acid efflux was different upon norepinephrine stimulation, lactate efflux was measured enzymatically from...
suggests that H+ measurements were somewhat confounded by differential rates of organic acid efflux (distinct from lactate) upon activation.

**Application of the method**

**ATP production rates show patient-derived glioma xenografts acquire an artificial glycolytic phenotype during in vitro cell culture**

A clear benefit of measuring ATP production rates is the ability to extend beyond commonly used OCR:ECAR ratios to directly quantify changes in the balance between oxidative phosphorylation and glycolysis. To illustrate this, we analyzed tumor cells from patient-derived glioma xenografts either immediately harvested and purified from mice (~3 h. isolation) or matched cells propagated in in vitro gliomaspheres (>1 week culture period). In vivo evidence suggests that the enhanced aerobic glycolysis observed in some glioma cell culture models may be driven, in part, by artifacts of cell selection or in vitro growth conditions (Marin-Valencia et al., 2012). To explore this, we again confirmed that our measurements of lactate using H+ efflux matched enzymatic measurements for these patient-derived cultures (Fig 4A). We then examined the information that can be gained from ATP production rates as opposed to OCR:ECAR ratios. In five different cell lines, in vitro cultured glioma cells showed a significantly reduced OCR:ECAR ratio compared to matched cells assayed immediately after purification from corresponding murine xenografts (ex vivo), indicating a relative shift towards glycolysis in in vitro cultured cells (Fig 4B).

Quantifying ATP production rates, however, extends this qualitative analysis. Glioma cells analyzed ex vivo just after isolation and purification are highly dependent on oxidative phosphorylation (~70% of total ATP). When cultured in vitro, however, cells undergo a remarkable shift in metabolism, increasing their energy demand and switching to glycolysis as the dominant ATP-generating pathway (Fig 4C). Importantly, there is only a slight, statistically insignificant trend towards increased oxidative phosphorylation, suggesting a specific reprogramming of glycolysis acquired upon in vitro culture and not simply an increased energy demand met by both pathways. The result shows that calculating ATP production from oxidative phosphorylation and glycolysis yields insights absent in qualitative, ratio-based analyses, and further supports an in vivo bioenergetic role of mitochondria in glioblastoma (Maher et al., 2012; Mashimo et al., 2014).

**ATP production rates demonstrate oxidative phosphorylation and glycolysis are differentially engaged upon pro-inflammatory macrophage activation**

We then applied our calculation to study pro-inflammatory macrophage activation, a form of immune cell activation associated with substantial metabolic alterations. Upon classical, “type I” activation [LPS (± interferon-γ)], murine BMDMs profoundly remodel energy metabolism away from oxidative phosphorylation (van den Bossche et al., 2016; Fig 5A). ATP production is shifted to glycolysis and

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**Figure 4. ATP production rates capture changes OCR:ECAR ratios cannot in primary cancer cell models.**

A. Lactate as measured by the XF Analyzer matches enzymatic lactate measurements for a panel of patient-derived, gliomasphere (GS) cell lines after in vitro culture (n ≥ 3 biological replicates for each line).

B. OCR:ECAR ratios for patient-derived glioblastoma cells cultured in vitro or assayed immediately after harvesting from mouse xenographs (ex vivo). Cell lined used were GS25, GBM39, GS114, GS116, GS122 (n = 5 biological replicates).

C. ATP Production rates from cell types as in (B) (n = 5 biological replicates).

All data are mean ± S.E.M. Statistics were analyzed by a two-tailed Student’s t-test. *P < 0.05; **P < 0.01. Source data are available online for this figure.
mitochondria are repurposed to generate signals that support the pro-inflammatory response (West et al., 2011; Lampropoulou et al., 2016; Mills et al., 2016). By measuring this switch with a qualitative OCR:ECAR ratio, metabolic reprogramming with LPS was indistinguishable from respiratory chain inhibition with complex I inhibitor rotenone (Fig 5B, inset). However, quantifying ATP production rates per cell revealed an increased energy demand associated with LPS activation, discriminating between mitochondrial dysfunction and mitochondrial repurposing during macrophage activation (Figs 5B and EV5).

Quantifying ATP production rates over a range of LPS concentrations further highlights the depth of information gained from the measurements. Qualitative OCR:ECAR ratios in response to increasing LPS concentrations (1–30 ng/ml) suggest a uniform, coordinated shift away from oxidative phosphorylation towards glycolysis to meet ATP demands upon activation (Fig 5C, inset). Cellular ATP production rates, on the other hand, surprisingly reveal these two pathways are engaged independently. Increases in glycolysis and overall ATP utilization are observed with treatments as low as 1 ng/ml LPS, whereas rates of mitochondrial ATP production are not
compromised until LPS concentrations rise above 10 ng/ml (Fig 5C). Additionally, rates of maximal respiration are not compromised at low concentrations of LPS that substantially increase glycolysis, further demonstrating that the increase in glycolytic ATP is not simply a response to impaired mitochondrial oxidative metabolism (Fig 5D).

Indeed, the time-dependence of energetic activation further reveals that regulation of mitochondrial and glycolytic ATP production are disengaged: glycolysis increases within 2 h of activation while rates of oxidative phosphorylation and maximal respiration are maintained up to at least 4 h (Fig 5E and F). The results show that hallmark metabolic changes observed during cell adaptation and activation. For example, in vivo stable isotope tracing has shown that in vitro models of glioblastoma and other cancers can overestimate the contribution of glycolysis to energy metabolism (Marin-Valencia et al., 2012; Faubert et al., 2017; Momcilovic et al., 2019). In support of this, matched samples of patient-derived glioblastoma cells clearly show the acquisition of an artificially high ATP demand driven almost entirely by a five-fold increase in glycolysis in cells cultured in vitro relative to those assayed immediately after isolation from murine orthotopic xenografts.

Additionally, the assay reveals that cells can adjust the balance of ATP production differently depending on the type of activation and biological context. As an example, acute activation of the Na⁺/K⁺-ATPase in primary cortical neurons increases the ATP demand in a way that does not broadly change the proportion of ATP produced by oxidative phosphorylation and glycolysis. Despite an acute, 2.5-fold increase in ATP production, the balance between the two pathways remained roughly constant with about 85% of the total ATP produced by mitochondria. It is sometimes stated that the Na⁺/K⁺-ATPase and other plasma membrane ATPases preferentially rely on ATP produced by glycolysis (Epstein et al., 2014; Sepp et al., 2014). However, our results do not support this relationship, at least in two-dimensional cell culture models not subject to microenvironmental gradients.

Unlike the instantaneous increase in ATP demand elicited by plasma membrane depolarization, immune cell activation shows that cells can redistribute the balance between oxidative phosphorylation and glycolysis when activating appropriate signaling cascades. Both T cell antigen receptor (TCR)-linked activation of human T cells as well as TLR-mediated activation of murine macrophages demonstrated that the increase in ATP utilization upon cell activation was increasingly met by glycolysis relative to the naive state. In T cells this increased glycolytic flux supports generation of biosynthetic intermediates necessary for cell growth and expansion (Maciver et al., 2013). Given that many types of TLR activation cause growth arrest in murine macrophages, the observed increase in glycolysis serves a different role. For example, the increase in glycolysis is likely associated with increased flux through the pentose phosphate pathway to support cytoplasmic NADPH production and fuel the anti-microbial NADPH oxidase (Cathcart, 2004). In fact, the approach highlights that regulation of glycolysis and oxidative phosphorylation are disengaged during the pro-inflammatory activation of macrophages. Rather than a coordinated process where mitochondrial ATP production decreases alongside a matched increase in glycolysis, pro-inflammatory stimuli can substantially increase glycolysis without changing rates of oxidative phosphorylation.

Discussion

ATP fluxes during cell activation

Our empirical approach to adjust proton efflux rates for respiratory acidification enables quantification of lactate efflux using OCR and ECAR. Furthermore, obtaining quantitative lactate readouts from the XF Analyzer allows estimation of cellular ATP production rates. ATP production rates from both oxidative phosphorylation and glycolysis provide additional insight into how ATP utilization is distributed during cell adaptation and activation. For example, in vitro models of glioblastoma and other cancers can overestimate the contribution of glycolysis to energy metabolism (Marin-Valencia et al., 2012; Faubert et al., 2017; Momcilovic et al., 2019). In support of this, matched samples of patient-derived glioblastoma cells clearly show the acquisition of an artificially high ATP demand driven almost entirely by a five-fold increase in glycolysis in cells cultured in vitro relative to those assayed immediately after isolation from murine orthotopic xenografts.

Additionally, the assay reveals that cells can adjust the balance of ATP production differently depending on the type of activation and biological context. As an example, acute activation of the Na⁺/K⁺-ATPase in primary cortical neurons increases the ATP demand in a way that does not broadly change the proportion of ATP produced by oxidative phosphorylation and glycolysis. Despite an acute, 2.5-fold increase in ATP production, the balance between the two pathways remained roughly constant with about 85% of the total ATP produced by mitochondria. It is sometimes stated that the Na⁺/K⁺-ATPase and other plasma membrane ATPases preferentially rely on ATP produced by glycolysis (Epstein et al., 2014; Sepp et al., 2014). However, our results do not support this relationship, at least in two-dimensional cell culture models not subject to microenvironmental gradients.

Unlike the instantaneous increase in ATP demand elicited by plasma membrane depolarization, immune cell activation shows that cells can redistribute the balance between oxidative phosphorylation and glycolysis when activating appropriate signaling cascades. Both T cell antigen receptor (TCR)-linked activation of human T cells as well as TLR-mediated activation of murine macrophages demonstrated that the increase in ATP utilization upon cell activation was increasingly met by glycolysis relative to the naive state. In T cells this increased glycolytic flux supports generation of biosynthetic intermediates necessary for cell growth and expansion (Maciver et al., 2013). Given that many types of TLR activation cause growth arrest in murine macrophages, the observed increase in glycolysis serves a different role. For example, the increase in glycolysis is likely associated with increased flux through the pentose phosphate pathway to support cytoplasmic NADPH production and fuel the anti-microbial NADPH oxidase (Cathcart, 2004). In fact, the approach highlights that regulation of glycolysis and oxidative phosphorylation are disengaged during the pro-inflammatory activation of macrophages. Rather than a coordinated process where mitochondrial ATP production decreases alongside a matched increase in glycolysis, pro-inflammatory stimuli can substantially increase glycolysis without changing rates of oxidative phosphorylation.
Adjusting proton production rates to quantify lactate efflux

In line with previous reports (Mookerjee et al., 2015), our empirical approach determined that glycolytic and respiratory acidification were the only dominant contributors to the extracellular acidification rate under normal assay conditions. By and large, other reactions that could result in a net pH change of the medium and alter the efflux rate either (i) did not have with fluxes comparable to those of respiratory and glycolytic acidification, or (ii) occurred proportionally with respiratory CO₂ evolution or lactate efflux (e.g., CO₂ evolution from the pentose phosphate pathway) and fell within a reasonable margin of experimental error. Cell types or experimental conditions that may deviate from this framework are addressed in the “Best practices” section of the Discussion.

Although we demonstrate that the H⁺ efflux rate can quantitatively measure lactate efflux after accounting for respiratory CO₂, the empirical values obtained did not follow the expected stoichiometry for either lactate:H⁺ or H⁺:CO₂ and were inconsistent with prior work (Mookerjee et al., 2015). These deviations from expected stoichiometry presented here, however, have theoretical and experimental
support. The value of 1.53 ± 0.12 for the lactate:H\(^+\) can be partially explained by incomplete coverage of the plate well by the fluorometric sensor in the XF cartridge, as cells in the center of the well have a far greater effect on the readings than those on the rim.

In fact, the expected lactate:H\(^+\) value of ~1 is obtained when cells are only within a circle connecting the three microplate risers, roughly corresponding to the XF96 fluorometric sensor area. The demonstration that cells directly under the measurement sensor are preferentially, if not exclusively, read by the cartridge suggests that there is no systematic error with the enzymatic lactate measurements. Additionally, the result implies a 1:1 lactate:H\(^+\) ratio should never have been expected, particularly in older instruments where the fluorometric sensors consist of two small spots (Gerencser et al., 2009) and not the larger smears found in newer instruments. Seahorse XF Wave software now incorporates this correction into the Proton Efflux Rate (PER) parameter for all multi-well platforms, accounting for the buffering power of the medium and incorporating the geometric corrections obtained from this work.

The H\(^+\):O\(_2\) values from our analysis also deviate from theoretical stoichiometries expected from the complete oxidation of known substrates. Nonetheless, the obtained values change with the appropriate directionality and magnitude in response to oxidizing varying energy substrates in both intact cells and isolated mitochondria. Moreover, lactate efflux rates calculated from the XF Analyzer using an H\(^+\):O\(_2\) of 0.38 ± 0.05 to adjust for respiratory CO\(_2\) matched enzymatic lactate assays, suggesting there was no systemic error in measuring buffering power of the experimental medium.

The discrepancy between the obtained and expected H\(^+\):O\(_2\) values persists in isolated mitochondria, where the experiments are highly controlled and the metabolic reactions are well defined. In this system, the apparent H\(^+\):O\(_2\) values for a truncated reaction where mitochondria are offered pyruvate and malate to generate citrate (theoretical H\(^+\):O\(_2\) = 1 (Mookerjee et al., 2015)) are more than 3-fold lower than what is expected in principle and previously observed. The result is likely due, in part, to the different ways the XF Wave software calculates rates of H\(^+\) efflux and O\(_2\) consumption. While the reported rates of H\(^+\) production are a straightforward measurement, the instrument reports rates of O\(_2\) consumption only after an empirical correction to deconvolute the effects of back-diffusion of ambient oxygen into the measurement microchamber (Gerencser et al., 2009). This includes use of an ‘apparent chamber volume’ that is 3-times greater than the actual measurement chamber volume to account for oxygen storage in the walls of the polystyrene microplate, and the algorithmic constants were determined by matching respiration rates obtained from synaptoneurosomes in the XF Analyzer with values obtained from a platinum-based oxygen electrode (Gerencser et al., 2009).

Crucially, these corrections would necessarily account, at least partially, for respiration in material along the rim of the plate unseen by the measurement sensor because they are clamped to values obtained from a Clark-type electrode. However, no such corrections for material unseen by the measurement sensor are made for rates of medium acidification. As such, it is expected that rates of H\(^+\) production are consistently underestimated relative to rates of O\(_2\) consumption, and that the observed H\(^+\):O\(_2\) ratios fall below theoretical predictions. Put simply, while software corrections adjust OCR to reflect the full well, this is not the case when measuring H\(^+\) production.

**Assumptions and limitations in calculating ATP production rates**

Notwithstanding the deviation from expected values, the consensus values obtained for lactate:H\(^+\) and the H\(^+\):O\(_2\) accurately adjust for respiratory CO\(_2\) and can be used to quantitatively reflect lactate efflux. They can therefore be used to convert oxygen consumption rates and H\(^+\) efflux rates into estimates of ATP production as detailed in Datasets EV2-EV4.

Any approach to estimate an ATP production rate, be it the empirical approach presented here or ones based on theoretical principles (Mookerjee et al., 2017), are subject to limitations and rely on multiple assumptions. Our empirical approach uses an average H\(^+\):O\(_2\) value for use with standardized experiment medium for cellular bioenergetics: DMEM supplemented with glucose, glutamine, and pyruvate. This approach, however, precludes the ability to estimate ATP supply changes upon shifts in cellular substrate preference (e.g. from fatty acids to glucose) in response to external cues. In principle, this could be done by strictly defining the medium composition and applying different H\(^+\):O\(_2\) values based on assumptions or measurements of what substrates are being oxidized.

Additional limitations associated with our approach are (i) use of an approximate P:O value and (ii) an underestimation of glycolytic ATP as described in the results. Both result from conducting experiments in a composite medium more like cell culture conditions with multiple oxidizable substrates available. It is therefore difficult to know, and perhaps unwise to assume, the fractional contribution of individual substrates to the oxygen consumption rate.

We therefore chose to use an approximate P:O ratio and not to account for glycolytic ATP produced during the complete oxidation of glucose. This approach did not significantly alter measurements of glycolytic ATP in A549 lung adenocarcinoma cells, but the experimental conditions substantially increased rates of oxidative phosphorylation and respiratory capacity. The pre-set calculations of the Seahorse Analytical Software Tools follow this framework: they incorporate average H\(^+\):O\(_2\) and lactate:H\(^+\) values, use an approximate P:O ratio, and ignore the glycolytic ATP contribution from complete glucose oxidation.

**Best practices for calculating of ATP production rates**

Many of the best practices associated with this particular assay are shared with Seahorse XF assays in general. These include optimizing the plating density for suitable rates, ensuring all measurements are conducted with rates at relatively stable steady-states, using the inner 60 wells of the XF96 microplate to avoid temperature and evaporative effects, and following best practices for normalization (Divakaruni & Jastroch, 2022).

When calculating the buffering power of the experimental medium, it is always best practice to use a non-volatile acid (e.g. H\(_2\)SO\(_4\)) or two distinct calibrations (e.g. one with HCl and one with H\(_2\)SO\(_4\)) to minimize instrument and operational error. The buffering power should be checked or recalculated when critical parts of the measurement process are changed – such as switching cartridge lot numbers, batches of medium, or instruments – but does not necessarily need to be calculated alongside each experimental run. In the 96-well instrument, it is often convenient to use...
columns 1 and 12 of the microplate for these calculations as is shown in Dataset EV1.

Additionally, it may be that the approximations used for this method are meaningfully inaccurate in specific cases depending on the experimental hypothesis and quantitative rigor required. For example, rates of lactate efflux may be particularly low when using acutely isolated primary cells, and therefore the non-zero background rate of H\(^+\) efflux (Figs 1E and 2C) may represent a substantial component of the signal and skew results. Furthermore, conditions where the rate of lactate efflux is matched by organic acid efflux of the same magnitude may also be less amenable to using consensus values of lactate:H\(^+\) and H\(^+\):O\(_2\). Indeed, this is apparent in noradrenaline-stimulated adipocytes known to release fatty acids: our XF Analyzer calculations based on H\(^+\) release estimated an almost two-fold increase in lactate efflux, whereas the enzymatic assay showed a 1.5-fold increase (Fig EV4). Additionally, under extreme, non-physiological conditions such as neurons depolarized in the absence of glucose, release of glutamate and other neurotransmitters resulted in a profound increase in acidification entirely independent of lactate efflux.

As such, any system in which the investigator believes that non-lactate acid efflux could prohibitively alter the conclusions should consider independently calculating lactate efflux with other methods such as enzymology or mass spectrometry. However, the empirical approach presented here provides the framework for researchers to calculate lactate:H\(^+\) and H\(^+\):O\(_2\) values tailored to any (monolayer) model system or experimental condition.

### Materials and Methods

#### Reagents and Tools table

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Methods and Protocols

Animals and primary cells

All animal procedures were performed in accordance with the NIH Guide for the Care and Use of Laboratory Animals, and approved by the UCLA Animal Research Committee (ARC-2020-027).

Murine bone marrow-derived macrophages (BMDMs)

BMDMs were cultured as previously described (Hsieh et al., 2020). Briefly, macrophages were isolated from femurs of male C57BL/6J mice (Jackson 000664) and maintained at 37°C in a humidified 5% CO₂ incubator. Cells were treated with 3 ml RBC lysis buffer (Sigma-Aldrich) to remove red blood cells for 5 min, centrifuged at 386 g for 5 min, and resuspended in BMDM culture medium. Culture medium consisted of high-glucose DMEM (Gibco 11965) supplemented with 10% heat-inactivated fetal bovine serum (FBS), 2 mM L-glutamine, 100 U/ml penicillin, 100 μg/ml streptomycin, 500 μM sodium pyruvate, and 5% (v/v) conditioned medium containing macrophage colony stimulating factor (M-CSF) produced by CMG cells to induce differentiation to BMDMs. BMDMs were allowed to differentiate for 6 days, changing medium after 4 days, and then plated for experiments.

Rat cortical neurons

Cortical neurons were generated from embryonic day 18 Sprague–Dawley rats as described previously (Kushnareva et al., 2005) and maintained at 37°C in a humidified 5% CO₂ incubator. Cells were plated onto poly-d-lysine-coated wells of black-walled 96-well plates for enzymatic assays or Seahorse XF96 plates. Cells for 96-well plate-based assays were seeded at 2.5 x 10⁵ cells/well and maintained in maintenance medium composed of 200 μl Neurobasal (Thermo Fisher Scientific) medium supplemented with 1x B27 serum-free supplement (Thermo Fisher Scientific), 2 mM GlutaMAX (Thermo Fisher Scientific), 100 U/ml penicillin, and 100 μg/ml streptomycin. Half the medium was replaced every 3–4 days. Experiments in 96-well plates were conducted at day in vitro (DIV) 13–17.

Human T cells

Frozen stocks of human naïve CD4⁺ T cells were purchased from Lonza. Cells were thawed and permitted to recover in RPMI 1640 (Gibco, #11875-085) supplemented with 10% (v/v) FBS at 37°C, 5% CO₂ for 18–24 h. After recovery, cells were spun at 250 g for 10 min and resuspended to a final density of 4 x 10⁶ cells/ml in Agilent Seahorse XF RPMI Medium pH 7.4 (#103576-100) supplemented with 10 mM glucose, 2 mM glutamine and 1 mM pyruvate (XF Assay Media). Fifty microliters of the naïve T-cell suspension (2 x 10⁵ cells) were plated in each well of a poly-D-lysine-coated Seahorse XF96 microplate. Plates were spun at 200 g for 1 min to adhere cells to the plate. One hundred and thirty microliters of warmed XF Assay Media was then added to each well, and plates were either run in the XF Analyzer or used to measure intracellular ATP concentration with the Cell-Titer Glo Assay (Promega). ATP measurements were taken before and after activation with magnetic Dynabeads coated with anti-CD3/CD28 antibodies (Thermofisher, 4:1 beads/cell).

Additional primary cells preparations

Neonatal rat ventricular myocytes (Rubio et al., 2009) and rat cortical astrocytes (Kim & Magrané, 2011) were isolated as previously described. Murine splenocytes were isolated from dissected spleens from C57BL/6 mice. Tissue was mechanically dissociated in HBSS and passed through a 40 μm strainer. Cells were washed and resuspended in red blood cells lysis buffer (Stemcell # 07850) and incubated on ice for 10 min. Cell were washed again in HBSS, passed through a 70 μm filter, counted, and used immediately. For Seahorse XF experiments cells were resuspended in XF Assay Media and seeded at 1.5 x 10⁵ cells/well.

Mitochondrial isolation

Hearts from C57BL/6 mice were drained of blood and homogenized with a hand-held Polytron followed by a Potter-Elvejhem Teflon-on-glass tissue grinder in ice-cold MSHE buffer [70 mM sucrose, 210 mM mannitol, 5 mM HEPES, 1 mM EGTA, and 0.5% (w/v) fatty acid-free BSA] (Kubli et al., 2013). The homogenate was centrifuged at 500 g for 10 min at 4°C. The supernatant was then collected and centrifuged at 12,000 g for 10 min at 4°C to obtain a mitochondrial pellet. The pellet was washed in MSHE and centrifuged at 12,000 g for 10 min at 4°C, and this wash step was repeated. Finally, the pellet was resuspended in a small volume (~20 μl) of MSHE, and the protein concentration was determined by Bradford assay.

Cultured cells

All cell lines were obtained from American Type Culture Collection (ATCC) and cultured as suggested by the supplier except when otherwise notified. Each formulation of medium was supplemented with 10% (v/v) fetal bovine serum (FBS), 2 mM GlutaMAX, 100 U/ml penicillin, and 100 μg/ml streptomycin unless otherwise specified. All cells were maintained at 37°C in a humidified 5% CO₂ incubator. Cell lines were maintained in the medium listed parenthetically: A549 [DMEM/F12 (Gibco 11330)]; HepG2 [MEM (Gibco 11095)]; HCT116 [RPMI (Gibco 11875)], C2C12 [DMEM (Gibco 11960 + 2 mM glutamine + 1 mM pyruvate)], A431 [DMEM (Gibco 11960) + 2 mM glutamine + 1 mM pyruvate], BAEC [DMEM (Gibco 11054)], BT474 [ATCC Hybri-Care Cat.#46-x], H460 [RPMI (Gibco 11870), 1 mM HEPES, 4 mM glutamine, 5% FBS], HUVEC [EGM Endothelial Cell Growth Medium (Lonza CC-3124)], Jurkat [RPMI (Gibco A10491-01)], MCF7 [RPMI (Gibco 21870 + 2 mM glutamine + 1 mM pyruvate)], MCF-10A [MEM supplemented with Kit CC-3150 (Lonza) and 100 ng/ml cholesta toxin (Sigma)], MDA-MB231 [RPMI (Gibco 21870 + 2 mM glutamine + 1 mM pyruvate)], PC12 [RPMI (ATCC 30-2001+ 2 mM glutamine + 10% Horse Serum + 5% FBS)], Raw 264.7 [DMEM (Gibco 11960) + 2 mM glutamine + 1 mM pyruvate].

3T3-L1 pre-adipocytes and differentiation

3T3-L1 pre-adipocytes were maintained below ~70% confluency in DMEM (Gibco 11965) supplemented with 10% (v/v) bovine calf serum (BCS), 100 U/ml penicillin, and 100 μg/ml streptomycin. For differentiation, cells were plated on Day 2 and were allowed to reach confluency over 48 h. Differentiation was induced on Day 0 by addition of fresh medium with FBS replacing BCS and supplemented with the following: 0.5 mM IBMX, 0.25 μM dexamethasone, 1 μg/ml insulin, and 100 nM rosiglitazone. On Day 2, medium was replaced to DMEM + FBS supplemented with only 1 μg/ml insulin, and thereafter insulin was removed when replacing medium every 2 days.
**Immortalized brown adipocytes**

Immortalized human brown adipocytes were maintained as described previously (Panic et al., 2020). Briefly, cells were cultured in DMEM supplemented with 25 mM glucose, 10% (v/v) fetal bovine serum, 4 mM glutamine, 1 mM sodium pyruvate, 10 nM HEPES, and 100 U/ml penicillin with 100 μg/ml streptomycin. Cells were plated at 1 × 10^8 cells/well and reached total confluency after approximately 24 h. Medium was then changed to induction medium consisting of the growth medium as before supplemented with 1 μM rosiglitazone, 0.5 mM IBMX, 0.5 μM dexamethasone, 125 μM Indomethacin, 1 nM T3, and 4 nM human insulin. After 2 days, medium was changed to growth medium supplemented with only rosiglitazone and insulin (concentrations as before), and this medium composition was used to change the fluid every other day for an additional 5–6 days.

**Seahorse XF analysis**

All oxygen consumption and extracellular acidification measurements were conducted using an Agilent Seahorse XF96 or XF24 Analyzer. Experiments were conducted at 37°C and at pH 7.4 (intact cells) or 7.2 (isolated mitochondria). For experiments with cultured cells, only the inner 60 wells were used, and the outer rim was filled with 200 μl of PBS throughout the incubation to minimize variance in temperature and evaporative effects across the plate (Lundholt et al., 2003). All respiratory parameters were corrected for non-mitochondrial respiration and background signal from the instrument with addition of 200 nM rotenone and 1 μM antimycin A. Where appropriate, oligomycin was used at 2 μM unless otherwise specified, and FCCP concentrations were titrated to determine an optimal concentration for a given experiment. Unbuffered DMEM assay medium is composed of DMEM (Sigma #5030; pH 7.4) supplemented with 31.6 mM NaCl, 3 mg/l phenol red, and 5 mM HEPES unless otherwise indicated.

**XF analysis primary cortical neurons**

All experiments with primary cortical neurons were conducted in custom medium lacking glutamine as described earlier.

**Calculation of lactate:H+ and H+-O2**

Rates of medium acidification without contribution from respiration were calculated as follows: DMEM assay medium was supplemented with 5 mM glucose and varying concentrations of 2-deoxyglucose (2-DG) and 5 mM iodoacetate (IA) where indicated in the figure. Medium was also supplemented with 200 μM rotenone (to block complex I), 1 μM antimycin A (to block complex III), and 2 μM oligomycin (to block ATP hydrolysis and prevent an energy crisis). Rates were calculated for 75 min, and the spent medium was immediately harvested from each plate and frozen at −80°C for later calculation of endpoint lactate concentration by enzymatic assay (Mookerjee et al., 2015). Endpoint lactate values were used to calculate lactate efflux rates.

Rates of medium acidification in the absence of glycolysis were calculated as follows: DMEM assay medium was supplemented with 5 mM pyruvate and 5 mM glucose (no glucose was provided) along with 2 mM 2-DG and 50 μM IA (to block residual glycolysis and glycolenolysis). 500 U/ml carbonic anhydrase was added to ensure rapid CO2 hydration, though it was later determined to have a negligible effect on rates of acidification. Respiratory rates were titrated by addition of oligomycin, FCCP (500 nM or 1 μM), and rotenone with antimycin A. The medium was also supplemented with varying concentrations of both UK5099 and BPTES (3 nM to 3 μM for both) to further titrate the respiratory rate. Experiments with primary cortical neurons were conducted in custom medium lacking glutamine as described earlier.

**Patient-derived glioblastoma cultures**

Patient-derived high-grade gliomas were isolated, purified, and maintained as cell lines or implanted as orthotopic xenografts according to previously published protocols (Gosa et al., 2019). Cultured cells (in vitro) and tumor cells acutely purified from xenografts (ex vivo) were seeded onto Cell-Tak-coated XF96 plates (25 μg/well; Corning). Cells were plated at either 4.0 × 10^4 or 1.0 × 10^5 based on cell size and the initial oxygen consumption rate, and spun at 600 g for 5 min to adhere cells to the well.

**Cell activation assays**

Cortical neurons were plated at 2.5 × 10^4 cells/well and acutely activated with 1.5 μM veratridine added via the injector port of the instrument (Choi et al., 2009). Where indicated, cells were pretreated with 2 nM ouabain immediately prior to the assay. When activating the NMDA receptor, 100 μM NMDA was acutely given via the injector port to neurons with or without 15 min. pre-treatment of 10 μM MK-801. For these experiments, the custom assay medium was supplemented with 2 mM β-hydroxybutyrate as substrate, as pyruvate could not be used due to complications with measuring glutamate in the medium by a linked enzymatic assay to confirm depolarization and glutamate efflux. For acute, T cell activation, murine anti-CD3/CD28 Dynabeads (Gibco) were added via the injector port as previously described (Gubser et al., 2013).

**LPS-mediated activation of BMDMs**

BMDMs were plated at 5.0 × 10^4 cells/well for 48 h, prior to cyto-kine treatment, which lasted for 24 h, unless otherwise specified. Lipopolysaccharide (LPS) was purchased from PeproTech, Inc. (New Jersey, USA) and offered to cells at 50 ng/ml unless otherwise specified in the manuscript.

**H+-O2 in isolated heart mitochondria**

Isolated rat heart mitochondria from male Sprague–Dawley rats between 3 and 4 months old were plated in XF96 plates (1.5 μg/well) or XF24 (4 μg/well) and spun 2,000 g for 20 min to adhere mitochondria to the well. Assays were conducted in MAS medium (220 mM mannitol, 70 mM sucrose, 10 mM KH2PO4, 5 mM MgCl2,
1 mM EGTA, and 2 mM HEPES (pH 7.2]) supplemented with 0.2% (w/v) fatty acid-free BSA. For all conditions, uncoupler-stimulated respiration was measured in the presence of 2 μM oligomycin and 4 μM FCCP to eliminate effects of ATP synthesis or hydrolysis on the acidification rate. Rates of respiration and acidification were measured under three conditions: (i) pyruvate/malate (P/M): 10 mM pyruvate with 1 mM malate and 2 mM dichloroacetate; (ii) pyruvate/malate with an inhibitor mix to block various enzymes to run a truncated TCA cycle (P/M/truncated): 10 mM pyruvate with 1 mM malate, 2 mM dichloroacetate, 60 μM fluorocitrate (to block isocitrate dehydrogenase), 2 mM malonate (to block succinate dehydrogenase), and 1 mM aminooxyacetate (to block transaminase activity), and (iii) succinate/rotenone (S/R): 10 mM succinate with 2 μM rotenone to consume oxygen without release of CO₂ to acidify the experimental medium.

**Quantification and statistical analysis**

All statistical parameters, including the number of replicates (n), can be found in the figure legends. Statistical analyses were performed using Graph Pad Prism 5 software for Mac OS X. Data are presented as the mean ± SEM unless otherwise specified. Individual pairwise comparisons were performed using two-tailed Student's t-test. For experiments involving two or more groups, data were analyzed by one-way, repeated measures ANOVA followed by Dunnett’s post hoc multiple comparisons tests. Data were assumed to follow a normal distribution (no tests were performed). Values denoted as follows were considered significant: *P < 0.05; **P < 0.01; ***P < 0.001.

**Step-by-step protocol**

The protocol provided here is written alongside the Datasets EV1–EV5 for the calculation of ATP production rates. Established, time-tested protocols are available for conducting standardized respirometry assays with the Seahorse XF Analyzer (Pelletier et al., 2014) as well as guidelines for best practices (Divakaruni et al., 2014; Divakaruni & Jastroch, 2022). As such, this protocol focuses specifically on the calculation of the buffering power of the experimental medium and subsequent ATP production rates from a Seahorse XF assay. These calculations are amenable to standard protocols for respirometry profiling (i.e. measurements of OCR and ECAR in response to sequential additions of oligomycin, FCCP, and rotenone with antimycin A.). The measurement times at which data points are collected follow standard best practices (Divakaruni et al., 2014; Divakaruni & Jastroch, 2022), and examples are provided in the Datasets EV1–EV4. Any deviations from standard Seahorse XF analysis are explicitly noted.

**Assay preparation**

1. If using adherent cells, plate cells of interest on XF96 microplates at the optimal, previously determined density day(s) prior to the assay according to standard best practices. Only plate cells in the inner 60 wells of the microplate to mitigate deleterious effects from temperature and humidity gradients. Cells grown in suspension are to be acutely spun down onto an adhesive-coated microplate immediately prior to the assay. For all assays, columns 1 and 12 of the microplate must not contain cells, as these wells will be used for acid calibrations and determination of the buffering power of the medium.

2. Prepare assay medium by dissolving DMEM powder into 1 l water and adding 1.85 g NaCl (31.6 mM; to control for osmolarity from lack of bicarbonate), 600 μl phenol red (3 mg/l final), and 5 ml HEPES (5 mM final). Fill to 1 l and adjust pH to 7.4 at 37°C.

3. To this base medium add oxidizable substrates of interest. For the experiments presented here, assay medium is composed of this base supplemented with 8 mM glucose, 2 mM glutamine, and 2 mM pyruvate.

4. Prepare injectate solutions in assay medium for standard respirometry profiling (e.g. oligomycin, FCCP, rotenone, and antimycin A) according to established protocols.

5. Prepare acid solutions to be injected in each port (25 μl of either 2.5 mM HCl or 1.25 mM H₂SO₄).
Load injector ports A–D in columns 2–11 with injectates/effector compounds from Step #4.

Load injector ports A-D in columns 1 and 12 with HCl or H2SO4 solution from step #5. The calculations presented are for 25 μl loaded in each port with a starting assay volume of 150 μl.

Follow manufacturer’s instructions and/or best practices for warming the Seahorse XF assay cartridge and preparing assay template. In general, a measurement period of 3 min and 2–3 min of mixing is ideal for this assay to allow for stabilization of the pH readings to assess the buffering power.

While the sensor cartridge is calibrating, wash the cells (including background wells) a minimum of three times according to manufacturer’s instructions and/or best practices. For columns 1 and 12 used for the acid calibrations, use an aspirator and completely remove the medium, and repeat by filling the well with assay medium and fully aspirating. This is to ensure no remaining growth medium or PBS will affect the buffering power calculations. The example shown here uses a starting assay volume of 150 μl, but the calculations can be adjusted to accommodate any starting volume appropriate for the instrument.

Run the Seahorse XF assay according to manufacturer’s instructions and/or best practices.

Normalize the data according to best practices and available equipment. The data presented here are normalized using nuclear staining (Hoescht) and post hoc high-content imaging of the XF microplate (Perkin-Elmer Operetta), but this may not be feasible for all. Guidelines for other, practical means of normalization are discussed in Divakaruni & Jastroch (2022).

**Calculation of buffering power (Dataset EV1)**

Calculate the average value of the last 5 pH recordings prior to the injection of an acid solution (or injectates used in background wells) as shown in rows 3–30.

Calculate the change in pH after each injection. Correct for any background drift as shown in cells F32-38

Plot the change in pH against the amount of acid added to the ground wells) as shown in rows 3–30.

While the sensor cartridge is calibrating, wash the cells (including background wells) a minimum of three times according to manufacturer’s instructions and/or best practices. For columns 1 and 12 used for the acid calibrations, use an aspirator and completely remove the medium, and repeat by filling the well with assay medium and fully aspirating. This is to ensure no remaining growth medium or PBS will affect the buffering power calculations. The example shown here uses a starting assay volume of 150 μl, but the calculations can be adjusted to accommodate any starting volume appropriate for the instrument.

**Conversion of ECAR to pmol H+/min (Dataset EV2)**

15 Calculate the average rate of the last two ECAR reading prior to the first injection. This averaged ECAR is then divided by the calculated buffering power yielding the proton production rate (see equation in worksheet). Repeat for each technical replicate. Sample calculations of H+ production rates for macrophages treated with 50 ng/ml of LPS are presented.

**Calculation of required OCR parameters (Dataset EV3)**

16 Calculate the basal mitochondrial respiration rate, ATP-linked respiration rate, and proton leak-linked respiration rate using the average of the last two measurements prior to injection.

Parameters and best practices are defined in (Divakaruni et al., 2014; Divakaruni & Jastroch, 2022) are determined for each technical replicate presented here. Sample calculations of OCR parameters for macrophages treated with 50 ng/ml of LPS are presented.

**Calculation of ATP production rates (Dataset EV4)**

As detailed in the manuscript text, apply the following equations to the OCR and H+ production rate parameters obtained in EV2 and EV3 to calculate ATP production rates from oxidative phosphorylation (ATP_{OxPhos}) and glycolysis (ATP_{Glyco}). Sample calculations of ATP production rates for macrophages treated for 24 h. with 50 ng/ml of LPS are presented.

\[
\text{ATP}_{\text{OxPhos}} = 5.45 \cdot \text{OCR}_{\text{ATP}} + 0.1 \cdot \text{OCR}_{\text{Leak}}
\]

\[
\text{ATP}_{\text{Glyco}} = 1.53 \cdot (\text{H}^+_{\text{Total}} - 0.38 \cdot \text{OCR}_{\text{Mito}})
\]

\[
\text{ATP}_{\text{Total}} = \text{ATP}_{\text{OxPhos}} + \text{ATP}_{\text{Glyco}}
\]

**Data availability**

This study includes no data deposited in external repositories.

**Expanded View** for this article is available online.

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**Author contributions**

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Disclosure and competing interests statement

NR, GWR, PS, and AN are currently employees and shareholders of Agilent Technologies. BPD, and DAF were previously employees and shareholders of Agilent Technologies at the time of this study. ASD has previously served as a paid consultant for Agilent Technologies.

References


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