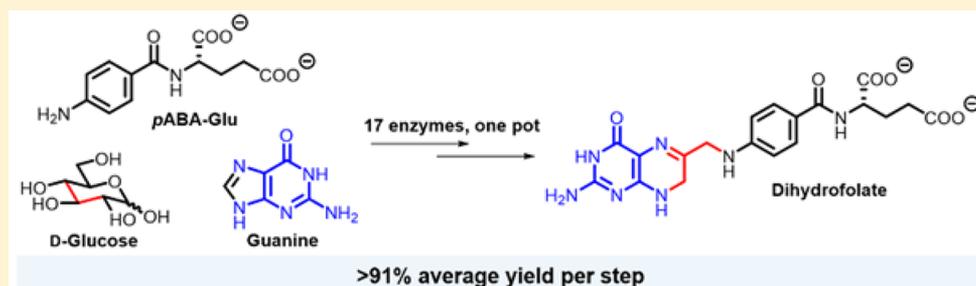


Chemoenzymatic Assembly of Isotopically Labeled Folates

Antonio Angelastro, William M. Dawson,[†] Louis Y. P. Luk, E. Joel Loveridge,[‡] and Rudolf K. Allemann^{*†}

School of Chemistry, Cardiff University, Park Place, Cardiff CF10 3AT, United Kingdom

Supporting Information



ABSTRACT: Pterin-containing natural products have diverse functions in life, but an efficient and easy scheme for their in vitro synthesis is not available. Here we report a chemoenzymatic 14-step, one-pot synthesis that can be used to generate ^{13}C - and ^{15}N -labeled dihydrofolates (H_2F) from glucose, guanine, and *p*-aminobenzoic acid-L-glutamic acid. This synthesis stands out from previous approaches to produce H_2F in that the average yield of each step is >91% and it requires only a single purification step. The use of a one-pot reaction allowed us to overcome potential problems with individual steps during the synthesis. The availability of labeled dihydrofolates allowed the measurement of heavy-atom isotope effects for the reaction catalyzed by the drug target dihydrofolate reductase and established that protonation at N5 of H_2F and hydride transfer to C6 occur in a stepwise mechanism. This chemoenzymatic pterin synthesis can be applied to the efficient production of other folates and a range of other natural compounds with applications in nutritional, medical, and cell-biological research.

INTRODUCTION

Pterin is a common motif found in natural products. Folate, the essential vitamin that fuels the one-carbon cycle for the biosynthesis of nucleotide and amino acid building blocks, was one of the first natural products found to contain pterin.^{1,2} The metabolic importance of pterins is illustrated by their integration into enzyme cofactors such as molybdopterin and tetrahydrobiopterin.^{3–5} Pterin natural products are also used as pigments in the butterflies *Catopsilia argante* and *Appias nero*,⁶ whereas biopterin- α -glucoside serves as a natural sunscreen that protects cellular contents from photoinduced damage in photosynthetic cyanobacteria.⁷

5,6,7,8-Tetrahydrofolic acid (H_4F), which in addition to the pterin ring system contains *p*-aminobenzoic acid (*p*ABA) and L-glutamic acid (Glu), is required for the biosynthesis of metabolites that are key for cell survival and replication.² A one-carbon unit in different oxidation states can be added at N5 and/or N10 of H_4F and used to produce metabolites such as thymidylate, purines, glycine, serine, and S-adenosylmethionine (SAM) (Figure 1).⁸ Because of the central importance of folate biochemistry for cell replication and survival, dihydrofolate reductase (DHFR), thymidylate synthase (TS), and serine hydroxymethyltransferase (SHMT) have long been exploited as important drug targets in the treatment of bacterial infections,⁹ malaria,^{10,11} and cancer,^{12,13} and the DHFR-targeting drugs trimethoprim, proguanil, pyrimethamine, and methotrexate are

listed as essential medicines by the World Health Organization (WHO).¹⁴ Nevertheless, as with many clinically used drugs, resistance to antifolates has begun to emerge,^{15–17} and investigation of the enzymes of the one-carbon cycle is an important part of inhibitor design strategies.¹⁸

Detailed mechanistic insight into enzyme-catalyzed reactions is often obtained by isotopic labeling and measurement of kinetic isotope effects (KIEs)^{19–22} or spectroscopic analysis.^{23,24} Information derived from regio- and stereospecific substrate labeling has been used to design inhibitors with dissociation constants in the micro- to picomolar range.²⁵ However, the use of these techniques to investigate folate-dependent enzymes is hindered by the absence of a general and efficient method to specifically label atoms of the pterin ring system, particularly at N5, C6, C7, and C9, which are directly linked to the chemistry of the catalyzed reactions. Folate and its derivatives can be synthesized by connecting the pterin, *p*ABA, and glutamate groups in sequential order,²⁶ and several synthetic strategies to incorporate an isotopic label into pterin in a regiospecific manner have been reported.^{27–30} Pterins have been synthesized by condensing guanidine or dihydroxyacetone with the respective heterocyclic starting materials, and N5-, C2-, and C6-labeled folates have been made previously.^{27,28} However, because symmetric

Received: June 19, 2017

Published: August 18, 2017

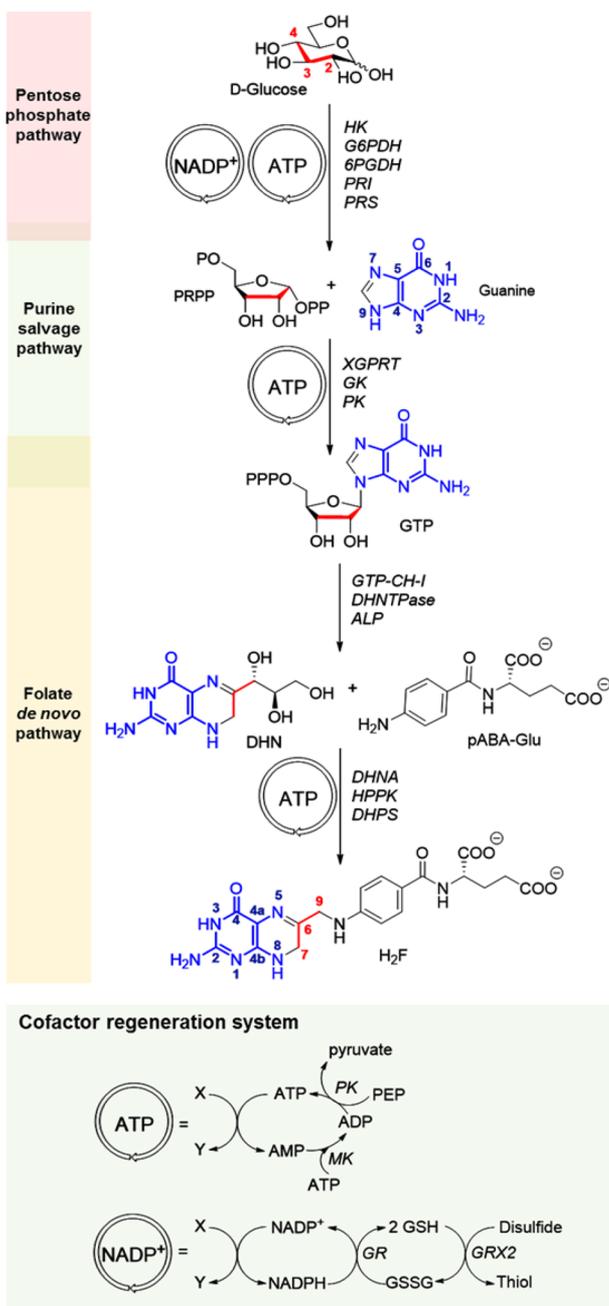


Figure 2. Strategy for the synthesis of 7,8-dihydrofolate (H_2F). Guanosine triphosphate (GTP) was made via the pentose phosphate and purine salvage pathways under hexokinase (HK), glucose 6-phosphate dehydrogenase (G6PDH), 6-phosphogluconate dehydrogenase (6PGDH), phosphoriboisomerase (PRI), ribose-phosphate pyrophosphokinase (PRS), xanthine guanine phosphoribosyl transferase (XGPRT), guanylate kinase (GK), and pyruvate kinase (PK) catalysis. In the folate de novo pathway, GTP is converted to 7,8-dihydroneopterin (DHN) under catalysis by GTP cyclohydrolase I (GTP-CH-I), 7,8-dihydroneopterin pyrophosphatase (DHNTPase), and alkaline phosphatase (ALP); DHN is then converted to H_2F under catalysis by dihydroneopterin aldolase (DHNA), 6-hydroxymethyl 7,8-dihydropterin pyrophosphokinase (HPPK), and dihydropterate synthase (DHPS). The additional enzymes myokinase (MK), glutathione reductase (GR), and glutaredoxin 2 (GRX2) are used for the regeneration of ATP and $NADP^+$.^{44,45} The pterin atoms of H_2F derived from glucose (red) and guanine (blue) are highlighted. Details of each biosynthetic step are described in Figure S2.

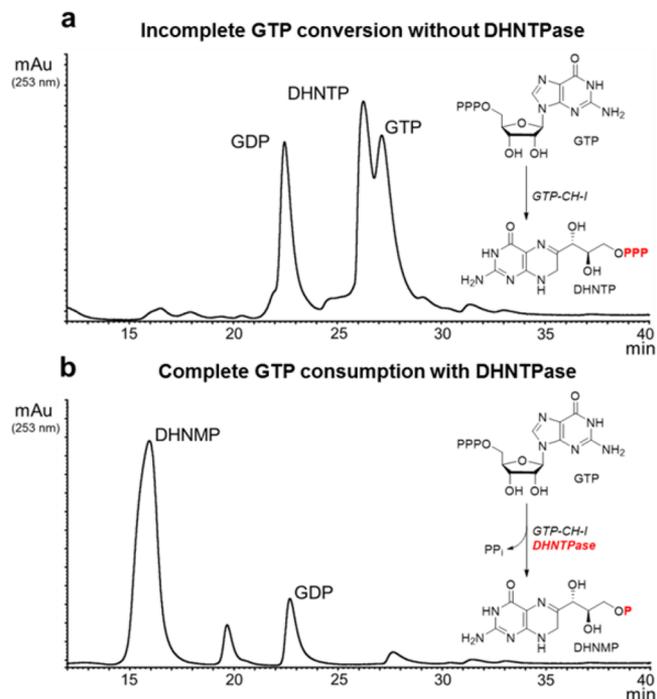


Figure 3. Conversion of guanosine triphosphate (GTP) into 7,8-dihydroneopterin triphosphate (DHNTp) and monophosphate (DHNMP). Shown are anion-exchange chromatography analyses of GTP incubated for 2 h at 37 °C with (a) GTP cyclohydrolase I (GTP-CH-I), which shows partial conversion to DHNTP, and (b) both GTP-CH-I and 7,8-dihydroneopterin triphosphate pyrophosphohydrolyase (DHNTPase), resulting in essentially complete conversion to DHNMP. Production of guanosine diphosphate (GDP) is most likely due to the nonenzymatic hydrolysis of GTP during incubation.

p-aminobenzoyl-L-glutamate (*p*ABA-Glu). In the biosynthetic scheme of H_2F (Figure 2), C2, C3, and C4 of glucose correspond to C7, C6, and C9 of H_2F and N1, C2, N3, C4, C5, C6, N7, and N9 of guanine correspond to N3, C2, N1, C4b, C4a, C4, N5, and N8 in H_2F . GTP is the key intermediate in this synthetic pathway, connecting the purine salvage pathway to de novo folate biosynthesis. Accordingly, a minimum of 14 enzymes need to be assembled *in vitro* to produce H_2F .

The GTP biosynthetic pathway is composed of enzymes from the pentose phosphate and purine salvage pathways (Figures 2 and S2). D-Glucose serves as the starting material, which is transformed into phosphoribose pyrophosphate (PRPP) in five steps that are catalyzed by hexokinase (HK), glucose 6-phosphate dehydrogenase (G6PDH), 6-phosphogluconate dehydrogenase (6PGDH), phosphoriboisomerase (PRI), and ribose-phosphate pyrophosphokinase (phosphoribosylpyrophosphate synthetase, PRS).⁴⁴ PRPP is then combined with guanine to form GMP under xanthine guanine phosphoribosyl transferase (XGPRT) catalysis. The resulting GMP is converted to the corresponding nucleotide triphosphate in reactions catalyzed by guanylate kinase (GK) and pyruvate kinase (PK). Since HK and GK use ATP as the phosphate source, PK can also function as the recycling enzyme. On the other hand, PRS uses ATP as the pyrophosphate source, so myokinase (MK) was included to regenerate ATP from AMP.⁴⁵ A significant amount of $NADP^+$ is also needed for GTP biosynthesis. Hence, the recently developed glutathione reductase (GR)/glutaredoxin 2 (GRX2) recycling system was used to regenerate the oxidized cofactor.⁴⁴ The GR/GRX2 system uses disulfides like 2-hydroxyethyl

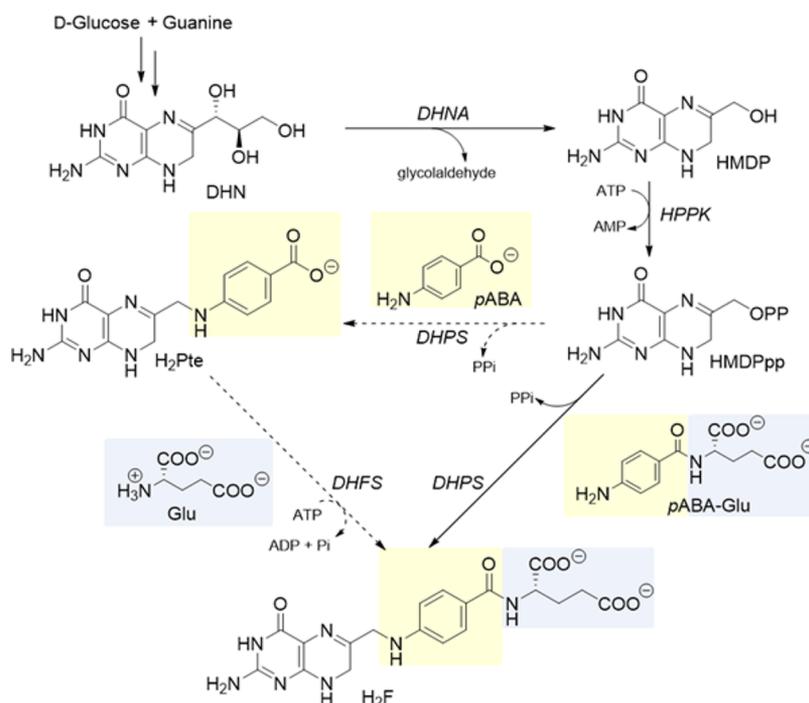


Figure 4. Conversion of 7,8-dihydroneopterin (DHN) to dihydrofolate (H_2F). Dihydroneopterin aldolase (DHNA) catalyzes the transformation of DHN to 6-hydroxymethyl-7,8-dihydropterin (HMDP), which is pyrophosphorylated by 6-hydroxymethyl 7,8-dihydropterin pyrophosphokinase (HPPK). The resulting 6-hydroxymethyl-7,8-dihydropterin pyrophosphate (HMDPpp) is converted to H_2F by dihydropterate synthase (DHPS), whose promiscuity allows the use of preassembled *pABA*-Glu as the substrate and the omission of dihydrofolate synthase (DHFS) in the *in vitro* synthetic pathway.

disulfide (HED or oxidized β -mercaptoethanol) or cystine as regenerating reagents and produces thiols as useful byproducts that protect the enzymes and intermediates from oxidative damage.

The conversion of GTP to DHNTP by GTP-CH-I marks the entry point of the folate *de novo* pathway. It has been reported that potassium and magnesium ions are positive allosteric effectors that can increase the rate of the GTP-CH-I reaction up to 5-fold.⁴⁶ The addition of these cations, however, was insufficient, as the reaction was found to be incomplete, giving a poor yield of DHNTP (Figure 3a). In folate *de novo* biosynthesis, dephosphorylation of DHNTP to 7,8-dihydroneopterin monophosphate (DHNMP) by DHNTP pyrophosphohydrolase (DHNTPase) is the biochemical step followed by the GTP-CH-I reaction (Figure 2).^{47,48} Knockout of the DHNTPase gene significantly impairs folate metabolism in *E. coli*,⁴⁸ which suggests that DHNTP phosphohydrolysis is a key regulatory step in folate metabolism. In other words, the activity of GTP-CH-I is most likely inhibited by its own product, DHNTP, which therefore needs to be immediately converted to DHNMP in order to sustain the activity of GTP-CH-I. In the presence of DHNTPase, GTP-CH-I showed a marked rate enhancement (Figure S3), with nearly complete conversion of GTP and a high yield of DHNMP (Figure 3b).

Additional enzymes are needed to convert DHNMP into folate. To the best of our knowledge, the natural enzyme responsible for the conversion of DHNMP to 7,8-dihydroneopterin (DHN) is unknown,^{37,48} and alkaline phosphatase (ALP) was used instead as a surrogate. DHN is subjected to a retro-aldol reaction catalyzed by dihydroneopterin aldolase (DHNA) to yield 6-hydroxymethyl-7,8-dihydropterin (HMDP),^{49,50} which then reacts with ATP in the presence of 6-hydroxymethyl 7,8-dihydropterin pyrophosphokinase (HPPK) (Figure 4).⁵¹

In *E. coli*, the resulting intermediate, 6-hydroxymethyl-7,8-dihydropterin pyrophosphate (HMDPpp), is first condensed with *pABA* and then with glutamate, catalyzed by dihydropterate synthase (DHPS)⁵² and dihydrofolate synthase (DHFS),⁵³ respectively, to finally generate H_2F . However, DHPS accepts preassembled *pABA*-Glu as a substrate,⁵⁴ so DHFS is not required in the *in vitro* reaction. The entire H_2F synthetic pathway requires only one purification step of the product, but ALP needs to be removed by ultrafiltration prior to the addition of DHNA, HPPK, and DHPS because the phosphatase can also catalyze the phosphorolysis of ATP and HMDPpp. Two additional modifications were made to further improve the overall yield. A N_2 -filled glovebox system was used because all reduced pterin-containing compounds, DHNTP, DHNMP, DHN, HMDP, HMDPpp, and the final product H_2F , are oxygen-sensitive.⁴¹ Also, cystine was found to be the preferred reagent over HED for the $NADP^+$ regeneration system operated by GR and GRX2. Perhaps β -mercaptoethanol made from the reduction of HED interferes with other enzymatic reactions, such as the chelation of Zn^{2+} in GTP-CH-I. The total turnover numbers for the regeneration of ATP from ADP by pyruvate kinase and from AMP by pyruvate kinase/myokinase are both ~ 100 ,⁵⁵ while the total turnover number for our GR/GRX2-based $NADP^+$ recycling system can reach 5×10^5 .⁴⁴ In general, a typical biosynthetic cascade produced 6.6 mg of H_2F from 9 mg of glucose in an overall yield of 30%, i.e., the average yield of each chemical transformation is in excess of 91%.

Synthesis of Selectively Labeled Folates. Five isotopically labeled H_2F s were synthesized using the newly developed *in vitro* pathway (Figure 5a). Liquid chromatography–high-resolution mass spectrometry (LC-HRMS) analysis of $[6-^{13}C]H_2F$, produced from $[3-^{13}C]D$ -glucose, showed an increase of ~ 1 amu; ^{13}C NMR spectroscopy revealed a singlet at 152 ppm.

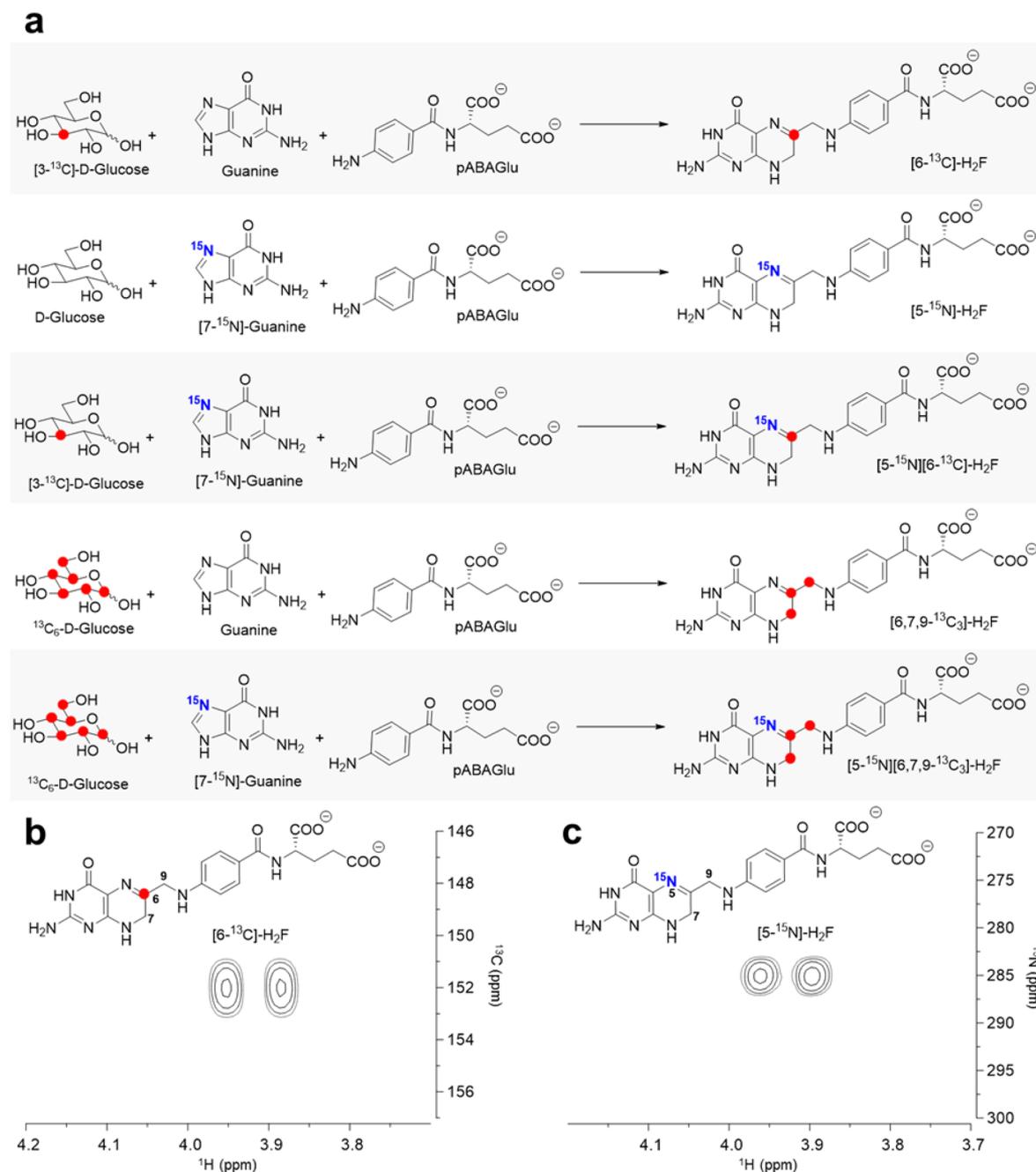


Figure 5. H₂Fs labeled with stable isotopes on the pterin ring system. (a) Patterns of isotopic distribution are determined by the starting material isotopic enrichment used during H₂F biosynthetic assembly. (b) ¹H,¹³C HMBC and (c) ¹H,¹⁵N HMBC spectra of [6-¹³C]H₂F and [5-¹⁵N]H₂F. Protons attached to C7 (3.95 ppm) and C9 (3.87 ppm) correlate to C6 (152 ppm) or N5 (285 ppm). ¹H NMR spectra of all compounds and additional 2D NMR characterization of [5-¹⁵N][6-¹³C]H₂F, [6,7,9-¹³C₃]H₂F and [5-¹⁵N][6,7,9-¹³C₃]H₂F are reported in Figures S12–S15. HRMS data for all of the compounds are given in Table S3 and Figures S16–S21. Isotopic enrichment was calculated to be at least 97% from the mass spectrometric data (as illustrated in Figures S22–S24).

Additionally, long-range coupling between 6-¹³C and protons on C7 and C9 of [6-¹³C]H₂F were observed in the ¹H,¹³C HMBC spectrum (Figure 5b).

To incorporate an ¹⁵N label into the pterin moiety at N5, [7-¹⁵N]guanine was used. The resulting product showed ~1 amu increase in LC-HRMS analysis and an ¹⁵N signal at 285 ppm coupling to C7 and C9 protons in the ¹H,¹⁵N HMBC spectrum (Figure 5c).

When [3-¹³C]D-glucose and [7-¹⁵N]guanine were combined to produce [5-¹⁵N][6-¹³C]H₂F, a mass increase of ~2 amu was measured. The ¹³C NMR spectrum showed a doublet at 152 ppm

with a coupling constant ¹J_{CN} of 7.5 Hz; long-range coupling between the protons on C7 and C9 was observed in the ¹H,¹³C HMBC and ¹H,¹⁵N HMBC spectra (see the Supporting Information), indicating that both N5 and C6 of H₂F were isotopically enriched.

Similarly, [6,7,9-¹³C₃]H₂F and [5-¹⁵N][6,7,9-¹³C₃]H₂F were synthesized from ¹³C₆-D-glucose and [7-¹⁵N]guanine, and their identities were confirmed by HRMS and NMR spectroscopy (see the Supporting Information).

Heavy-Atom Kinetic Isotope Effects on the Reaction Catalyzed by EcDHFR. The preparation of ¹³C- and ¹⁵N-labeled

dihydrofolates allowed the measurement of heavy-atom isotope effects for the reactions catalyzed by dihydrofolate reductase (DHFR), a key enzyme in one-carbon metabolism and a validated target for the treatment of bacterial infections, malaria, and cancer.⁵⁶ DHFR catalyzes the reduction of H₂F to H₄F via transfer of the pro-*R* hydride from C4 of NADPH to the *Re* face on C6 accompanied by protonation of N5 of H₂F (Figure 6).^{57,58}

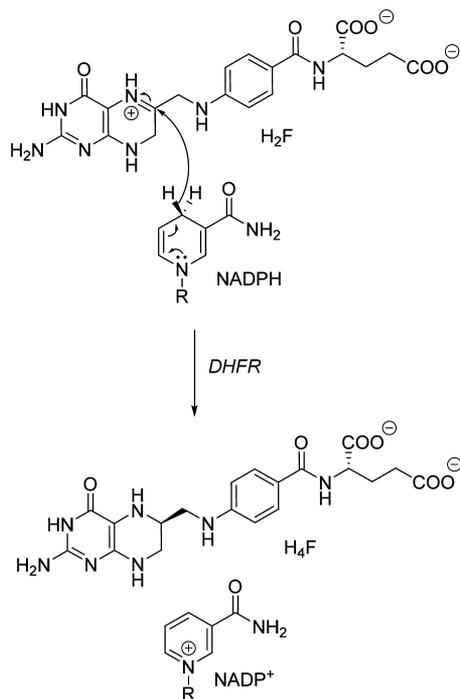


Figure 6. Reduction of 7,8-dihydrofolate (H₂F) to 5,6,7,8-tetrahydrofolate (H₄F) catalyzed by dihydrofolate reductase (DHFR).

Several aspects of the reaction mechanism warrant additional investigation. In particular, the transition state structure and the order of chemical transformation events have not been fully determined.^{59,60} The active site of DHFR provides a favorable environment for protonation of N5 by elevating the pK_a from 2.6 to 6.5 and using an active-site water as the proton source.^{61,62} Solvent and hydrogen KIE measurements combined with site-directed mutagenesis have suggested a stepwise mechanism in which protonation precedes hydride transfer.^{63,64} However, D₂O increases the viscosity of the reaction buffer relative to H₂O,^{65,66} and site-directed modification can alter the catalytic behavior of an enzyme.⁶⁷ Thus, additional mechanistic investigations are needed to establish the order of events. Since isotopic substitution does not alter the chemistry of the reaction but only the kinetics, [6-¹³C]H₂F and [5-¹⁵N]H₂F were used to measure the ¹⁵N and ¹³C heavy-atom kinetic isotope effects. Pre-steady-state kinetic measurements at 15 °C by fluorescence resonance energy transfer from the active-site tryptophan in DHFR to the reduced cofactor yield first-order hydride transfer rate constants with an accuracy of up to 0.7% (Figures S25–S27 and Table S4). While a ¹³C KIE of 1.015 ± 0.006 was observed for the reduction of [6-¹³C]H₂F, the corresponding ¹⁵N KIE for [5-¹⁵N]H₂F was essentially unity (0.999 ± 0.006) under the same conditions. To confirm this finding, [5-¹⁵N][6-¹³C]H₂F was used to probe both positions at the same time, and the measured value for the corresponding multiple heavy-atom KIE was 1.014 ± 0.008, which is statistically identical to that obtained when the substrate was labeled with ¹³C only.

The observed ¹⁵N KIE on hydride transfer indicates that protonation of N5 is not isotopically sensitive, likely because it is not rate-limiting under pre-steady-state conditions (as the reaction is essentially irreversible,⁵⁸ the observed KIE will tend to unity rather than to the equilibrium isotope effect).²² On the other hand, the measured ¹³C KIE indicates that the hydride transfer step is rate-determining. This strongly suggests a stepwise mechanism. If the protonation and hydride transfer steps were concerted, ¹⁵N- and ¹³C-labeled H₂Fs should both yield measurable KIEs;^{19,21,22} this interdependence may also lead to an additive effect in the multiple heavy-atom isotope effect measurement with the double-labeled substrate.^{19,21,22} In other words, our results suggest that the pre-steady-state kinetic measurement at pH 7.0 reveals only the step of hydride transfer because protonation of N5 is in rapid equilibrium and the ensemble of reaction-ready conformations is mostly populated with protonated H₂F. Importantly, these results confirm the validity of previous solvent KIE and site-directed mutagenesis studies, which also concluded that the sequence of chemical events (protonation and hydride transfer) is distinct and strictly ordered.^{64,68} Overall, the results provided here strongly support a mechanism where protonation and hydride transfer are independent of each other and occur in a stepwise fashion.

CONCLUSIONS

Dihydrofolate was produced enzymatically in an easy one-pot, high-yielding reaction sequence from glucose, guanine, and *p*ABA-Glu that required only a single purification step. Potential problems with individual steps during the synthesis could be overcome through the use of a one-pot reaction. This methodology can be used to generate dihydrofolates labeled in specific positions with stable isotopes with average overall yields of >30%, facilitating many applications in cell biology and mechanistic enzymology.^{27–29,61,69,70} For the first time, heavy-atom KIEs for the DHFR-catalyzed reduction of H₂F could be measured to provide strong support for a stepwise reduction of the substrate in which protonation at N5 and hydride transfer from C4 of the NADPH to C6 of protonated dihydrofolate proceed independently. This chemoenzymatic pterin synthesis can be integrated into other enzymatic procedures to generate folate derivatives^{31,32} and other high-value natural products that are not easily accessible by conventional synthesis.⁷¹ It can be applied to nutritional, medical, and cell-biological research to address questions of in vivo bioavailability and to explore the kinetics of folate metabolism in intact cells and organisms.^{70,72–76}

ASSOCIATED CONTENT

Supporting Information

The Supporting Information is available free of charge on the ACS Publications website at DOI: 10.1021/jacs.7b06358.

Full experimental procedures and supplementary figures and tables (PDF)

AUTHOR INFORMATION

Corresponding Author

*allemannrk@cardiff.ac.uk

ORCID

William M. Dawson: 0000-0003-2710-6879

E. Joel Loveridge: 0000-0002-8528-4019

Rudolf K. Allemann: 0000-0002-1323-8830

Present Addresses

[†]W.M.D.: School of Chemistry, University of Bristol, Cantock's Close, Bristol BS8 1TS, United Kingdom.

[‡]E.J.L.: Department of Chemistry, Swansea University, Singleton Park, Swansea SA2 8PP, United Kingdom.

Notes

The authors declare no competing financial interest.

ACKNOWLEDGMENTS

We are grateful to Dr. James R. Williamson (Scripps Research Institute) for his gift of *prsA*. The help of Dr. Rob Jenkins and Mr. Thomas Williams with mass spectrometry is gratefully acknowledged. This work was supported by Cardiff University through a President's Research Scholarship to A.A. and the UK's Biotechnology and Biological Sciences Research Council (BBSRC) through Grants BB/J005266/1 and BB/L020394/1 to R.K.A.

REFERENCES

- (1) Hoffbrand, A. V.; Weir, D. G. *Br. J. Haematol.* **2001**, *113*, 579–589.
- (2) Fox, J. T.; Stover, P. J. *Vitam. Horm.* **2008**, *79*, 1–44.
- (3) Basu, P.; Burgmayer, S. J. *Coord. Chem. Rev.* **2011**, *255*, 1016–1038.
- (4) Schwarz, G.; Mendel, R. R.; Ribbe, M. W. *Nature* **2009**, *460*, 839–847.
- (5) Thöny, B.; Auerbach, G.; Blau, N. *Biochem. J.* **2000**, *347*, 1–16.
- (6) Pfeleiderer, W. *Adv. Exp. Med. Biol.* **1993**, *338*, 1–16.
- (7) Noguchi, Y.; Ishii, A.; Matsushima, A.; Haishi, D.; Yasumuro, K.; Moriguchi, T.; Wada, T.; Koderu, Y.; Hiroto, M.; Nishimura, H.; Sekine, M.; Inada, Y. *Mar. Biotechnol.* **1999**, *1*, 207–210.
- (8) Ducker, G. S.; Rabinowitz, J. D. *Cell Metab.* **2017**, *25*, 27–42.
- (9) Murima, P.; McKinney, J. D.; Pethe, K. *Chem. Biol.* **2014**, *21*, 1423–1432.
- (10) Nzila, A.; Ward, S. A.; Marsh, K.; Sims, P. F.; Hyde, J. E. *Trends Parasitol.* **2005**, *21*, 292–298.
- (11) Nzila, A.; Ward, S. A.; Marsh, K.; Sims, P. F.; Hyde, J. E. *Trends Parasitol.* **2005**, *21*, 334–339.
- (12) Locasale, J. W. *Nat. Rev. Cancer* **2013**, *13*, 572–583.
- (13) Yang, M.; Vousden, K. H. *Nat. Rev. Cancer* **2016**, *16*, 650–662.
- (14) *WHO Model Lists of Essential Medicines*, 19th ed.; World Health Organization: Geneva, Switzerland, 2015.
- (15) Zhao, R.; Goldman, I. D. *Oncogene* **2003**, *22*, 7431–7457.
- (16) Müller, I. B.; Hyde, J. E. *Mol. Biochem. Parasitol.* **2013**, *188*, 63–77.
- (17) Sköld, O. Sulfonamides and Trimethoprim. In *Antimicrobial Drug Resistance*; Mayers, D. L., Ed.; Humana Press: New York, 2009; Vol. 1, pp 259–269.
- (18) Copeland, R. A.; Harpel, M. R.; Tummino, P. J. *Expert Opin. Ther. Targets* **2007**, *11*, 967–978.
- (19) Cleland, W. W. *Arch. Biochem. Biophys.* **2005**, *433*, 2–12.
- (20) Vardi-Kilshtain, A.; Nitoker, N.; Major, D. T. *Arch. Biochem. Biophys.* **2015**, *582*, 18–27.
- (21) Cook, P. F. *Isot. Environ. Health Stud.* **1998**, *34*, 3–17.
- (22) O'Leary, M. H. *Annu. Rev. Biochem.* **1989**, *58*, 377–401.
- (23) Wharton, C. W. *Nat. Prod. Rep.* **2000**, *17*, 447–453.
- (24) Palmer, A. G. *Acc. Chem. Res.* **2015**, *48*, 457–65.
- (25) Schramm, V. L. *Acc. Chem. Res.* **2015**, *48*, 1032–1039.
- (26) Maunder, P.; Finglas, P.; Mallet, A.; Mellon, F.; Razzaque, M.; Ridge, B.; Vahteristo, L.; Witthoft, C. *J. Chem. Soc., Perkin Trans. 1* **1999**, 1311–1323.
- (27) Cocco, L.; Groff, J. P.; Temple, C.; Montgomery, J. A.; London, R. E.; Matwiyoff, N. A.; Blakley, R. L. *Biochemistry* **1981**, *20*, 3972–3978.
- (28) Selinsky, B. S.; Perlman, M. E.; London, R. E.; Unkefer, C. J.; Mitchell, J.; Blakley, R. L. *Biochemistry* **1990**, *29*, 1290–1296.
- (29) Cheung, H. T. A.; Birdsall, B.; Frenkiel, T. A.; Chau, D. D.; Feeney, J. *Biochemistry* **1993**, *32*, 6846–6854.
- (30) Cowart, M.; Falzone, C. J.; Benkovic, S. J. *J. Labelled Compd. Radiopharm.* **1994**, *34*, 67–71.
- (31) Saeed, M.; Tewson, T. J.; Erdahl, C. E.; Kohen, A. *Nucl. Med. Biol.* **2012**, *39*, 697–701.
- (32) Curthoys, N. P.; Scott, J. M.; Rabinowitz, J. C. *J. Biol. Chem.* **1972**, *247*, 1959–1964.
- (33) Moran, R. G.; Keyomarsi, K.; Colman, P. D. *Methods Enzymol.* **1986**, *122*, 309–312.
- (34) Gräwert, T.; Fischer, M.; Bacher, A. *IUBMB Life* **2013**, *65*, 310–322.
- (35) Fischer, M.; Thöny, B.; Leimkühler, S. The biosynthesis of folate and pterins and their enzymology. In *Comprehensive Natural Products II*; Mander, L.; Liu, H.-W., Eds.; Elsevier: Oxford, U.K., 2010; Vol. 7, pp 599–648.
- (36) Rebelo, J.; Auerbach, G.; Bader, G.; Bracher, A.; Nar, H.; Hösl, C.; Schramek, N.; Kaiser, J.; Bacher, A.; Huber, R.; Fischer, M. *J. Mol. Biol.* **2003**, *326*, 503–516.
- (37) Burg, A. W.; Brown, G. M. *Chem. Commun.* **1968**, *243*, 2349–2358.
- (38) Yim, J. J.; Brown, G. M. *J. Biol. Chem.* **1976**, *251*, 5087–5094.
- (39) Illarionova, V.; Eisenreich, W.; Fischer, M.; Haussmann, C.; Romisch, W.; Richter, G.; Bacher, A. *J. Biol. Chem.* **2002**, *277*, 28841–28847.
- (40) Schramek, N.; Bracher, A.; Bacher, A. *J. Biol. Chem.* **2001**, *276*, 2622–2626.
- (41) Dantola, M. L.; Vignoni, M.; Capparelli, A. L.; Lorente, C.; Thomas, A. H. *Helv. Chim. Acta* **2008**, *91*, 411–425.
- (42) Schultheisz, H. L.; Szymczyna, B. R.; Scott, L. G.; Williamson, J. R. *ACS Chem. Biol.* **2008**, *3*, 499–511.
- (43) Tolbert, T. J.; Williamson, J. R. *J. Am. Chem. Soc.* **1996**, *118*, 7929–7940.
- (44) Angelastro, A.; Dawson, W. M.; Luk, Y. P. L.; Allemann, R. K. *ACS Catal.* **2017**, *7*, 1025–1029.
- (45) Gross, A.; Abril, O.; Lewis, J. M.; Geresh, S.; Whitesides, G. M. *J. Am. Chem. Soc.* **1983**, *105*, 7428–7435.
- (46) Schoedon, G.; Redweik, U.; Frank, G.; Cotton, R. G.; Blau, N. *Eur. J. Biochem.* **1992**, *210*, 561–568.
- (47) Suzuki, Y.; Brown, G. M. *J. Biol. Chem.* **1974**, *249*, 2405–2410.
- (48) Gabelli, S. B.; Bianchet, M. A.; Xu, W.; Dunn, C. A.; Niu, Z. D.; Amzel, L. M.; Bessman, M. J. *Structure* **2007**, *15*, 1014–1022.
- (49) Mathis, J. B.; Brown, G. M. *J. Biol. Chem.* **1970**, *245*, 3015–3025.
- (50) Wang, Y.; Li, Y.; Yan, H. *Biochemistry* **2006**, *45*, 15232–15239.
- (51) Derrick, J. P. *Vitam. Horm.* **2008**, *79*, 411–433.
- (52) Weisman, R. A.; Brown, G. M. *J. Biol. Chem.* **1964**, *239*, 326–331.
- (53) Griffin, M. J.; Brown, G. M. *J. Biol. Chem.* **1964**, *239*, 310–316.
- (54) Richey, D. P.; Brown, G. M. *Biochim. Biophys. Acta, Gen. Subj.* **1970**, *222*, 237–239.
- (55) Chénault, H. K.; Simon, E. S.; Whitesides, G. M. *Biotechnol. Genet. Eng. Rev.* **1988**, *6*, 221–270.
- (56) Schweitzer, B. I.; Dicker, A. P.; Bertino, J. R. *FASEB J.* **1990**, *4*, 2441–2452.
- (57) Charlton, P. A.; Young, D. W.; Birdsall, B.; Feeney, J.; Roberts, G. C. K. *J. Chem. Soc., Perkin Trans. 1* **1985**, 1349–1353.
- (58) Fierke, C. A.; Johnson, K. A.; Benkovic, S. J. *Biochemistry* **1987**, *26*, 4085–4092.
- (59) Shrimpton, P.; Allemann, R. K. *Protein Sci.* **2002**, *11*, 1442–1451.
- (60) Shrimpton, P.; Mullaney, A.; Allemann, R. K. *Proteins: Struct., Funct., Genet.* **2003**, *51*, 216–223.
- (61) Chen, Y. Q.; Kraut, J.; Blakley, R. L.; Callender, R. *Biochemistry* **1994**, *33*, 7021–7026.
- (62) Wan, Q.; Bennett, B. C.; Wilson, M. A.; Kovalevsky, A.; Langan, P.; Howell, E. E.; Dealwis, C. *Proc. Natl. Acad. Sci. U. S. A.* **2014**, *111*, 18225–18230.
- (63) Stone, S. R.; Morrison, J. F. *Biochemistry* **1984**, *23*, 2753–2758.
- (64) Liu, C. T.; Francis, K.; Layfield, J. P.; Huang, X.; Hammes-Schiffer, S.; Kohen, A.; Benkovic, S. J. *Proc. Natl. Acad. Sci. U. S. A.* **2014**, *111*, 18231–18236.
- (65) Cioni, P.; Strambini, G. B. *Biophys. J.* **2002**, *82*, 3246–3253.
- (66) Schowen, K. B.; Schowen, R. L. *Methods Enzymol.* **1982**, *87*, 551–606.
- (67) Peracchi, A. *Trends Biochem. Sci.* **2001**, *26*, 497–503.

- (68) Loveridge, E. J.; Behiry, E. M.; Swanwick, R. S.; Allemann, R. K. *J. Am. Chem. Soc.* **2009**, *131*, 6926–6927.
- (69) Deng, H.; Callender, R.; Howell, E. *J. Biol. Chem.* **2001**, *276*, 48956–48960.
- (70) Mönch, S.; Netzel, M.; Netzel, G.; Rychlik, M. *Anal. Biochem.* **2010**, *398*, 150–160.
- (71) McCarty, R. M.; Somogyi, A.; Lin, G.; Jacobsen, N. E.; Bandarian, V. *Biochemistry* **2009**, *48*, 3847–3852.
- (72) Gregory, J. F., III; Williamson, J.; Liao, J.-F.; Bailey, L. B.; Toth, J. *P. J. Nutr.* **1998**, *128*, 1896–1906.
- (73) Gregory, J. F., III. *J. Nutr.* **2001**, *131*, 1376S–1382S.
- (74) Gregory, J. F., III; Quinlivan, E. P. *Annu. Rev. Nutr.* **2002**, *22*, 199–220.
- (75) Kopp, M.; Morisset, R.; Koehler, P.; Rychlik, M. *PLoS One* **2016**, *11*, e0156610.
- (76) de Ambrosis, A.; Vishnumohan, S.; Paterson, J.; Haber, P.; Arcot, J. *Eur. J. Clin. Nutr.* **2017**, *71*, 103–106.